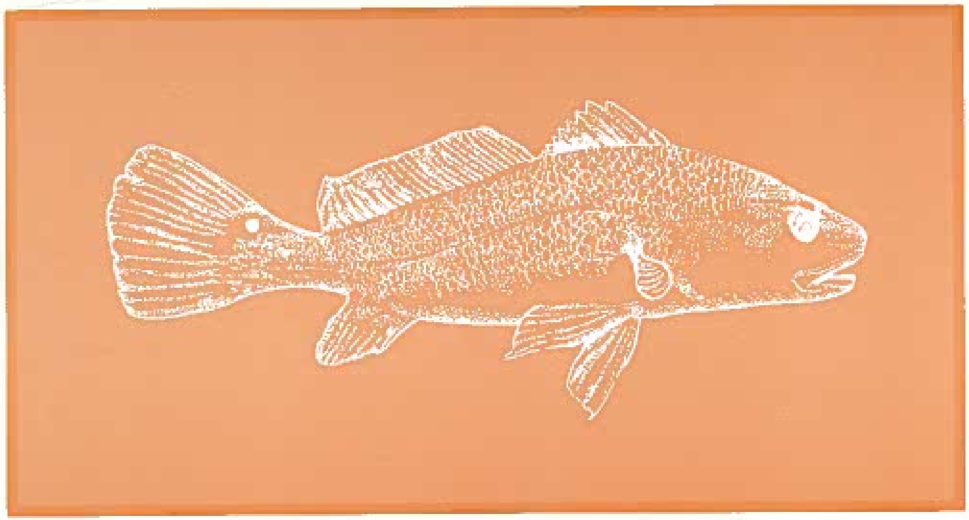
Red Drum Aquaculture



Aquaculture

Compilers
Dr. George W. Chamberlain
Dr. Russell J. Miget
Michael G. Haby

TEXAS A&M SEA GRANT COLLEGE PROGRAM

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There is an accompanying publication that resulted from a scientific conference held in Corpus Christi in 1987. This publication, **Red Drum Aquaculture, Proceedings of a Symposium on the Culture of Red Drum and Other Warm Water Fishes**, may be ordered for $6.00 from the following:

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Red Drum Aquaculture

Compilers
Dr. George W. Chamberlain
Texas Agricultural Extension Service

Dr. Russell J. Miget
Texas Marine Advisory Service

Michael G. Haby
Texas Marine Advisory Service

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The Life History of Red Drum

Gary C. Matlock
Texas Parks and Wildlife Department
4200 Smith School Road
Austin, Texas 78744

The red drum (*Sciaenops ocellatus* Linnaeus) is a quasi-catadromous sciaenid (Rounsefell, 1975) that ranges from Tuxpan, Mexico in the Gulf of Mexico (Gulf) to Massachusetts in the Atlantic Ocean (Hildebrand and Schroeder, 1928; Bigelow and Schroeder, 1954; Simmons and Breuer, 1962). Important directed fisheries for red drum have existed since the 1700s, but are mainly limited today to the Carolinean province (Briggs, 1974; Matlock, 1980). In Texas, red drum are harvested primarily in estuaries where they once comprised as much as 35 percent of the commercial fish landings. Recreational fishermen may retain five red drum per day in Texas.

Major conflicts have occurred over allocating red drum for 100 years (Matlock 1980). Recreational fishermen blame inadequate catches on commercial overfishing, and commercial fishermen accuse sport fishermen of having inadequate skill. Management usually attempted to reduce conflict by addressing recreational concerns without appreciably affecting commercial harvests (Matlock, 1980). Prior to the 1970s, the success of each regulation was rarely examined, and procedures for selecting them were seldom specified. Size limits were enacted in the 1920s to protect juvenile and adult fish (Pearson, 1929), and seasonal and area net closures were expanded over time to protect spawning adults (Heffernan and Kemp, 1980).

In Texas, additional regulations resulted until the sale of native red drum was prohibited in September 1981 because of overfishing (Matlock, 1982). Management's goal was modified from reducing conflict to obtaining optimum yield (OY) (Anonymous, 1982).

Identity and Morphology

Eggs and Yolk-sac Larvae

Only laboratory–spawned specimens have been described (Johnson *et al.*, 1977; Holt *et al.*, 1981a, 1981b), but both eggs and yolk-sac larvae are similar to those of other sciaenids in that they are small and lack pigmentation, which makes field identifications unreliable below the Family level (Marley, 1983). The spherical eggs are 0.86-0.98 mm in diameter, and usually contain one colorless oil droplet about 0.2.-0.4 mm in diameter (Johnson et al. 1977), have a perivitelline space less than 2 percent of the egg diameter, and have a clear, unsculptured chorion.

Larvae are 1.71.-1.79 mm standard length at hatching. [Illustrations of red drum eggs and larvae are presented in *Growth and Development of Red Drum Eggs and Larvae* elsewhere in this book.]

Larvae

Red drum larvae are difficult to identify because they are morphologically similar to Atlantic croaker (*Micropogonias undulatus*) except for pigmentation and anal fin spines. Sciaenids considered to be one species based on descriptions of Pearson (1929) and Hildebrand and Schroeder (1928) were caught in the Texas surf, reared in the laboratory, and later identified as both Atlantic croaker and red drum (Simmons and Breuer, 1962). However, recent descriptions of laboratory-spawned red drum larvae (Johnson *et al.*, 1977) are consistent with previous pigmentation descriptions (Pearson, 1929; Lippson and Moran, 1974; Hildebrand and Cable, 1934); myomere and anal fin ray descriptions (Pearson, 1929; Miller and Jorgensen, 1973); illustration of a sharp opercular spine (Jannke, 1971); and illustration of caudal fin elements (Topp and Cole, 1968). Fin development and pigmentation of laboratory-reared fish (Johnson *et al.*, 1977) were not consistent with the description of a similar sized individual given by Powles and Stender (1978). Descriptions of laboratory-reared fish are probably the most reliable because they are based on many fish from known parents.

Juveniles, Sub-Adults and Adults

External morphology and swim bladder and otolith shapes identify juveniles, sub-adults and adults. The young have two short tube-like diverticula on the carrot-shaped swim bladder, characteristics of the suprageneric group *Sciaenops* (Chao, 1978). These diverticula remain as fish mature and a pair of sac-like projections develop dorsolaterally in the anterior swim bladder. These projections fit into a body wall

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From "The Basis for the Development of a Management Plan for Red Drum in Texas," the author's Ph.D. dissertation for Texas A&M University, College Station, Texas.

cavity between the third and fourth pleural ribs and may be involved in sound reception in older fish. The otolith's sagitta has the sciaenid characteristics of a tadpole-shaped sulcus in its inner surface but is enlarged and slightly rectangular. Chao (1978) summarized the external morphology as: snout with five upper and five marginal pores; lower jaw with five pores; no barbel or lower jaw; mouth inferior; teeth villiform in bands; and gill rakers short.

The juveniles and adults differ externally in caudal fin shape and external color. Caudal fins are pointed in young and become slightly concave in adults (Pearson, 1929). Large black blotches are distributed over the sides and back in fish 100 mm (Hildebrand and Schroeder, 1928). A pronounced chromatophore enlargement at the upper caudal fin base appears at about 36 mm and remains throughout life. Lateral blotches enlarge until the fish reach about 150 mm, then fade and disappear. Adults are elongate, silvery reddish fish with an elevated back (Hildebrand and Schroeder, 1928; Pearson, 1929). The head is long, rather low, with a bluntish snout and a larger inferior mouth; body shape may be an adaptation to shallow surf habitat (Chao, 1978).

Reproductive Cycle

Spawning

Courtship behavior

Red drum are dioecious, and males court females before spawning. Drumming and nudging intensifies prior to spawning and may be a major stimulus for spawning, at least in the laboratory (Guest and Lasswell, 1978).

Spawning periodicity

Spawning occurs from August through January and peaks in September or October. This conclusion seems supported by both field and laboratory evidence, much of which is not well documented. Sexually mature adults, larvae, and small fish (<150 mm) occur mainly in fall (Table 1). Moreover, only simulated fall photoperiod and temperatures induced spawning in the laboratory (Arnold *et al.*, 1977; Roberts *et al.*, 1978a). The date of peak spawning varies among years, but bag seine collections in bays indicate October 1 as a reasonable hatching date for modeling purposes (Matlock, 1984).

Spawning areas

No study has comprehensively examined spawning areas, but probable areas can be suggested by a process of elimination using the criterion that the absence of red drum from larval collections probably indicates no spawning at the collection sites.

Spawning apparently does not occur in bays or the offshore gulf. Larvae do not occur beyond 20 km (18 m depths) from gulf beaches (Houde *et al.*, 1979; Arnold *et al.*, 1976; Finucane *et al.*, 1977). They are also absent from Aransas, Corpus Christi, and Alazan Bays (Hoese, 1965; Holland *et al.*, 1973, 1974; Dokken, 1981) where grass beds, channels, shorelines, areas near serpulid rock formations, and deep water were extensively, although not randomly, sampled. In Mississippi, larval abundance increased as distance from bays increased out to 20 km (Laroche, personal comment). On the Atlantic coast, bay and offshore collections also lack larval red drum (Pearson 1941; Massmann et al. 1961, 1962; Herman 1963; Crocker 1965; Dovel 1967), although most of these collections were made north of Virginia in areas where few red drum now occur.

Spawning seems to occur in the nearshore gulf, probably near bay-gulf passes. Many larvae (2.11 mm) have been captured in bay-gulf passes (Compton, 1965; Jannke 1971; King, 1971). In contrast, only one larval red drum was caught in two years of ichthyoplankton sampling in the Gulf (18 to 135 m deep) off Texas; that larva was caught at the shallowest (18 m) station (Arnold *et al.*, 1976; Finucane *et al.*, 1977). These recent studies confirm the nearshore gulf spawning site originally proposed by Pearson (1929) and supported by Simmons and Breuer (1962) and Yokel (1966) because they caught spent adults along gulf beaches and felt juveniles were most abundant near passes. Their evidence for this was weak, however, because: 1) the occurrence of ripe fish in bays was ignored, as were adult movements before and after spawning; 2) juvenile catches were not adjusted for distribution of sampling effort, although most collections were near bay-gulf passes; 3) spawning was not observed, and eggs were not collected; and 4) the offshore gulf was not sampled. Although "ripe" fish have been reported in bays (Pearson, 1929; Yokel, 1966), this may not indicate where spawning occurs because fish like red drum cruise at about three times their length per second (Harden Jones, 1968). So, a 750-mm adult could travel 194 km/d. Moreover, the term "ripe" was not defined and may or may not indicate imminent spawning.

Hydrographic considerations also suggest gulf spawning, probably easterly or northerly of estuaries. The larvae reported from passes are probably transported by nearshore gulf surface currents which are strongly along shore to the south and southwest and have an onshore component during fall. In addition, spawning outside estuarine nurseries is consistent with the triangular migration pattern typically exhibited by marine fish in which larvae are transported from spawning areas to spatially separate nurseries, and adults occupy separate feeding or overwintering grounds (Harden Jones, 1968).

Recruitment and Nurseries

Egg and larval transport

Gulf surface currents probably carry eggs and larvae from nearshore spawning areas to estuarine nurseries. Evidence to support this includes: 1) red drum eggs float in gulf salinities (25 to 35‰) (Holt *et al.*, 1981b); 2) nearshore surface currents in the gulf during fall are onshore and strongly along shore in a south to southwest direction (Watson and Behrens, 1970; Armstrong *et al.*, 1976); and 3) larvae concentrate at the surface in Texas passes (King, 1971). The same conclusion was reached for the sand seatrout, *Cynoscion arenarius*, (Shlossman and Chittenden, 1981) another estuarine sciaenid. Moreover, Standard and Chittenden (1984) and Murphy and Chittenden (1982) have suggested that spawning in non-estuarine fishes such as the banded drum, *Larimus fasciatus*, and the Gulf butterfish, *Peprilus burti*, is also correlated with south/southwest along-shore current transport at that time of year and that it may reflect a general correlation between spawning periodicity and current transport. However, gulf hydrology is not understood to the extent that existing physical oceanographic models can predict exactly where larvae caught in bay-gulf passes were spawned. (Brucks, personal communication).

Subsurface currents may also be involved in larval transport within passes and bays, but supporting evidence is weak and the relationship may depend on the type of estuary. Mansueti (1960) suggested that distribution within Chesapeake Bay, a two-layer flow with vertical mixing type estuary (Bowden, 1967), was determined by subsurface currents, although he did not have adequate current data. Along the gulf, where most estuaries are the vertically homogeneous type, vertical distribution of larvae is inconsistent. Jannke (1971) reported greatest catches of larvae near bottom in Florida, but King (1971) found greatest catches at the surface off Texas. Reasons for this apparent difference in the gulf are not clear, although wind conditions may strongly influence mixing in vertically homogeneous estuaries.

Nursery locations

No study has comprehensively examined nursery locations, but probable nurseries can be suggested by a process of elimination using the criterion that the absence of juveniles and sub-adults from a location probably indicates that area is not a nursery.

Red drum do not use the offshore gulf as a nursery. Despite extensive collections in depths from 4 to 400 m, juvenile and sub-adult fish have rarely, if ever, been captured with: 1) shrimp trawls (Hildebrand, 1954; Springer and Bullis, 1956; Miller, 1965; Chittenden and McEachran, 1976; Chittenden and Moore, 1977; Bryan *et al.*, 1982; Ross *et al.*, 1982); 2) fish trawls (Cody *et al.*, 1977); 3) midwater trawls (C.E. Bryan, personal communication); 4) longlines (Cody *et al.*, 1981; G. Graham, Texas A&M University unpublished data); 5) rods and reels (C.E. Bryan, personal communication; McEachron and Matlock, 1983); 6) fish traps (C.E. Bryan, personal communication); or 7) purse seines (Knapp, 1950; Christmas *et al.*, 1960; Dunham, 1972).

Juveniles and sub-adults only occasionally occur in the surf zone. Chittenden (personal communication) captured 10 fish about 300 mm on hook and line within 8 m of shore off Big Shell Beach on Padre Island in August 1980, a week after Hurricane Allen. This appears unusual, however, and may be related to that hurricane because more extensive seine collections rarely yield juveniles in the surf (Gunter, 1958; McFarland, 1963; Juneau, 1975; Modde and Ross, 1981).

Red drum utilize estuarine nurseries. Many scientists have captured large numbers of juveniles and sub-adults in estuaries (Mansueti, 1960; Yokel, 1966; Jannke, 1971; Matlock *et al.*, 1978; Hegen, 1982; Matlock, 1983; Holt *et al.*, 1983). A tolerance to low salinities that increases with larval age (Crocker *et al.*, 1981) may be an adaptation to estuarine conditions.

Distribution and Movements

Juveniles

Juvenile red drum occur in all Gulf and Atlantic estuaries from Chesapeake Bay to the Laguna Madre of Mexico. However, their abundance among Gulf estuaries varies, depending mainly on surface area (Yokel, 1966). Salinity may also affect distribution, probably by affecting survival in early life stages (Neff *et al.*, 1981; Wohlschlag, 1981). Although rain and river inflow cause salinities to be lower along the upper coast of Texas (17 +/- 4 ‰ annually in Galveston Bay) than on the lower coast (35 +/- 6 ‰ in upper Laguna Madre) (Matlock, 1984), juvenile abundance is similar.

Bay-gulf passes interconnect estuarine nurseries with Gulf spawning grounds and areas occupied by adults. Each estuary in Texas, except the upper Laguna Madre and San Antonio Bay, is connected to the Gulf by at least one pass (Brown *et al.*, 1974). These two bays are connected to the Gulf indirectly by the Intracoastal Waterway. Additional connections may be created when hurricanes create new passes or temporarily re-open old ones.

Little information exists on distribution of juveniles within estuaries. They apparently prefer shorelines, shallow water (<1.3 m), and sea-grass meadows. Collections in open-bay areas seldom yield juveniles (Mansueti, 1960; Jannke, 1971), but shoreline collections typically do (Hegen, 1982). Although reasons for their distribution are not known because the literature essentially describes only their presence and absence, red drum occur throughout the bays, over all substrates, in vegetated and nonvegetated areas, adjacent to human developments, in marshes, and in channels and rivers during winter (Simmons and Breuer, 1962; Yokel, 1966; Perret *et al.*,

Table 1. Summary of reported red drum spawning and peak spawning dates.

State	Study area	Author	Spawning Period	Spawning Peak	Basis	Matlock's comments
Texas	Aransas Bay	Pearson (1929)	Oct through Dec	Oct	Capture of larvae (5-60 mm) near bay-gulf	Few details given on sampling periods, locations and sampling design; method of back calculating from juvenile length to spawning date not given.
Texas	Aransas Bay	Gunter (1945)	Sept through mid-Nov	Not given	Capture of juveniles (size not given)	Method of back calculating from juvenile length to spawning date not given.
Texas	Aransas Bay	Miles (1951)	Sept through mid-Nov	Not given	Capture of juveniles (size not given)	Few details given.
Texas	Upper Laguna Madre	Simmons and Breuer (1962)	Nov through Jan	Nov in 1952 and 1953; Dec in 1955; Jan in 1950, 1953, 1954, 1955 and 1960	Capture of fish (50-125 mm TL) in April	Method of back calculating from juvenile length to spawning date not given.
Texas	Aransas Bay	Compton (1965)	Oct	Oct	Capture of 61 larvae (2-11 mm TL) in a bay-gulf pass in 1964.	Monthly sampling in 1964; October only in 1961, 1962 and 1963 (Bradley and Compton 1963, Compton and Bradley 1964).
Texas	Aransas Bay	King (1971)	mid-Aug through Oct	Sept in 1968; Oct 1969	Capture of 1,450 larvae (5-7 mm TL) in a bay-gulf pass	At least 3 consecutive sampling days each week, January 1968-March 1970.
Texas	Galveston Bay to lower Laguna Madre	Matlock and Weaver (1979a)	Oct through Jan	Not given	Capture of 61 larvae (11-24 mm TL) and 41 fish (<42 mm TL)	Sampling conducted Oct 1977-May 1978; spawning before Oct would not have been detected.
Mississippi	Mississippi Sound	Loman (1978)	Aug through Jan	Sept or Oct	Appearance of larvae (size not given) in nekton samples	Few details given.
Florida	Tampa Bay	Sykes and Finucane (1965)	Sept through Feb	Not given	Capture of 449 fish <117 mm TL	Monthly sampling Aug 1961 - Nov 1962.
Florida	Cedar Key and Bayport	Kilby (1955)	Sept through Nov	Not given	Capture of 151 juveniles 12-145 mm SL	Few details given.
Florida	Tampa Bay	Springer and Woodburn (1960)	Sept through Oct	Not given	Capture of 82 juveniles 20-126 mm SL	Monthly sampling Oct 1957-Dec 1958 for small and juvenile fish.
Florida	Southwestern Coast	Yokel (1966)	Sept through Oct	Oct	Capture of sexually developing fish	Few details given.

State	Study area	Author	Spawning Period	Spawning Peak	Basis	Matlock's comments
Florida	East Coast	Yokel (1966)	Sept through Jan	Not given	Capture of juveniles (size not given) in May and June	Few details given.
Florida	Buttonwood Canal, Everglades National Park	Roessler (1967)	Sept through Oct	Not given	Capture of fish 30-80 mm TL in Nov-Mar and growth of 20 mm/mo based on length frequencies	Monthly sampling Jan 1963-Dec 1964.
Florida	Little Shark River, Everglades National Park	Jannke (1971)	mid-Sept through mid-Feb	Oct	Capture of larvae <10 mm SL	Plankton net used on flood tide at river's mouth, Jan 1966-Dec. 1967.
North Carolina	Not given	Yokel (1966)	July through Dec	Sept or Oct	Capture of fish <76 mm TL	Few details given.
Maryland and Virginia	Chesapeake Bay	Mansueti (1960)	Aug through Nov	Not given	Capture of 171 fish 20-85 mm TL	Monthly sampling of several years; method of back calculating from juvenile length to spawning date not given.
Eastern U.S. Coast States	Not given	Welsh and Breder (1924)	Late fall - early winter	Not given	Capture of small fish	Few details given.

Table 2. Summary of minimum distances traveled (km) by recaptured red drum in Florida, 1961-1966. Data are from Ingle *et al.* (1962), Topp (1963), Beaumariage (1964, 1969), and Beaumariage and Wittich (1966). Original data are presented in four zones.

Year of tagging	Number tagged	1961		1962		1963		1964		1965		1966	
		10	>10	10	>10	10	>10	10	>10	10	>10	10	>10
1961	308	149	13	3	0	0	0	0	0	0	0	0	0
1962	171			70	9	3	0	0	0	0	0	0	0
1963	40					9	5	0	0	0	0	0	0
1964	153							55	5	0	0	0	0
1965	18									9	1	0	1

1980). Holt *et al.* (1983) found fish (6- 27 mm SL) prefer the area between sea grass (*Halodule* sp.) and non-vegetated bottoms.

Sub-adults

Sub-adults occur in the Gulf only occasionally. Recreational fishermen catch sub-adults in the surf in the fall but only infrequently (Weixelman, 1982). Few studies report sub-adults from the surf zone or the Gulf (see Nursery Areas section for references). However, McFarland (1963) seined three 440-mm fish in the Gulf during the day in summer 1960 and speculated more might be caught at night.

Sub-adults apparently remain in estuaries throughout the year and do not migrate seasonally between estuaries and gulf. Many tagged fish have been recaptured in winter in estuaries (Osburn *et al.*, 1982; Green *et al.*, 1985). In addition, fishery-independent gill and trammel net catch rates (Gunter, 1945; Matlock *et al.*, 1978; Hegen and Matlock, 1980; Hegen, 1981), recreational catch rates (Heffernan *et al.*, 1977; Breuer *et al.*, 1977), and commercial landings (Hamilton, 1980, 1981, 1982) either remain the same throughout the year or generally decline after recruitment of new year classes in fall.

The distances that sub-adults move are small, and these fish normally remain within one estuary. Fish are usually recaptured within 10 km of the tagging site (Table 2; Carr and Chaney, 1976; Adkins *et al.*, 1979; Osburn *et al.* 1982), so red drum do not exhibit the broad random movement that Moe (1972) reported for sciaenids in general. More than 77 percent of 1,339 tag returns were from the estuary where tagging occurred (Osburn *et al.*, 1982); 95 percent of all recaptures included fish that had only moved to adjacent estuaries.

Little information exists on distribution of sub-adults within estuaries, although temperature-induced movements do occur between shallow and deep water. Like the juveniles, sub-adults are distributed throughout each estuary. However, their habitat preferences are unknown. Vegetation and reef types and amounts vary among bays, but relative abundance generally does not (Matlock, 1984). Sub-adults are more abundant along shorelines or shallow water (<1.3 m) than in open bay in fall and spring, but will temporarily move to water >1.3 m to escape temperature extremes in winter (Matlock *et al.*, 1978; Matlock, 1979). Matlock *et al.* (1978) reported highest gill net catches adjacent to deep holes just prior to passage of cold fronts indicating these fronts move fish to deep water within estuaries. Pearson (1929), Gunter (1945), and Miles (1951) reported red drum emigrate from estuaries to the Gulf in winter, but this was based only on absence of fish in shoreline net sets then and appears wrong based on Matlock *et al.* (1978). Gunter's (1945) trammel net catches, moreover, were equally high in winter (3.4 fish/set) and summer (3.1 fish/set). Saline gradients may help orient localized movements required for behavioral thermoregulation (Owens *et al.*, 1981).

In the laboratory sub-adults are most active at night. Owens *et al* (1981) reached this conclusion by continuous electronic monitoring of movements, but the validity of extrapolation to wild fish is unknown.

Adults

Red drum permanently emigrate from estuaries to the gulf when sexually mature. Sexual maturity is probably reached in 3.5 years (Matlock, 1986). Adults (>750 mm) rarely occur in the estuaries (Matlock, 1984), but commonly occur in the Gulf (Weixelman, 1982; Ross *et al.*, 1982). This presence in the Gulf seems permanent because they are absent from bays. Moreover, Simmons and Breuer (1982) found that all of 30 returns from 134 fish (400-460 mm) tagged at Cedar Bayou, Texas, in April were from the Gulf.

Adult red drum in the Gulf are distributed to at least 113 km from shore. Most occur within 16 km of shore off Texas, but current literature is inadequate to describe in detail the distribution of adults in the Gulf. Charter boats catch these fish off Louisiana near oil rigs to 34 km offshore in 13 m deep water (Dugas *et al.*, 1979), and on reefs 113 km off Texas (Texas Parks and Wildlife Department, unpublished data). Fish were caught in purse seines about 19-21 km off the Chandeleur Islands, La., in 18 m deep water (National Marine Fisheries Service, unpublished data) and with fish trawls, bottom longlines, and shrimp trawls within 24 km off Texas (Cody *et al.*, 1977; Cody and Avent, 1980; Cody *et al.*, 1981; Ross *et al.*, 1982).

Feeding Behavior and Diet

Feeding

Red drum feed throughout the water column, but mainly at the bottom. This conclusion is based on the following: 1) they eat mostly bottom organisms in the wild; 2) fish 279-312 mm FL (fork length) eat live food in aquaria only when it is on a substrate (Yokel, 1966); and 3) fish searching for food in tanks apparently orient their body to the bottom by contacting it with their lower jaw and small pelvic fin ray extensions (Yokel, 1966). Prey are sucked up from the bottom by a rapid expansion of the branchial region or captured by biting the substrate.

Red drum feeding in shallow water often exhibit "tailing" behavior. In this activity their caudal and dorsal fins are out of the water as they feed on the bottom (Gunter, 1945; Simmons and Breuer, 1962; Yokel, 1966). Schools of tailing fish can be easily sighted which increases their vulnerability to bay fishermen.

Occasionally red drum feed at the surface. Schools have been sighted close to Gulf beaches feeding on other fish at the surface (Rohr, 1968). I have seen schools of red drum feeding at the surface on Gulf menhaden (*Brevoortia patronus*) at heated water discharges in Trinity Bay. Fish demonstrating this behavior are also easily sighted which probably increases their susceptibility to fishermen.

Diet

Red drum feed indiscriminantly within at least three ontogenetic stages and, after yolk-sac absorption, their diet is varied. Overstreet and Heard (1978) found 59 taxa in juveniles, sub-adults and adults from Mississippi Sound. The most common taxa were crustaceans and fishes, as found in many other studies (Table 3). Boothby and Avault (1971) considered red drum to be omnivorous feeders because 69 percent of 197 stomachs examined contained more than two foods. Even marsh rats have been reported (Pearson, 1929). As with most sciaenids, prey availability and size probably determines diet (Chao and Musick, 1977; Boothby and Avault, 1971).

Larvae begin exogenous feeding within four days of spawning and eat mainly zooplankton. The yolk-sac provides nourishment for the first four days post-spawn (Johnson *et al.*, 1977). Bass and Avault (1975) determined preference by simultaneously sampling zooplankton and fish and found fish <30 mm prefer calanoid and cyclopoid copepods while those 30-70 mm prefer mysids. Copepods are the common food of fish <25 mm regardless of geographic location (Colura and Hysmith, 1976; Odum, 1971). However, red drum 25-50 mm will utilize what is available in the absence of mysids (Colura and Hysmith, 1976).

Juveniles mainly eat small bottom invertebrates and young fish. Fish 60-100 mm rely heavily on amphipods (Bass and Avault, 1975). Penaeid shrimp, callinectid crabs, and fish (menhaden, gobies, mullet, killifish, and eels) predominate in fish larger than 100 mm (Table 3).

Adults eat mainly fish. Fish occur more frequently in comparison to invertebrates as red drum grow (Pearson, 1929; Yokel, 1966; Overstreet and Heard, 1978).

Male and female diets seem similar. Boothby and Avault (1971) reported no noticeable diet difference between sexes, but presented no supporting data. Red drum diet apparently changes among seasons and geographic locations. However, the change may be caused more by prey availability or size than by season. For example, shrimp and fish replaced crabs in winter in similarly sized (190-780 mm) Mississippi Sound fish (Table 3) when crabs were less abundant (Overstreet and Heard, 1978). Similar change in taxa occurred in summer at Hopedale, La. (Table 3). As fish grew through summer in earthern ponds polychaetes and amphipods replaced copepods (Table 3). In Caminada Pass, La., diet changed dramatically (Table 3) within seasons as prey availability and size changed (Bass and Avault, 1975).

Red drum diet may differ between day and night depending on fish size. Fish 35-40 mm ate mainly mysids in both day and night (Bass and Avault, 1975). However, fish 90-115 mm ate mainly palaemonid shrimp in the day and fish at night.

In summary, red drum diet varies with their size, temporally, and spatially probably because of prey availability and size of prey change.

Parasites and Diseases

The literature on red drum parasites and diseases deals primarily with their identification. [In Section IV of this manual, S.K. Johnson describes the life history and control of several parasites and diseases common to red drum.] Thirty-one organisms have been found on or in red drum, but copepods and cestodes are most frequent (Table 4). The occurrence of many species may be related to the omnivorous feeding habits of red drum (Yokel, 1966).

Infections have generally not been quantified. Yokel (1966) concluded that red drum are not heavily parasitized, but the studies he summarized generally failed to give the number of fish examined or infected or the number of parasites found.

Estimates of Life History Parameters

Fecundity

Females probably produce more than 500,000 eggs annually. Existing estimates are 551,600,-3,420,552 eggs/female (Table 5). However, they are based on few fish, undescribed techniques, or do not consider fish size.

Growth of Juveniles and Sub-Adults

Much information exists on juvenile and sub-adult growth. Their growth has been determined by: 1) measuring length changes in laboratory and pond-reared fish (Colura *et al.*, 1976; Arnold *et al.*, 1977; Johnson *et al.*, 1977; Roberts *et al.*, 1978b; Trimble, 1979; Hein and Shepard, 1980; Crocker *et al.*, 1981; Holt *et al.*, 1981a; Hysmith *et al.*, 1983) and in tagged wild fish (Simmons and Breuer, 1962; Matlock and Weaver, 1979b; Perret *et al.* 1980; McKee, 1980); 2) length-frequency analysis of wild fish (Pearson, 1929; Miles 1951; Simmons and Breuer, 1962; Bass and Avault, 1975); 3) counting rings (annuli) on scales of wild fish (Welsh and Breder, 1924; Pearson, 1929; Wakefield and Colura, 1983), and 4) counting rings on otoliths of wild fish (Thieling and Loyacana, 1976; Rohr, 1980). However, growth is still not accurately known because the application of laboratory and pond studies to the natural environment may not be valid, and most studies on wild fish (Table 6) are unreliable because they are based on few fish, inadequate description of procedures, inadequate statistical analysis, gear selection bias, questionable data, and failure to adjust for time of annulus formation.

Table 3. Percentage of red drum intestinal tracts containing each of ten types of food items. Length data from Mississippi Sound, Hopedale, La., and Georgia is FL.

Location/ Date	Total length range (mm)	Number of fish with food	Fish	Shrimp	Crabs	Stoma-topods	Amphi-pods	Cope-pods	Mol-lusks	Echino-derms	Poly-chaetes	Mysids	Reference
Aransas Bay, TX 2/27-4/27	60-680	235	16-32	62-78	7-23	0	0	0	2-18	0	0	0	Pearson (1929)
Mississippi Sound, MI Spring 1977	190-780	34	41	24-32	71	18	0	0	0	9	15	0	Overstreet and Heard (1978)
Mississippi Sound, MI Winter 1976	190-780	26	27	31-46	65	0	4	0	0	0	0	0	Overstreet and Heard (1978)
Mississippi Sound, MI Fall 1976	190-780	14	50	71-79	36	21	14	0	0	0	0	0	Overstreet and Heard (1978)
Mississippi Sound, MI 11/61	992-1049	3	0	0	100	0	0	0	0	0	0	0	Rohr (1968)
Sapelo Island Ga. (Gulf) 6/70-8/70	430-1020	16	19-63	0	31-83	0	0	0	6-19	38-64	0	0	Overstreet and Heard (1978)
Lake Pontchar-train, LA 7/53-5/55	184-625	12	17	0	62	0	1	0	0	0	0	0	Darnell (1958)
Aransas Bay, TX 3/41-11/42	245-745	199	25	48-68	44-54	0	4	0	0	0	0	0	Gunter (1945)
Aransas Bay, TX (9/49-8/50	60-162	123	20	79	13	1	0	0	2	0	8	0	Miles (1950)
Earth ponds, Palacios, TX 5/75-9/75	25	38	0	22	22	0	0	56	0	0	0	0	Colura and Hysmith (1976)
Clear Lake TX 1/58-12/58	58-274	51	25-50	<25	<25	0	0	0	0	0	<25	0	Diener *et al.* (1974)
Biloxi Marsh Complex, LA 7/60-6/68	Not given	110	23-36	16-23	39-68	0	6	0	0	0	1	0	Fontenot and Rogilio (1970)
East Bay, TX 6/54-7/54	Not given	4	50	75	25	0	0	0	0	0	0	0	Reid (1956)

Location/ Date	Total length range (mm)	Number of fish with food	Fish	Shrimp	Crabs	Stoma- topods	Amphi- pods	Cope- pods	Mol- lusks	Echino- derms	Poly- chaetes	Mysids	Reference
East Bay, TX Summers 1954 and 1955	484-523	6	33	<100	0	0	0	0	0	0	0	0	Reid *et al.* (1956)
Hopedale, La. Winter 1967	307-1100	54	81	33	39	0	0	0	0	0	0	0	Boothbay and Avault (1971)
Hopedale, La. Spring 1968	307-1100	61	83	38	31	0	0	0	0	0	0	0	Boothby and Avault (1971)
Hopedale, La. Summer 1968	307-1100	79	86	73	61	8	0	0	0	0	0	0	Boothbay and Avault (1971)
Hopedale, La. Fall 1967 and 1968	307-1100	155	65	57	57	0	0	0	0	0	0	0	Boothby and Avault (1971)
Flamingo, Ever- glades, National Park, FL 1/61-9/61	Not given	179	16-34	16-69	16-60	2	0	0	0	0	2-8	0	Yokel (1966)
10,000 Islands, FL 1/61-9/61	Not given	190	22-41	7-27	12-71	0	0	0	0	0	2	0	Yokel (1966)
Caminada Pass, LA Winter 1971-72	10-23	5	0	0	0	0	0	100	0	0	0	0	Bass and Avault (1975)
Caminada Pass, LA Winter 1971-72	25-35	93	0	0	0	0	0	81	0	0	0	19	Bass and Avault (1975)
Caminada Pass, LA Winter 1971-72	36-47	140	5	0	0	0	7	13	0	0	0	67	Bass and Avault (1975)
Caminada Pass, LA Winter 1971-72	48-59	95	5	1	0	0	23	3	0	0	0	65	Bass and Avault (1975)
Caminada Pass, LA Winter 1971-72	60-71	58	16	2	1	0	55	0	0	0	1	23	Bass and Avault (1975)
Caminada Pass, LA Winter 1971-72	72-82	31	30	0	0	0	28	0	0	0	6	1	Bass and Avault (1975)
Caminada Pass, LA Winter 1971-72	83-94	26	57	5	1	0	21	0	0	0	6	0	Bass and Avault (1975)
Caminada Pass, LA Winter 1971-72	95-106	19	6	22	8	0	17	0	0	0	14	0	Bass and Avault (1975)
Caminada Pass, LA Winter 1971-72	107-118	16	33	40	18	0	10	0	0	0	2	0	Bass and Avault (1975)

Table 3. (continued)

Location/ Date	Total length range (mm)	Number of fish with food	Fish	Shrimp	Crabs	Stoma-topods	Amphi-pods	Cope-pods	Mol-lusks	Echino-derms	Poly-chaetes	Mysids	Reference
Caminada Pass, LA Winter 1971-72	119-130	13	19	60	16	0	2	0	0	0	0	0	Bass and Avault (1975)
Caminada Pass, LA Winter 1971-72	131-142	9	27	17	14	0	1	0	0	0	0	0	Bass and Avault (1975)
Caminada Pass, LA Winter 1971-72	143-153	9	21	66	13	0	0	0	0	0	0	0	Bass and Avault (1975)
Caminada Pass, LA Winter 1971-72	154-165	10	52	35	4	0	0	0	0	0	4	0	Bass and Avault (1975)
Caminada Pass, LA Winter 1971-72	166-177	5	6	21	6	0	0	0	0	0	3	0	Bass and Avault (1975)
Caminada Pass, LA Winter 1971-72	178-189	3	30	15	15	0	0	0	0	0	0	0	Bass and Avault (1975)
Caminada Pass, LA Winter 1971-72	190-200	5	13	25	63	0	0	0	0	0	0	0	Bass and Avault (1975)
Caminada Pass, LA Winter 1971-72	201-212	2	2	0	0	0	0	0	0	0	0	99	Bass and Avault (1975)
Caminada Pass, LA Winter 1971-72	213-224	2	0	17	33	0	0	0	0	0	0	50	Bass and Avault (1975)
Rockport and Pt. Aransas, TX 6/49-9/49	Not given	360	18-28	45-65	8-17	0	0	0	1	0	0	0	Kemp (1949)
Arroyo Colorado TX 12/67-6/68	120-150	5	60	40	20	0	0	0	0	0	0	0	Bryan (1971)
Galveston-Pt. Aransas, TX 6/48-8/48	Not given	>754	19	64	43	0	0	0	0	0	0	0	Knapp (1950)

Table 4. **Summary of literature reporting parasites from red drum (NG = not given).**

Parasite	Infection: Site	Infection: Extent	Fish: Number examined	Fish: Total length (mm)	When examined	Capture: Geographic location	Capture: Date	Reference
Bacteria (anaerobic)								
Catenbacterium sp. (Tentative identification)	Brain, liver, kidney, blood	NG	NG	200-300	At necropsy	Port Aransas Galveston	5/73 (Probably)	Henley and Lewis (1976)
Protozoa								
Henneguya ocellata	Epitheleum of intestine and pyloric caeca	60 cysts	447	185-745	At necropsy	Everglades National Park and Florida Bay, Fla.	1/61-9/61	Iversen and Yokel (1963)
Myxobolus (Henneguya) sp.	Epitheleum	NG	NG	NG	NG	Beaufort, NC	NG	Linton (1901)
Nematoda								
Ascaris sp. (Adult)	NG	9 worms	NG	NG	At necropsy	Sandy Hook, NJ (Fulton Market, NY	9-1886	Linton (1901)
Ascaris sp. (Immature) (Probably same species as adult)	Peritoneum (encapsulated)	NG	NG	NG	At necropsy	Sandy Hook, NJ (Fulton Market, NY	9-1886	Linton (1901)
Contracaecum sp.	Mesenteries of guts	NG	NG	NG	NG	Everglades National Park, Fla.	9/60-12/61	Yokel (1966)
Contracaecum collieri	NG	NG	7	NG	After preservation	Galveston, TX	NG	Chandler (1935)
Dichelyne fastigatus	Intestine	2 parasites	7	NG	After preservation	Galveston, TX	NG	Chandler (1954)
Heterakis sp.	NG	NG	NG	NG	NG	Beaufort, NC	NG	Linton (1905)
Platyhelminthes								
Trematoda								
Bucephaloides sp.	NG	Found in all fish	6	NG	NG	Grand Isle, LA	NG	Sparks (1958)
Bucephaloides megacirrus	Intestine	NG	NG	NG	NG	Alligator Harbor, Fla.	NG	Riggin and Sparks (1962)
Fimbriatus fimbriatus Von Wicklen	NG	NG	NG	NG	NG	Grand Isle, LA	NG	Sparks (1958)
Diostomum vitellosum Linton	NG	NG	NG	NG	NG	Beaufort, NC	NG	Linton (1905)
Diostomum areolatum Rudolphi	NG	NG	NG	NG	NG	Beaufort, NC	NG	Linton (1905)
Diostomum tenue Linton	NG	NG	NG	NG	NG	Beaufort, NC	NG	Linton (1905)
Cestoda								
Poecilancistrium robustus (Chandler)	Muscles	NG	NG	NG	NG	Chokoloskee, Fla.	9/60-12/61	Yokel (1966)
Poecilancistrium robustum (Chandler)	Muscles	60.0%	5	NG	At necropsy	Gulf of Mexico off Pt. Aransas, TX	6/58-8/58	McFarland (1959)

Table 4. (continued)

Parasite	Infection: Site	Infection: Extent	Fish: Number examined	Fish: Total length (mm)	When examined	Capture: Geographic location	Capture: Date	Reference
Poecilancistrium caryophyllum	Musculature	10.5%	19	40-510	At necropsy	Mississippi Sound	6/72-11/75	Overstreet (1977)
Scolex polymorphus	Muscles	NG	NG	NG	NG	Beaufort, NC	NG	Linton (1905)
Annelida								
Hiurdinea								
Myzobdella lugubria	NG	NG	NG	NG	After preservation	NG	NG	Sawyer *et al.* (1975)
Arthropoda								
Eucrustacea								
Copepoda								
Braachiella gulosa Wilson	Gills	NG	NG	NG	NG	Tampa & Sarasota Bays, Fla.	9/60-2/61	Yokel (1966)
Braachiella gulosa Wilson	Operculum (inside)	NG	NG	NG	NG	Chokoloskee, Fla.	9/60-12/61	Yokel (1966)
Brachiella gulosa Wilson	NG	NG	NG	NG	NG	Wilmington, NC	9/60-12/61	Yokel (1966)
Brachiella gulosa Wilson	NG	NG	NG	NG	NG	Pt. Aransas, TX	6/52-7/52	Causey (1953)
Brachiella gulosa Wilson	Gills	3 parasites from 1 fish	NG	NG	NG	Pt. Aransas, TX	5/51-7/51	Pearse (1952)
Brachiella gulosa Wilson	Gills	NG	NG	NG	NG	Woods Hole, Mass.	1931	Wilson (1932)
Brachiella gulosa Wilson	Operculum	NG	NG	NG	NG	Washington, DC (Fish market)	NG	Wilson (1915)
Brachiella gulosa Wilson	Gills and Operculum	NG	NG	NG	NG	Lemon Bay, Fla.	NG	Bere (1936)
Brachiella intermedia	NG	NG	NG	NG	NG	Wilmington, NC	9/60-12/61	Yokel (1966)
Brachiella intermedia	Operculum	NG	NG	NG	NG	Chokoloskee, FL	9/60-12/61	Yokel (1966)
Brachiella intermedia	Gills and Operculum	NG	NG	NG	NG	Lemon Bay, Fla.	NG	Bere (1936)
Echetus typicus Kroyer	Operculum (inside)	NG	NG	NG	NG	Chokoloskee, Fla.	9/60-12/61	Yokel (1966)
Echetus typicus Kroyer	NG	NG	NG	NG	NG	Wilmington, NC	9/60-12/61	Yokel (1966)
Echetus typicus Kroyer	Gills	NG	NG	NG	NG	Pt. Aransas, TX	6/52-7/52	Causey (1953)
Echetus typicus Kroyer	NG	18 parasites	6	NG	NG	Pt. Aransas, TX	5/51-7/51	Pearse (1952)
Echetus typicus Kroyer	Gill cavity	NG	NG	NG	NG	Lemon Bay, Fla.	NG	Bere (1936)
Echetus typicus Kroyer	Operculum	NG	NG	NG	NG	Washington, DC (fish market)	NG	Wilson (1905)
Lernaeenicus Radiatus (LeSueur)	NG	NG	NG	NG	NG	Wilmington, NC	9/60-12/61	Yokel (1966)
Anilocera laticauda Milne Edwards	NG	1 parasite	6	NG	NG	Pt. Aransas, TX	5/51-7/51	Pearse (1952)

Parasite	Infection: Site	Infection: Extent	Fish: Number examined	Fish: Total length (mm)	Fish: When examined	Capture: Geographic location	Capture: Date	Reference
Caligus repax Milne Edwards	NG	NG	NG	NG	NG	Upper Laguna Madre, TX	NG	Simmons (1957)
Caligus bonito Wilson	NG	NG	NG	NG	NG	Upper Laguna Madre, TX	NG	Simmons (1957)
Caligus haemulonis Kroyer	Mouth	NG	NG	NG	NG	Pt. Aransas, TX	6/52-7/52	Causey (1953)
Caligus haemulonis	NG	2 parasites	6	NG	NG	Pt. Aransas, TX	5/51-7/51	Pearse (1952)
Lernanthropus paenulatus Wilson	Gills	NG	NG	NG	NG	Pt. Aransas, TX	6/52-7/52	Causey (1953)
Caligus sciaenops n. sp.	Gills	2 parasites from 1 fish	6	NG	NG	Pt. Aransas, TX	5/51-7/51	Pearse (1952)
Lernanthropus longipes Wilson	NG	2 parasites	2	NG	NG	Pt. Aransas, TX	5/51-7/51	Pearse (1952)

Table 5. Summary of available information on red drum fecundity.

Author	Study location	Source of fish	Number involved	Total length (mm)	Fecundity (eggs/female)	Matlock's comments
Roberts *et al.* (1978a)	Florida	Laboratory	4	Not given	2,107,617	Estimating techniques and precision not given; number of spawning fish unknown but ≤2.
Roberts *et al.* (1978b)	Florida	Laboratory	8	Not given	551,600	Estimating techniques and precision not given; number of spawning fish unknown but ≤2
Pearson (1929)	Texas	Aransas Bay	1	900	3,382,886 or 3,420,552	Volumetric and weight subsample techniques used; error not estimated for either method.
Miles (1951)	Texas	Aransas	1	Not given	2,500,000	No details given.

Table 6. Summary of information on red drum growth rate (blanks indicate no estimates given).

Environment	State	Reference	Growing period	Length of rearing period (d)	Initial size or age	Total length growth rate (mm/d)	Matlock's comments
Laboratory	Texas	Arnold *et al.* (1977)	Not given	570	44 mm TL	0.70-1.14	Growth was 1.14 mm/d in first 180 d and 0.70 mm/d in last 390 d; no other details given.
Raceways	Texas	Crocker *et al.* (1981)	July-Aug 1979	30	72 mm TL	1.7 1.3	Analysis of covariance used to test for differences in growth between salinities, but variance homogeneity assumption apparently violated so conclusion of significant difference is questionable; growth rate exceeded 1.0 mm/d regardless; 1.0 mm/d regardless; >93% survival in both treatments.
Ponds	Alabama	Trimble (1979)	Oct. 1976-May 1979	136-946	2 d	Not given; presented weight data only	Disease problems rampant; data not statistically analyzed; incomplete detail on procedures used to estimate size at stock stocking, aampling techniques, and growth in weight estimates. I estimated growth at 0.80 mm/d after converting weight to length and fitting regression to mean length (r = 0.973).
Ponds	Louisiana	Hein and Shepard (1980)	Oct. 1978-Jan. 1979	79	27.2 mm TL	0.92	19 of 1,000 fish measured initially and all 1,000 placed in one 0.1-ha pond; 86 fish measured at harvest.
Ponds	Texas	Colura *et al.* (1976)	Aug - Nov 1975	27-37	2-6 d	1.02-1.66	No adjustments for stocking rate variations (156,000-880,000 larvae/ha); stocking rate estimating procedures not given; estimating procedures for mean size at stocking or harvested not given; survival in ponds very low (<10%); few details given.
Ponds (received heated power plant effluent)	Texas	Luebke and Strawn (1973)	June 8-Nov. 6, 1972	151	272-295 mm TL	0.76-0.85	Estimating procedure not clearly defined; only 13% mortality. I used linear regression and estimated growth at 0.70 mm/d (r = 0.99).

Environment	State	Reference	Growing period	Length of rearing period (d)	Initial size or age	Total length growth rate (mm/d)	Matlock's comments
Ponds	Texas	Hysmith *et al.* (1983)	Nov. 7, 1975-Apr. 28, 1976	108-173	41 mm TL	0.66±0.04 (Fed) 0.35±0.06 (Unfed)	Found no significant influence of stocking density (5,000, 10,000, 15,000 fish/ha) on growth but did not significantly higher growth in fish fed artificial diet than in those not fed; no indication of reduced growth in winter; few details on sampling techniques used to obtain measured fish.
Power plant cooling lake	Texas	McKee (1980)	Nov. 1975-Nov. 1977	Not given	366-837 mm TL	0.49±0.06	Based on 27 recaptured tagged fish; growth rate (Y) decreased significantly with increased size at tagging, according to Y = 0.75925 0.00246 X ($X = SL_{mm}$ at tagging).
Wild	Florida	Perret *et al.* (1980)	1961-1965	Not applicable	282-655 mm TL	0.04-0.66	Based on data from 12 recaptured tagged fish published by Ingle *et al.* (1962), Topp (1962), Beaumariage and Wittich (1966); no statistical analysis conducted.
Wild	Florida	Roessler (1967)	Jan. 1963-Dec. 1964	Not applicable	30 mm TL	0.67	Based on 106 juveniles, 94.3% of which were caught in Dec-Feb; no statistical analysis conducted.
Wild	Louisiana	Bass and Avault (1975)	Oct. 1971-May 1972	Not applicable	16 mm SL	0.62	I estimated growth using linear regression fit to mean size on each sampling data (Oct. 30 was considered day 0); r = 0.989.
Wild	Texas	Matlock and Weaver (1979b)	Nov. 1975-Sept. 1976	Not applicable	275-815 mm TL	0.43±0.08	Based on 110 recaptured tagged fish from Texas bays; no significant difference in growth among bays; no apparent change in growth with increased size at tagging but no staistical analysis conducted; data obtained from fishermen.
Wild	Texas	Simmons and Breuer (1962)	Feb-Dec	Not given	50 mm	1.00	I estimated growth based on plot of model lengths of bag seine caught fish in upper Laguna Madre; no other details given.
Wild	Texas	Matlock *et al.* (1986)	June 1979-May 1980	350	41 mm TL	1.03±0.05	Based on 48 recaptured stocked fish from St. Charles Bay; artificially reared juveniles stocked out of phase with wild fish so identifiable by size; fish grew through two summers in first year so growth rate should be greater than wild fish.

Although much growth information exists, few mathematical expressions relating length and age of red drum have been developed. Rohr (1980) Wakefield and Colura (1983) estimated parameters in the von Bertalanffy equation using otoliths and scales from Louisiana and Texas. However, Rohr published only an abstract, and Wakefield and Colura collected fish from a few bays only.

Juveniles in Texas reach 90-95 mm at 6 months and 115-150 mm at 8 months (Matlock, 1984). Juveniles generally grew in size continuously and in an exponential manner during their first 300 days without slowing during winter. Sub-adults reach 330-360 mm at 18 months, and 550-600 mm at 26 months (Matlock, 1984). These growth estimates agree closely with that based on the von Bertalanffy equation for Texas fish (based on Pearson's 1929 data) but not for South Carolina fish (Matlock, 1984). Values for K, L_{∞}, and t_0 were 0.295, 1068 mm, and + 0.144/year (Texas) and 0.449, 945 mm, and -0.324/year (South Carolina). Predicted length at age for the von Bertalanffy equations were 106, 352, and 480 mm (Texas) and 292, 528, and 636 mm (South Carolina) at 6, 18, and 26 months.

Seemingly exponential growth explained more than 68 percent of the variation in juvenile lengths (Matlock, 1984). Their growth may not really be exponential, however, because gear selection overestimates length early and underestimates it late in each phase. The estimate that size at 0.8/year is 160-240 mm (Matlock, 1984) may be too low, but it agrees with the von Bertalanffy length estimate of 190 mm at 0.8/year using Pearson's data. Mean daily growth during the first 8 months of life (0.53-0.80 mm) is similar to published rates (Table 6) that approximate 0.6 mm/d in the wild and 1.0 mm/d in controlled laboratory, raceway, or pond environment where growth may be higher.

Reported values of K in the von Bertalanffy equation are 0.3 (Pearson's data), 0.4 (Rohr 1980), 0.35-0.52 (Wakefield and Colura 1983), and 0.45 (Thieling and Loyacano's data). The von Bertalanffy equation based on Pearson's data may be most reasonable because: 1) L_{∞} from it is below but reasonably close to the largest fish on record in Texas; 2) length at ages one and two years estimated from it are consistent with lengths from bag seines and trammel net collections; 3) his data were based on the greatest size range; and 4) he included fish collected in the gulf where the largest fish occur. Wakefield and Colura's (1983) and Thieling and Loycano's (1976) estimates were based on data from estuaries and may reflect size bias because old large fish emigrate to the Gulf whereas Rohr's estimate may reflect unreliable otolith readings in old fish. These authors estimate L_{∞} as being less than 950 mm which may reflect inadequate representation of old fish, because many fish larger than 950 mm occur in the Gulf.

Mortality

Much work has been done on red drum mortality. However, except for recent work on total mortality by Matlock and Weaver (1979b) and Green *et al.* (1985), parameters needed for yield assessments have not been emphasized. Past work has emphasized causes of infrequent massive kills (Gunter, 1941, 1952; Gunter and Hildebrand, 1951; Simmons, 1957; Simmons and Breuer, 1962; Bryan, 1971; Harrington, 1973; Cardeilhac *et al.*, 1981; Amos, 1981), influence of temperature and salinity on hatching success and larval mortality (Holt *et al.*, 1981a; Crocker *et al.*, 1981), larval cannibalism (Arnold *et al.*, 1977), effects of the IXTOC I oil well blowout on larval morphology and survival (Rabalais *et al.*, 1981), and parasites and diseases of age one year and older fish (Yokel, 1966) The available literature on red drum predators provides little insight into mortality because few predators have been identified (Gunter, 1942, 1945; Kemp, 1949; Reid *et al.*, 1956; Darnell, 1958; Caldwell and Caldwell, 1972; Diener *et al.*, 1974; Moffett *et al.*, 1979; Overstreet and Heard, 1982).

Instantaneous annual total mortality (Z) of red drum in Texas bays approximate 1.4 to 1.9. An estimate of 1.6 by Matlock (1984) was based upon catch curve analysis and agrees with reported values of Z = 1.39 and 1.90 + 0.13 based on tagging (Matlock and Weaver, 1979a; Green *et al.*, 1985). However, these estimates appear extremely high for such an apparently long-lived fish, based on theoretical estimates of M (0.3-0.5) using the relationship between mortality and maximum age (Royce, 1972). Catch curve and tagging data analysis may overestimate Z if there is: 1) handling mortality of tagged fish or tag loss; 2) gear selection in which the mesh becomes less and less capable of capturing fish as they grow in size; or 3) change in availability due to seasonal emigration away from shorelines in bays or an emigration to the gulf as the sub-adults mature.

Handling mortality is probably minimal because tagged fish were handled carefully and Elam (1971) found mean mortality of only 6.8 percent for tagged sub-adults held in ponds for 35-424 days. Tag loss was <13 percent. Together, these sources of error may add up to 7 to 20 percent underestimate of Z.

Gear selection likely occurred because trammel nets are selective. Its magnitude is unknown, but recruitment of sub-adults was knife-edged (e.g. occurred in a few months), the effects of which would anchor the left end of a catch curve and emphasize gear selection of larger fish (Matlock, 1984). Change in availability due to emigration must have occurred. Its magnitude also is not known, but 100 percent of the sub-adults emigrate to the Gulf when they reach sexual maturity; they must spend two to four years as sub-adults assuming they reach sexual maturity in three to five years, and spend one year as juveniles.

Their average annual change in availability (Z) due to emigration to the Gulf can be estimated using Royce's (1972) approach, substituting the duration of the sub-adult stage for life span; doing so, average annual values of Z are 0.92-1.53. This range of values approximates values of Z estimated via catch curve analysis and suggests simple emigration to the gulf could account for much of the large values of Z found by catch curve analysis if emigration occurred constantly for three to five years. This would be true for any catch curve analysis, whether the fish were tagged or not, assuming they emigrate equally and continuously to the Gulf. However, emigration is knife-edged, not continuous. Therefore, Royce's (1972) approach probably yields gross underestimates of Z in bays when maximum age instead of maximum time spent in bays is used.

Although emigration to the Gulf occurs, fishing may cause most mortality of sub-adults in bays. Based on tag recaptures, at least 17 percent of the sub-adults are caught by fishermen in bays within one year after release (Green *et al.*, 1985). However, this is a gross underestimate of bay tag recaptures because only 29 percent of the recaptures get reported by recreational fishermen (Matlock, 1981). Nonreporting may even be higher than 29 percent for commercial fishermen because they intentionally discard tags (Matlock, 1984).

Red drum are quite susceptible to fishing. Their schooling and tailing behavior may make them easily located and captured. Reported tag recaptures in Florida have been over 47 percent when rewards of $10,000 were given for returned tags (Beaumariage, 1969).

Additional Research

All five gulf states, several universities, and the federal government, including National Marine Fisheries Service are conducting additional research on the life history of red drum in the Gulf of Mexico. Several private companies are also involved in aquaculture research of red drum. Much information has been generated since 1984 to improve the life history parameter estimates, but the basic life history description presented herein remains unchanged. The reader is referred to the U.S. Secretary of Commerce's Red Drum Fishery Management Plan for additional information.

Personal Communications

Mr. John Brucks, National Marine Fisheries Service, National Space Technology Laboratory, NSTL Station, Miss., pers. commun., February 1983.

Mr. C.E.. Bryan, Texas Parks and Wildlife Department, Austin, Tex., pers commun., April 1983.

Dr. Mark E. Chittenden, Jr., Texas A&M University, College Station, Tex., pers. commun., April 1983.

Ms. Joanne L. Laroche, Gulf Coast Research Laboratory, Ocean Springs, Miss., pers. commun., April 1983.

Literature Cited

Adkins, G., J. Tarner, P. Bowman, and B. Savoie. 1979. A study of commercial finfish in coastal Louisiana. **La. Dep. Wildl. Fish., Tech. Bull.** 29:62.70.

Amos, A.F. 1981. Redfish kill in the Gulf of Mexico: June-July 1981, R/V KATY CRUISE REDFISH—1, hydrographic observations. Univ. Tex. Mar. Sci. Inst., Port Aransas, Texas. (Unpublished manuscript).

Anonymous. 1982. Texas Parks and Wildlife Department, Coastal Fisheries Plan, FY 1982 progress and FY 1983 plan. PWD Report 3000-74, 34 pp.

_________ 1983. Saltwater finfish research and management in Texas, a report to the Governor and the 68th Legislature. Tex. Parks Wildl. Dep., PWD Report 3000-154, 48 pp.

Armstrong, R., M. Devine, J.A. Martin, R.F Temple, A. Vastro, C. Warsh and R. Whitaker. 1976. *n*: W.B. Jackson (editor), **Environmental Studies of the South Texas Continental Shelf, Vol. II. Physical oceanography.** NOAA Final Report to Bur. Land Management, Interagency Agreement No. 08550-IA5-19, 290 pp.

Arnold, C.A., C.W. Caillouet, Jr., M.E. Chittenden, Jr., J.H. Finucane, R. Juhl, J.A. Martin, J.D. McEachran, E.L. Nakamura, E.T. Park, R.F. Temple and W.L. Trent. 1976. p. 1-425. *In*: W.B. Jackson (editor), **Environmental Studies of the South Texas Outer Continental Shelf 1975, Vol. 1. Plankton and fisheries.** NOAA Final Rep. to Bur. Land Mngmnt., Interagency Agreement No. 08550-IA5-19, 425 pp.

Arnold, C.R., J.D. Williams, A. Johnson, W.H. Bailey and J.L. Lasswell. 1977. Laboratory spawning and larval rearing of red drum and southern flounder. **Proc. Annu. Conf. S.E. Assoc. Fish Wildl. Agencies.** 31:437-441.

Bass, R.J. and J.W. Avault, Jr. 1975. Food habits, length-weight relationships, condition factor, and growth of juvenile red drum, *Sciaenops ocellata*, in Louisiana. **Trans. Am. Fish. Soc.** 104:35-45.

Beaumariage, D.S. 1964. Returns from the 1963 Schlitz tagging program. **Fla. Board Conser. Tech. Ser.** 43, 38 pp.

_________ 1969. Returns from the 1965 Schlitz tagging program including a cumulative analysis of pervious results. **Fla. Dep. Nat. Resour. Mar. Lab. Techy. Ser.** 59, 38 p.

Beaumariage, D.S. and A.C. Wittich. 1966. Returns from the 1964 Schlitz tagging program. Fla. Board Conser. Tech. Ser. No. 47, 51 p.

Bere, R. 1936. Parasitic copepods from Gulf of Mexico fish. **Am. Midl. Nat.** 17(3):577-625.

Bigelow, H.B. and W.C. Schroeder. 1954. Fishes of the Gulf of Maine. **U.S. Fish. Wildl. Serv. Fish. Bull.** 53(74):1-577.

Boothby, R.N. and J.W. Avault, Jr. 1971. Food habits, length-weight relationships, and condition factor of the red drum, *Sciaenops ocellata*, in southern **Louisiana. Trans. Am. Fish. Soc.** 100:290-295.

Bowden, K.F. 1967. Circulation and diffusion. p.15-36. *In*: G.H. Lauff (editor), **Estuaries, Amer. Asso. Advanc. Sci. Publ.** 83, Washington, D.C.

Bradley, E. and H. Compton. 1963. Survey of larval and post-larval finfish in Aransas and Corpus Christi Channels and in the inshore Gulf of Mexico. Tex. Game Fish Comm., Proj. Rep. 1961-1962, Proj. No. MF- R-4, 7 p.

Breuer, J.P., R.L. Benefield, M.G. Weixelman, A.R. Martinez, and I. Nava. 1977. Survey of finfish harvest in selected Texas bays, segment II. Tex. Parks. Wildl. Dep., Coast. Fish. Rep. Proj. No. 2- 231-R-2, 116 pp.

Briggs, J.C. 1974. **Marine zoogeography**. McGraw-Hill, N.Y., 475 p.

Brown, L.F., Jr., R.A. Horton, J.H. McGowen, C.W. Kreitler, and W.L. Fisher. 1974. Natural hazards of the Texas coastal zone. Bur. Econ. Geol. Univ. Tex., Austin, Tex. 13 pp.

Bryan, C.E. 1971. An ecological survey of the Arroyo Colorado, Texas, 1966. 169. **Tex. Parks Wildl. Dep., Tech.** Ser. 10, 28 pp.

Bryan, C.E., T.J. Cody, and G.C. Matlock. 1982. Organisms captured by the commercial shrimp fleet on the Texas brown shrimp (*Penaeus aztecus* Ives) grounds. **Tex. Parks Wildl. Dep., Tech. Ser.** No 31, 26 pp.

Caldwell, K.D. and M.C. Caldwell. 1972. **The world of the bottlenosed dolphin.** J.B. Lippencott Co., Phila. and N.Y., 157 pp.

Cardeilhac, P.T., C.F. Simpson, F.H. White, N.P. Thompson, and W.E. Carr. 1981. Evidence for metal poisoning in acute deaths of large red drum *Sciaenops ocellata*. **Bull. Environm. Contam. Toxicol.** 27:639-644.

Carr, W.E.S. and T.B. Chaney. 1976. Harness for attachment of an ultrasonic transmitter to the red drum, *Sciaenops ocellata*. Fish. Bull, U.S. 74(4):998.

Causey, D. 1953. Parasitic copepoda of Texas coastal fisheries. Pub. Inst. Mar. Sci., Univ. Tex. 3(1):5.16.

Chandler, A.C. 1935. Parasites of fishes in Galveston Bay. **Proc. U.S. Nat. Mus.** 83(2977):123.157.

__________ 1954. Nematoda. p. 357.358. In: P. Galtsoff (coordinator), Gulf of Mexico, its origin, waters, and marine life. **U.S. Fish. Wildl. Serv., Fish. Bull.** 55.

Chao, L.N. 1978. A basis for classifying western Atlantic Sciaenidae (Teleostei: Perciformes). U.S. Dep. Commer., NOAA Tech. Rep. NMFS Circ. 415, 64 pp.

Chao, L.N. and J.A. Musick. 1977. Life history, feeding habits, and functional morphology of juvenile sciaenid fishes in the York River Estuary, **Virginia. Fish. Bull, U.S.** 75(4):657-702.

Chittenden, M.E.,Jr. and D. Hoore. 1977. Composition of the ichthyofauna inhabiting the 110-meter bathymetric contour of the Gulf of Mexico, Mississippi River to the Rio Grande. N.E. Gulf Sci. 1(2):106-114.

Chittenden, M.E., Jr. and J.D. McEachran. 1976. **Composition, ecology, and dynamics of demersal fish communities on the Northwestern Gulf of Mexico continental shelf, with a similar synopsis for the entire Gulf.** Sea Grant Publ. TAMU-SG-76-208, 104 pp.

Christmas, J.Y., G. Gunter, and E.L. Whatley. 1960. Fishes taken in the menhaden fishery of Alabama, Mississippi, and eastern Louisiana. **U.S. Dep. Interior, Fish Wildl Ser., Spec. Sci. Rep.**-Fish. No. 339, 10 pp.

Cody, T.J. and R.H. Avent. 1980. Shrimp and finfish investigations in the Northwestern Gulf of Mexico. Part III. Assessment of bottom longline fishing off the central Texas coast. **Tex. Parks. Wildl. Dep., Coast. Fish. Branch, Mngmnt. Data Ser.**, 24 p. (Unpublished manuscript).

Cody, T.J., B.E. Fuls, G.C. Matlock, and C.E. Bryan. 1981. Assessment of bottom longline fishing off the central Texas coast, a completion report. **Tex. Parks Wildl. Dep., Coast. Fish. Branch, Mngmnt. Data Ser.** 22, 51 pp.

Cody, T.J., K.W. Rice, and C.E. Bryan. 1977. Foodfish and potential foodfish captured in a large-mesh fish trawl along the central Texas coast. Tex. Parks Wildl. Dep., Coast. Fish. Branch, 49 p. (Unpublished manuscript).

Colura, R. and B. Hysmith. 1976. Fingerling production of spotted seatrout, *Cynoscion nebulosus*, and red drum, *Sciaenops ocellata*, in saltwater ponds. **Annu. Rep. Mar. Fish. Res. Sta., Tex. Parks Wildl. Dep.**, Palacios, Tex., 39 pp.

Colura, R.L., B.T. Hysmith and R.E. Stevens.1976. Fingerling production of striped bass (*Morone saxatilis*), spotted seatrout (*Cynoscion nebulosus*) and red drum (*Sciaenops ocellata*) in saltwater ponds. **Proc. 7th World Maricult. Soc.**p. 79-92.

Compton, H. 1965. Survey of the fishes found in the inshore Gulf of Mexico and of post-larval fishes in Aransas, Port Mansfield and Port Isabel Ship Channels. **Tex. Fish Game Comm., Proj. Rep.**, Proj. No. MR-R-6, p. 383-413.

Compton, H. and E. Bradley. 1964. A study of the post-larval penaeid shrimp entering Aransas Bay. **Tex. Parks Wildl. Dep., Coast. Fish. Branch, Proj. Rep.** 1962-1963, p. 127-143.

Crocker, P.A., C.R. Arnold, J.A. DeBoer, and J.D.. Holt. 1981. Preliminary evaluation of survival and growth of juvenile red drum (*Sciaenops ocellata*) in fresh water. **J. World Maricult. Soc.** 12(1):122.134.

Crocker, R.A. 1965. Planktonic fish eggs and larvae of Sandy Hook estuary. **Chesapeake Sci.** 6(2):92-96.

Darnell, R.M. 1958. Food habits of fishes and larger invertebrates of Lake Pontchartrain, Louisiana, an estuarine community. Publ. Inst. Mar. Sci., Univ. Texas. 5:353.416.

Diener, R.A., A. Inglis and G. B. Adams. 1974. Stomach contents of fishes from Clear Lake and tributary waters, a Texas estuarine area. **Contrib. Mar. Sci.** 18:7-17.

Dokken, Q.R. 1981. Spatial and temporal distribution and species composition of larval fish populations within Alazan Bay, Texas. M.S. Thesis, Corpus Christi State Univ., 61 pp.

Dovel, W. 1967. Fish eggs and larvae of the Magothy River, Maryland. **Chesapeake Sci.** 8(2):125-129.

Dugas, R., V. Guillory and M. Fisher. 1979. Oil rigs and offshore sport fishing in Louisiana. **Fisheries.** 4(6):2-10.

Dunham, F. 1972. A study of commercially important estuarine-dependent industrial fishes. **La. Wildl. Fish. Comm., Tech. Bull.** 4, 63 pp.

Elam, L. 1971. Evaluation of fish tagging methods. Tex. **Parks Wildl. Dep., Coast. Fish. Proj. Rep.**, p. 39-55.

Finucane, J.H., L.A. Collins and J.D. McEachran. 1977. *In*: W.B. Jackson (editor), Environmental studies of the South Texas outer continental shelf, ichthyoplankton-mackerel eggs and larvae. NOAA Final Rep. Bur. Land Mngmnt, Interagency Agreement No. AA550-1A7-3, 484 pp.

Fontenot, B.J., Jr. and H.E. Rogilio. 1970. A study of estuarine sport fishes in the Biloxi marsh complex Louisiana. L. Wildl. Fish. Comm., D.J. Proj. F-8 Compl. Rep., 172 pp.

Green, A.W., H.R. Osburn, G.C. Matlock and H. E. Hegen. 1985. Estimated survival rates for red drum in Northwest Gulf of Mexico bays. **Fish. Res.** 3:263-277.

Guest, W.C. and J.L. Lasswell. 1978. A note on courtship behavior and sound production of red drum. **Copeia.** 1978(2):337-338.

Gunter, G. 1941. Death of fishes due to cold on the Texas coast, January, 1940. **Ecology.** 22:203-208.

Gunter, G. 1942. Contributions to the natural history of the bottlenose dolphins, *Tursiops truncatus* (Montague), on the Texas coast, with particular reference to food habits. **J. Mamm.** 23(3):267-276.

Gunter, G. 1945. Studies on marine fishes of Texas. Publ. Inst. Mar. Sci., Univ. Tex. 1(1):1-190.

Gunter, G. 1952. The import of catastrophic mortalities for marine fisheries along the Texas coast. **J. Weldl. Mngmnt.** 16(1):1-7.

Gunter, G. 1958. Population studies of the shallow water fishes of an outer beach in South Texas. Publ. Inst. Mar. Sci., Univ. Tex. 5:186-193.

Gunter, G. and H.H. Hildebrand. 1951. Destruction of fishes and other organisms on the south Texas coast by the cold wave of January 28-February 3, 1951. **Ecology** 32:731-735

Hamilton, C.L. 1980. Texas commercial harvest statistics, 1978-79. Tex. Parks Wildl. Dep., Coast. Fish. Branch, Mngmnt. Data Ser. 12, 32 pp.

__________ 1981. Texas commercial harvest statistics, 1979-80. Texas Parks Wildl. Dep., Coast. Fish. Branch, Mngmnt. Data Ser. 26, 42 pp.

__________ 1982. Texas commercial harvest statistics, 1977-81. Tex. Parks Wildl. Dep., Coast. Fish. Branch, Mngmnt. Data Ser. 45, 59 pp.

Harden Jones, F.R. 1968. **Fish migration.** Edward Arnold Ltd., London, 325 pp.

Harrington, R. 1973. Fish graveyard no longer traps victims. **Tex. Parks Wildl. Magazine,** July:16-17.

Heffernan, T.L., A.W. Green, L.W. McEachron, M.G. Weixelman, P.C. Hammerschmidt and R.A. Harrington. 1977. Survey of finfish harvest in selected Texas bays. Tex. Parks Wildl. Dep., Coast. Fish. Branch Proj. No. 2-231-R-I, 116 pp.

Heffernan, T.L. and R.J. Kemp. 1980. Management of red drum resources. **Proc. Colloquium on the Biology and Management of Red Drum and Seatrout, Gulf States Mar. Fish. Comm.,** Oct. 19-21, 1978, Tampa, Fla.

Hegen, H.E. 1981. Monitoring of coastal finfish resources for sport fish management, October 1979-September 1980. **Tex. Parks Wildl. Dep., Coast. Fish. Branch, Mngmnt. Data Ser.** 28, 228 pp.

__________ 1982. Monitoring of coastal finfish resources for sport fish management, October 1980. September 1981. **Tex. Parks Wildl. Dep., Coast. Fish. Branch, Mngmnt. Data Ser.** 33, 186 pp.

Hegen, H.E. and G.C. Matlock. 1980. Monitoring of coastal finfish resources for sport fish managment, October 1978-September 1979. **Tex. Parks Wildl. Dep., Coast. Fish. Branch, Mngmnt. Data Ser.** 17, 245 pp.

Hein, S.H. and J.A. Shepard. 1980. Growth of juvenile red drum, *Sciaenops ocellata,* in a quarter-acre pond. *In*: Contributions of the Marine Research Laboratory-1978. La. Dep. Wildl. Fish. Mar. Res. Lab., Grand Terre Island, **La., Tech. Bull.** 31, 86 pp.

Henley, H.W. and D.H. Lewis. 1976. Anaerobic bacteria associated with epozootics in grey mullet (*Mugil cephalus*) and redfish (*Sciaenops ocellata*) along the Texas Gulf Coast. **J. Wildl. Dis.** 12:448-453.

Hermann, S.S. 1963. Planktonic fish eggs and larvae of Narragansett Bay. **Limnology Oceanog.** 8(1):103-109.

Hildebrand, H.H. 1954. A study of the brown shrimp (*Penaeus aztecus* Ives) grounds in the Western Gulf of Mexico. Publ. Inst. Mar. Sci., Univ. Tex. 3(2):1-366.

Hildebrand, S.F. and L.E. Cable. 1934. Reproduction and development of whitings or kingfishes, drum, spot, croaker and weakfishes or sea trouts of the United States. **Bull. U.S. Bur. Fish.** 48(16):41-117.

Hildebrand, S.F. and W.C. Schroeder. 1928. Fishes of Chesapeake Bay. **Bull. U.S. Bur. Fish.** XLIII:1-388.

Hoese, H.D. 1965. Spawning of marine fishes in the Port Aransas, Texas area as determined by the distribution of young larvae. Ph.D. Dissertation, Univ. Tex., 144 pp.

Holland, J.S., N.J. Maciolek, R.D. Kalke and C.H. Oppenheimer. 1973. A benthos and plankton study of the Corpus Christi, Copano and Aransas Bay systems. I. **Report on the methods used and data collected during the period September 1972-June 1973.** Unpubl. 1st Annu. Rep. to Texas Water Develop. Board, Univ. Tex. Mar. Sci. Inst. Port Aransas, Tex. 118 pp.

__________ 1974. A benthos and plankton study of the Corpus Christi, Copano, and Aransas Bay systems, II. **Report on data collected during the period July 1973-April 1974.** Unpubl. 2nd Annu. Rep. to Tex. Water Develop. Board, Univ. Tex. Mar. Sci. Inst., Port Aransas, Tex., 121 pp.

Holt, J., G. Godbout and C.R. Arnold. 1981a. Effect of temperature and salinity on egg hatching and larval survival of red drum, *Sciaenops ocellata.* **Fish. Bull., U.S.** 79(3):569-573.

Holt, J., A.G. Johnson, C.R. Arnold, W.A. Fable, Jr. and T.D. Williams. 1981b. Description of eggs and larvae of laboratory reared red drum, *Sciaenops ocellata.* **Copeia.** 1981:751-756.

Holt, S.A., C.L. Kitting and C.R. Arnold. 1983. Distribution of young red drums among different sea-grass meadows. **Trans. Am. Fish. Soc.** 112:267-271.

Houde, E.D., J.C. Leak, C.E. Dowd, S.A. Berkley and W.J. Richards. 1979. Ichthyoplankton abundance and diversity in the eastern Gulf of Mexico. Rep. Bur. Land Mngmnt., Contract No. AA550-CT7-28, U.S. Dep. Commer. Nat. Tech. Info. Serv., PB-299839, 546 pp.

Hysmith, B.T., R.L. Colura and G.C. Matlock. 1983. Effects of stocking rate and food type on growth and survival of fingerling red drum. **Proc. Warmwater Fish. Cult. Workshop, Annu. Meet. World Maricult. Soc.,** March 1-4, 1982, Charleston, SC (in press).

Ingle, R.M., R.F. Hutton and R.W. Topp. 1962. Results of the tagging of saltwater fishes in Florida. **Fla. Board Consev. Tech. Ser.** 38, 57 pp.

Iversen, E.S. and B. Yokel. 1963. A myxosporidian (sporozoan) parasite in the red drum. **Bull. Mar. Sci. Gulf Caribb.** 18:449-453.

Jannke, T.E. 1971. Abundance of young sciaenid fishes in Everglades National Park, Florida, in relation to season and other variables. **Univ. of Miami Sea Grant Tech. Bull.** 11, 128 pp.

Johnson, A.G., W.A. Fable, Jr., T.D. Williams and C. R. Arnold. 1977. Description of reared eggs and young larvae of the red drum, *Sciaenops ocellata.* p. 118-140. *In*: Marine Fish Propagation Study, Fed. Aid Proj. F-31-R, Compl. Rep., Tex. Parks Wildl. Dep.

Juneau, C.L., Jr. 1975. An inventory and study of the Vermilion Bay-Atchafalaya Bay complex, Phase II; biology. p 21-67. *In*: **An inventory and study of the Vermilion Bay-Atchafalaya Bay complex. La. Wildl. Fish. Comm., Tech. Bull.** 13.

Kemp, R.J. 1949. Report on stomach analysis on June 1, 1949 through August 31, 1979. Tex. Game, Fish, Oyster Comm., **Mar. Lab. Annu. Rep.** (1948-1949):101-127.

Kilby, J.D. 1955. The fishes of two Gulf coastal marsh areas of Florida. **Tulane Study. Zool.** 2(8):176-247.

King, B.D., III. 1971. Study of migratory patterns of fish and shellfish through a natural pass. **Tex. Parks Wildl. Dep. Tech. Ser.** 9, 54 pp.

Knapp, F. T. 1950. Menhaden utilization in relation to the conservation of food and game fishes of the Texas Gulf coast. **Trans. Am. Fish. Soc.** 79:137-144.

Linton, E. 1901. Parasites of fishes of the Woods Hole Region. **Fish. Bull., U.S.** 19:405-492.

________ 1905. Parasites of fishes of the Beaufort, North Carolina region. **Fish. Bull, U.S.** 24:323-428.

Lippson, A.J. and R.L. Moran. 1974. **Manual for identification of early developmental stages of fishes of the Potomac River Estuary.** Env. Tech. Center, Martin Marietta Corp., Baltimore, MD., 282 pp.

Loman, M. 1978. Other finfish. p. 143-147. *In:* J.Y. Christmas (editor), **Fisheries assessment and monitoring, Mississippi.** P.L. 88-309, 2-215-R, Compl. Rep., Gulf Coast Res. Lab., Ocean Springs, Miss.

Luebke, R.W. and K. Strawn. 1973. The growth, survival, and feeding behavior of redfish (*Sciaenops ocellata*) in ponds receiving heated discharge water from a power plant. **Proc. 4th Annu. Workshop World Maricult. Soc.**, p. 143-154.

McEachron, L.W. and G.C. Matlock. 1983. Harvest estimate of the Texas Chartered boat fishery. **Mar. Fish. Rev.** 45(1):11-17.

McFarland, W. 1959. Distribution and infestation of the parasite *Poecilancistrium robustrum* in some Texas coast fishes. **Tex. Game Fish Comm. Proj. Rep.** (1958-1959):11 pp.

McFarland, W.N. 1963. Seasonal change in the number and biomass of fishes from the surf at Mustang Island, Tex. Publ. Inst. Mar. Sci., Univ. Tex. 9:91.105.

McKee, D.A. 1980. A comparison of the growth rate, standard length-weight relationships and condition factor of red drum, *Sciaenops ocellata* (Linnaeus), from an electric generating station's cooling lake and the natural environment. M.S.Thesis, Corpus Christi State Univ., 53 pp.

Mansueti, R.J. 1960. Restriction of very young red drum, *Sciaenops ocellata*, to shallow estuarine waters of Chesapeake Bay during late autumn. **Chesapeake Sci.** 1:207-210.

Marley, R.D. 1983. Spatial distribution patterns of planktonic fish eggs in lower Mobile Bay, Alabama. **Trans. Am. Fish. Soc.** 112:257-266.

Massmann, W .H., J.J. Norcross and E.B. Joseph. 1961. Fishes and fish larvae collected from Atlantic plankton cruises of R/V PATHFINDER, December 1959-December 1960. **Va. Inst. Har. Sci., Va. Fish. Lab., Spec. Sci. Rep.** 26, 22 pp.

Massmann, W. H., E.B. Joseph and J.J. Norcross. 1962. Fishes and fish larvae collected from Atlantic plankton cruises of R/V PATHFINDER, March 1961-March 1962. **Va. Inst. Mar. Sci., Spec. Sci. Rep.** 33, 23 pp.

Matlock, G.C. 1979. The use of fish trawl to sample large fish. **Tex. Wildl. Dep., Coast. Fish. Branch, Mngmnt. Data** Ser. 2, 12 pp.

________ 1980. History and management of the red drum fishery. **Proc. Colloquium on the Biology and Management of Red Drum and Seatrout,** Gulf States Mar. Fish. Comm., Atlantic States Mar. Fish. Comm., Oct. 19-21, 1978, Tampa, Fla.

Matlock, G.C. 1981. Nonreporting of recaptured tagged fish by saltwater recreational boat anglers in Texas. **Trans. Am. Fish. Soc.** 110:90.92.

________ 1982. The conflict between user groups of red drum and spotted seatrout in Texas. p. 101-108. *In:* R.H. Stroud (editor), **Marine Recreational Fisheries, Proc. 7th Annu. Mar. Rec. Fish. Symposium,** Fort Lauderdale, Fla., May 10-11, 1982.

________ 1983. Angler origin and retention rate interpretation. **N. Am. J. Fish. Mngmnt.** 3:438-444.

Matlock, G.C. 1984. A basis for the development of a management plan for red drum in Texas. Ph.D. Dissertation, Texas A&M Univ., College Station, Tex. 291 pp.

Matlock, G.C. 1986. Red drum sex ratio and size at sexual maturity, **Tex. Parks Wildl. Dep., Coast. Fish. Branch Mngmnt. Data Ser.** No. 85, 6 pp.

Matlock, G.C. and J.E. Weaver. 1979a. Assessment and monitoring of Texas coastal finfish resources. Tex. Parks Wildl. Dep., Coast. Fish. Branch, Rep. Proj. No. 2-313-R, 247 pp.

________ 1979b. Fish tagging in Texas bays during November 1976-September 1976. Tex. Parks Wildl. Dep., **Coast. Fish. Branch, Mngmnt. Data Ser.** No. 1, 136 pp.

Matlock, G.C., R.J. Kemp, Jr. and T.L. Heffernan. 1986. Stocking as a management tool for a red drum fishery, a preliminary evaluation. **Tex. Parks Wildl. Dept., Coast. Fish. Branch, Mngmnt. Data Ser.** No. 75, 27 pp.

Matlock, G.C., J.E. Weaver, L.W. McEachron, J.A. Dailey, P.C. Hammerschmidt, H.E. Hegen, R.A. Harrington and G.M. Stokes. 1978. Trends in finfish abundance in Texas estuaries as indicated by gill nets. **Tex. Parks Wildl. Dep., Coast. Fish.**, 271 pp.

Miles, D.W. 1950. The life histories of the spotted seatrout. *Cynoscion nebulosus*, and the redfish, *Scianeops ocellatus*. Annu. Rep. Tex. Game, Fish, Oyster Comm., Mar. Lab., p. 1-38.

Miles, D.W. 1951. The life histories of the seatrout, *Cynoscion nebulosus*, and the redfish, *Sciaenops ocellatus*, sexual development. Tex. Game, Fish, Oyster Comm., Annu. Rep. 1 September 1950-31 August 1951, 1 pp.

Miller, G.L. and S.C. Jorgenson. 1973. Meristic characters of some marine fishes of the western Atlantic ocean. Fish. Bull., U.S. 71:301-312.

Miller, J.M. 1965. A trawl survey of the shallow Gulf fishes near Port Aransas, Texas. Publ. Inst. Mar. Sci., Univ. Tex. 10:80-107.

Modde, T. and S.T. Ross. 1981. Seasonality of fishes occupying a surf zone habitat in the northern Gulf of Mexico. **Fish. Bull., U.S.** 78:911-922.

Moe, M.A., Jr. 1972. Movement and migration of south Florida fishes. Fla. Dep. Nat. Resour. Tech. Ser. 69, 25 pp.

Moffett, A.W., L.W. McEachran and J.G. Key. 1979. Observations on the biology of sand seatrout (*Cynoscion arenarius*) in Galveston and Trinity Bays, Texas. **Contrib. Mar. Sci.** 22:163-172.

Murphy, M. and M.E. Chittenden, Jr. 1982. Aspects of the life history of the Gulf butterfish (*Prilus burti*) (Unpublished manuscript).

Neff, J.N., M.P. Coglianse, L.A. Reitsema, S. Anderson and W. McCullouch. 1981. Brine toxicity bioassays on redfish. Vol. V (Part A). *In:* W.B. Jackson (editor), **Shrimp and redfish studies; Bryan Mound brine disposal site off Freeport, Texas, 1979-1981.** NOAA Tech. Memorandum NMFS-SEFC-69, 82 pp.

Odum, W.E. 1971. Pathways of energy flow in a south Florida estuary. **Univ. Miami Sea Grant Tech. Bull.** 7, 162 pp.

Osburn, H.R., G.C. Matlock and A.W. Green. 1982. Red Drum (*Sciaenops ocellatus*) movement in Texas bays. **Cont. Mar. Sci., Univ. Tex.** 24:85-97.

Overstreet, R.M. 1977. *Poecilancistrium caryophyllum* and other trypanorynch cestode plerocercoids from the musculature of *Cynoscion nebulosus* and other sciaenid fishes in the Gulf of Mexico. **J. Parasitol.** 63(5):780-789.

Overstreet, R.M. and R.W. Heard. 1978. Food of the red drum *Sciaenops ocellata* from the Mississippi Sound. **Gulf Res. Rep.** 6(2):131-135.

________ 1982. Food contents of six commercial fishes from Mississippi Sound Gulf **Res. Rep.** 7:137-151.

Owens, D.W., K.A. Jones and B.J. Galloway. 1981. Brine avoidance/attraction bioassays on redfish. Vol. V(Part B). *In*: W.B. Jackson (editor), **Shrimp and redfish studies; Bryan Mound Brine disposal sites off Freeport, Texas, 1979-1981.** NOAA Tech. Memorandum NMFS-SEFC-69, 58 pp.

Pearse, A.S. 1952. Parasitic crustacea from the Texas coast. Publ. Inst. Mar. Sci., Univ. Tex. 2(2) :5-43.

Pearson, J.C. 1929. Natural history and conservation of the redfish and other commercial sciaenids on the Texas coast. **Bull. Bur. Fish.** 44:129-214.

_________ 1941. The young of some marine fishes taken in lower Chesapeake Bay, Virginia, with special reference to the gray sea trout, *Cynoscion regalis* (Bloch). **U.S. Dep. Interior, Fish Wildl. Ser., Fish Bull.** 50:79.102.

Perrett, W.S., J.E. Weaver, R.O. Williams, P.L. Johansen, T.D. McIlwain, R.C. Raulerson and W.M. Tatum. 1980. Fishery profiles of red drum and spotted seatrout. **Gulf States Mar. Fish. Comm.** 6, 60 pp.

Powles, H. and B.W. Stender. 1978. Taxonomic data on the early life history stages of Sciaenidae of the South Atlantic Bight of the United States. **South Carolina Mar. Res. Center Tech. Rep.** 31 pp.

Rabalais, S.C., C.R. Arnold and N.S. Wohlschlag. 1981. The effects of IXTOC I oil on the eggs and larvae of red drum (*Sciaenops ocellata*). **Tex. J. Sci.** 33:33-38.

Reid, G.K., Jr. 1955. A summer study of the biology and ecology of East Bay, Texas. Part II. The fish fauna of East bay, the Gulf beach, and summary. **Tex. J. Sci.** 7(4):430-453.

Reid, G.K., A. Inglis and H.D. Hoese. 1956. Summer foods of some fish species in East Bay, Texas. **Southwest Nat.** 1(3):100- 104.

Riggin, G.I., Jr. and A.K. Sparks. 1962. A new gasterostome, *Bucephaloides megacirrus*, from the redfish *Sciaenops ocellata*. **Proc. Helm. Soc.** 29:27-29.

Roberts, D.E., Jr., B.V. Harpster and G.E. Henderson. 1978a. Conditioning and induced spawning of the red drum (*Sciaenops ocellata*). **Proc. Ninth Annu. Meet. World Maricult. Soc.,** p. 333-343.

Roberts, D.E., Jr., L.A. Norey, III, G.E. Henderson and K.R. Halscott. 1978b. The effects of delayed feeding, stocking density and food density on survival, growth and production of larval red drum *Sciaenops ocellata*. **Proc. Ninth Annu. Meet World Maricult. Soc.,** p.333-343.

Roessler, M. 1967. Observations on the seasonal occurrence and life histories of fishes in Buttonwood Canal, Everglades National Park, Fla. Ph.D. Dissertation, Univ. of Miami, 155 p.

Rohr, B.A. 1968. Surface schooling of red drum. **Underwater natur.** 5(2):21-23.

Rohr, B.A. 1980. Use of hard parts to age Gulf of Mexico red drum, *Sciaenops ocellata*. **Proc. Colloquium on the Biology and Management of Red Drum and Seatrout,** p. 15.

Ross, J.L., F.S. Pavela and M.E. Chittenden, Jr. 1982. Seasonal occurrence of black drum, *Pogonias cromis*, and red drum, *Sciaenops ocellatus*, off Texas. **N.E. Gulf Sci.** (In press).

Rounsfell, G.A. 1975. **Ecology, utilization and management of marine fishes.** C.V. Mosby Co., St. Louis, No., 516 pp.

Royce, W.F. 1972. **Introduction to the fishery sciences.** Academic Press, N.Y., 351 p.

Sawyer, R.T., A.R. Lawler and R.M. Overstreet. 1975. Marine leeches of the eastern United States and the Gulf of Mexico with a key to the species. **J. Nat. Hist.** 9:633-667.

Shlossman, P.A. and M.E. Chittenden, Jr. 1981. Reproduction, movements, and population dynamics of the sand seatrout, *Cynoscion arenarius*. **Fish. Bull., U.S.** 79:649-669.

Simmons, E.G. 1957. An ecological survey of the upper Laguna Madre of Texas. Publ. Inst. Mar. Sci., Univ. Tex. 4(2):156-200.

Simmons, E.G. and J.P. Breuer. 1962. A study of redfish, *Sciaenops ocellata* Linnaeus, and black drum, *Pogonias cromis* Linnaeus. Publ. Inst. Mar. Sci., Univ. Tex. 8:184.211.

_________ 1982. Fish tagging on the Texas coast. **Tex. Parks Wildl. Dept., Coast. Fish. Proj. Rep.** 1976, p. 66-107.

Sparks, A.K. 1958. Some digenetic trematodes of fishes of Grand Isle, Louisiana. **Proc. La. Acad. Sci** 8. 20:71-82.

Springer, S. and H.R. Bullis, Jr. 1956. Collections by the Oregon in the Gulf of Mexico. LIfe of crustaceans, mollusks and fishes identified from collections made by the exploratory fishing vessel Oregon in the Gulf of Mexico and adjacent seas 1950 through 1955. **Spec. Sci. Rep. U.S. Fish. Wildl. Serv.** 196:1-34.

Springer, V.G. and K.D. Woodburn. 1960. An ecological study of the fishes of the Tampa Bay Area. Fla. State Board Conser., **Prof. Paper Ser.** 1:104 pp.

Standard, G. and M.E. Chittenden, Jr. 1984. Reproduction, movements and population dynamics of the banded drum, *Larimus fasciatus*, in the Gulf of Mexico. **Fish. Bull.** 82:1-27.

Sykes, J.E. and J.H. Finucane. 1965. Occurrence in Tampa Bay, Florida, of immature species dominant in Gulf of Mexico commercial fisheries. **Fish. Bull., U.S.** 65(2):369-379.

Thieling, D.L. and H.A. Loyacano, Jr. 1976. Age and growth of red drum from a saltwater marsh impoundment in South Carolina. **Trans. Am. Fish. Soc.** 105:41-44.

Topp, R. 1963. The tagging of fishes in Florida, 1962 program. Fla. Board Conser., **Mar. Res. Lab., Prof. Paper Ser.** 5:76 pp.

Topp, R.W. and C.F. Cole. 1968. An osteological study of the sciaenid Genus, *Sciaenops* Gill (Teleosteri, Sciaenidae). **Bull. Mar. Sci.** 18:902-945.

Trimble, W.C. 1979. Yield trials for red drum in brackish-water ponds, 1976-1979. **Proc. Annu. Conf. S.E. Assoc. Fish. Wildl. Agencies.** 33:432-441.

Wakefield, C.A. and R.L. Colura. 1983. Age and growth of red drum in three Texas bay systems. **Proc. Annu. Meet. Tex. Chap. Am. Fish. Soc.,** Vol. 5. (In press).

Watson, R.L. and E.W. Behrens. 1970. Nearshore surface currents, south-eastern Gulf coast. **Contrib. Mar. Sci., Univ. Tex.** 15:133.144.

Weixelman, M.G. 1982. The fall red drum Gulf of Mexico pier fishery off Galveston Bay, Texas. **Tex. Parks Wildl. Coast. Fish. Branch, Mngmnt. Data Ser.** 42, 23 pp.

Welsh, W.W. and C.M. Breder, Jr. 1924. Contributions of the life histories of Sciaenidae of the eastern United States coast. Bull. **U.S. Bur. Fish.** 39:141-201.

Wilson, C.G. 1905. North American parasitic copepods belonging to the Caligidae. **Proc. U.S. Nat Mus.** 28:479-672.

_________ 1915. North American parasitic copepods belonging to the Lernaropodidae, with a revision of the entire family. **Proc. U.S. Nat. Mus.** 47:565-729.

_________ 1932. The copepods of the Woods Hole region. Bull. **U.S. Nat. Mus.** 158(20):1-635.

Wohlschlag, D.E. 1981. Natural factors affecting recruitment. p. 177-189. *In*: H. Clepper (editor), **Marine Recreational Fisheries 6. Proc. 6th Annu. Mar. Rec. Fish. Symposium,** Houston, Tex., March 19-20, 1981, 216 pp.

Yokel, B.J. 1966. Contributions on the biology and distribution of red drum, *Sciaenops ocellata*. Master's Thesis, Univ. of Miami, 160 pp.

Status of the Commercial and Recreational Fishery

Wayne E. Swingle
Gulf of Mexico Fishery Management Council
Lincoln Center, Suite 881
5401 W. Kennedy Blvd
Tampa, Florida 33609-2586

Red drum has been a highly sought-after commercial and recreational species throughout the development of Gulf coast fisheries. Commercial harvest and trade in red drum has been reported from the Gulf as early as 1776 in Florida. By 1888 when the first Gulf-wide statistics were reported, commercial harvest was approximately 1.5 million pounds. Commercial landings for the Gulf ranged generally between two to three million pounds from that time until 1970, increased to five million pounds for 1975, and declined to about two million pounds thereafter. (See Table 12-1 for more recent landings).

Recreational participation in the fishery became a significant factor by at least the early 1900s as documented by requests to state fishery management agencies for regulation of the commercial fishery. Recreational participation and harvest greatly escalated after World War II and has continued to increase over the following years.

Red drum is an estuarine dependent species that generally spends its first three or four years within the estuaries and nearshore waters. Historically, the fishery has been generally conducted within the states' jurisdiction on the juvenile population.[1] Catches from federal waters historically consisted of a small incidental bycatch by commercial shrimp vessels and relatively small recreational catches, principally from the Gulf waters off Louisiana, Mississippi and Alabama.

Because red drum were targeted by both recreational and commercial fishermen within the estuarine areas of the states, the juvenile fishery was subjected to intense fishing pressure and extreme competition existed between user groups for the available resource. This has resulted in overfishing of the resource, regulatory actions to reduce the excessive fishing pressure, and political solutions to competition between user groups, in some areas.

Recreational and Commercial Landings

Region-wide statistics, of even a moderate degree of reliability, on recreational landings did not become available for the Gulf fishery until 1979. Table 12-1 includes reported commercial landings and estimated recreational landings for 1979 through 1985, with preliminary data for 1986. Over this seven-year period, estimated recreational landings ranged between 6 and 11 million pounds and reported commercial landings between 2.5 and 13.5 million pounds. These data for recreational landings have such a high degree of variability associated with the point estimates, especially for the Exclusive Economic Zone (EEZ) component, that it is difficult to speculate on trends in that segment of the fishery. Recreational landings data collected by the states, such as Texas, have a much greater reliability and precision but have not been collected by other states on an annual basis and, therefore, are not useful to detect regional trends. The principal trends obvious in Table 1 are the increased commercial landings from federal waters of the EEZ for 1984 through 1986 and from state waters for 1986 Overall total landings by both groups appear relatively stable, with a jump in landings for 1986. This may suggest to you that the status or condition of the fishery is good or at least stable. However, this is definitely not the case. The stock is being overfished and overfishing has been occurring for many years.

Regulation

Federal[2]

The Council's scientific advisory committee and NMFS stock assessment scientists after reviewing new and historical data on the fishery have concluded that the high rate of inshore fishing on juvenile fish has, or will, reduce the spawning stock below a level at which recruitment overfishing will occur. When recruitment overfishing occurs it means that there will be insufficient spawning fish producing eggs and larvae to maintain the stock, that the whole fishery will collapse and that it may require decades with no fishing to recover or it may not recover at all. These state, federal and university scientists have concluded that the inshore exploita-

[1] *Life history and biological information, as well as detailed the fishery are discussed in the Fishery Management Plan for Red Drum available from NMFS.*

[2] *Current federal regulation prohibits harvest or possession from federal waters.*

Table 1. Reported commercial and estimated recreational red drum landings (thousands of pounds) in the Gulf of Mexico, 1979-1986.

Year	Recreational[1] State Waters[3]	Recreational[1] EEZ	Recreational[1] Total[4]	Commercial[2] State Waters	Commercial[2] EEZ	Commercial[2] Total[4]	State Waters	EEZ	Total
1979	8,536	34	8,570	2,691	80	2,771	11,227	114	11,341
1980	6,863	1,282	8,145	2,681	48	2,729	9,544	1,330	10,.874
1981	5,351	306	5,657	2,717	31	2,748	8,068	337	8,405
1982	10,259	475	10,734	2,348	77	2,425	12,607	552	13,159
1983	5,397	2,065	7,462	2,881	206	3,087	8,278	2,271	10,549
1984	4,964	1,491	6,425	3,347	987	4,334	8,281	2,478	10,759
1985	6,212	324	6,536	2,886	3,457	6,343	9,098	3,781	12,879
1986[2]	3,484	232	4,080	5,346	8,184	13,535	10,223	7,268	17,615
Total[3]	47,552	5,977	53,524	19,551	4,886	24,437	67,103	10,863	77,960
Average[3]	6,793	854	7,647	2,793	698	3,491	9,580	1,552	11,138

[1] Source: Marine Recreational Fishery Statistics Survey data provide to NMFS Southeast Fisheries Center by D. Duel, Dec. 3, 1986. [2]Source: NMFS Landing Statistics, 1979-1985; 1985 data are preliminary.[3]Landings in state waters include landings for which the area of capture is unknown. [4] May not equal column totals due to rounding off.

[2] Preliminary data subject to change, Texas data and headboat data not available.

[3] for 1979-1985.

tion rates are and have been higher than the level which would maintain the spawning stock, even if no offshore fishing occurs on the spawning stock. They have indicated that in order to assure that overfishing does not occur the stock must not be reduced below 20 to 40 percent of the spawning stock size that existed before any fishing occurred.

Therefore, the Gulf-wide status or condition of the fishery is not good and rather significant regulatory actions are and will be required to assure the spawning stock is restored and maintained and to improve the productivity of the inshore fisheries. These regulatory actions will result in restricted harvest levels for fishermen in the near-term in order to increase long-term productivity from the fishery.

The Gulf states are taking and planning actions to regulate the inshore fishery to allow a minimum escapement level of 20 percent of the fish that would have escaped had there been no fishery. This means that 20 percent of each year-class of fish should be allowed to survive in the inshore fishery so they may migrate to the offshore spawning stock when they reach three or four years of age or approximately 30 inches in length. Considering that annual escapement levels have been as low as one percent of each year-class as early as 1964 for Florida and much below 20 percent for other states more recently, even attaining a 20 percent escapement level will require several years for some state regulatory agencies.

NMFS through implementation of a Secretarial Fishery Management Plan (FMP) and the Council through an amendment to that plan have taken actions to prohibit directed harvest of red drum from the offshore spawning stock, allowing only small allocations for incidental bycatch from the EEZ by recreational and commercial users. The Council's amendment will prohibit such directed harvest until the escapement goal of 20 percent is reached by the states. It also prohibits any retention of red drum from federal waters off Texas and Florida where data suggested the historical levels of escapement to the spawning stock were lowest. Therefore, for the immediate future, total landings by recreational and commercial users from federal waters will not exceed 625,000 pounds, annually.

State[3]

Actions by the states to increase escapement of juvenile fish to the spawning stock will similarly reduce harvest for inshore waters. Texas Parks and Wildlife Department, who recognized overfishing was occurring in their fishery in the mid-1970s, implemented progressively more restrictive rules to reduce fishing harvest levels. Based on a legislative mandate, they implemented a prohibition on sale of red drum in 1981. Currently their rules provide for a recreational bag limit of five fish that must be between 18 and 30 inches in length. Restrictions such as

[3]*State rules have been changing rapidly in efforts to increase escapement of juveniles to 30 percent (the current escapement goal). Each state should be contacted for current rates.*

these not only will increase escapement, but will also over time increase the poundage available from the inshore fishery. Texas also has been stocking hatchery-reared red drum to increase recruitment to the fishery.

Florida has currently closed its red drum fishery to all harvest by emergency rule that will be extended by permanent rule while they develop conservation measures to increase the escapement level. Alabama prohibits sale of red drum from its waters and is considering a reduced bag limit and size limits for their fishery. Mississippi limits commercial catch by a 200,000 pound annual quota and recreational catch by size limits and a bag limit of ten fish. Louisiana's legislature is considering bills which would implement commercial quotas and more restrictive recreational bag and size limits.

The status of the fishery is, therefore, in somewhat of a flux and probably will remain so for several years. Fortunately, however, a great deal of additional scientific information is being collected on the fishery that will give periodic bench marks to better assess the status of the stock and whether regulatory restrictions should be increased or relaxed.

Development of an Aquaculture Industry – The Catfish Industry

Thomas L. Wellborn, Jr., Professor Emeritus
Department of Fisheries and Aquaculture
University of Florida

The channel catfish (*Ictalurus punctatus*) is the most important commercially cultured species of fish in the United States. In 1986 there were 133,100 acres of ponds in production of farm-raised catfish (Table 1) primarily channel catfish. Most of this production is located in Mississippi where there are 85,139 water acres devoted to this enterprise (Wellborn *et al.*, 1987).

For any aquacultural commodity to develop into a viable commercial enterprise there is a certain logical sequence of events or stages that must occur. The commercial catfish industry can serve as a model for the development of other aquatic species into commercially viable aquacultural commodities. The channel catfish is the most recent and the most successful aquatic animal to be cultured on a commercial scale. These events or stages in the development of the commercial catfish industry are discussed. Each of these events or stages serves as a foundation for those following, although it must be recognized that they are not discrete events in themselves but rather are interwoven in a complex manner.

1. Hatchery: Develop technology for spawning.
2. Feed: Develop a suitable and economical feed for the extensive production of "seed stock" and food-size fish.
3. Education: Develop extension education and applied research program.
4. Marketing: Develop markets for food-size fish.
5. Year-round production: intensive production for market expansion.
6. Industry infrastructure: develop infrastructure needed to support a viable expanding industry.

Table 1. Surface acres of water in commercial catfish ponds in production in the United States.

Alabama	14,500[a]	Mississippi	85,139[j]
Arkansas	8,414[b]	Missouri	2,500[k]
California	2,300[c]	North Carolina	50[l]
Florida	254[d]	Oklahoma	1,240[m]
Georgia	6,000[e]	South Carolina	250[n]
Idaho	120[f*]	Tennessee	4,000[o]
Kansas	1,790[g]	Texas	700[p]
Kentucky	200[h]	**Total**	**133,157**
Louisiana	5,700[i]		

a. J.W. Jense, Pers. Comm.
b. USDA Crop & Livestock Reporting Service
c. F. Conte, Pers. Comm.
d. R.L. Busch, 1985
e. G.W. Lewis, Pers. Comm.
f. G.W. Klontz, Pers. Comm.
g. Bush, 1985
h. G.L. Jensen, Pers. Comm.
i. G.L. Jensen, Pers. Comm.
j. Wellborn *et al.* 1987
k. L.C. Belusz, Pers. Comm.
l. R.L. Busch, 1985.
m. R.W. Altman, Pers. Comm.
n. T.E. Schwedler, Pers. Comm.
o. T. Hill, Pers. Comm.
p. J.T. Davis, Pers. Comm.

*Raceway production converted to equivalent surface acres of ponds.

Economic Aspects

Although it was not done until late in the development of the farm-raised catfish industry, economists should be involved in every phase of applied research relating to the production, processing and marketing of a new aquaculture species. Knowing the cost of each item needed for producing, processing and marketing of a particular species is invaluable in directing research to the areas that can have the greatest impact on these costs. Research by Dr. John E. Waldrop and his associates and students (Foster and Waldrop, 1972; Waldrop and Smith, 1980) on the economics of catfish production had an immeasureable impact on the development of the catfish industry in Mississippi.

People involved in the development of an aquatic species as a commercial enterprise must always keep in mind the difference between biological feasibility and economic feasibility. Too often in the past researchers and those responsible for technology transfer have forgotten this and as a consequence farmers and investors have lost money. This, in fact, helped serve as a hindrance to the development of catfish farming in several states.

Catfish Farming Industry Development

Hatchery

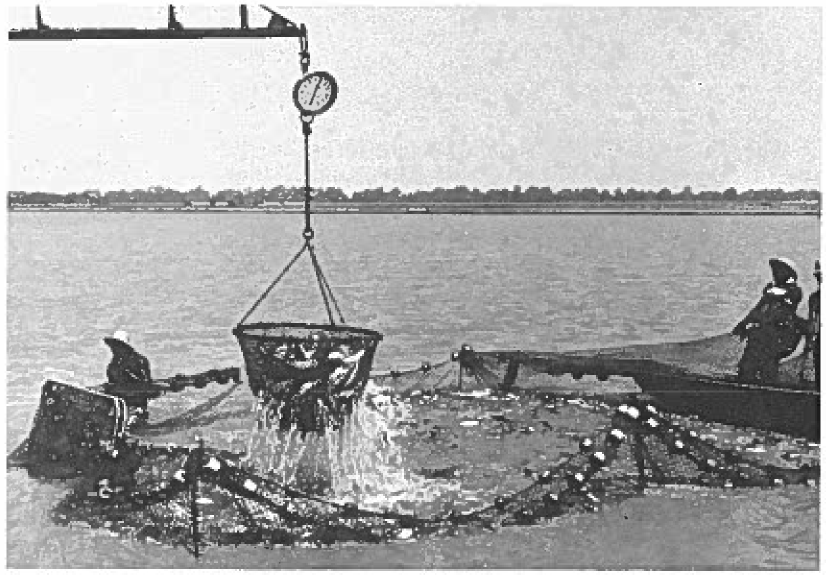
Attempts to determine the spawning requirements of channel catfish started in the early 1900s (Leary, 1910). These attempts to successfully spawn channel catfish were continued by a number of fishery scientists and federal and state fish hatchery workers (Clapp, 1929; Mobley, 1931; Lenz, 1947). By 1950 the basic methods for spawning channel catfish had been developed and are still in use today. Thus, it took about five decades to get through the first phase and only about three and a half decades to reach a viable, economically important industry worth an estimated $300 to $350 million to farmers in 1986.

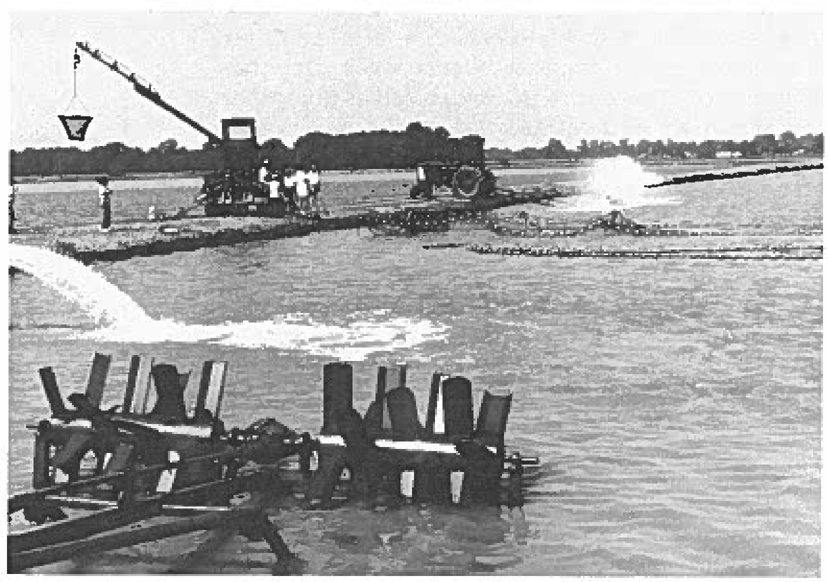
In addition to learning how to spawn and hatch eggs, research must be directed at the development of the technology needed to domestic brood stock. A viable commercial industry can seldom develop if brood stock must be captured annually from the wild.

Feed

Early attempts to raise fingerling and food-size catfish centered around the use of inorganic fertilizers to increase the natural productivity of the ponds and the use of a variety of food items to supplement the natural food available (Morris, 1939; Swingle, 1954). Maximum annual yields using fertilization only was 92 pounds per acre. Feeding soybean cake increased this yield to only 247 pounds per acre per year (Swingle, 1954). These production levels were too low to form the basis for a viable commercial enterprise.

Swingle (1956) conducted further experiments on the feasibility of commercial producing channel catfish using high stocking rates; feeding an artificial feed composed of soybean oil meal, peanut oil meal, fish meal and distillers dried solubles; and fertililzing with an inorganic fertilizer. Maximum production achieved with this method was 1,242 pounds per acre. Swingle (1959), Prather (1961) and others continued research on production technology, including feed formulations, needed for a successful commercial catfish farming enterprise. Although these early diets were





Catfish harvest from a commercial pond in the Mississippi delta.

not nutritionally complete, they gave satisfactory results at the low stocking densities (1,000 to 2,000 fish per acre) then used.

Education

Because aquaculture is a relatively new farming enterprise in the United States and requires totally different managerial skills from those normally acquired in the production of domestic livestock and row crops, extension education programs and assistance are necessary for training people on how to produce, process and market fish. Applied research is also necessary to develop the information needed to improve production, processing and marketing technology.

Early assistance to catfish farmers was provided by Auburn University and the Arkansas Game and Fish Commission. But Meyer *et al.* (1967) pointed out that "the high number of novices in fish culture has taxed assisting agencies to the limit and further expansion of the industry can only increase the need for technical assistance."

The value of extension aquaculture education programs can be illustrated by using those developed in Mississippi for the catfish industry as a model. Commercial production of food-size catfish started in Mississippi in 1965 with the construction of a 40-acre pond in Sharkey County (Wellborn, 1983). At that time there were already 9,500 acres of commercial catfish ponds in Arkansas (Meyer *et al.*, 1967). By 1968 the acres in catfish production were expanding rapidly in the state and farmers went to Mississippi State University, the Land Grant University, and requested assistance. As a result of this request, Mississippi State University hired its first extension fisheries specialist and fisheries researcher. Extension education programs for catfish farmers in Mississippi started in 1969 with a water quality management workshop. Since that time numerous extension education programs on all phases of catfish farming, including processing and marketing, have been conducted by specialists, agents and home economists of the Mississippi Cooperative Extension Service. Extension programs were also conducted for personnel of lending institutions to educate them as to what was involved in the commercial production of catfish. Additionally, the Mississippi Cooperative Extension Service Fish Disease Diagnostic Laboratory provided absolutely essential educational programs and assistance to fish farmers on disease diagnosis, treatment recommendations and water quality assessment.

The role of the extension worker is to provide educational opportunities to citizens of the state and assist them in solving problems to enhance the quality of their life. The extension worker also serves as the liaison between clientele and researchers at the university. They translate applied research information into layman's terms and take it to their clientele. Extension workers identify problem areas on which the researcher should focus. Mississippi State University has used the multidisciplinary approach and has involved all of the appropriate subject matter departments, both in the Mississippi Cooperative Extension Service and the Mississippi Agricultural and Forestry Experiment Station, in assisting the catfish farming industry in the state.

The research by many different scientists (Cruz, 1975; Lovell, 1971; Prather and Lovell, 1972; Prather and Lovell, 1973; Tiemeier *et al.*, 1965; Wilson, 1973), on the nutritional requirements of channel catfish was invaluable since it allowed the formulation of an economical, nutritionally complete feed needed for intensive production. Extension education programs and applied research certainly played a major role in the development of the catfish industry in Mississippi.

Marketing

Initially the food-size catfish produced were sold to local live markets. As acreage increased in the late 1960s these local markets were soon saturated and farmers had to look for other outlets for their fish. Because of the pressure to sell their catfish, some farmers started small-scale processing on their farms and began to sell dressed catfish to local restaurants and grocery stores. In the late 1960s several large companies became interested in the processing and marketing of farm-raised catifsh as a means of diversification. Many of these initial small processing plants failed for a variety of reasons.

With the advent of these small processing plants came efforts to expand beyond local markets to regional and national markets. These early efforts met with limited success because of the production methods used. Basically, large fingerlings were stocked in the late winter or early spring, fed during the spring, summer and early fall, and then harvested. Thus, catfish were available for sale only during part of the year. This production method proved to be a major handicap in developing markets if catfish farming was to become an industry and expand beyond some local and regional markets. The growth of catfish processing is shown in Figure 1.

Year-round Production

The sporadic supply of catfish, lack of processing capacity and suitable market outlets plagued catfish farmers until about 1974 (Wellborn, 1983; Waldrop, 1986). Two things occurred then that had a major impact on catfish farming in Mississippi. First, Paul Smith of Yazoo City, Miss., initiated an innovative production method enabling farmers to supply food-size fish throughout the year; and, secondly, some Mississippi catfish farmers formed a cooperative to build and operate a feed mill that produced a catfish feed of a consistently uniform, high quality. Because of these two factors, Mississippi catfish farmers claim "industry status" starting in 1974.

The development of the multiple harvest or top-

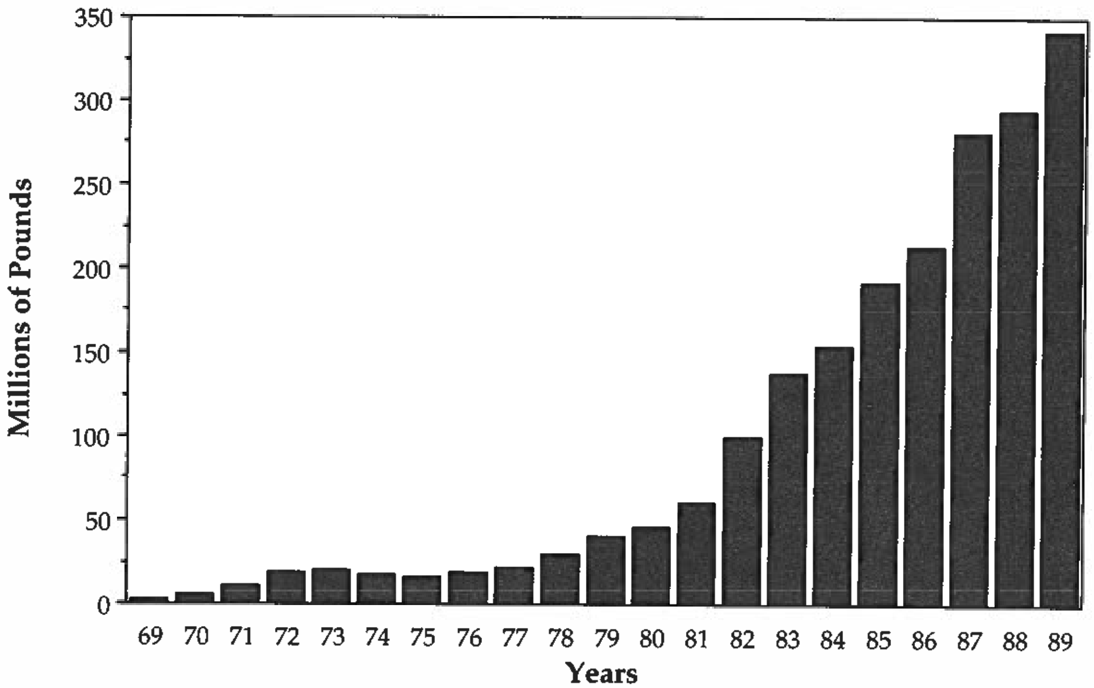


Figure 1. Millions of pounds of live farm-raised catfish processed by catfish processing plants from 1969 through 1989 (USKA Catfish, Crop Reporting Board, Economic and Statistics Service).

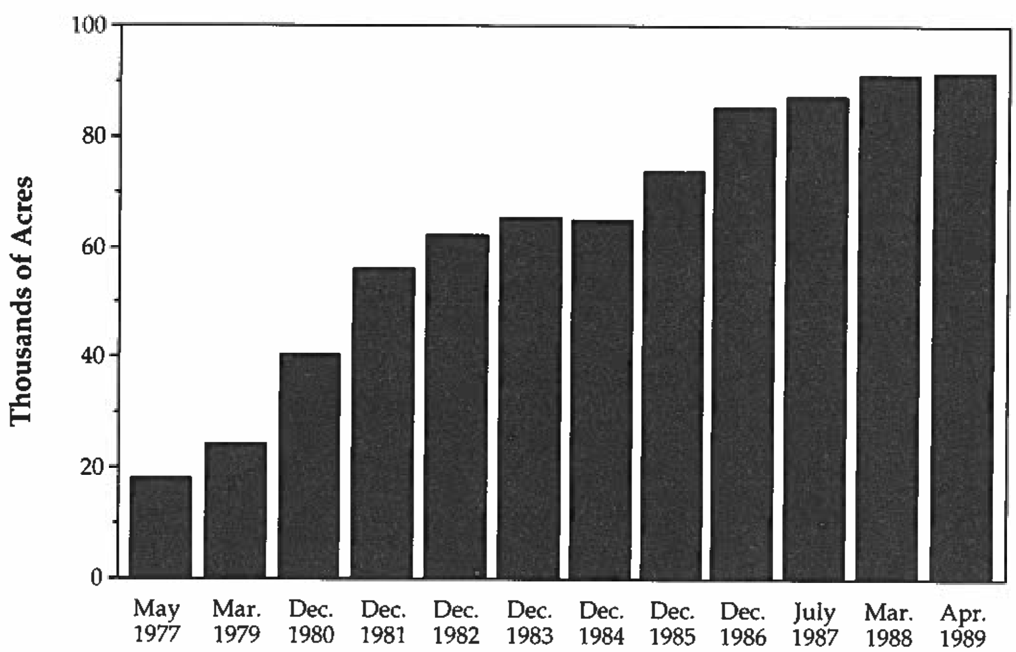


Figure 2. Water acres in production of farm-raised catfish in Mississippi from May 1977 to April 1989.

ping system allowed farmers to provide processing plants with catfish year-round. Thus, processing plants were able to expand existing markets and, more importantly, develop new markets that required fish all year.

Building of the catfish feed mill by the farmer cooperative allowed catfish farmers to ensure the quality of feed needed for best growth and feed conversions at reasonable prices. Feed was formulated according to the best knowledge available and was changed only when applied research has demonstrated the value of changing it.

Industry Infrastructure

Critical to the development of catfish farming as a full-fledged industry in Mississippi was the bringing together of all of the component parts, i.e., natural resources, production, extension and applied research programs, feed mills, processing capability, equipment and chemical suppliers, and marketing, in one geographic area. This needed infrastructure developed in Mississippi as the result of the close working relationship between innovative farmers, business men, Mississippi Cooperative Extension Service, Mississippi State University, and appropriate state and federal agencis.

The growth of the farm-raised catfish industry is evidenced by the increase of the pounds of farm-raised catfish processed annually since 1969 (Figure 1) and the increase in water acres in Mississippi in the production of farm raised catfish (Figure 2). Many different factors, both overt and subtle, influence the success of any new enterprise. Failure to keep in mind that the ultimate goal of a farmer, business man, or entrepeneur is making a profit is one of the greatest impediments to the development of a new enterprise. From what I know, it appears that redfish culture as a commercial enterprise is in stage two of development. Redfish can be spawned with a minimum of trouble, although I do not know if cost of production has been determined. Suitable and economical feeds for the production of fingerlings and food-size fish are still being developed.

It is apparent from the information available in the manual and from the strong attendance at the Red Drum Aquaculture Conference, that progress is being made in developing extension education and applied research programs, markets for the product, and production methods. I feel that the fledgling redfish insdustry can certainly benefit from looking closely at the farm-raised catfish industry to learn where it had successes and failures.

Literature Cited

Busch, R.L. 1985. Channel catfish culture in ponds. *In* C.E. Tucker, Ed. **Channel Catfish Culture**, Elsevier Science Publishing Co., Inc. N.Y., N.Y. 66 pp.

Clapp, A. 1929. Some experiments in rearing channel catfish. **Trans. Amer. Fish Soc.** 59:114-117.

Cruz, E.M. 1975. Determination of nutrient digestibility in various classes of natural and purified feed materials for channel catfish. Ph.D. Dissertation, Auburn Univ. 82 pp.

Foster, T.H. and J.E. Waldrop. 1972. Cost-size relationships in the production of pond-raised catfish for food. **Ms Agricult. & Forest. Exp. Station Bull.** 792, 69 pp.

Leary, J.L. 1910. Propagation of crappie and catfish. **Trans. Amer. Fish Soc.** 39 (1909): pp. 143-148.

Lenz, Gerhard. 1947. Propagation of Catfish. **Prog. Fish-Cult.** 9(4):231- 233.

Lovell, R.T. 1971. Calcium and phosphous requirements of channel catfish. **Fisheries Ann. Rep. Ala. Agr. Exp. Sta.**, Auburn Univ., 37 pp.

Meyer, F.P., D.L. Gray, W.P. Mathis, J.M. Martin and B.R. Wells. 1967. Production and returns from the commercial production of fish in Arkansas during 1966. **Proc. 21st Annual Conference of the S.E. Assoc. Game and Fish Comm.**: pp. 525-531.

Mobley, Ben E. 1931. The culture of channel catfish (*Ictalurus punctatus*). Trans. Amer. Fish Soc. 61:171-173.

Morris, A.G. 1939. Propagation of channel catfish. **Prog. Fish-Cult.**, No. 44:23-27.

Prather, E.E. 1961. A comparison of production of albino and normal channel catfish. **Proc. 15th Ann. Conf. S.E. Assoc. Game & Fish Comm.**, pp. 302-303.

Prather, E.E. and R.T. Lovell. 1972. Effect of vitamin fortification in Auburn No. 2 fish feed. **Proc. S. E. Assoc. Game and Fish Comm.** 25:479-483.

Prather, E.E. and R.T. Lovell. 1973. Response of intensively fed channel catfish to diets containing various protein-energy ratios. **Proc. S. E. Game and Fish Comm.** 27: 455-459.

Swingle, H.S. 1954. Experiments on commercial fish production in ponds. **8th Ann. Conf. S.E. Assoc. Game and Fish Comm.**: pp. 69-74.

Swingle, H.S. 1956. Preliminary results on the commercial production of channel catfish in ponds. **Proc. 10th Ann. Conf. S.E. Game & Fish Comm.**: pp. 160-162.

Swingle, H.S. 1959. Experiments on growing fingerling channel catfish to marketable size in ponds. **Proc. 12 Ann. Conf. S.E. Game & Fish Comm.**: pp. 63-72.

Tiemeier, O.W., C.W. Deyoe and S. Weardon. 1965. Effects on growth of fingerling channel catfish of diets containing two energy and two protein levels. **Trans. Kansas Acad. Sci.** 68:180-186.

Waldrop, J.E. 1986. An overview of the farm-raised catfish industry in the United States-an agricultural economists view. *In* Bilio, M., H. Rosenthal and C.J. Sinderman, (Eds.). **Realism in Aquaculture: Achievement, constraints, perspectives. Euporean Aquaculture Soc.** pp. 519-533.

Waldrop, J.E. and R.D. Smith. 1980. An economic analysis of producing pond-raised catfish for food in Mississippi: A January 1980 update. **Dept. of Agricult. Econ. Res. Rep.** (MAFES) 103.

Wellborn, T.L., Jr. 1983. The catfish story: farmers, state services create new industry. **1983 Yearbook of Agriculture**, USDA, pp. 298- 305.

Wellborn, T.L., R.M. Durborow, M.D. Crosby and P.W. Taylor. 1987. Status of fish farming in Mississippi-December 1986. **For Fish Farmers, MS Cooperative Extension Service**, 7 pp. mimeo.

Wilson, R.P. 1973. Absence of ascorbic acid synthesis in channel catfish, *Ictalurus punctatus*, and blue channel catfish, *Ictalurus furcatus*. **Comp. Biochem. Physiol.** 46B: 635-638. Production in the United States.

Broodstock Collection

Richard E. Tillman
County Marine Agent
Texas Marine Advisory Service
Rockport, Texas 78382
and
Joe T. Surovik
County Marine Agent
Texas Marine Advisory Service
Port Lavaca, Texas 77979

This chapter will describe some techniques on how to catch, then transport brood red drum to the hatchery site. It cannot be emphasized too strongly that adequate time be budgeted for this task. When spawning tanks are ready, the hatchery operator should determine the closest source of bull reds and begin talking with fishing guides to identify local bull red fishermen. These people are more often accessible at the local coffee shop or tackle store than at their homes. Also, listen for reports of bull reds being caught near jettied passes and in the surf just off the beach.

The size of a red drum is directly related to its age and sexual maturity. Most researchers feel that it takes four years for a red drum to reach sexual maturity. Pearson (1929) found that red drum are sexually mature at approximately 750 mm (29.5 inches), although smaller "ripe" fish have been reported (Gunter, 1949).

Fishing Regulations

Fishing regulations should be consulted before attempting to take fish from state waters, or transport fish to the hatchery, i.e. from one state to another. Several states have adopted a maximum size restriction for red drum as a conservation measure. In such states it will be necessary to obtain a collecting permit to source oversized brood fish.

In Texas, for example, a permit will be issued only after Texas Parks and Wildlife Department personnel have reviewed the layout of the hatchery, and determined that appropriate handling procedures will be followed.

To source brood fish in federal waters first contact the National Marine Fisheries Service. Their regional office address is: The U.S. Department of Commerce; National Oceanic and Atmospheric Administration; National Marine Fisheries Service; Region IV Office; 9450 Koger Blvd.; St. Petersburg, Fla., 33702; 813/893-3722.

See Appendix B for list of permitting agencies.

Location of Adult Red Drum

Ideally, brood fish should be obtained from waters as near as possible to the hatchery's water source. Placing brood fish in similar quality water allows them to recover more rapidly from the stress of handling.

Migratory Habits

Large redfish school in nearshore waters in the early fall to spawn (see Life History by G. Matlock). These large breeders are often found in shallow water near natural and jettied passes at this time of year. Bull red fishermen pride themselves on their ability to catch large red drum, and have proven to be good indicators of the presence of brood-sized stocks.

These breeding fish remain in nearshore Texas waters as late as December or early January, then move to deeper waters (90 to 120 feet) as the water cools.

Broodstock Capture Methods

Several techniques have been used to capture brood red drum. These include hook and line, modified longline, and beach seine.

Hook and Line

Hook and line technique can be used anytime of the year, and in most locations—on the beach, nearshore, and offshore. Tackle should include a 3/0 to 4/0 light tackle gamefish reel with 30/40# test monofilament line and a medium to heavy action six-foot rod. (Smaller tackle tends to stress the fish because of the extended time required to land it.) Use either a 7/0 short shanked beak hook or a 3/0 to 7/0 circle tuna hook.

The terminal rigging is the same whether fishing from a boat or the beach. Use an egg sinker heavy enough to keep the bait on the bottom.

Live bait is preferred, with fresh dead the next choice, and frozen the least desired. Most effective live baits have included 6- to 10-inch mullet (eye or dorsal fin hooked, depending on the currents), and 10- to 15- count shrimp (hooked under the rostrum or horn).

Dead baits include the same size mullet and shrimp. Cut the mullet in half and hook the meat away from the cut end. Hook the shrimp mid-body.

When fishing from a boat, bounce the bait along the bottom. When fishing from the beach, locate and fish deep holes. (These can be determined by observing the pattern of breaking waves).

The seaward end of jettied passes, about 100 yards off the rocks, has been productive along the Texas coast. When moving down the jetties, the beach side (vs. the channel) has consistently produced more brood fish. Fish the outgoing tide. Strong currents move forage fish out of the passes where large redfish feed on them.

It cannot be stressed enough how important it is to take special care of captured brood fish. Fight the fish firmly, but gently. Get the fish to the boat (or beach) as soon as possible, but do not aggravate the battle. Boat and dehook carefully (or cut the leader), then gently place the fish in the transport container. Maintain oxygen above 5ppm by diffusing compressed oxygen through an airstone.

Chances for infection increase as excessive mucus is removed through rough handling. Fish are best carried wrapped in a wet towel. Covering the eyes with the towel seems to calm them down.

The Longline Method

This method is thought to result in the least stressed/damaged animal; however, it is more capital intensive. This is actually a "short" longline fished off the beach – less than one mile in length and fished in 10 to 40 feet of water.

The mainline is 1000# test braided nylon or 700# monofilament. Stainless snaps are attached to 2 to 10 feet long gangions (200# monofilament) rigged with 3/0 to 7/0 circle tuna hooks. Gangions are spaced 10 to 25 feet apart on the mainline. Baited gangions are commonly stored in tubs or longline racks. For best results these lines should be set nearshore, from the beach to 30 feet from August through October; and 30 to 60 feet from November through January.

Best baits for longline fishing are the same as those listed for hook and line fishing.

It is important to run the line frequently. This results in less stress on hooked fish, prevents shark attacks, and alerts commercial trawlers to the location of the line.

Commercial fishermen may be contracted to longline for brood redfish. Local Marine Advisory agents can assist in contacting these fishermen, as well as help in obtaining the necessary permits to retain oversized fish.

Beach Seining Method

It is possible to catch broodstock with a seine, but this technique requires a trained fishing crew with proper gear including:

a. A minimum 300-foot seine, 6- to 8-feet deep, with 2- to 3-inch mesh. Leglines should be extended 200 feet on each end.
b. 16-foot flat bottom skiff (net boat). Larger boat with transport tank if the beach is not accessible to vehicles.
c. Fish live car—4'x6'x4', 2-inch mesh with cork flotation—to hold fish until transported to the hatchery.
d. 6 to 8 people.

First determine when brood-sized fish are on the beach in your particular area. Peak occurrence is usually September through October. These large fish tend to congregate closer to the beach in rough weather, and are not as easily frightened by fishermen or the net. The best beach seining for bull redfish on the Texas coast has been in late September in 3- to 5-foot seas.

When beach seining, first locate deep cuts (3 to 6 feet) between bars parallel to the beach. Two people then swim one end of the seine to shore, while the net shift remains seaward of the surf. After getting one end of the seine on the beach, the boat pays out the remaining net as it runs with the current parallel to the beach.

The seine is then pulled with the current for approximately 500 feet (depending on longshore current and wave action).

The beach crew should be careful to keep the net at the water's edge as they walk it down the beach to prevent fish from escaping through shallow water.

The boat is beached by catching a breaking wave and the net is immediately removed and pulled up the beach. The four man crew then "purses" the net by constantly pulling it up onto the beach. Brood fish, which have gathered in the center of the net are best collected while still in the shallow water by using a net made of wide, soft webbing (1/4 inch ACE knotless nylon).

If the surf is too rough to risk this capture technique, pull the fish onto the beach. Wrap the fish in wet towels (cover the eyes) when moving to the live car or transport tank.

Abdominal Gas Formation

When fish are brought up quickly from deep water, gas may form in the abdomen. The mechanism for this gas formation is not clear, but it may cause the fish to turn belly up in the hauling tank. This condition may be avoided by landing the fish more slowly. However, this may subject the animal to excessive stress while on the line. The stress may be more harmful than the inflated gas.

To remove the gas from the abdomen a needle should be inserted just forward of the anal vent until gas begins escaping. The fish may be held beneath the water as the gas escapes, the additional water pressure will aid in its escape. Following this minor surgery the fish should be able to swim more upright and breath properly.

If the fish will be placed in a deep holding tank (10 ft.+) within one to two hours, it may be best not to bleed the abdominal gas but see if the fish can compensate by sounding.

Transport

Transportation stress should be reduced as much as possible to ensure faster recovery time after delivery to the hatchery. If the brood stock is captured from a boat, a hauling tank measuring 39-50 x 18-30 x 24-30 must be on board, and equipped with airstones and enough oxygen for the return trip to the dock. It is advised that the airstones be secured to an inside bottom edge of the tank to reduce additional stress on the fish. If it is possible to pad the end of the tank this will also reduce stress.

An intermediate holding facility, close to the point of capture, might be used prior to a long distance haul (12 hours or more). Fish should be held long enough to become thoroughly accustomed to the hauling tank.

At least 5 ppm oxygen should be maintained in the hauling tank. Compressed oxygen diffused through an airstone not only saturates the water, but also dissipates toxic ammonia. If fish will be in the tank for five hours or more the water should be slowly cooled to about 70°F. This reduces the activity of the fish, plus it allows the water to absorb more oxygen.

Antibiotics can be added to the transport water to reduce the risk of bacterial infection. Acriflavine (1 to 2 ppm) and Furacin (5 to 15 ppm) have been successfully used as bactericides.

If the temperature or salinity of the receiving water is more than a few degrees (or parts per thousand) different from the hauling tank water, the waters should be slowly (3 to 4 hours) mixed until they are uniform. Fish may then be transferred.

New fish should be isolated in separate spawning tanks or holding ponds until they are determined to be disease free.

While transferring fish from the hauling tank they should be sexed (see Daniel Roberts' "Photoperiod/ Temperature Control in the Commercial Production of Red Drum (*Sciaenops ocellatus*) Eggs") and identifying marks noted so that the proper ratio of males to females can be placed in the spawning tanks.

Literature Cited

Gunter, G. 1945. Studies on marine fishes of Texas. **Publ. Inst. Mar. Sci., Univ. Tex.** 1(1):1.190.

Pearson, J.C. 1929. Natural history and conservation of the redfish and other commercial sciaenids on the Texas coast. **Bull. Bur. Fish.** 44:129-214.

Hormone Induced Strip-spawning of Red Drum

Robert L. Colura
Texas Parks and Wildlife Department
and
Perry R. Bass Marine Fisheries Research Station
Star Route Box 38
Palacios, Texas 77465

The manipulation of photoperiod and temperature to induce spawning has become the preferred method for routine production of red drum eggs. Strip-spawning, the manual removal and mixing of the sex products, however, offers several important advantages over photoperiod and temperature conditioning: spawning may be conducted in small, inexpensive tanks (200 to 600 gallons); natural spawning seasons which do not coincide can be synchronized; and the cost of long-term broodstock maintenance is eliminated.

On the other hand, strip-spawning has several disadvantages: the method is labor-intensive; substantial technical expertise is necessary to ensure that fertilization follows ovulation; and broodfish may not survive manual stripping of gametes.

Broodstock used for strip-spawning are generally collected by hook and line or net during the natural spawning season. However, hormone-induced strip-spawning methods similar to those used for striped bass (Stevens, 1966; Bayless, 1972) have also been used with red drum.

Hormone-induced strip-spawning is the only suitable method to produce hybrids (because of the limited interbreeding between different species). For example, personnel of the Perry R. Bass Marine Fisheries Research Station have cross fertilized black drum and red drum to produce hybrids that have demonstrated mariculture potential (Henderson-Arzapalo and Colura, 1984). If a black drum–red drum cross is to be produced, broodstock must be matured using photoperiod and temperature methods prior to strip-spawning as their natural spawning seasons do not coincide.

Evaluation of Broodstock Maturity

Red drum broodstock collected during the spawning season (mid-August to mid-October) are almost always eligible for hormone induced strip-spawning. However, the gonadal condition of all fish should be verified prior to hormone injection. Males may be examined for spermiogenesis by applying pressure along the sides and belly of the fish to extrude milt. Intra-ovarian tissue samples may be obtained from females by inserting a 1- to 2-mm diameter glass tube into the oviduct and withdrawing a small amount of tissue for microscopic examination (Stevens, 1966).

Ovarian samples will generally contain three oocyte stages. Primary oocytes are approximately 0.1 mm in diameter, transparent in appearance, and nucleated. As eggs begin to mature, yolk is deposited near the center of the egg during vitellogenesis. Yolk deposition causes an opaque appearance around the nucleus while the egg periphery remains clear. At this stage, ova diameter ranges 0.15-0.30 mm.

As vitellogenesis continues, the periphery also becomes opaque and the eggs assume a pale yellow color. The vitellogenic eggs measure $\geq$ 0.50 mm in diameter.

Hormone Injection

Once eggs complete vitellogenesis ($\geq$ 0.50 mm), earlier oocyte stages are not obvious in the intraovarian sample. When vitellogenic ova dominate the intraovarian sample, an intramuscular injection of approximately 500 to 600 IU of human chorionic gonadotropin (HCG) per kg of body weight will induce ovulation in approximately 24 to 30 h at 25°C. (Generally, it is not necessary to inject males.) During the final stages of egg maturation, ova will double in size (approximately 1.0 mm diameter), become transparent, and develop one to several oil globules.

When ovulation occurs, ova will be easily extruded by light pressure on the abdomen. Ova can then be manually stripped into a container and fertilized by stripping milt from a male. A measured volume of salt water (28-32 percent) should then be added and the ova stirred for approximately one minute. After mixing, the total volume of eggs and water is determined and subsamples of the egg-water mixture are counted to estimate the total number of eggs.

Procedures for Spawned Eggs

The mixture is subsequently transferred to a well aerated 37-liter (10-gallon) aquarium for 1 to 2 hours before ova are examined for mitotic division. If 70 percent or more of the ova examined exhibit mitotic division, eggs may be placed directly in the incubator and treated as eggs obtained by photoperiod and temperature-induced spawning. If fertilization is 30 to 70 percent, aeration in the aquarium should be discontinued for several minutes. Live eggs will float at the water surface, while dead eggs will settle to the bottom. Dead material is then removed by siphoning. If less than 30 percent of the eggs are fertilized, the spawn should be discarded.

These procedures generally produce approximately 1 to 3 million eggs from an 11 to 14 kg (25 to 30 pounds) female. However, keep in mind that hormone induced strip-spawning is labor intense, requires substantial technical skill, and may result in loss of broodfish. Nevertheless, hormone induced strip-spawning provides an alternative spawning method for red drum as well as a method to produce hybrids that would otherwise be unavailable to aquaculturists.

Literature Cited

Bayless, J.D. 1972. Artificial propagation and hybridization of striped bass, *Morone saxatilis* (Walbaum). **South Carolina Wildlife Resources Dept.** 135 p.

Henderson-Arzapalo, A. and R.L. Colura. 1984. Black drum x red drum hybridization and growth. **J. World Maricult. Soc.** 15:412-420.

Stevens, E. 1966. Hormone induced spawning of striped bass for reservoir stocking. **Prog. Fish-Cult.** 28:19-28.

Photoperiod/Temperature Control in the Commercial Production of Red Drum (*Sciaenops ocellatus*) Eggs

Daniel E. Roberts, Jr.
Florida Department of Natural Resources
Bureau of Marine Research
100 Eighth Avenue S.E.
St. Petersburg, Florida 33701

Commercial production of marine finfish has been limited by developments in the controlled maturation and spawning of broodstock. Successful fingerling production and grow-out rely upon a predictable supply of healthy eggs and larvae. Lack of such seedstock has played a significant role in the failure of mariculture ventures in Florida in the past 15 years.

This paper reviews the physiological basis of red drum maturation/spawning theory and technique and includes an outline of practical applications for the commercial aquaculturist.

Background Information

Finfish, like many other aquatic and terrestrial species, exhibit rhythmic physiological and behavioral patterns generically known as bio-rhythms. Fish reproduction (maturation, mating, and spawning) is often rhythmic and strongly correlated with the inter-related seasonal cycles of solar insolation, water temperature and food supply. This naturally leads to a period of the year which can be defined as the spawning season. The farther one proceeds from the equator, the more distinct the annual cycle and the more pronounced the spawning season.

Whereas the spawning season occurs once annually and during a rather restricted period in higher latitudes, it is often difficult to define in the tropics where seasonality is sometimes better defined in terms of "wet" and "dry" seasons.

To the fisherman or fish-farmer who observes the gonads of his fish as he cleans them, the spawning season may appear brief. But the period of gametogenesis and maturation, or ripening of the eggs and sperm, is more protracted and may take many months to culminate in spawning. Furthermore, the factors which control the onset of ripening are usually very different than those which control spawning itself. In subtropical and temperate latitudes, where red drum are usually found, the controlling factors for maturation and spawning are day length, water temperature, and food quality and abundance. These factors are termed exogenous (i.e., outside the organism).

There is mounting evidence that many fish species, perhaps even red drum, may also respond to cycles under endogenous (inside the organism) control. However, if endogenous control of red drum maturation and spawning exists, it can usually be entrained, if not overridden, by application of the appropriate exogenous controls.

Observations in the mid-late 1970s that red drum drastically modify their natural spawning cycle when under the influence of artificial exogenous factors, have led to development of a reproduceable technique for maturation and spawning of this valuable fish. Maturation and spawning can now be controlled very accurately in red drum. The quality of the resulting hatchery-produced eggs and fry is unsurpassed by any other hatchery-spawned marine finfish species. This egg production protocol and high survivability is the major technological basis allowing development of the red drum aquaculture industry.

Ambient Photothermal Cycles

Light : dark cycles are usually termed as photoperiod. Photoperiod is defined as the number of hours of light in a 24 hour period. In this discussion, the definition is further restricted in that the period of light is one continuous segment. This same definition applies to natural photoperiods as well.

Red drum complete their maturation and spawning cycle within photoperiods of 9 to 16 hours light and temperatures from 17°C to 30°C. Data for ambient conditions are available from several sources. When measured over time (starting from winter and ending the following winter), the natural annual photoperiod increases in spring, plateaus in summer, decreases in autumn, and plateaus again in winter. Temperature follows a similar seasonal pattern.

During the winter phase, the reproductive cycle is in a state of recovery and replication – recovery from the previous spawning season, and replication of sex cells, which have been depleted. During the spring acceleration phase, the replication of sex cells continues and enters a state of rest prior to the summer plateau. During the summer plateau, the sex cells of the female begin to make and store yolk, a process that becomes very obvious toward the end of the summer plateau.

During this same period male sex cells are undergoing a transition from a non-motile sex cell to a sperm capable of swimming. The fall phase of the cycle is the most conspicuous, as the gonads become very large, the sex cells ripen, and spawning begins.

Physiology

How does changing day-length and water temperature affect reproduction in fishes? The process is complex. Changes in photothermal energy are perceived by fish via sensory organs in the skin, eyes, and possibly the pineal organ (Figure 1). This information is passed to the brain where it is processed and evaluated. It then reaches an organ at the base of the brain, the hypothalamus, which converts neural signals into chemical information in the form of releasing hormones. These hormones from the hypothalamus stimulate production of gonadotropin (GTH) in the pituitary gland. GTH, released into the blood, reaches the gonadal tissue where it mediates production of certain steroids which control a variety of important reproductive functions (Figure 1).

Controlled Photothermal Cycles

Maturation Regime

The original controlled maturation regime for red drum condensed the natural cycle, from winter plateau to fall maturation, to 120-days (Fig. 2). Minimum temperature was above 20°C, and maximum was above 30°C. Daily temperatures fluctuated considerably, often warming during the cool fall phase and cooling during the warm spring phase. The 10-day average curve, however, showed a smooth transition from one season to another.

A variation of this initial regime, currently used at the John Wilson Marine Fish Hatchery (JWMFH), employs a lower winter minimum temperature of 15°C (Figure 3). The cost to achieve this lower temperature, especially during ambient summer, can be significant. The need to achieve this low temperature has yet to be demonstrated; it may indeed be necessary with certain broodstocks.

The ideal time to begin the controlled regime is during the winter plateau. During this phase, sex cells (gametes) are at the starting point of the reproductive cycle. But it is not always possible to begin a controlled regime at this point, especially if the hatchery is new and is just collecting brooders. In this case, the cycle is adjusted to ambient conditions at the time and place of collection. The following is a prototypic method for induced maturation of red drum in a hatchery. (Individual hatcheries may have to vary certain procedures due to biological variation in broodstock, systems design, etc.)

Procedure

1. Acclimate broodstock to winter photothermal conditions (17°C:9 HL).
2. Assure that broodstock water quality parameters are stable; do not stress biological filter by sudden changes in temperature in preparation for winter conditions.
3. Get fish on feed gradually by feeding about 3 percent body weight per day about five days per week.
4. After fish are on feed, the water quality parameters are within tolerable levels, and all other timed events are scheduled, follow the maturation table in Figure 4. Remember, fish can remain at checkpoint A until ready to begin the regime.
5. Proceed from checkpoint A to checkpoint B over 40 days. Time change is +7.5 minutes per day; temperature change is +0.27°C per day. Conditions at checkpoint B are temperature 28°C and 14 HL.
6. Proceed from checkpoint B to checkpoint C over 30 days. Time change is + 4.0 minutes per day, and temperature change is + 0.067°C per day.
7. Checkpoint C is summer. If for some reason you want to program a hold in the regime, this checkpoint, like checkpoint A, is a good place to do so. Conditions at checkpoint C are 16 HL, and 30°C. If no hold is planned, proceed to checkpoint D over 30 days.
8. Time and temperature change from checkpoint C to checkpoint D are -10 minutes and -0.17°C per day, respectively. Conditions at checkpoint D are 12 HL and 25°C.
9. From checkpoint D to checkpoint E is another 20 days bringing the photoperiod from from 12 HL to 10 HL at a rate of 6 minutes per day and temperature from 25°C to 23°C at -0.1°C per day.
10. Checkpoint E is the end of the maturation regime. Sex cells should be ripe or nearly ripe by the end of the 120-day regime.

Other regimes have been used successfully. Arnold *et al.* (1977), first induced maturation using a one year cycle in indoor tanks with a captive broodstock. Roberts *et al.* (1978d) succeeded in using a 90-day cycle, allowing an extended summer plateau. Experiments with 90-day maturation cycles are ongoing in Florida.

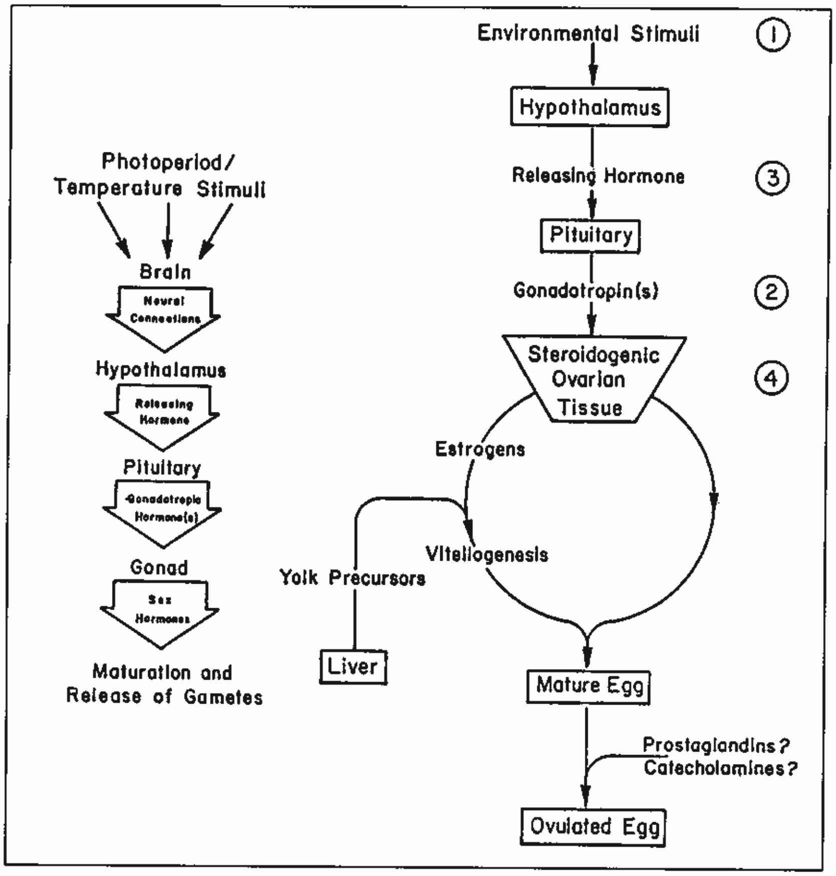


Figure 1. (A) Links in the physiological pathway from fish's perception of photothermal information to maturation and spawning. (B) Hormonal links in the physiological pathway from female fish's perception of photothermal (environmental) information to ovulation of eggs. Circled numbers refer to stages where the process can be influenced by artificial manipulation of hormones. (Modified from Harvey and Hoar, 1979).

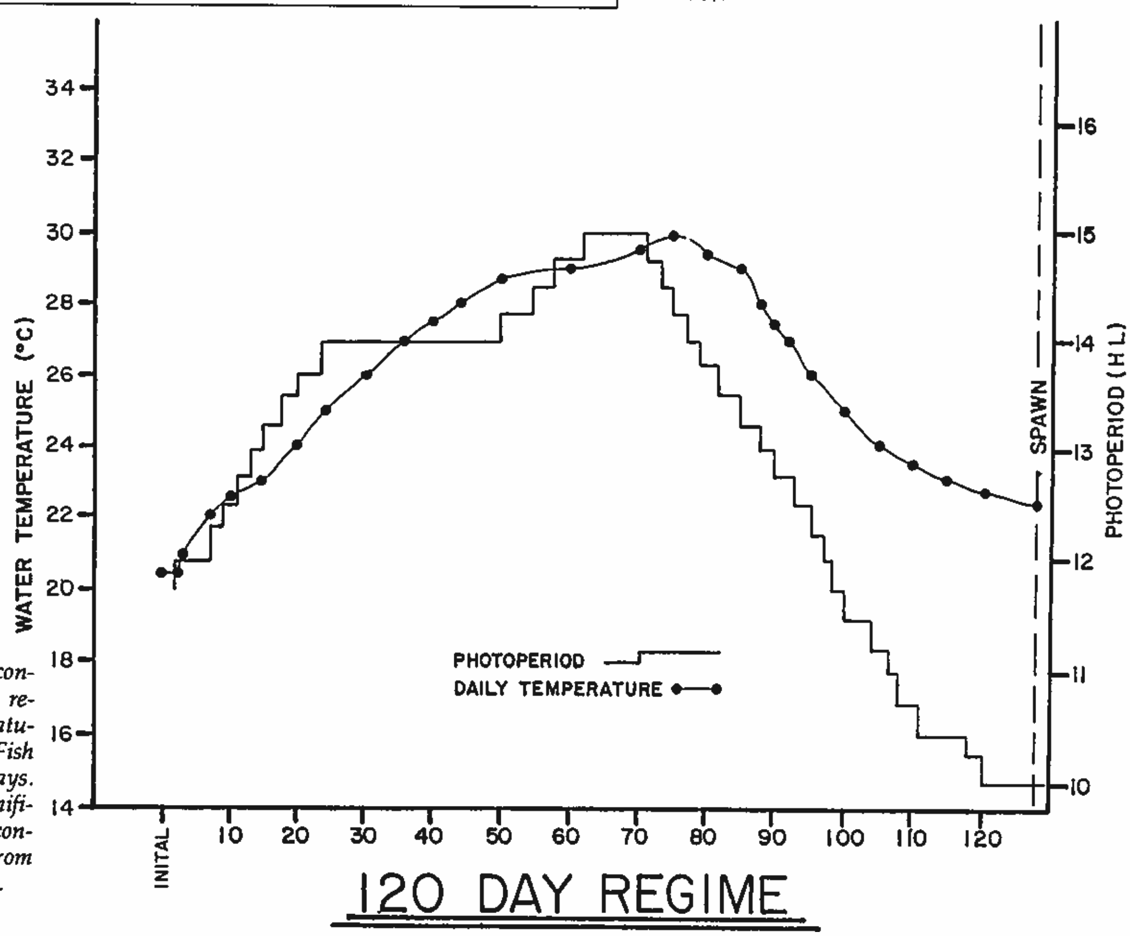


Figure 2. Original condensed photothermal regime (120 days) for maturation of red drum. Fish spawned after 129 days. Maturation was significantly advanced over controls at 120 days. (From Roberts et al., 1978a).

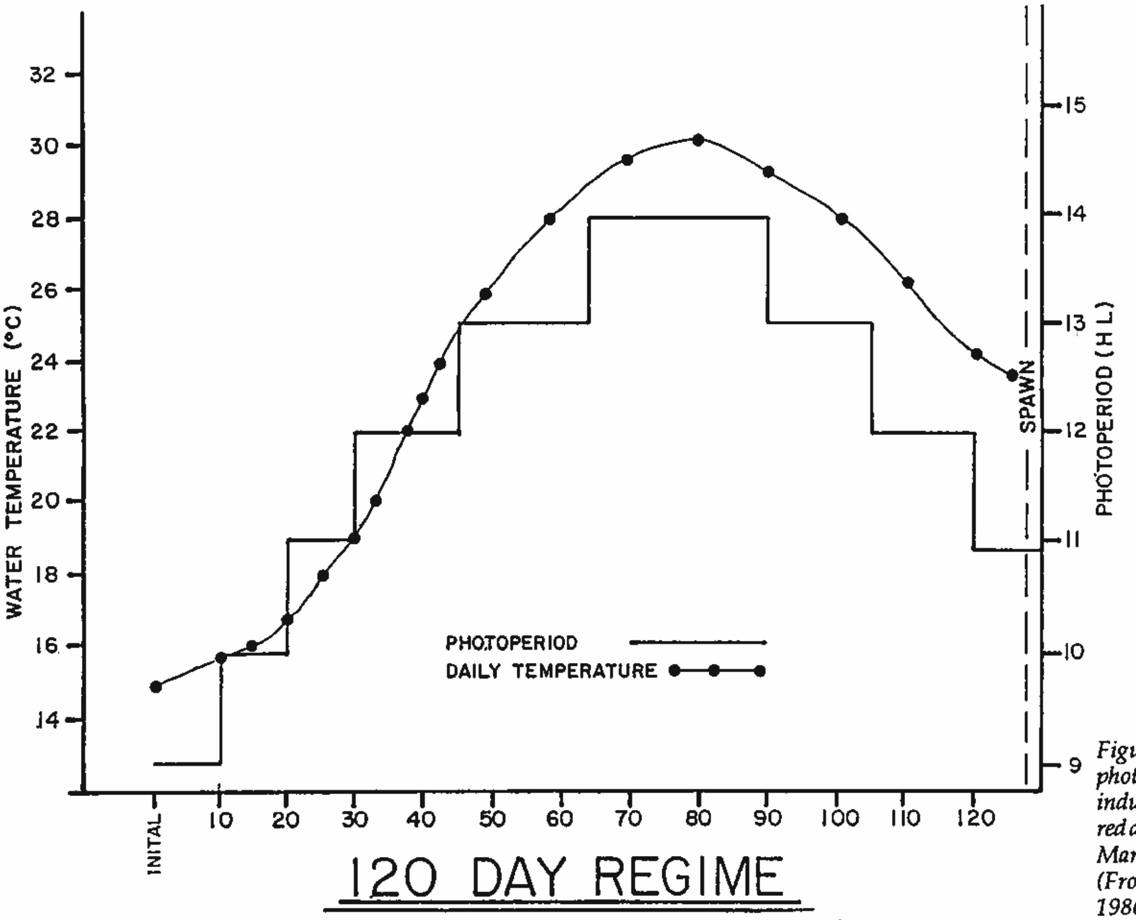


Figure 3. Controlled photothermal regime for induced maturation of red drum at John Wilson Marine Fish Hatchery. (From McCarty et al., 1986).

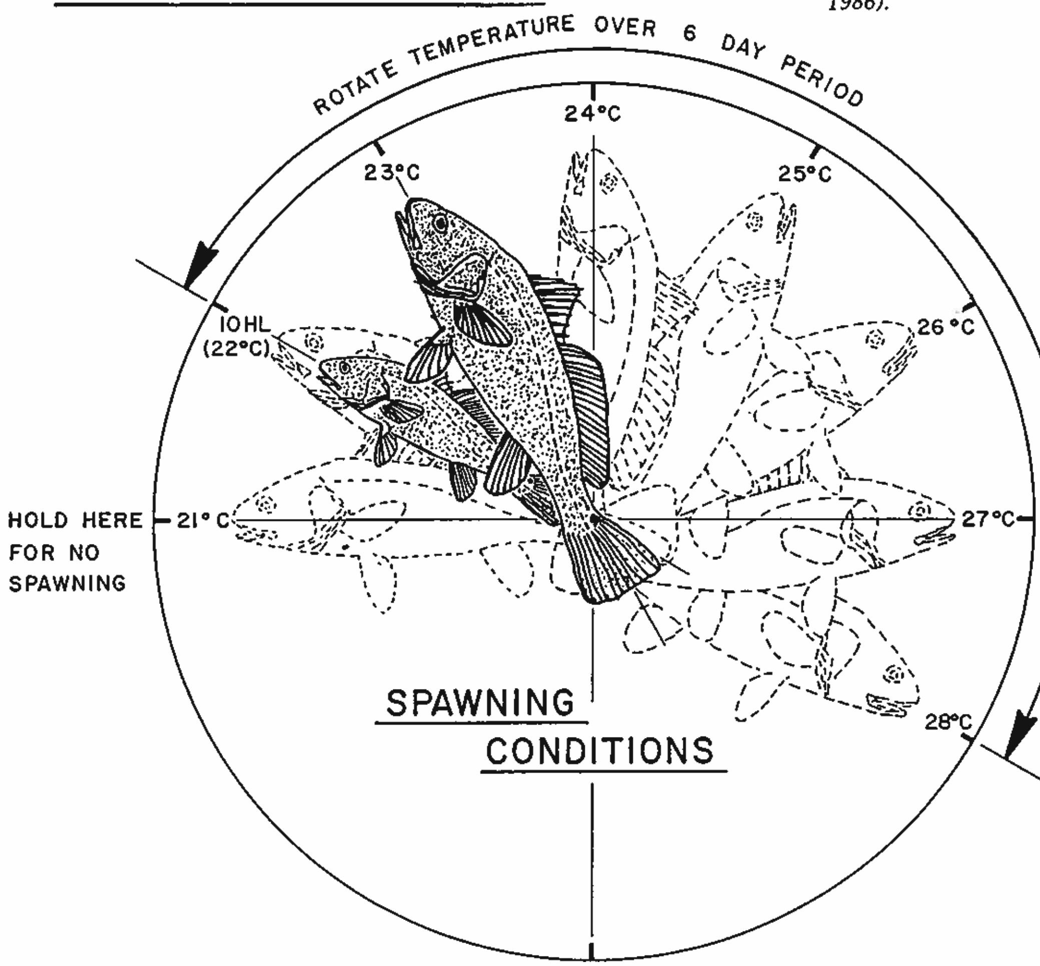


Figure 5. Temperature fluctuations to induce spawning in mature red drum broodstock. Photoperiod remains constant at 10 HL.

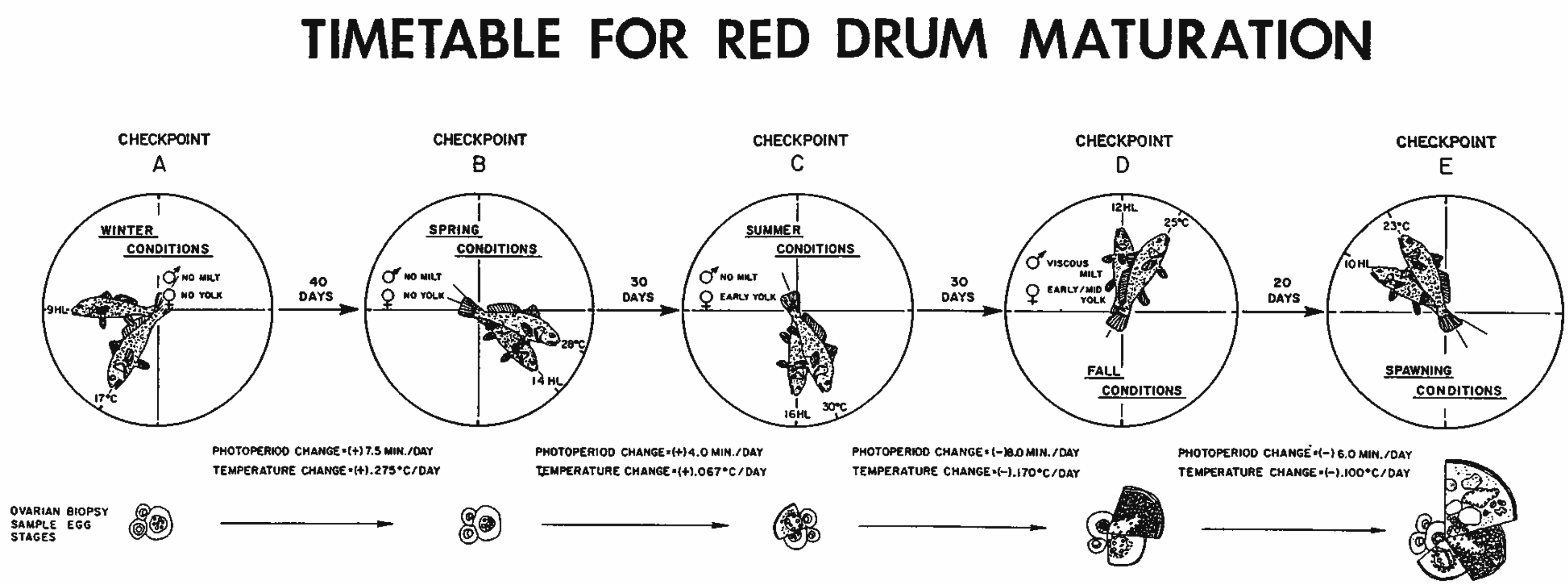


Figure 4. Timetable for red drum maturation. Demonstration of photoperiod and temperature changes necessary to induce reproductive maturation of red drum from winter condtion (checkpoint A) to spawning condition (checkpoint E), and diagramatic sample of oocyte stages for each checkpoint from ovarian biopsies of females.

Spawning Regime

Once spawning conditions (10HL:23°C) are reached controlled spawning can be achieved by a variety of methods. In fact the methodology used to control spawning becomes less consistent. Variation from one group of broodstock to the other is observed. This variation may be due to differences in fish size, holding tank size, holding tank shape and other factors. Some broodstock will continue to spawn every day for months at the spawning regime of 10HL:23°C. Others require a gentle manipulation of water temperature from 23°C to 28°C. Usually, spawning can be stopped for short periods of time by lowering temperature 1 to 2 degrees below the spawning temperature. This also has varied from one broodstock to another. Generally, however, an individual broodstock will respond the same, repeatedly, even though that group responds differently than other broodstock groups.

Procedure

1. Wait at least 10 days after reaching the spawning regime. Fish may begin to spawn on their own. If fish spawn on their own proceed to step 3. If they do not, proceed to step 2 (Figure 5).
2. Gently raise/lower the water temperature through the range from 22°C to 28°C over a period of six days(1.0°C/day:.042°C/hour) until pawning occurs (Figure 5). Drumming, darkening of males from mid-line up, and presence of red, swollen vents in females are indicative of spawning. When spawning occurs hold photothermal conditions and proceed to step 3.
3. Allow fish to spawn for three to four days if they spawn every day. Stop fish from spawning after 3 to 4 days by rapidly (.08 to .20°C/hour) lowering water temperature to 21°C. The rate will change depending on the temperature fish actually spawn.
4. When hatchery requirements change and spawning for a particular broodstock is to be terminated, use a 30-day termination schedule to get back to winter plateau (9HL:17°C).

Verification

The most common verification procedure used to test the efficacy of the maturation regime is ovarian biopsy. A plastic tube connected to a syringe (Figure 6) is inserted in the oviduct and a small core sample of the female gonad is withdrawn and examined under a microscope. The 5-ml hypodermic syringe with stainless steel cannula, 16 cm in length, is attached to 10 to 12 cm of 1-mm diameter polyethylene tubing. A 1-mm diameter hole is drilled into the barrel of the syringe. Finger valving of the hole controls internal pressure. The ripening oocytes from biopsies at each checkpoint are depicted in Figure 2. The plastic syringe is also useful in determining sex of broodstock.

Other verification procedures are being developed. However, at this time, the ovarian biopsy is still the simplest, most reliable method. This method can certainly be performed by most technicians.

Males are usually more precocious than females, and are also good indicators of maturation. Gentle palpitation of the abdomen in front of the vent will cause ripe males to expel milt (fluids and ripe spermatozoa) from the ureter duct. Sometimes this mixture may be clear (hydrated), sometimes it may be white and thick (anhydrous).

Verification usually requires anesthetizing fish for handling. This procedure can be done on a regular basis during the regime, but should not be done more frequently than 3- to 4-week intervals. Tricane methane sulfate (MS-222) at a dosage of 122 mg/L has been used successfully to anesthetize broodstock for handling. Fish are anesthetized in a suitable container and then placed in a respirator for evaluation procedures.

Control of Photothermal Regimes

Control of environmental cycles can be accomplished with several kinds of electromechanical devices. Reverse cycle room air conditioners with remote thermostats and electric timers are suitable. Such a system can be interfaced with audible and visual alarms. Monitoring requires a technician to change timer tabs, verify performance, record temperature, etc. Electric timers fail.

A very practical, reliable system for controlling cycles was developed at the Florida Bureau of Marine Research – a Computerized Environmental Monitoring Control System (CEMACS) monitors and controls parameters in red drum maturation and spawning tanks (regimes, water quality, water temperature, sunrise, sunset, photoperiod, etc.) Data are stored in digital format. Control functions are checked by monitoring systems to assure programmed functions are executed. Malfunctions are re-executed. If repeated failure occurs, an alarm system alerts staff by phone.

The prototype developed in Florida consists of an Apple II Plus microcomputer with two disk drives, two 16-channel analog-to-digital/digital-to-analog convertors, a clock board, a modem, a BSR remote module controller, printer and expansion chassis (Figure 7). The microcomputer is the core of the operations control. Operation programs are stored in drive 1; data are stored in drive 2. Continuous assessment of culture conditions is available via monitor. CEMACS automatically recovers from power failures by running specified programs. Power interruptions seldom occur with a back-up generator. How-

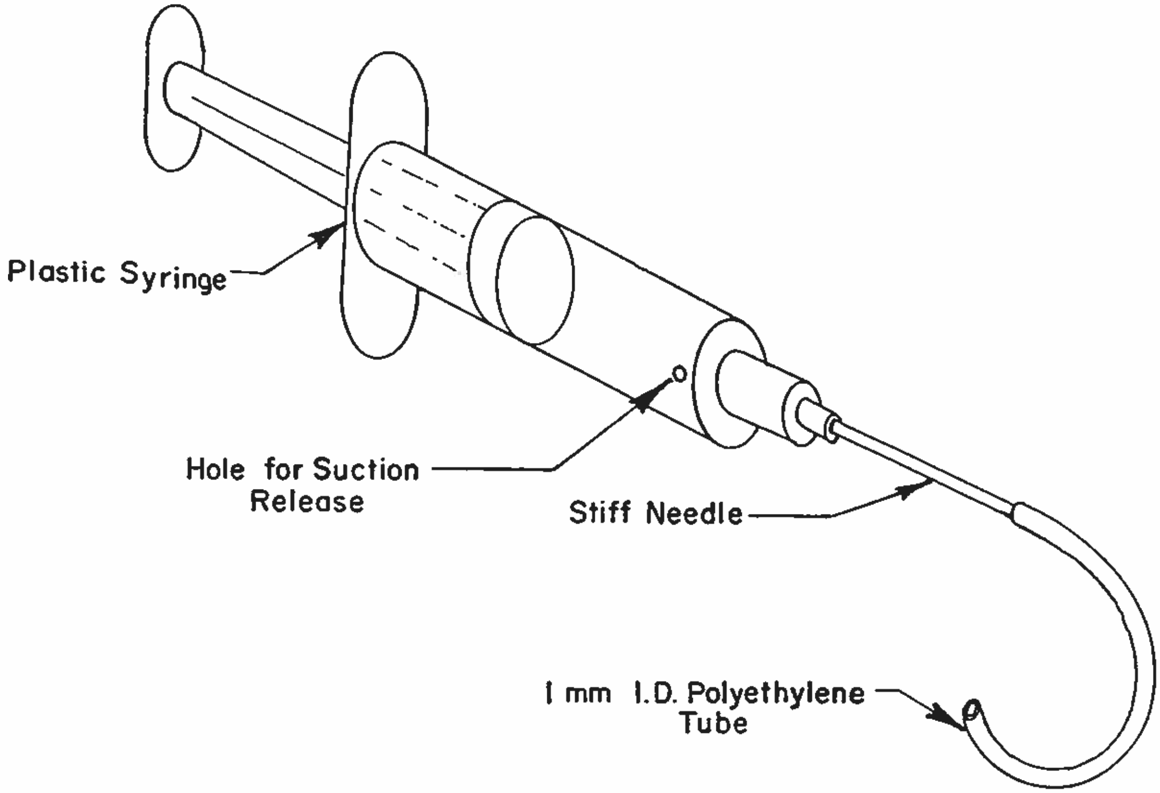


Figure 6. Plastic 5ml syringe modified for extraction of oocytes from ovaries of female red drum. (Modified from Hoff, 1976).

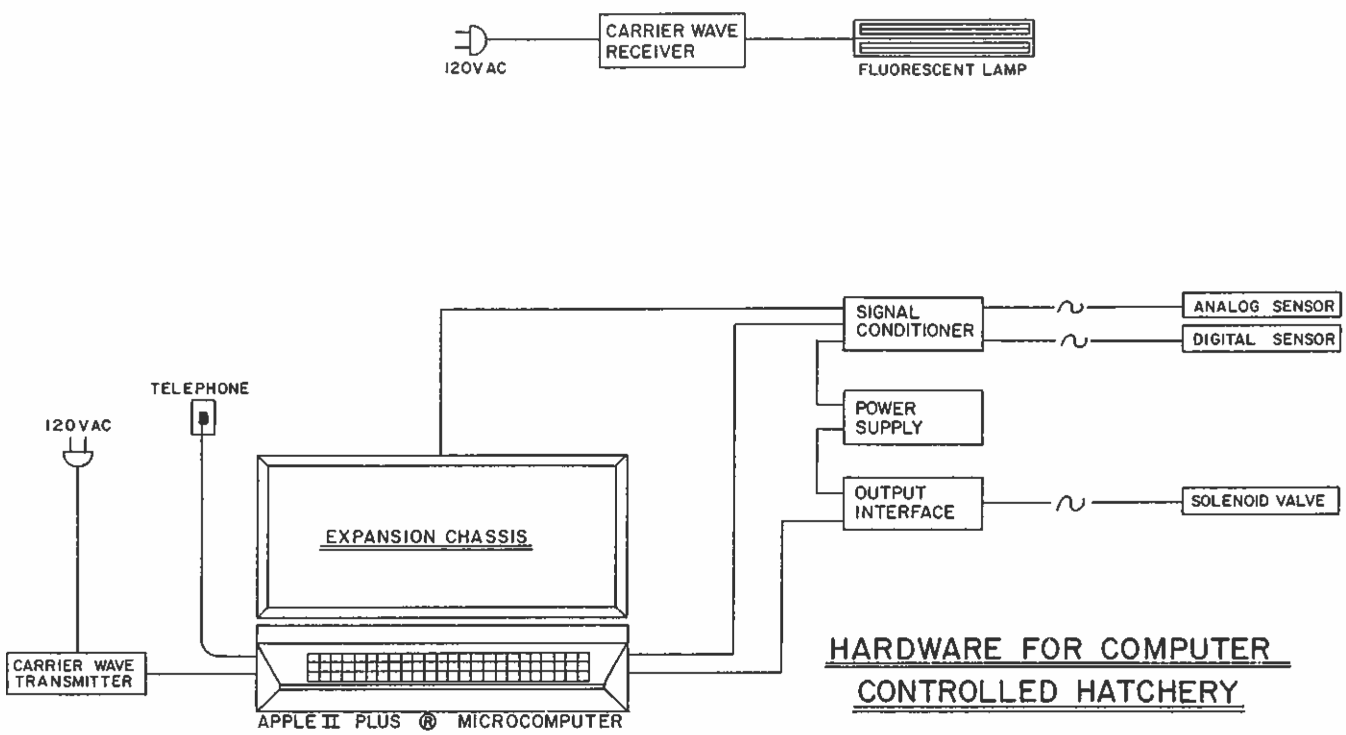


Figure 7. Hardware for computer controlled red drum hatchery. Sensing and controlling systems (analog sensor, digital sensor, solenoid valve, carrier wave transmitter and receiver). Translation and conversion systems (signal conditioner, power supply, output interface). Data and program storage systems (Apple II Plus microcomputer).

ever, when they do, the real-time clock (auxillary battery powered) allows re-sequencing and start up of control software. The computer opens and closes solenoid valves that regulate chilled brine or hot water from the HVAC system. Teflon heat exchangers transfer heat/cool from the HVAC to fish holding system seawater. Temperature sensors feed back to the microprocessor continuously comparing water temperature to pre-programmed standards. Temperature is maintained within an acceptable range (better than controlled switches). These switches are activated by high frequency signals sent via building electrical wiring. Fluorescent lights, switched on and off by this system provide photoperiod. Sunrise and sunset are controlled by gradual lighting of incandescent lights 15 minutes before photoperiod begins and 15 minutes after photoperiod ends, respectively.

Summary

The methods described herein have taken 10 years to develop and represent the work of many scientists and aquaculturists in Texas, Alabama, South Carolina and Florida. Some of this work is summarized in Table 1. The intent of this section is to provide a working method for maturation and spawning of red drum using photothermal control. This technique is new and must be continually refined. If you use it, help improve it. This can only be done through communication of results in practical formats such as this.

Acknowledgements

I am very appreciative for the assistance and support of the following people without whose help this and other publications would not be possible: Drs. Anthony Macieroski and Anne Henderson-Arzapalo, Mr. Eugene McCarty, Mr. Robert Colura, Texas Parks and Wildlife Department, Austin, Tex., Drs. Karen Steidinger and William Falls, Mr. William Halstead, Mrs. Ruth Reese and Ms. Marie Wooten, and Ms. Patricia Boyett, Florida Department of Natural Resources, Bureau of Marine Research, St. Petersburg, Fla., and Mr. Frank DeBerardinis, F. DB Drafting Service, St. Petersburg, Fla.

Relevant Literature

Arnold, C.R., J.D. Williams, A. Johnson, W.H. Bailey and J.L. Lasswell. 1977. Laboratory spawning and larval rearing of red drum and southern flounder. Presented at 31st Annual Conference, **Southeast Association of Game and Fish Commission.** 10 pp.

Table 1. Summarized methods for photothermal induction of maturation and spawning of red drum at the John Wilson Marine Fish Hatchery, Perry R. Bass Fisheries Research Station and Florida Bureau of Marine Research.

	JWMFH	Perry R. Bass	Florida Bureau Marine Research
Tank size (metric tons)	13	10-20	5-20
Filtration system	RBC, FB, SH, S, UV	PT, TF	S, CB, DE
Fish/Tank	4	4-6	4-6
Sex ratio (M:F)	2:2	2:2, 2:1, 1:2	3:3, 3:2, 2:2
Mean fish size (Kg)	13	11	11
Regime duration (days)	150	120	120
Max. regime temperature (°C)	30.0	30.0	30.0
Min. regime temperature (°C)	15.0	18.0	18.0
Max. regime photoperiod (HL)	14.0	14.0	16.0
Min. regime photoperiod (HL)	9.0	10.0	9.0
Spawning period (days)	22	30	100
Spawning temperature (°C)	24	26	25
Spawning photoperiod (HL)	11	10	9
Mean no. eggs/spawn period (millions)	20	20	100
Biopsy interval (weeks)	4	12	2-4
Feeding regime (% bw/day)	1.07	3.00	2.85

CB-Conventional biofilter
DE-Diatomaceous earth
FB-Fluidized bed
PT-Packed tower
S-Sand
SH-Shell
TF-Trickle filter
RBC-Rotating biological contactor
UV-Ultraviolet filtration

Gardner, M.B. and J. McCauley. 1982. Microcomputers in aquaculture: valuable management tools. **Aquacult. Mag.** Vol. 8, No. 6, Sept.-Oct.:22-26.

Harvey, B.J. and W.S. Hoar. 1979. Theory and practice of induced breeding in fish. International Development Research Centre Publication. Ottawa, Canada.

Haydock, I. 1971. Gonad maturation and hormone-induced spawning of the Gulf croaker, *Bairdiella icistia*. **Fishery Bulletin** 69(1):157-181.

Hoar, W.S., D.J. Randall and E.M. Donaldson. 1983. **Fish Physiology. Vol. IX Reproduction, Part A, Endocrine Tissues and Hormones**. Academic Press, Inc., New York, New York. 483 pp.

Hoar, W.S., D.J. Randall and E.M. Donaldson. 1983. **Fish Physiology. Vol. IX Reproduction, Part B, Behavior and Fertility Control**. Academic Press, Inc., New York, New York. 477 pp.

Hoff, F.J., Jr. 1970. Artificial spawning of black sea bass, *Centropristis striatus melanus* Ginsburg, aided by chorionic gonadotrophic hormones. **Fla. Depart. Nat. Res. Mar. Res. Lab. Spec. Sci. Rep.** No. 25. 17 pp.

Hoff, F.H. Jr., C. Rowell and T.R. Pulver. 1973. Artifically induced spawning of the Florida pompano under controlled conditions. **Proc. Wor. Mar. Soc.** 3:53-64.

Hoff, F.H. Jr., J.A. Mountain, T.A. Frakes, T.R. Pulver and R.B. Futch. 1976. Controlled conditioning and spawning of the Florida pompano, *Trachinotus carolinus*, and southern sea bass, *Centropristis melana*. **Florida Department Natural Resources Marine Research Laboratory Federal Aid Progress Report** (completion), 2-113-R.

Kuo, C.M., C.E. Nash and N.H. Shehadeh. 1974. The effects of temperature and photoperiod on ovarian development in captive grey mullet, *Mugil cephalus Linne*. **Aquaculture** 3:25-43.

Landless, P. 1981. Fish and (silicone) chips. **Fish Farmer** 3(6):35.

Lasker, R. 1974. Induced maturation and spawning of pelagic marine fish at the Southwest Fisheries Center, La Jolla, **Calif. Proc. Wor. Mar. Soc.** 5:10

Leong, R. 1971. Induced spawning of the northern anchovy, *Engraulis mordax* Girard. **Fishery Bulletin** 69(2):357-360.

Lockwood, A.P.M., N.W. Jenkinson, S.R.L. Bolt, M.E. Dawson and A.C. Jenson. 1982. Microprocessor systems as an aid to estuarine studies in the laboratory. **Estuar. Coast. Shelf Sci.** 15(2):199-206.

McCarty, C.E., J.G. Geiger, L.N. Sturmer, B.A. Gregg and W.P. Rutledge. 1986. Marine finfish culture in Texas: a model for the future. *In*: **Fish Culture in Fisheries Management. American Fisheries Society**. 13 pp.

Plaia, W.C. 1985. A computerized environmental monitoring and control system for use in aquaculture. **Proc., W.M.S., 16th Ann. Mtg.**,1985, Orlando, Fl.

Reese, R.O., D.E. Roberts, Jr. and E. Truby. 1987. Red drum spawning: the 120-day cycle, vitellogenesis and maturation. **Proc. Wor. Aquacult. Soc. 18th Ann. Mtg.** 1987, Guayaquil, Ecuador.

Roberts, D.E., Jr., B.V. Harpster, W.K. Havens and K.R. Halscott. 1976. Facilities and methodology for the culture of the southern sea bass, *Centropristis melana*. **Proc. Wor. Mar. Soc.** 7:163-198.

Roberts, D.E. Jr., B.V. Harpster and G.E. Henderson. 1978a. Conditioning and induced spawning of the red drum (*Sciaenops ocellata*) under varied conditions of photoperiod and temperature. **Proc. Wor. Mar. Soc.** 9:311-332.

Roberts, D.E., Jr. and R.A. Schieder. 1983. Induced sex inversion, maturation, spawning and embryogeny of the protogynous grouper, Myctoperca microlepis. **Proc. Wor. Mar. Soc.** 14:639-649.

Roberts, D.E. Jr. 1985a. Induced maturation and spawning in the common snook, *Centropomus undecimalis*. **Proc. 38th Gulf and Caribbean Fish Inst.**, Nov. 1985. Fort-de-France, Martinique (In press).

________ 1985b. Reproductive maturation in common snook (*Centropomus undecimalis*) in closed recirculating systems: methodology and preliminary results. **Proc. Wor. Mar. Soc., 16th Ann. Mtg.**, 1985, Orlando, FL.

Salomon, C.H. and L.A. Collins. 1972. Hydrographic observations in Tampa Bay and adjacent waters. U.S. Department of Commerce, National Oceanic and Atmospheric Administration, NMFS-DR-90. 176 pp.

Schlieder, R. 1984. Environmentally controlled seawater systems for maintaining large marine finfish. **Prog. Fish. Cult.** 46(4):285.

Stamp, N.H.E. 1978. Computer technology and farm management economics shrimp farming. pp. 383-392. Avault, J.W. ed. *In*: **Proceedings of the Ninth Annual Meeting of the World Mariculture Society**.

Design and Operation of a Photoperiod/ Temperature Spawning System for Red Drum

Cecil E. McCarty
Texas Parks and Wildlife Department
4200 Smith School Road
Austin, Texas 78744

Although the basic biology of red drum (*Sciaenops ocellatus*) reproduction was known 10 years ago, culturists were unable to economically produce enough larvae at any one time to stock a commercial production facility.

Several important discoveries changed that. Arnold (1977) demonstrated the feasibility of using photoperiod and temperature variations to induce maturation in large numbers of broodstock. Roberts (1977) showed that captive fish would respond to an artificially compressed seasonal cycle. Finally, Colura (pers. comm.) combined the compressed photoperiod/temperature method of maturing red drum broodstock with the fertilized pond method of growing larvae to fingerling size at the Perry R. Bass Marine Fisheries Research Station.

These breakthroughs have paved the way for large-scale pond production of red drum. The John Wilson Marine Fish Hatchery (JWMFH), the first saltwater production fish hatchery on the Gulf coast, has refined these techniques and has made large-scale red drum production a reality.

Design of Spawning Facility

JWMFH has three spawning rooms, each 108 m² in area (Figure 1). Each room has four 13,000-liter, circular fiberglass spawning tanks. Each pair of spawning tanks shares a bio-disc (biological filtration) system consisting of a two-stage, rotating biological reactor plate (56 m² surface area) for bacterially-mediated oxidation of ammonia to nitrate (see Appendix, Section VI); a foam-fractionator, for removal of dissolved organic matter; and a buffering unit of crushed oyster shells to control pH.

Operation of Spawning Facility

The volume of each tank is cycled through the biodisc system every three hours. In addition, every 48 hours, tank water is pumped at 50 gpm for 12 hours through a rapid-sand filter (to remove large particulate matter) and an ultraviolet sterilization unit.

Four 8- to 18-kg brood fish – two males and two females – are placed in each spawning tank. Three times a week, brood fish are fed a diet of 50 percent shrimp, 25 percent beef liver at a rate of 2.5 percent of their body weight.

Annual variations in photoperiod and temperature are simulated over a modified 150-day maturation cycle (Figure 2) (Arnold *et al.*, 1977; Roberts *et al.*, 1978a). Spawning is induced at a water temperature of about 23° to 25°C, a photoperiod of 11 hours light/ 13 hours dark, and salinities in the range of 32 to 34 ppt.

The fertilized planktonic eggs flow into a collection box from which they are recovered, counted by volumetric methods (Bonn *et al.*, 1976) and transferred to incubators.

Using these techniques, red drum broodstock have been successfully induced to spawn at JWMFH since 1983 (McCarty *et al.*, 1986). In that period, 35 females have produced more than 152 million eggs. (Females average 265,000 eggs per night for 26 days.)

Eggs are incubated for 24 hours. Sixty-four percent of these eggs hatch and survive at least 36 hours.

These methods provide large quantities of healthy red drum larvae that have been stocked in rearing ponds at JWMFH and at the Freeport, Tx., satellite facility.

Literature Cited

Arnold, C.R., W.H. Bailey, T.D. Williams, A. Johnson and J.L. Lasswell. 1977. Laboratory spawning and larval rearing of red drum and southern flounder. **Proceedings Annual Conference of the Southeastern Association of Fish and Wildlife Agencies**, 31:347-440.

Bonn, E.W., W.M. Bailey, J.D. Bayless, K.E. Eriksson and R.E.Stevens (eds.). 1976. Guidelines for striped bass culture. Striped Bass Committee of the Southern Division, **American Fisheries Society**, Bethesda, Maryland.

McCarty, C.E., J.G. Geiger, L.N. Sturmer, B.A. Gregg and W.P. Rutledge. 1986. Marine finfish culture in Texas: A model for the future. *In*: R.H. Stroud (ed.), **The Role of Fish Culture in Fishery Management. An international symposium on the use of cultured fish in fishery management**. American Fisheries Society.

Roberts, D.E., B.V. Harpster and G.E. Henderson. 1978a. Conditioning and induced spawning of the red drum (*Sciaenops ocellata*) under varied conditons of photoperiod and temperature. **Proceedings, World Mariculture Society**, 9: 311-332.

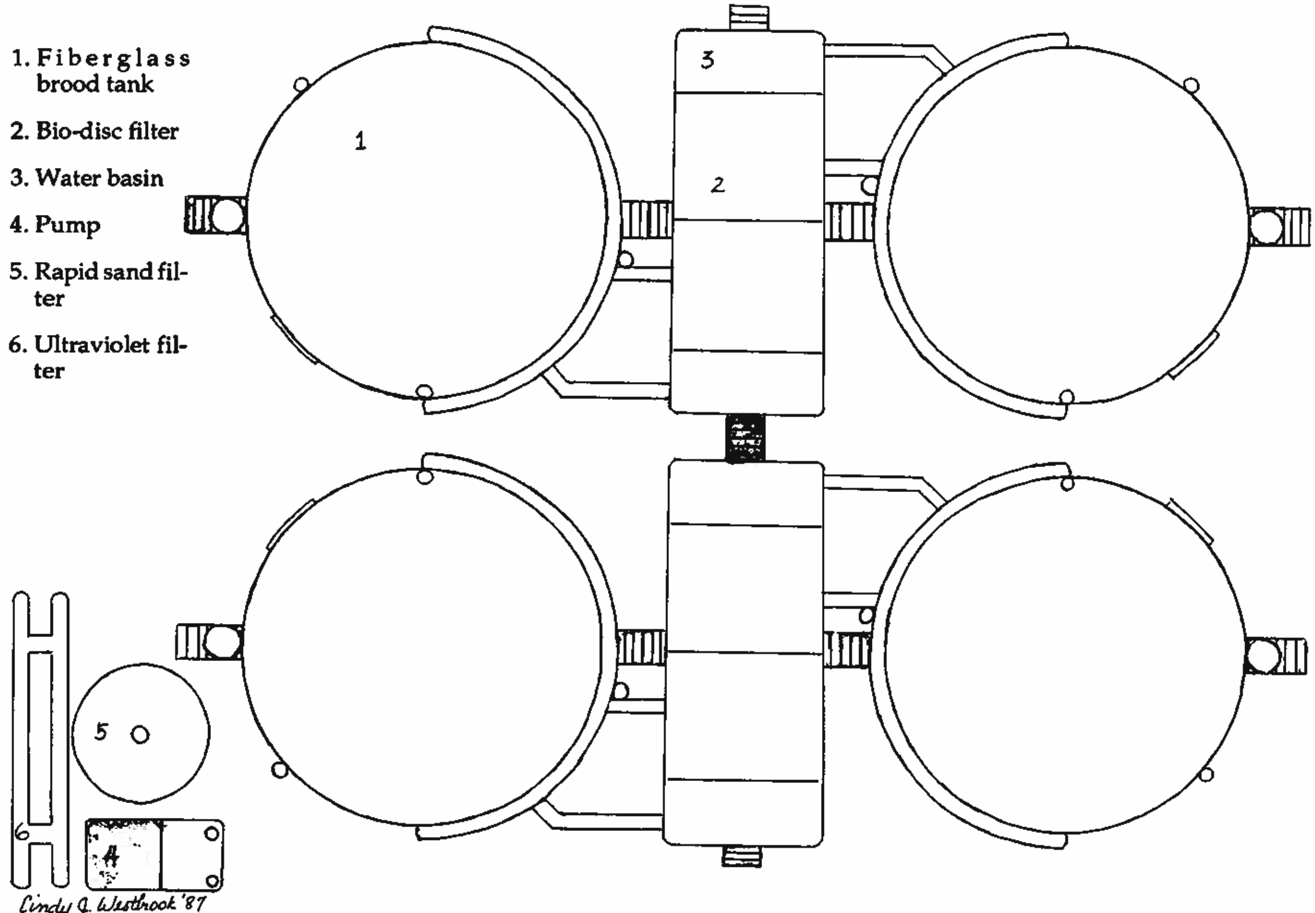


Figure 1. Typical red drum spawning room, John Wilson Marine Fish Hatchery.

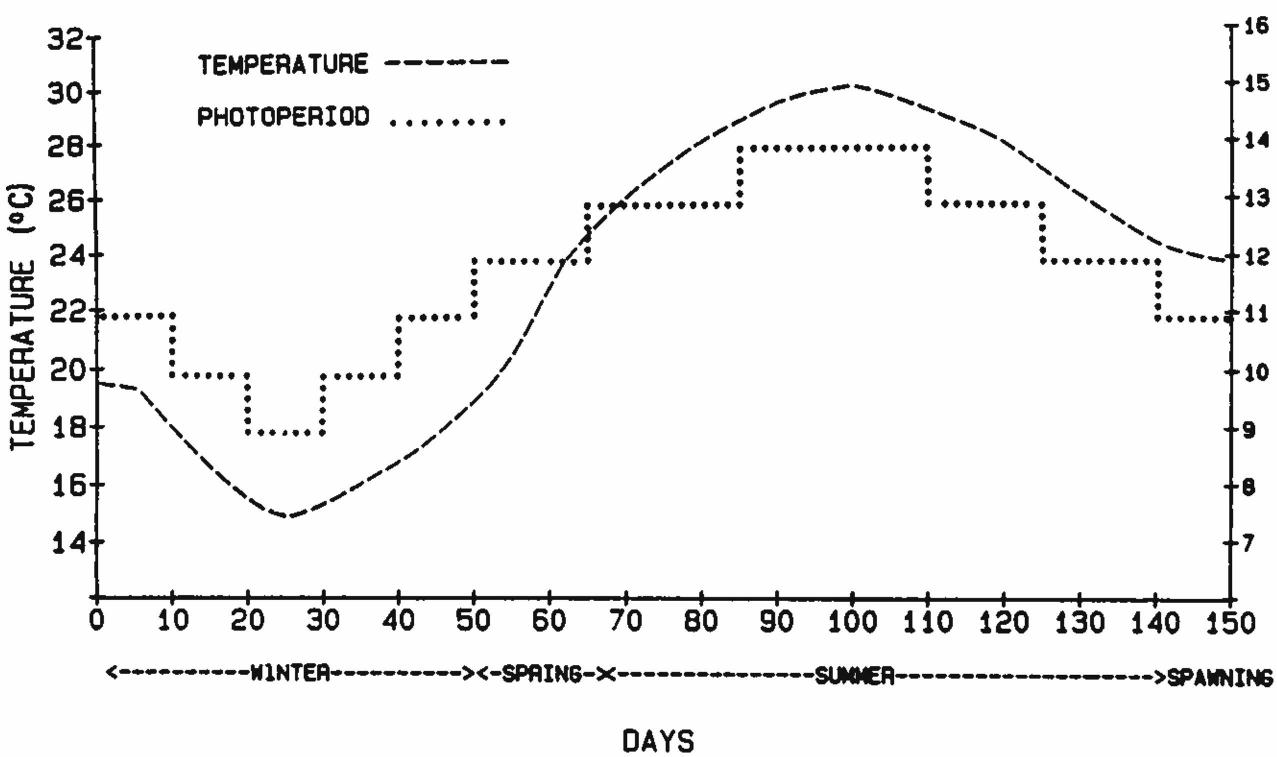


Figure 2. Photoperiod and temperature regime used to induce spawning of red drum.

Growth and Development of Red Drum Eggs and Larvae

G. Joan Holt
The University of Texas at Austin
Marine Science Institute
Port Aransas, Texas 78373

Larval rearing of marine fishes is an area of aquaculture that is in its infancy in the United States. To ensure success in this area, it is necessary to define the biological and physio-chemical requirements of the species of interest to the aquaculturists. For most larval fish, basic biology, including the effects of environmental factors on survival and growth, is poorly understood. Over the past few years, I have conducted numerous studies to examine the effects of physical and biological factors on survival and growth of larval and juvenile red drum (*Sciaenops ocellatus*). This research was carried out in order to enhance the success for growing larvae in culture, and to increase our knowledge of environmental tolerances and habitat needs of young red drum in their natural environment. Studies included 1) growth and developmental stage monitoring, 2) determining optimum temperature-salinity conditions, 3) evaluating effects of ammonia and nitrite and 4) feeding and diet development.

Research on red drum identified temperature as the most important factor affecting growth and development. Other factors found to enhance or detract from optimal growth in intensive culture include water quality, crowding, food density and nutritional quality of food.

Studies concerning these areas will be briefly described here.

Applications of these results to intensive culture of red drum are presented in Fingerling Production Technology (Section III-1).

Growth and Development

Red drum eggs, 0.9 to 1.0 mm in diameter, are spawned at night and develop into fully formed embryos ready to hatch in 18 to 25 hours at 24° to 28° C (Holt *et al.*, 1985). Egg developmental stages are similar for many warm water marine fishes with early cell divisions essentially complete by one and a half hours and an early embryo formed by 12 hours (Fig. 1). In an earlier publication we described development from hatching to two weeks (Holt *et al.*, 1981a). Figure 2 depicts a newly hatched yolk-sac larva, a three-day old larva nearly ready to begin feeding, and two older larvae. The larvae are transparent with no scales, are very sensitive to handling and have little tolerance of poor water quality at these stages.

Fin and scale development are correlated with age and size. Red drum larvae hatch without fully formed fins. Fin development initiates with the caudal (tail) fin, followed by the anal, dorsals and then pelvic fins (Holt and Arnold, 1986). The pectorals, first present as buds in the newly hatched larvae, are the last fins to develop a full complement of rays (Table 1 and Figure 3).

Scale development begins as a thickening of the skin on 9 to 9.5 mm SL larvae followed by the appearance of small scales along the posterior surface just in front of the tail (caudal peduncle). Scale development proceeds along the lateral line extending anteriorly on the lateral surface of the body finally covering both sides. By the time a fish reaches 25 mm SL, the complete adult pattern of body scales is formed.

Even under constant environmental conditions in rearing tanks, differential growth rates occur. Fish of the same age but of different sizes will be in different stages of development because developmental characters are more closely correlated with size than age. For example two-week old fish from the same spawn may vary in size from 4.5 to 10 mm, with the largest fish having completely developed fins (Figure 3) and

Table 1 Age (days) and smallest size (mm SL) when fins complete development for greenhouse reared red drum (composite of four different populations. Spring 1982 and 1983).

Fin	Age	Size
Caudal (primary)	10	5.0
Anal (II,8)	12	6.2
Dorsal (X, I, 23-25)	14	6.6
Pelvics (I, 5)	14	9.0 *
Pectorals (15-17)	16	10.0
Caudal (secondary)	21	17.5 **

*skin thickening
**scaled on lateral surface of body

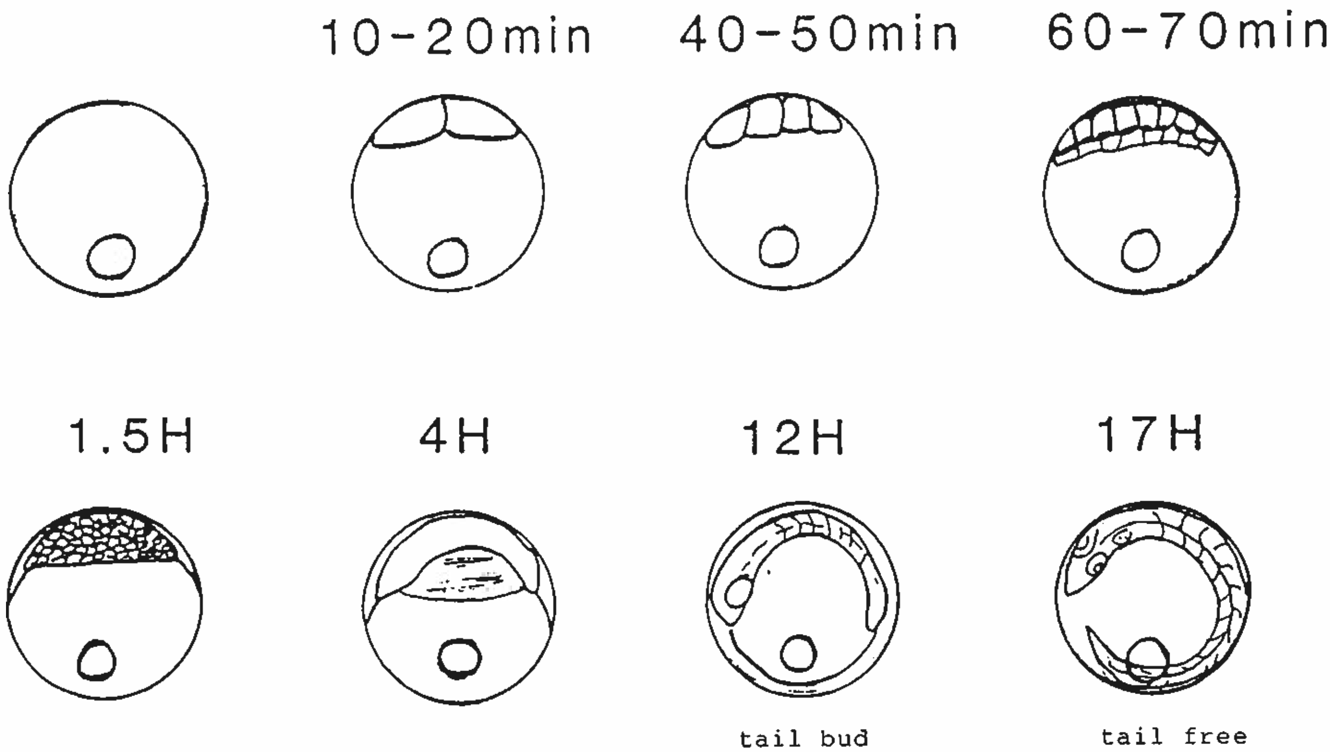


Figure 1. Timing of Sciaenid egg stages at 25°C.

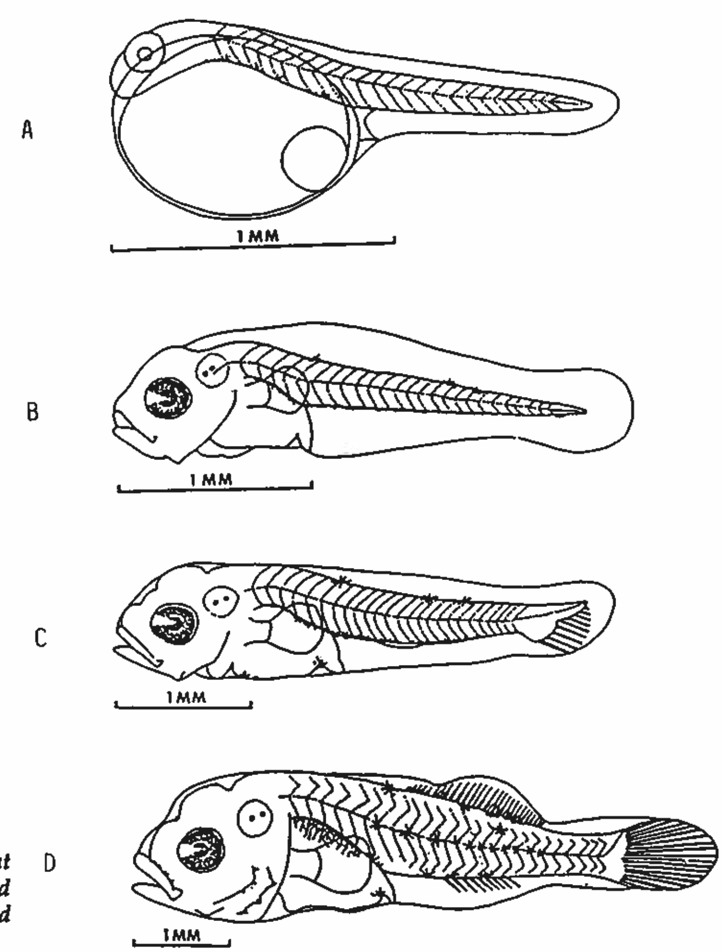


Figure 2. Lab-spawned larval red drum hatched and raised at 25°C. (A) 1 hour old yolk-sac larva (1.7 mm SL), (B) 3-day-old first feeding larva (2.5 mm SL), (C) 10 days old (4.2 mm SL) and (D) 2 weeks old (5.1 mm SL).

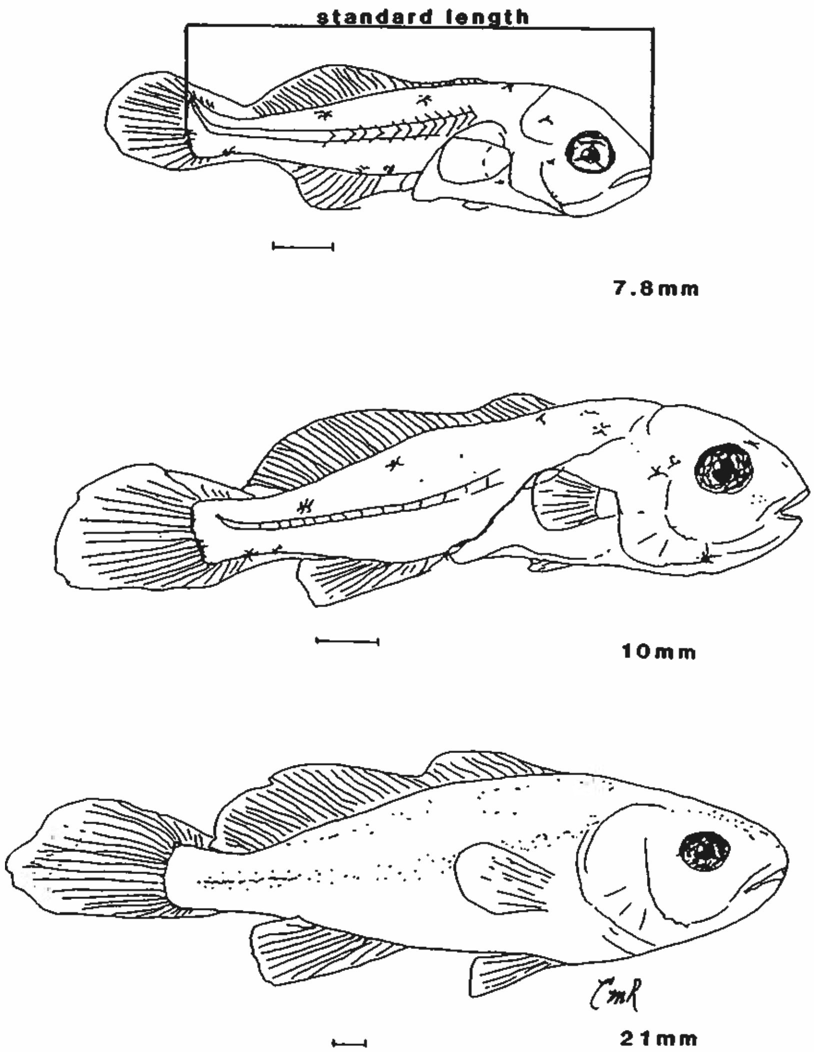


Figure 3. Developmental stages of red drum. Bar = 1mm.

the smallest with only slight development of the caudal fin (Figure 2).

Variation in size among siblings increases with age. Possible causes of this variability may be 1) genetic differences among the individuals in growth potential, 2) differential success at first feeding that results in some fish getting a head start on their siblings and therefore increasing in growth at a faster rate, or 3) competitive interactions among individuals in the rearing tank.

Temperature and Salinity

Optimal conditions for red drum eggs and larvae were found to be at temperatures of 25° to 30°C and salinities of 25 to 35 ppt (Holt *et al.*, 1981b). These ranges are very near the minimum and maximum values at which wild red drum eggs have been collected in south Texas (Holt *et al.*, 1988). Eggs hatch over a broad range of salinities (5 to 50 ppt) but larvae successfully develop to first feeding only at 10 to 40 ppt, with best survival at 25 to 30 ppt. Eggs spawned in normal seawater salinity (30 to 35 ppt) are positively buoyant, but if transferred to lower salinity water they begin to sink. At 20 ppt or lower, eggs quickly sink to the bottom and rapidly deplete the available oxygen unless well aerated. In natural populations or in culture ponds, high mortality of eggs developing on the bottom would be expected. Although salinity is important for early development through the yolk-sac stage, red drum thereafter acclimate to a wide range of salinities. For example, 6 mm SL larvae survive well at 5 ppt, and 13 mm SL larvae survive 96 hrs after direct transfer from 10 ppt to freshwater (Crocker *et al.*, 1981). Juvenile red drum, greater than 20 mm SL, have high survival in freshwater but growth rate is reduced. After 30 days red drum growth rate was significantly less in freshwater reared red drum than those reared in seawater (Croaker *et al.*, 1981).

Temperature is critical for larval survival and rates of growth and development. Time for egg hatching is temperature dependent as is duration of the yolk-sac stage which ranges from 40 hours at 30°C to 85 hours at 20°C (Holt *et al.*, 1981a). Optimum growing temperature is 27°C and in water temperatures of 20°C or lower, larval red drum become inactive, cease feeding, and eventually die. Survival rates are greatly increased when larvae are maintained at 25°C through first feeding (approximately 3 to 4 days) before being exposed to lower temperature. Growth rates are positively correlated with temperature; 14 day growth at 20°C was 0.1 mm per day, at 25°C 0.2 mm per day, and at 30°C 0.3 mm per day (Holt and Arnold, 1982).

Larval growth rates increase rapidly with age and reach 1 mm per day under ideal conditions of temperature, feeding and fish density. Development from a tiny newly hatched larva to a fully scaled juvenile therefore can occur in only a few weeks. That this rate depends to a large extent on temperature can be demonstrated by two examples (Figure 4) from red drum grown in intensive culture (20 ft. diameter tanks in a greenhouse). Larval fish grown at an average temperature of 28.5°C reached the juvenile stage in only three weeks, while those raised at an average temperature of 22.7°C took six weeks to reach the same size.

Ammonia and Nitrite

Red drum eggs are relatively insensitive to high ammonia levels, but concentrations of approximately 3.5 mg/1 (ppm) caused 100 percent mortality in newly

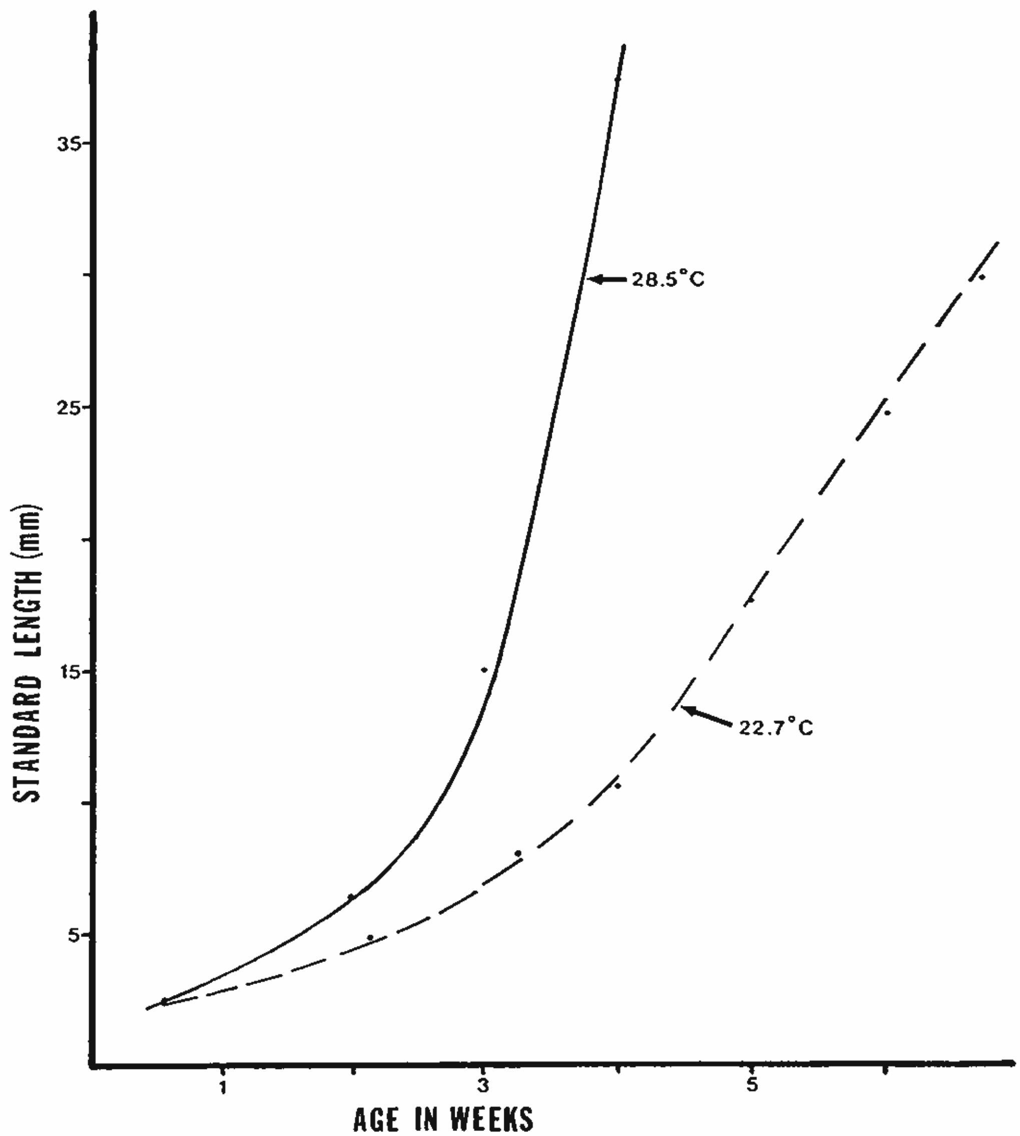


Figure 4. Red drum grown in 20-foot diameter tanks in a greenhouse under two temperature regimes. Solid line—temperature ranged from 27°C to 30°C; dashed line—temperature varied from 20°C to 24°C during the grow-out period.

hatched larvae after 10 days (Holt and Arnold, 1983). The toxicity of ammonia is directly related to the amount of un-ionized ammonia (NH3) present. Unionized ammonia increases with increased pH and temperature, thus toxic levels may be reduced by maintaining these parameters at the lowest acceptable levels. Nitrite toxicity is seldom a problem for red drum raised in a saltwater system. We found toxic levels for red drum larvae (100 mg/1 NO_2-N) were 100 times higher than the highest levels in our rearing tanks. Since high levels of dissolved chloride and calcium in saltwater decrease or inhibit nitrite toxicity, it could become a concern if red drum are raised in freshwater.

Feeding and Diet Development

Red drum require small sized food particles (50-100 um) at first feeding. Laboratory cultured rotifers (*Brachionus plicatalis*) described in Raising Food Organisms Intensive Larval Culture (Section III-24) are adequate for the first week of feeding. The young larvae eat only a few rotifers at a time throughout the day so rotifers must be available at all times. Also, since larvae do not have fins and are very small at this stage, they can not swim far. They do not have a large search volume so the density of rotifers needs to be high. I have found that maintaining from 3,000 to 5,000 rotifers per liter provides the best nutrition for red drum. If greater numbers of rotifers are present the larvae will not eat any more. Excess rotifers will remain in the tank for several days before they die, becoming progressively less nutritious food as they starve.

When red drum larvae are 10 days old, they require larger food and readily eat brine shrimp nauplii fed at a rate of 500 to 1,000 per liter. As larval red drum grow they need increasingly larger packages of food for optimum growth. After a few days feeding on brine shrimp, larvae may be weaned to chopped shrimp or commercially prepared diets. During this transition period mortality may be high as some fish do not rapidly accept these diets and cannibalism becomes prevalent. Recently I have been trying to develop a suitable diet to replace the rotifer/brine shrimp feeding system currently used.

Copepods are important natural food for larval red drum (Steen and Laroche, 1983) and several species of copepods including *Tisbe* have recently been utilized with great success in mariculture in Japan and Israel. The suitability of *Tisbe carolinensis* as food for larval red drum was evaluated and found to be of limited usefulness because small larvae would not feed on this copepod and larger larvae rejected *Tisbe* when other foods were available. The critical copepod density for red drum feeding was 1,000 to 2,000 per liter. This copepod could be valuable food for a limited period but since large batch culture of *Tisbe* requires as much effort as our current live feeding system it is not an attractive alternative.

Examination of commercially available diets are giving more encouraging results. Red drum larvae will readily accept non-living, non-moving food particles if they are in the water column. But so far, growth rates on commercially available larval diets are poor compared to those on live food. A combination of live rotifers and prepared diets for the first few days and then a switch to prepared diets alone looks like a promising compromise.

Conclusions

1. Egg hatching and larval rearing through the yolk-sac stage should be in 25° to 30°C and 25 to 35 ppt salinity seawater.
2. Gradual changes in salinity (approximately 5 ppt per day) can begin after first feeding and fish can be moved into freshwater after they are fully scaled (25 mm SL or 1 inch long).
3. The fastest growth occurs at high temperatures (30°C) but water quality is more difficult to maintain; 27°C is a good compromise.
4. Ammonia levels should be maintained below 1 ppm, especially at high temperature.
5. In seawater, nitrite should be below 10 ppm.
6. Live food diets (rotifers and brine shrimp) give the best survival and growth for larval red drum.

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Literature Cited

Arnold, C.R. 1984. Maturation and spawning of marine finfish. *In* Carl J. Sindermann (ed), **Proceedings of the seventh U.S.-Japan meeting on aquaculture, marine finfish culture,** Tokyo Japan, October 3-4, 1978, p. 25-27. NOAA Tech. Rep. NMFS 10.

Crocker, P.A., C.R. Arnold, J.A. DeBoer and J. Holt. 1981. Preliminary evaluation of survival and growth of juvenile red drum *Sciaenops ocellata* in fresh and salt water. **J. of World Maricult. Soc.** 12(1):122-134.

Holt, J., Allyn G. Johnson, C.R. Arnold, W.A. Fable, Jr., and T.D. Williams. 1981a. Description of eggs and larvae of laboratory reared red drum *Sciaenops ocellata*. **Copeia** 4:751-756.

Holt, J., R. Godbout and C.R. Arnold. 1981b. Effects of temperature and salinity on egg hatching and larvae survival of red drum *Sciaenops ocellata*. **Fishery Bulletin,** 79(3):569-573.

Holt, J., and C.R. Arnold. 1982. Effects of temperature on early development of red drum *Sciaenops ocellata*. Fifth Annual Larval Fish Conference, L.S.U., Baton Rouge, Louisiana.

Holt, G. J. and C.R. Arnold. 1983. Effects of ammonia and nitrite on growth and survival of red drum eggs and larvae. **Trans. Am. Fish. Soc.** 112:314-318.

Holt, G.J., S.A. Holt and C.R. Arnold. 1985. Diel periodicity of spawning in sciaenids. **Marine Ecology** - Progress Series, 27:1-7.

Holt, G.J. and C.R. Arnold. 1986. An overview of factors controlling growth and development in lab-cultured red drum. Presented at Second International Conference on Warm Water Aquaculture-Finfish, February 5-8.

Holt, S.A., G.J. Holt and L. Young-Abel. 1988. A procedure for identifying sciaenid eggs. **Contributions in Marine Science** Supplement to Vol. 30:99-107..

Steen, J.P., Jr. and J.L.Laroche. 1983. The food of red drum (*Sciaenops ocellatus*) larvae and early juveniles taken from Mississippi sound and the northern Gulf of Mexico. Shabica, S.V., N.B. Cofer &andE.W. Cake Jr., eds. 1983. **Proceedings of the northern Gulf of Mexico estuaries and barrier islands research conference.** 13-14 June 1983, Biloxi, Miss. U.S. Dep. Int. Natl. Park Serv. SE Reg. Off., Atlanta, Ga. 191 pp.

Red Drum Egg and Larval Incubation

Anne Henderson-Arzapalo
National Fisheries Research Center
Leetown Fish Culture Research Laboratory
Box 700
Kearneysville, West Virginia 25430

Description and Environmental Requirements

Red drum spawn planktonic, spherical eggs approximately 0.9 to 1.0 mm in diameter. Eggs are clear with a single, gold-colored oil droplet. Embryonic and larval development are described by Johnson (1978).

Hatching normally occurs in about 24 hours at 25° C. The alimentary tract is well-formed approximately 48 hours after hatching. Fry are approximately 1.74 mm in standard length at this stage.

Optimum salinity for egg incubation is between 28 and 35 ppt. At this salinity eggs are bouyant. At lower salinities, eggs sink; at higher salinities, they raft together on the surface. Gentle aeration is used to circulate eggs and water.

Temperature should be maintained between 22° to 30° C; ammonia concentrations, less than 0.5 mg/L (Holt and Arnold, 1983); and dissolved oxygen above 3.0 mg/L during incubation and hatching.

Egg Number Estimation

Volumetric methods are most commonly used to estimate the number of marine fish eggs in a sample (Bonn *et al.*, 1976; Bayless, 1972). Eggs are concentrated in a small known volume of water; a subsample is withdrawn and the eggs counted; then the number of eggs/sample is multiplied by the total volume to give the total number of eggs.

Total volume x # eggs/volume of subsample = # eggs

Once water-hardening of the eggs occurs, they can be handled with a fine mesh soft net. However, handling and transfer should be done in water as much as possible to reduce stress and mechanical damage to the eggs.

Eggs are usually concentrated in 500/ or 1000/mL graduated cylinders at The University of Texas Marine Science Institute, as well as at the Gulf Coast Conservation Association/Central Power and Light/Texas Parks and Wildlife Department (TPWD) Marine Development Center (MDC). As eggs float to the surface, the separation between eggs and water is clearly visible. The total volume of eggs [in milliliters (mL)] is observed and multiplied by 998 eggs/mL to estimate the total number.

At the TPWD Perry R. Bass Marine Fisheries Research Station (MFRS), eggs are counted somewhat differently. The eggs are first concentrated into a known volume, usually 20 liters. Eggs are then gently agitated with an airstone and the water stirred to ensure homogeneity of samples. Ten samples are removed with a 5-mL Hensen-Stemple pipette. Each sample is placed in a plastic petri dish and the eggs (or fry) counted. The mean number of eggs/mL is calculated, then multiplied by the volume of water.

Embryo Incuation

After estimating egg abundance, the spawn is transferred to an incubator. Eggs should be acclimated for several hours if there are large differences in temperature or salinity. Here, again, handling of eggs should be done in water as much as possible to reduce stress.

Incubators may be any size or configuration. At the TPWD, MDC and MFRS facilities, 1890-L cone-bottomed fiberglass tanks maintain up to 1 million red drum eggs per tank when water is exchanged (new water is flowed through). The cone shape of the tank allows dead material to settle to the bottom where it can be removed by siphoning. Gentle aeration (provided by airstones or airtubing) circulates eggs and water. Vigorous aeration can damage eggs and fry.

Eggs usually can be maintained without water exchange until they hatch. As hatching begins, however, ammonia concentrations increase and dissolved oxygen decreases. At TPWD hatcheries, a maximum water exchange rate of 1.0 L/min is usually adequate to maintain acceptable water quality.

The incubator drain is a PVC standpipe with most of the surface replaced by fine-meshed screen, such as saran or nitex (0.3- to 0.5-mm mesh). The large surface area of the screen coupled with low water exchange rates prevent fry from being trapped against the screen.

Dead material should be removed periodically to prevent water quality deterioration and fungal infection of eggs. Aeration and water exchange can be stopped for a few minutes, allowing most dead material to settle to the bottom where it is removed by siphoning. Water exchange and aeration are then resumed.

Fry are maintained in incubators approximately 48 hours after hatching, or until the yolk sac is absorbed and mouth parts and alimentary tract development is completed. Incubators are slowly drained and fry are concentrated in a smaller tank. At the MFRS, an 80-L polyethylene tank with a 15-cm diameter saran or nitex drainpipe is used as a collecting tank. Gentle aeration circulates fry and prevents impingement on the drain screen. Fry numbers are estimated by volumetric procedures similar to those described for eggs.

Red drum fry are fragile and should not be netted. Additionally, adequate water quality should be maintained throughout fry concentration and enumeration procedures. During transfer, fry should be gradually acclimated to pond water (or new tank systems) to prevent shock. Food must now be provided to the fry to prevent starvation.

Summary

Red drum eggs and fry are usually maintained in incubator tanks for approximately 72 to 96 hours, or until alimentary tract development is complete. Tank conditions should be maintained within 22° to 30°C, 28 to 35ppt salinity, dissolved oxygen more than 3.0 mg/L, and less than 0.5 mg/L of ammonia. Egg and fry numbers are estimated by volumetric procedures.

Literature Cited

Bayless, J.D. 1972. Artificial propagation and hybridization of striped bass, *Morone saxatilis* (Walbaum). South Carolina Wildlife Resources Dept.

Bonn, E.W., W.M. Bailey, J.D. Bayless, K.E. Erickson and R.E. Stevens (eds.). 1976. Guidelines for striped bass culture. Striped Bass Committee of the Southern Division, American Fisheries Society, Bethesda, Md.

Holt, G.J. and C.R. Arnold. 1983. Effects of ammonia and nitrite on growth and survival of red drum eggs and larvae. **Trans. Amer. Fish. Soc.** 112:314-318.

Johnson, G.D. 1978. Development of fishes of mid-Atlantic bight. **An atlas of egg, larval, and juvenile stages.** Volume IV:235-246.

Intensive Culture of Larval and Post Larval Red Drum

G. Joan Holt, C.R. Arnold and Cecilia M. Riley
With Special Assistance from Sherri Hatch
The University of Texas at Austin
Marine Science Institute
Port Aransas, Texas 78373

Methodology and procedures for intensive culture of red drum larvae in relatively small batch and recirculating systems are outlined herein. They are largely based on 10 years of research in our laboratory.

Egg Handling

Eggs are collected in 500-micron mesh nitex bags placed in the filter box under the outflow from the spawning tank (Figure 1). Dimensions for the bag are 35 cm (14 in) wide x 45 cm 18 in) long x 70 cm (28 in) high and should be varied according to the size of filter box used in each system. The bag should be tied to the top of the filter box in an open position with the major portion of the bag underwater.

In their natural environment red drum spawn just after dusk therefore, in the laboratory spawning occurs in the evening from 0-3 hours after the lights have been turned off. Eggs are collected the next morning when they are already in the tail-bud or tail-free embryo stage. The basket is untied from the filter box and rinsed into a bucket of seawater. The fertilized eggs float to the surface and the unfertilized eggs sink.

Eggs can be transferred by dipping them up with a beaker, bucket, or net if done very gently. The eggs are poured into a one-liter graduated cylinder, allowed to settle for approximately five minutes, and the number of milliliters of eggs measured. Each milliliter contains approximately 1,000 eggs.

The eggs are placed in a 10 parts per million formalin bath for one hour, then placed in hatching containers. The formalin bath is made by first preparing a stock solution of 10 ml of 37 percent formaldehyde in one liter of deionized or distilled water; add one ml of this stock solution for each liter of the final bath.

If there is a problem with bacterial or fungal growth, usually seen as white mucous threads or cloudy water, add formalin (at 25 parts per million) or an antibiotic (Erythromycin at 10 parts per million). For optimal egg development and hatching, all procedures should be carried out in seawater of the same temperature and salinity as the spawning tanks.

Eggs will begin hatching in the late afternoon since eggs hatch in less than 24 hours at temperatures above 25°C (77°F). Optimal temperatures for both spawning and rapid egg development range from 25° to 29°C (77° to 84°F). A low magnification microscope is necessary to monitor the eggs for viability and proper development.

It is advisable to monitor the hatch rate as a check on the quality of the brood stock. This can be done by counting out 50 eggs and putting them in a small container (100 ml) of the hatching seawater that is well aerated before use. Set up at least three containers for replication of the results, in the event that

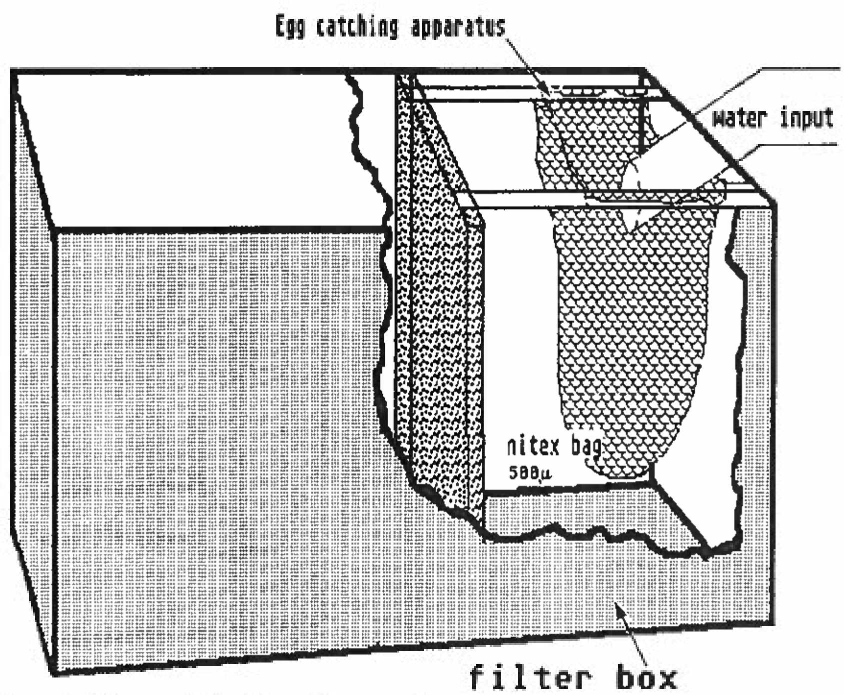


Figure 1. Diagram of filter box with egg catcher.

something goes wrong in one of these small static systems. The next morning, count the number of unhatched eggs. The hatch rate can be calculated as follows:

(1 minus the number of unhatched eggs divided by 50) x 100 = percent hatch, and the three replicates averaged. If brood stocks are doing well and hatching conditions are adequate, this rate will be between 90 and 100 percent.

Larval Culture System Design

Eggs may be placed directly in the tank or can be hatched and then transferred to the rearing tank. The latter allows easy removal of egg shells, which quickly contaminate the water, but requires care to ensure that temperature and salinity of both systems are the same before transfer and that the transfer does not physically damage the yolk-sac larvae.

Figure 2 shows a diagram of an experimental rearing tank currently being tested in our laboratory. The 150 l (40 gal.) fiberglass tank is designed so that water exchanges, if necessary for the maintenance of proper water conditions, can be made from the bottom while larvae float at the surface.

Ideal conditions for newly hatched larvae are water salinities and temperatures between 25-30 parts per thousand and 25° to 30°C, (77° to 86°F) respectively. Outside these ranges, survival may be reduced. Temperatures less than or equal to 20°C (68° F) may be counterproductive.

As the larvae grow and begin to feed, these conditions can be gradually relaxed, but growth rates will be reduced if the temperature is lowered, or if salinity is outside the range of 15 to 35 parts per thousand for the first four to six weeks.

Water Quality

Water quality must be maintained without loss or damage to the sensitive larvae. The water should be well aerated but not so forcefully that larvae are pinned against the wall; remember, they are very weak swimmers at first.

Total ammonia concentrations should be monitored regularly after feeding has begun (because the live prey also contribute ammonia to the system) and not allowed to exceed one part per million. Preferably, the ammonia level should remain below 0.5 ppm. Test kits to measure both ammonia and nitrite levels are available from many aquaculture supply houses or pet supply wholesalers.

Should water exchange become necessary, turn off the air for approximately 30 minutes, the larvae will come toward the surface and water can be siphoned from the bottom with very light suction. Small water exchanges (approximately 1/4 total volume) should be made until the original water quality is restored. Be sure the replacement water is the same temperature and salinity as the culture water and add it very slowly and gently. If seawater is being used, maintaining a pH of 7.8 to 8.2 should be no trouble and pH will not need to be monitored at this stage.

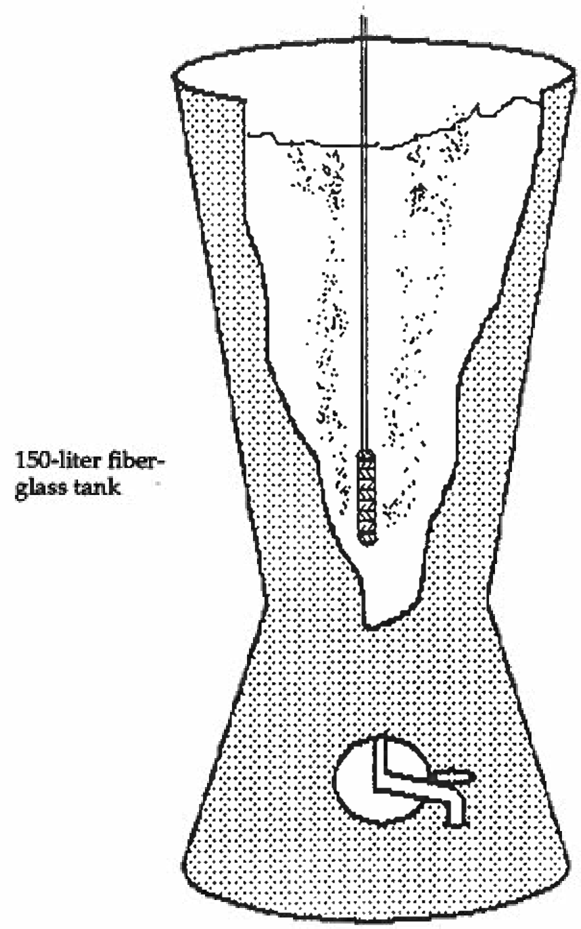


Figure 2. Example of an experimental larval rearing system.

Population Density

Fish density should be reduced as the larvae grow. During the first 10 days to two weeks, larvae can be maintained at 10 to 20 per liter, but after they have completely switched to feeding on brine shrimp nauplii, the fish density should be reduced to 1 to 2 per liter for two weeks, and then 1 per 2-liter for the final two weeks of larva culture.

At four to six weeks [25 to 30 mm (1 to 1.25 in) total length], the fish should be ready to be moved to the grow-out system. Until that time, fish should not be netted because they do not have scales to protect their skin. Changes in fish density must be accomplished without capturing the fish with nets.

Feeding

At approximately three days of age, larval red drum develop mouth parts and eyes. At this time their yoke-sac is depleted and they are ready to begin feeding. Because they are so small (2.5 mm (0.1 in) they are fed rotifers on days 3-10. Once they are larger they can be switched to brine shrimp (days 11-15) then switched to well blended fresh shrimp or artificial food for the remainder of the larval period (see Figure 3 and next section for more details).

Feeding methods for intensive systems need to be very dynamic to match the varied changes during larval development and growth from a 2-1/2-mm embryo to a 25-mm juvenile which occurs in four to six weeks. Feeding rates can be monitored and adjusted to changes in amount and food type with growth. Tank colors that provide sufficient contrast for prey detection by larvae include black, grey, blue and brown. Fiberglass tanks of various sizes and colors are available commercially but few have been tested as larval rearing systems.

Washing and Concentration of Rotifers

Pour rotifers through a funnel fitted with 63-micron nylon mesh. Most rotifers will be captured in the mesh while water and impurities will flow through. Rinse with low salinity seawater (if rotifers were cultured in low salinity) into a beaker to a volume of about 1/10 the original volume.

Counting Concentrated Rotifers

Because they are so concentrated, counting must be done in a sample dilution. Place 49 ml of seawater in a graduated cylinder then add 1 ml of well-mixed rotifer concentrate for a total volume of 50 ml; mix evenly.

Using a pipette, draw out 1 ml from the dilution, place it in a well-slide or counting wheel, and count individual rotifers (using a counter if necessary to keep track of number) under low magnification.

To determine the number of rotifers/ml in your total volume use this formula:

No./ml = No. of rotifers counted x 50 (No. of ml's in dilution)

You may want to adjust the volume of your dilution depending on the concentration of rotifers.

Feeding Larval Fish

Feeding usually begins on the third day after hatching. At the first feeding, 5 rotifers/ml total tank volume are added, regardless of fish density, using this formula:

$$\text{ml's of rotifer concentrate to add} = \frac{\text{No. ml in tank} \times 5}{\text{No. rotifers/ml in concentrated sample}}$$

Example: A 150 l (40 gal) fish tank, rotifer concentrate of 800/ml; need to feed fish 5 rotifers/ml on day 1, 150 x 1000 = 150,000 ml x 5 = 750,000/800 ml = 937.5 ml of rotifer concentrate needed to add to fish tank. Before subsequent feedings, it will be necessary to count the number of uneaten rotifers in each tank so that food density can be adjusted accordingly. To sample the tanks, withdraw 10 ml of water from the center of the tank directly in the path of greatest air flow. The rotifers in this sample are then counted by placing 1 ml onto a slide or counting wheel. Be sure the sample is well mixed. Three replicate counts should be taken then averaged. Now that you know how many rotifers remain, you can adjust the volume needed to return the food density to 5/ml using the following formula:

tank volume (ml) x (no. rotifers/ml needed)/ rotifer concentration

Example: A 150 l fish tank with 2 rotifers/ml left
Rotifers concentrated to 800/ml
Want a total of 5 rotifers/ml in the tank
150 x 1,000 x 3 = 450,000 / 800 = 562.5 ml rotifer concentrate needed

Larval red drum are fed rotifers two or three times a day at rates of 3-5/ml until they are nine to 10 days old. At that time they are switched to a diet of brine shrimp nauplii and fed 1/ml for the first day or two and then the brine shrimp should be reduced by half. Do not allow brine shrimp to build up in the tanks because nutritional value of the starved brine shrimp is greatly reduced in less than one day. An exception can be made if the fish are cultured with planktonic algae, since this is food for the brine shrimp.

Food density can be increased but ammonia levels must be monitored closely. Conduct daily counts on the brine shrimp much the same as you do the rotifer counts.

Beginning on day 15, the diet should be changed by adding a shrimp puree mixed with commercially available dry fry-food, while continuing to feed brine shrimp in reduced numbers. Changing red drum from a live to an inert diet is difficult and requires patience and persistence.

After about five days of mixed feedings, the brine shrimp can be discontinued and over the next two to three weeks the shrimp can be gradually reduced and the fish weaned to dry food in preparation for the move to the grow-out tank.

Although the feeding sounds simple, it is, in fact, a major obstacle to commercial production. The live food diets require a moderate level of knowledge and

Figure 3. Red Drum Feeding Schedule.

Age (days)	0		7		14		21
Size (mm)		2.5	3.5		5.0		10.0

Rotifers

Brine Shrimp

Shrimp and Art. Food

expertise to maintain healthy culture stocks that are available at the time and in the numbers required for larva culture.

For example if you want to raise 200,000 eggs through the rotifer stage at a density of 20 fish/liter you will need 10,000 liters of water. Feeding at an average rate of 3 rotifers/ml would require 30,000,000 (30 million) rotifers twice a day for 10 days, or a total of 600 million rotifers for one tank of fish.

Another problem occurs when larvae are changed to an inert diet, some fish never adapt, and some begin feeding on smaller fish which results in a great deal of mortality. Additionally, redfish do not grow as well on the commercially available dry diets as they do on shrimp or fish; nutrition studies are being carried out to develop a better diet.

Raising Food Organisms for Intensive Larval Culture: I. Algae

Granvil D. Treece
Sea Grant College Program
Texas A&M University
College Station, Texas 77843-4115
and
Nancy Wohlschlag
Division of Life Science
The University of Texas at San Antonio
San Antonio, Texas 78285

An area of major concern in the redfish hatchery is the provision of a suitable food source for the fish larvae. Unicellular microalgae, which provide the basis for natural food chains, also provide the basis for the food chain in the hatchery, giving good larval growth and survival.

Algae serve a number of purposes in the fish hatchery. Their presence in the larval fish culture system helps maintain desired water quality, and they utilize larval metabolic products in addition to providing the basis for the food chain. The food chain consists of the algae and the zooplankton which feed upon the algae and are in turn preyed upon by the fish larvae.

According to Guillard (1975), six different classes of algae are cultured to provide the basis of acceptable food chains for aquaculture reared animals. For the culture of red drum larvae, years of research and practical experience by a number of groups (National Marine Fisheries Service, University of Texas Marine Science Institute, Texas Parks and Wildlife Department and others) have indicated that two species of unicellular microalgae are both easy to grow and serve as excellent food sources. The species are *Tetraselmis chuii* and *Isochrysis* sp. (Tahiti isolate).

The present authors prefer the Tahitian strain of *Isochrysis* because it is easy to culture in the laboratory. Both species provide an excellent food source for zooplankton. Other comparisons of the two species show the desirability of *Isochrysis*, however.

Tetraselmis chuii is a green flagellate ranging from 10 to 15 micrometers in diameter (see Figure 1a). *Isochrysis* sp.(Figure 1b)is a smaller spherical-shaped brown, naked flagellate, approximately 3 to 5 micrometers in diameter, and capable of self-locomotion. The smaller size and the absence of a heavy cell wall may make it a more suitable food for larval herbivores than *Tetraselmis*. The Tahitian strain of Isochrysis prefers high temperatures, up to 30°C (a

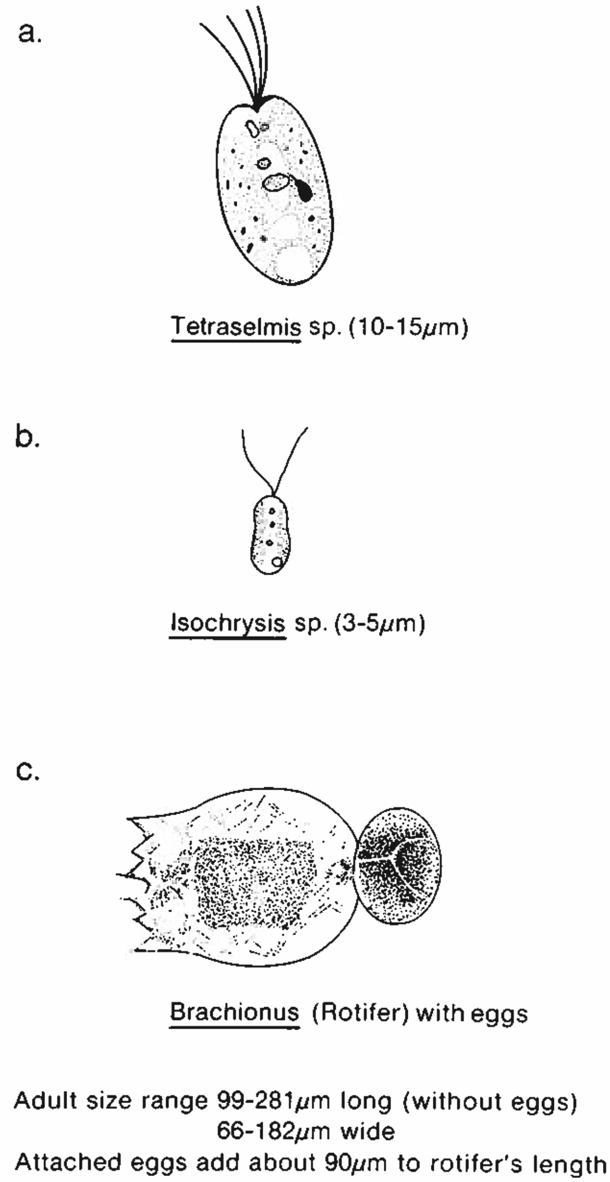


Figure 1. Some commonly cultured microalgae and zooplankton.

consideration in tropical culture conditions), and strong light. Cell counts commonly range between 1 and 7 million cells per ml. *Tetraselmis* cultures rarely achieve this density.

Tahitian *Isochrysis* shows only a slight variation in protein content (0.9-1.7 X 10-7 microgram atoms protein-nitrogen per cell). Being a naked flagellate, Tahitian *Isochrysis* or "T-Iso." as it is sometimes called, is motile and thus disperses well throughout the water column. *Tetraselmis*, on the other hand, has a tendency to settle out if the culture is not aerated. Mass cultures of *Isochrysis* also appear to selectively suppress growth of undesirable contaminants (such as bacteria) and thus are near"axenic" or close to being bacteria free for a longer period. In summary, Tahitian *Isochrysis* does very well indoors under the conditions described.

Once a hatchery is established, and culturing the above species is routine, additional species of algae from your specific site may be cultured. These algae may be better adapted to the specific environmental conditions at your particular location. Experiment with locally caught zooplankton (using copepods instead of rotifers, for example), only after a consistent *Isochrysis*- to *Brachionus*-to fish larvae food chain has been established. The zooplanktor *Brachionus* (Rotifer) is figured in 1c.

Culture Techniques

Obtaining Cultures

An algal stock culture (available from suppliers listed in the Appendix) is used to provide a dense "starter" culture (for the inoculation) of the species which has been chosen for mass culture as food for zooplankton. Rotifers, one example from the zooplankton, have traditionally been fed the two species of algae discussed. Other species can be used, but there is some risk involved in using untested species. The authors have attempted to culture rotifers using *Chaetoceros*, but had only limited success.

Growth Medium

Like other plants, algae require nutrients or fertilizers to achieve rapid growth. Dozens of nutrient formulations have been used for this purpose. We recommend that Guillard's f/2 algal culture medium be used since it has provided excellent results in the past and it can be purchased in a pre-mixed, concentrated form from a local aquaculture supply house or chemical dealer-distributor listed in the Appendix.

The f/2 algal culture nutrients are added to sterilized seawater and the resulting medium is recommended over other media at least through the carboy stage. Table 1 lists the nutrients in Guillard's f/2 algal culture medium. The richer Guillard's f medium has twice the amount of each nutrient per liter of seawater.

For mass culture of algae (indoors or outdoors) beyond the carboy stage, in an open vessel, a different growth medium may be used in order to save expense. In some of the earlier work done with larval fish food chains, the algae medium was a combination of fertilizers (ammonium sulphate and superphosphate) added to filtered seawater. One problem experienced with this method was that all of the fertilizer did not dissolve and go into solution for the algae to utilize. An alternate method is to use an organic fertilizer available in a liquid form. One recommended is called Fish Emulsion (suppliers are listed in the appendix).

Coupled with the right temperature and a continuous bright light, fish emulsions can promote excellent algae growth in seawater at a fraction of the cost of f/2 medium. Table 2 is a comparison of media costs.

Algae Growth Characteristics

Given the right conditions (nutrients, light, temperature etc.) algal cultures will generally exhibit a growth curve similar to that shown in Figure 2.

The objective is to keep the culture in the phase of most rapid cell division (exponential phase) and keep it from dying or "crashing" (Figure 2-II). The best way to do this is to constantly dilute the culture by draining one-third to one-half of it each day (using it as food for rotifers for example) and replacing the culture with new seawater and medium. Another way to keep the culture in the desired phase of growth is to transfer the existing culture into a larger, clean growth vessel containing additional fresh medium and seawater.

Table 1. Guillard's f/2 Algal Culture Medium (composition per liter of seawater)

Major Nutrients	
$NaNO_3$	75mg
NaH_2PO_4 - H_20	5 mg
Na_2SiO_3 - $9H_2O$	30mg[1]
Trace Metals	
Na_2 - EDTA	4.36mg
$FeCl_3$ - $6H_2O$	3.15mg
$CuSO_4$ - $5H_2O$	0.01mg
$ZnSO_4$ - $7H_2O$	0.022mg
$CoCl_2$ - $6H_2O$	0.01mg
$MnCl_2$ - $4H_2O$	0.18mg
Na_2MoO_4 - $2H_2O$	0.0006mg
Vitamins	
Thiamin - HCL	0.1mg
Biotin	0.5 micrograms
B_{12}	0.5 micrograms

[1]Silicate may be omitted for species other than diatoms.

Table 2. Comparison of Algae Culture Media Costs

Fish Emulsions (at $6/gal. or $1.59/liter)	Premixed f/2 Algae Food (at $22/gal. or $5.81/liter)	Guillard's f/2 (chemicals ordered separately, but not purchased in bulk)
$0.01/10 gal. media ($0.01/38 liters)	$0.05/10 gal. media ($0.05/38 liters)	$0.09/10 gal. media ($0.09/38 liters)

Inoculation and Transfer

Stock cultures can be maintained in small screw-cap test tubes – with the tops left slightly loose. The algae can be kept dormant by diluting the nutrient medium to f/8 (A 4-time dilution of the standard f/2 growth medium), then maintaining the culture at 24°C in a low-light environment. These conditions will restrict growth to a maintenance level, and there will be a minimal build-up of metabolic wastes.

Stock cultures should not be handled anymore than necessary, and they should be kept in a separate area where the traffic or work activities are minimal. This lessens the chance for contamination. Once a month, a small amount of the culture is transferred from the old tube into a new clean tube containing fresh sterile medium. This monthly transfer is best carried out under sterile conditions. Figure 3 depicts a typical algal transfer routine and production-line schedule.

Small flasks are inoculated from the stock culture tubes and incubated in a high-intensity light area. Once the cultures in the flasks are a few days old and their respective cell counts have increased considerably, they can be transferred to larger growth vessels, such as large flasks or carboys. After a similar period the carboy cultures can be transferred to even larger containers, or used as feed.

The objective is to keep the algal species which you are trying to culture as the dominant species in the culture, and to keep the culture in the correct growth phase until you are ready to use it for food. This should be accomplished with the least amount of contamination possible - by adding a pure inoculum culture to a relatively clean and sterilized, growth medium. Compromise on these simple precautionary measures will most assuredly lead to problems with algae culture.

Filtration and Sterilization

Seawater is generally filtered to one micron (one

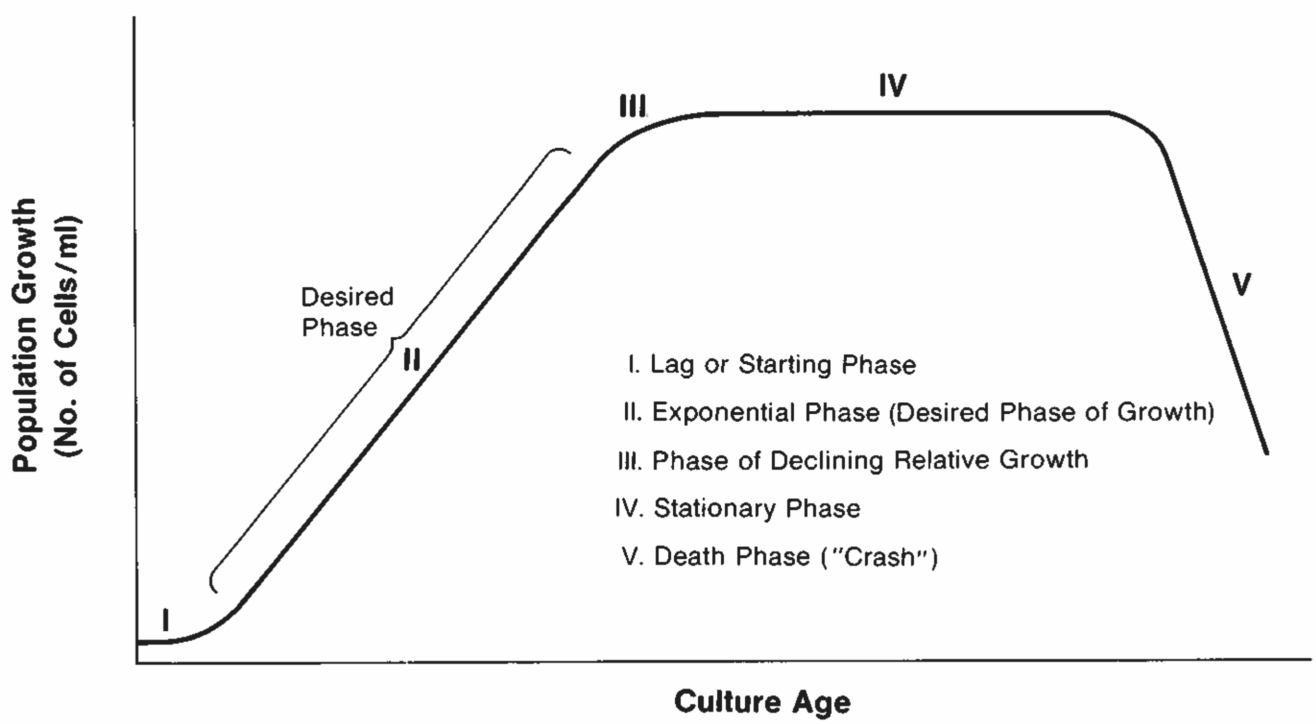


Figure 2. Typical phytoplankton culture growth.

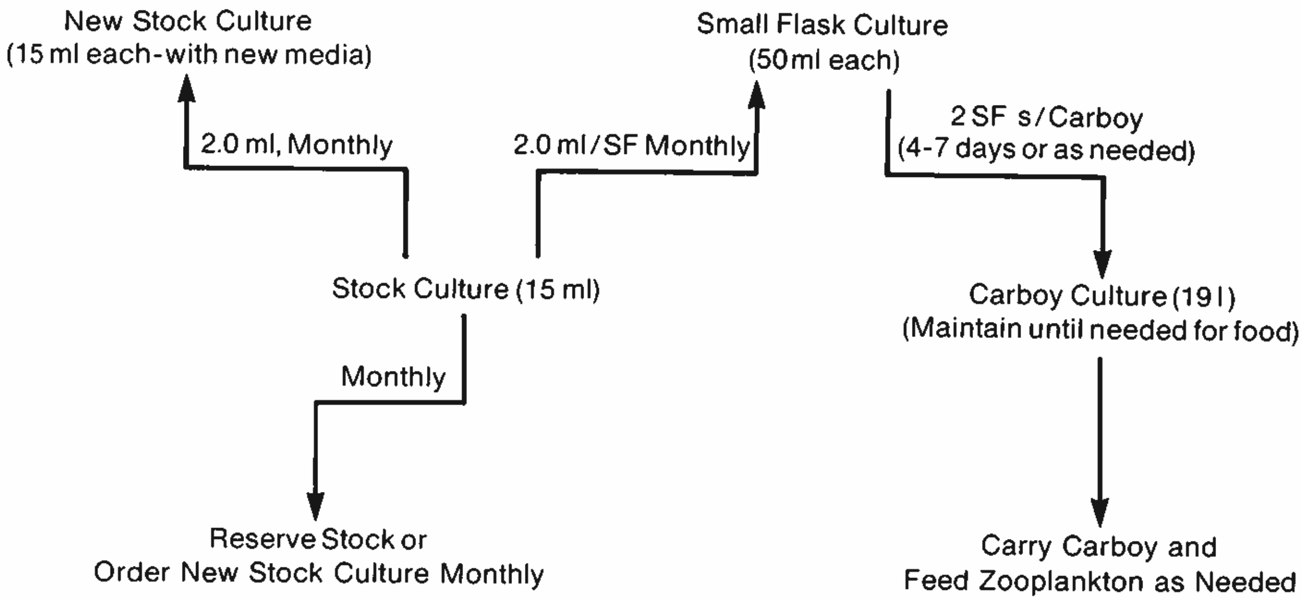


Figure 3. Algae Transfer routine.

micrometer) but it must be sterilized or treated before it is used in algae culture. The amount of filtration required is largely dependent on characteristics of the incoming seawater. The amount of filtration is, therefore, largely site-specific.

The method of sterilization selected is also site-specific. The following is a description of sterilization methods used for algal culture seawater.

Autoclaving

Autoclaving is the most effective and the most preferred, but is also the most expensive. This method involves heating to 120°C and 20 psi for a time period dependent on the volume (15 minutes for liquids less than one liter, 45 minutes for larger volumes like a 20 L. carboy). The glassware will last longer using this method, compared to direct heating methods. Carboy-size autoclaves cost approximately $3,000, and smaller, pressure cooker-type autoclaves, for sterilizing tubes and small flasks cost approximately $300. This method is most often used in research labs, and is not widely used in commercial operations, where a more cost effective method must be used.

Chlorination

A 2.5 ppm concentration of bleach (from a standard 5.25 percent hypochlorite solution) in seawater has been found to be an effective germicide. The treated seawater is left for 24 hours and then aerated to get rid of the remaining chlorine. Sodium thiosulfate is used in some hatcheries to neutralize the residual chlorine, but with the delicate trace metal balance found in seawater the addition of excess quantities of this chemical has caused detrimental side-effects.

Pasteurization

The steps involved in this method of water treatment are to heat a container of medium-enriched seawater (one liter or larger) to 73°C, hold at this temperature 10 to 15 minutes, then let cool overnight. The following day, reheat the enriched seawater to 73°C again and let cool to ambient temperature. Double heating destroys most microorganism spores which may have been formed during the first heat treatment. Low heat treatment is advantageous because it reduces trace metal precipitation and it is relatively inexpensive.

Filter Sterilization

Mechanical filtration through a sterile filter is a slow and expensive method, but can be used when small quantities of water are needed. One minor advantage of this method is that it prevents any precipitation of trace metal nutrients.

Other methods include the use of antibiotics and ultra-violet (UV) light. Antibiotics are more often used in controlling diseases in tanks. Likewise, UV light is more suitable as an ongoing way of controlling disease in flowing systems.

Summary

Selection of a sterilization technique should be based on site-specific resources. The method selected at one facility may not be appropriate at another. One of the first three sterilization methods listed (autoclaving, chlorination or pasterization) should meet the needs of most hatcheries. Occasional contamination of algae cultures is inevitable regardless of the method of sterilization needed. Be prepared to restart a culture from any point.

Precautions and Limitations

- Sterile procedures must be followed when transferring stock cultures on a monthly basis in order to maintain the culture in an "axenic," or bacteria free condition. An alternate method is to order new stock cultures periodically, but care should be taken to keep other cultures (in tubes, flasks and carboys) as bacteria free as possible.

Excessive contamination in hatchery cultures are also sources of problems. Bacterial contamination will shorten the life of an algae culture and will severely limit its production capacity. A contaminated culture will cause problems further up the food chain as well. Bacteria or protozoan contaminants may infect or attack the larvae which you are attempting to raise. A contaminated culture will appear dirty or will have excessive brown or blue-green growth on the sides of the vessel. Protozoan contamination is also a problem which can eventually cause the algae culture to "crash," i.e. die rapidly. Protozoans are easily recognized in the culture because they are usually larger and swim much faster than the algae. As a rule, a few contaminants cause no harm, but they do compete with the algae and some even prey on the algae. If proper culture procedures are followed,these contaminants will be slow in developing to critical numbers. An open,indoor algae culture which is kept in the exponential phase will usually last a month before it crashes and must be restarted.

- Zooplankton (rotifer) cultures should also be kept in the exponential growth phase for maximum production.
- Keep the algae system completely separate from the zooplankton production system. No plumbing should directly connect the two systems. If the zooplankton can back-track their way to the algae, the culture will be lost. Some hatcheries utilize a gravity feed system where the algae tank is located above the zooplankton tank. Algae are drained into the zooplankton, with no direct plumbing connection. This break is important.
- When culturing *Isochrysis*, cell counts should be sufficient in carboys to provide the food requirements for very large rotifer populations. Rotifers may be fed by carrying a carboy of algae into a separate rotifer culture room, and feeding manually by pouring a desired amount into the larger rotifer culture tank. Because of potential contamination, manual feeding is more desirable than having the larger algae tank (gravity-feed) system located in the same room. If the gravity system is used, then brushes and other equipment should not be used in both culture systems (each should have its own seperate cleaning equipment, to eliminate contamination). Technician's hands can also be a source of contamination which he or she should always keep in mind.
- Light or nutrient limitations, as well as contamination, can limit the productivity of the algae culture. High pH can also cause culture problems, especially if the mass culture is outdoors. A pH of nine to 10 would not be uncommon and can be lowered by adding a small amount of CO_2 gas to the air supply. Generally, indoor cultures have no pH problems because of relatively lower light intensity.
- Use Guillard's f/2-medium, or a similar substitute, when working with indoor cultures. When cost effectiveness comes into play (for example with large outdoor cultures), use fish emulsions or some less-expensive method of fertilizing the culture.

Summary Procedures for Algae Culture

- Obtain stock culture of selected species (perferable Tahitian *Isochrysis*) as described previously.
- Obtain growth medium as described previously.
- Follow algae transfer routine as shown in Figure 3.
- Perform stock culture transfer and monthly maintenance as follows:
  1. Materials needed: transfer hood (optional), sterile pipets, Bunsen burner or some other flame source, log book, screw cap test tubes, test tube holder or rack, spray disinfectant, a small autoclave or lightweight sterilizer and f/2 medium.
  2. Transfer Procedure
     a. Obtain clean stock culture tubes and fill with filtered seawater and proper amount of medium for f/2 culture (approximately 15 mls total volume per tube is adequate).
     b. Autoclave or otherwise sterilize tubes of media with caps loose. Remove tubes from sterilizer, tighten caps and allow tubes to cool to ambient temperature. The tubes are now ready to inoculate. One may wish to sterilize with a flame or use some other method as an alternative to autoclaving.
     c. Obtain stock cultures to be transferred.
     d. Disinfect work surface area by wiping with a cloth and a chemical disinfectant.
     e. Agitate old stock culture tube by swirling it (do not shake it), remove cap and flame the open end of tube.
     f. With sterile pipet, remove 2.0 ml of the old culture, flame open end and replace the tube cap loosely.
     g. Remove the cap of the new sterilized media tube and flame the open end in the same manner, then allow the inoculum from the pipet to drip into the fresh medium, flame the open end and replace the screw cap loosely.
     h. Agitate the new tube by swirling and place in a test tube on the top shelf of the Algae Culture Rack as seen in Figure 4.

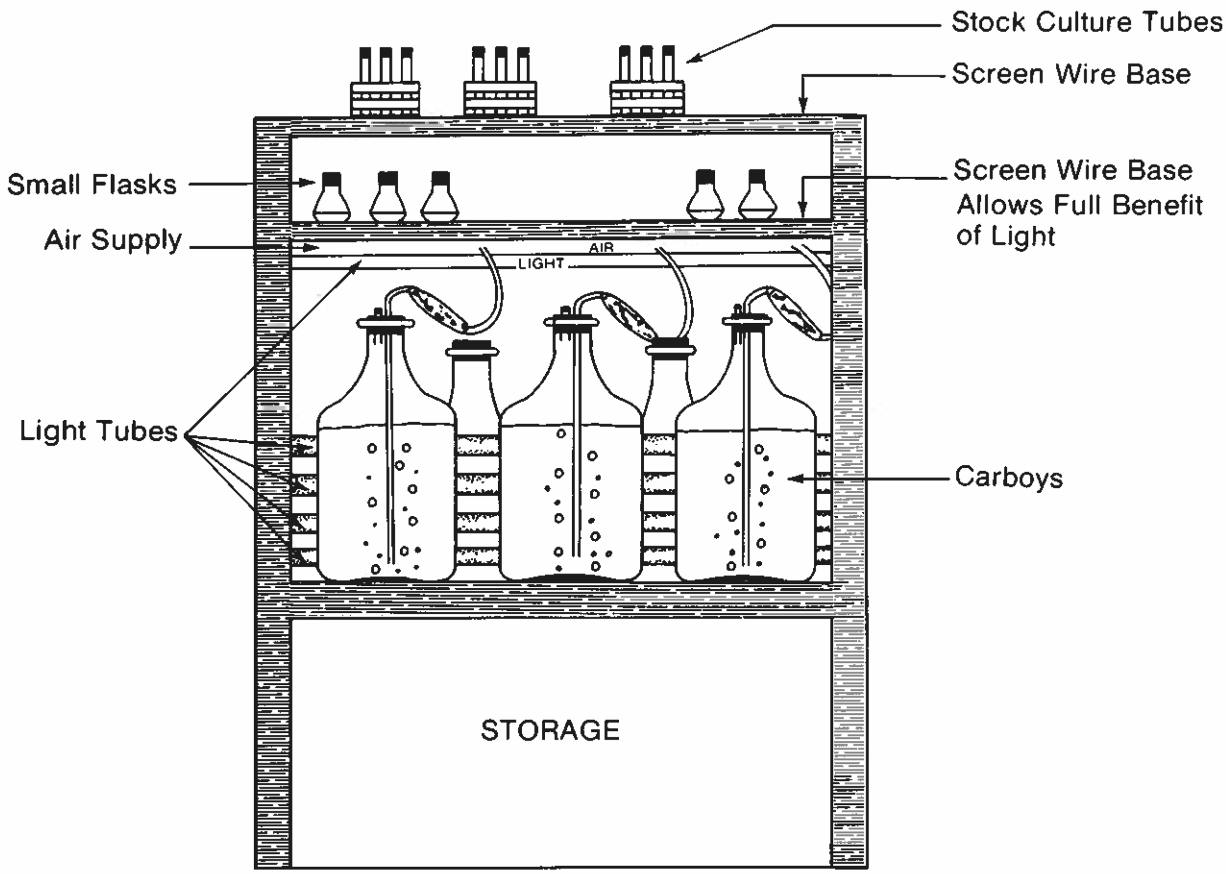


Figure 4. Typical algae culture rack.

- i. Repeat this routine (a through i) periodically.

- Place old stock culture in reserve (some hatcheries just order new stock culture tubes periodically).
- Transfer to small flasks. Small flasks' transfer are accomplished in the same manner except a greater quantity of culture inoculum is used (using approximately 2 mls to inoculate a small flask containing 50 mls of new sterilized medium; the 52 ml small flask is in turn used to inoculate a larger vessel, etc.).
- Procedure for the inoculation of a carboy
  1. Materials needed: autoclave (large enough to hold a 12 or 19 liter glass carboy), carboy with aeration device as shown in Figure 5, new culture medium, inoculum from at least two small flasks, aluminum foil, proper nutrients, heat resistant gloves and pipets.
  2. Procedure
     - a. Obtain clean, glass carboy.
     - b. Fill the carboy to the normal capacity mark (see Figure 5) with filtered seawater.
     - c. Add proper amount of nutrients (if using Fritz's f/2 medium, then add 2.5 mls "Solution A" and 2.5 mls "Sol. B" for 19 liters seawater).
     - d. Cover container loosely with aluminum foil or, if there is enough room in the autoclave, cover it with the apparatus shown in Figure 5; be sure to leave the cap on the inoculation tube loose or remove it during the sterilization period. Tie the air tube up above the water level or use a hose clamp to avoid accidental drainage due to siphoning.
     - e. Autoclave carboy and aeration apparatus on a liquid cycle using a slow exhaust. If you have a manually operated sterilizer, then allow 45 minutes sterilization once the autoclave reaches a temperature of 120°C and 20 psi.
     - f. After autoclaving, seal the bottle and let cool overnight. If time is of the essence then a low aeration level may be started in the bottle to assist the cooling process. Let cool to ambient temperature.
     - g. Transfer carboy to the algae rack (Figure 4) and connect the air supply. This will bring the pH back to the normal culture range (7.8 to 8.2).

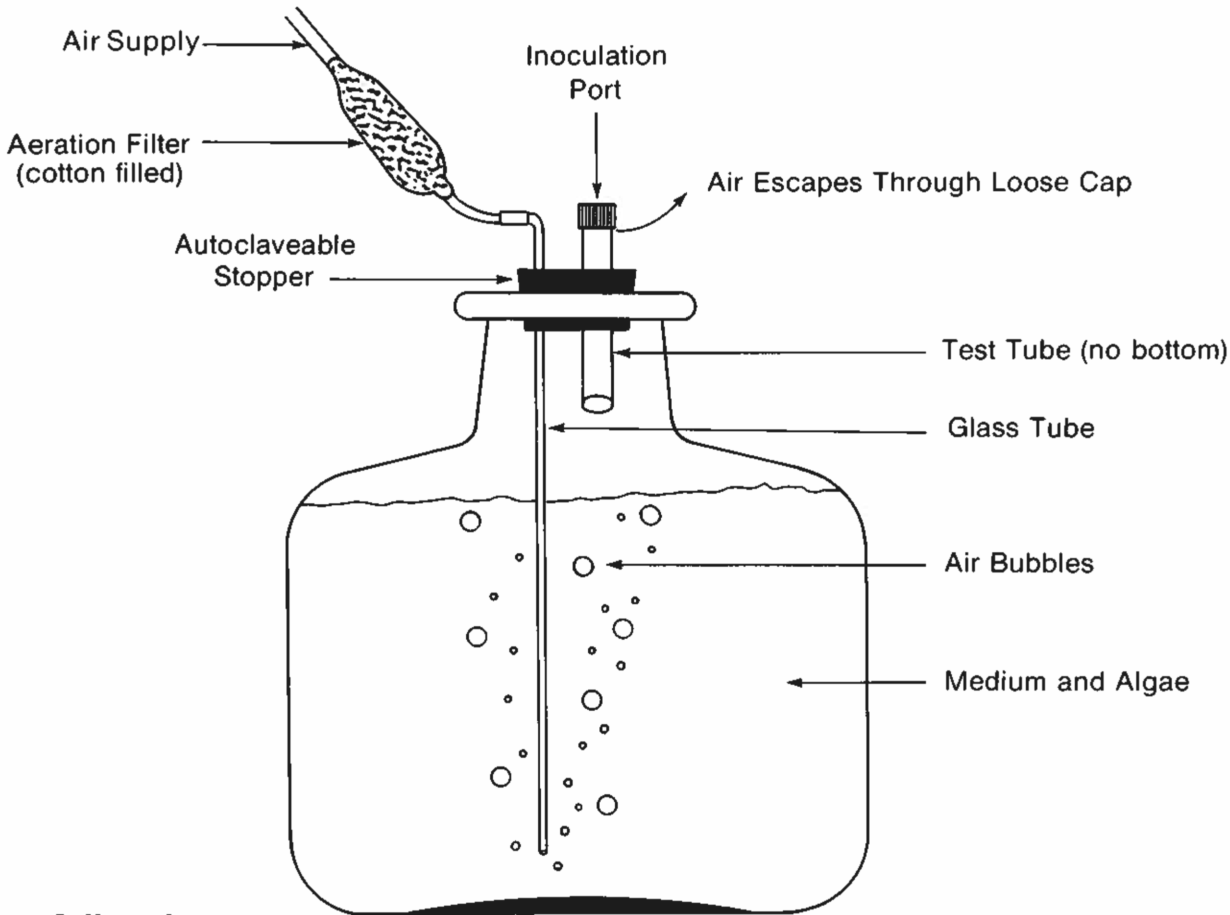


Figure 5. Glass carboy

h. Inoculate the carboy by pouring the contents of at least two small culture flasks through the inoculation port (some culturist prefer to inoculate a carboy of this size with at least one liter of inoculum).

- Alternate culture methods

If the chlorination treatment method is selected instead of autoclaving, then the procedures are slightly different for carboy inoculation.

1. Materials needed: 5-gallon, clean, drinking-water bottle (plastic or glass), filtered seawater, chlorine, sterilized nutrients, and inoculum from at least two small flasks.
2. Procedure
   a. Obtain a clean, 5-gallon drinking-water bottle and fill with filtered seawater (some hatcheries utilize disposable plastic bags inserted inside the bottle).
   b. Add chlorine as described in the section on chlorination.
   c. Let sit for 24 hours, with no aeration.
   d. Aerate the treated seawater (overnight to be safe).
   e. Add the proper amount of sterilized nutrients.
   f. Inoculate with pure culture from at least two small flasks.

Chlorination can also be used to sterilize the carboy and the medium can be pre-treated by pasteurization before adding to the carboy.

1. Materials needed: 5-gallon bottle, pasteurized medium, and inoculum from at least two small flasks.
2. Procedure
   a. Sterilize bottle with chlorine and rinse with sterile water.
   b. Add chlorine treated seawater that has been aerated (as described previously).
   c. Add medium.
   d. Inoculate.

- Cell counting and analysis of algal cultures

Analysis of algal cultures for feeding zooplankton in a commercial fish hatchery should only involve visual inspection of the cultures. For all practical purposes, cell counting using the simple haemacytometric technique can be avoided, but occasional microscopic observation of the cultures is necessary to keep ahead of the ever-encroaching problem of contamination.

If algae cell counting is deemed necessary, then use a haemacytometer. Other more expensive methods used for counting algae are: Coulter counter, turbidometer, and spectrophotometer.

Summary

A typical algae room arrangement can be seen in Figures 6 and 7. This particular operation utilizes the chlorination technique and a variety of culture vessels (including the glass drinking-water bottles). Note the clean, uncluttered appearance; this is important to minimize contamination. This particular algae room was home-made, (i.e. constructed of easily obtainable, inexpensive materials). With the general description of the fundamentals of algae culture presented here and with the simplified procedures given above, the newcomer can master the concepts involved in growing algae.

Substitutes such as yeast have been used for algae, and micro-particulate and micro-encapsulated diets offer future hope for replacement of algae in the fish hatchery, but each system has problems at the present time. Until new diets are developed and tested, the culture of algae and rotifers will remain the method of choice in most hatcheries.

Literature Cited

Arnold, C.R. *et.al.* 1977. Methods and techniques for spawning and rearing spotted seatrout... (section on production of algae). In the Final Report, **Federal Aid in Fisheries Restoration Act.. Federal Aid Project F-31-R, State-wide** Fisheries Research Objective 21: Marine Fish Propagation Study.

Fox, J.M. 1983. Intensive algal culture techniques. **CRC Handbook of Mariculture,** Vol. I: Crustacean Aquaculture, p. 15-41.

Griffith, G.W. *et.al.* A mass culture method of *Tetraselmis* sp. - A promising food for larval crustaceans. Unpublished manuscript, NMFS Gulf Coastal Fisheries Center, Galveston, Texas. 9 pp.

Guillard, R.R.L. 1975. Culture of phytoplankton for feeding marine invertebrates, **Culture of Marine Invertebrate Animals,** Plenum Press, New York.

McLachlan, J. 1973. Growth media-marine, Handbook of Phycological Methods, **Culture Methods and Growth Measurement,** Stein, J.R., Ed., Cambridge University Press, London.

Starr, R.C. 1978. The culture collection of algae at the University of Texas at Austin. **Journal of Phycology,** Vol. 14, pp. 47-100.

Starr, R.C. 1978. Additions to the UTEX collection since 1978. Available upon request from Richard C. Starr, Dept. of Botany, The University of Texas at Austin, Austin, Texas 78712.

Stein, J.R. 1973. **Handbook of Phycological Methods, Culture and Growth Measurement,** Cambridge University Press, London. 448 pages.

Appendix

Stock Cultures

The University of Texas at Austin, Department of Botany (Dr. Richard C. Starr), Austin, Tex., 78713-7640 (Phone 512/471-4019) provides a variety of algae cultures at a cost of $10 per tube for academic institutions and $25 per tube for commercial enterprises.

A commercial source of zooplankton cultures and some algae cultures is: Carolina Biological Supply Company, 2700 York Road, Burlington, N.C. 27215 (Phone 800/334-5551). Consult their current catalog for available cultures and prices. [A culture of the marine rotifer, *Brachionus* (see Figure 1c.), costs $5.95 plus shipping.]

Growth Medium

Fritz f/2 Algae Food is manufactored by Fritz Chemical, P.O. Drawer 17040, Dallas, Tex. 75217 (Phone 214/289-1791). The cost is $22/gallon.

One fluid ounce of their solution A mixed with one fluid ounce of solution B makes 60 gallons of algae growth medium; one gallon of solution A mixed with one gallon of solution B will make 7,600 gallons of medium.

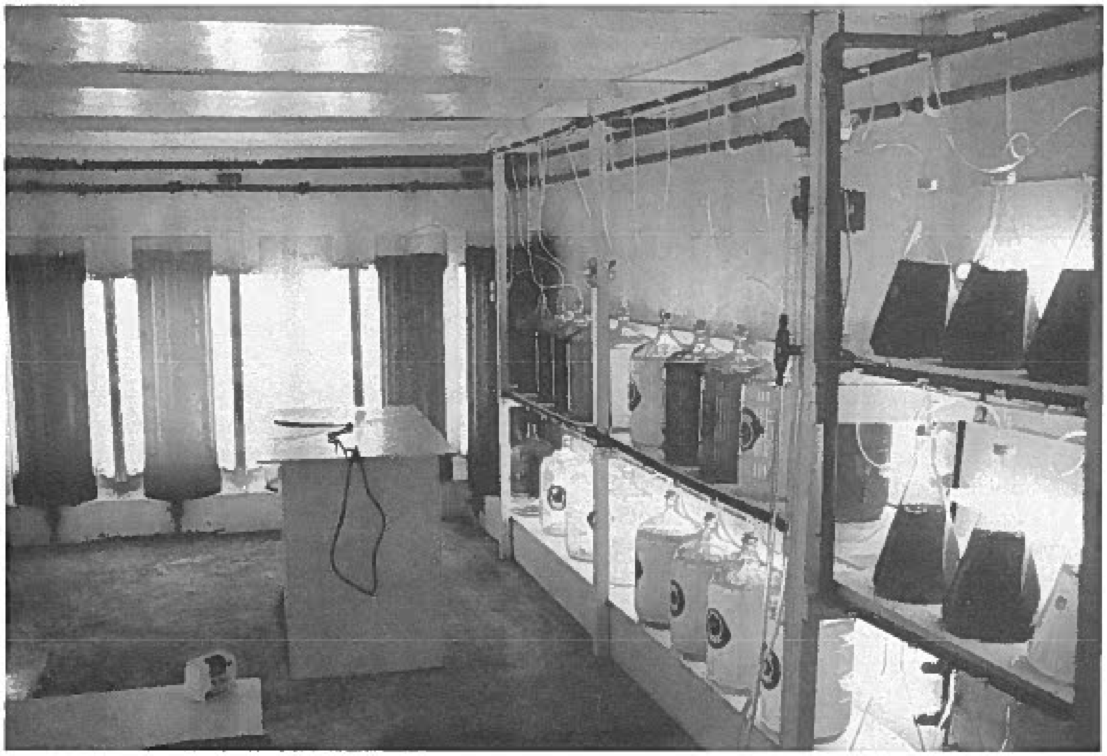
The cost of this averages out to be $0.0057 per gallon of medium, or about a nickle for every 10 gallons (exclusive of shipping charges). This figure is of the same order as an estimate made by Fox (CRC Handbook of Mariculture, Vol. I: Crustacean Aquaculture, 1983, pp. 15-41). He calculated that Guillard's f/2 medium (made from "scratch") costs $0.726 for each 300 liters, or about $0.0091 per gallon (including shipping).

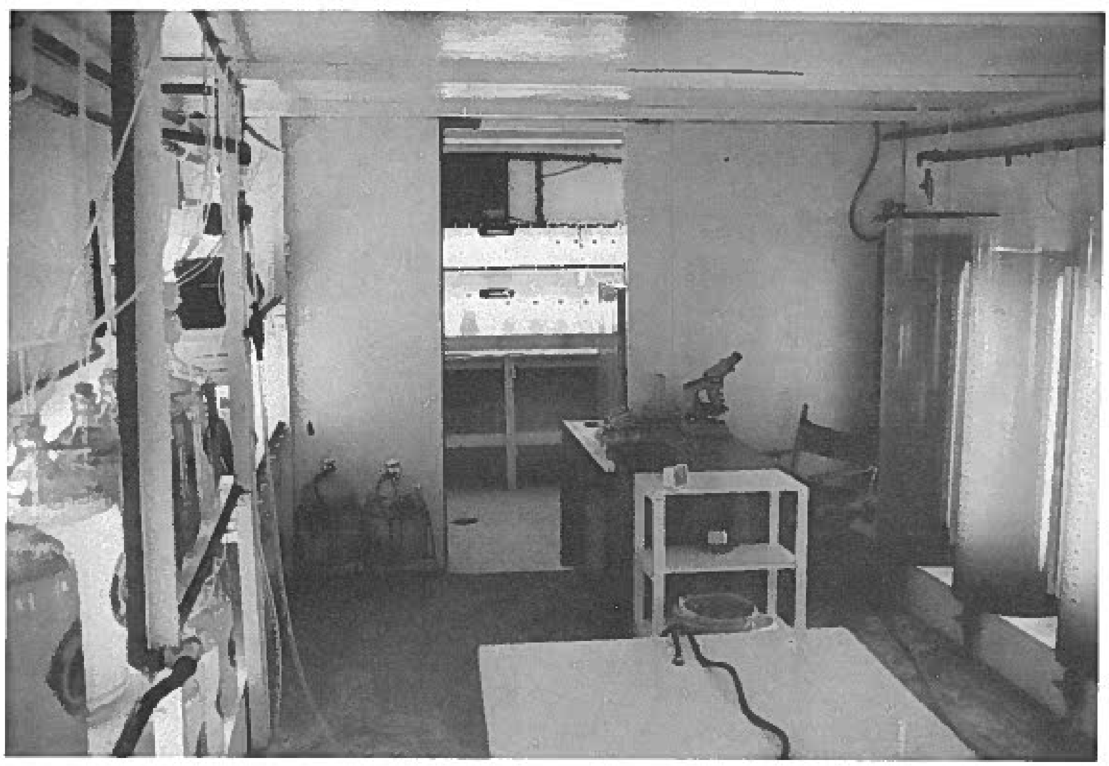
Fish Emulsions

A major supplier of fish emulsions is Alaska Fish Fertilizer Co., Washington (Phone 800/325-2585). They will inform you of their fish emulsion representatives in your area. For Texas, contact either Frank Catalani, Lake Dallas, Tex. (Phone 817/497-5111) or Mike Harris, Silsbee, Tex. (Phone 409/755-6201). They will tell you who the wholesalers and retailers for their product are in your specific area.

In the Houston area, Magnolia Seed, 3202 McKinney, Houston, Tex. (Phone 713/227-3631), has the Alaskan Fish Fertilizer Company's brand (Atlas). In the Corpus Christi area, Curry's Nursery, Aransas Pass, Tex. (Phone 512/758-5354) carries a variety of brand named fish emulsions with a variety of prices as well.

Fish emulsion brand names include Ortho, Greenlight, Atlas and Dexol. On a retail basis, the least-expensive we've found is Dexol, at $6/gal. (in a case of 4). Harpool Seed Company, Austin, Tex., offers a reduced rate, but has a 300-gallon minimum order.





Figures 6 and 7. Typical algae room arrangement.

Raising Food Organisms for Intensive Larval Culture: II. Rotifers

N.S. Wohlschlag, Li Maotang and C.R. Arnold
The University of Texas at Austin
Marine Science Institute
Port Aransas, Texas 78373

The rotifer, *Brachionus plicatilis*, is an important food organism for the mass cultivation of larval fish in fish hatcheries around the world. It is a cosmopolitan, euryhaline species, and is extensively used as a first food for larval fish. The culture and use of *B. plicatilis* as larval fish food was first developed and studied in Japan in the early 1960s. Since then, many different methods for culturing the rotifer have been developed and used (Scott and Baynes, 1978).

The first methods used phytoplankton as the primary food source. Later, yeast was used as a supplimental food source (Hirata, *et al.*, 1983) and an increase in rotifer population was achieved. Artificial diets and enrichment supplements also have been used with mixed results.

B. plicatilis is well suited to mass culture because of its life cycle. It is a planktonic filter feeder that feeds on organic particles that are brought to its mouth by the movements caused by the corona, which is the ciliated organ on the head region that characterizes the rotifers. It also is the rotifers means of locomotion.

The reproductive characteristic that allows *B. plicatilis* to be an ideal mass culture organism is that it reproduces parthenogenetically asexually for most of its life cycle, producing only the larger females. These females produce a thin-shelled egg that is not fertilized and only produces amictic females until environmental factors trigger the production of mictic females. Mictic eggs are thin-shelled and, if not fertilized, they produce male rotifers. If mictic eggs are fertilized, a resistant shell is secreted that enables the egg to withstand harsh environmental conditions. These eggs are called resting or dormant eggs that allow the rotifer population to overwinter. When conditions are favorable, the resting eggs hatch into amictic females and the cycle is repeated.

Parthenogenetic reproduction allows large numbers of individuals to be produced in a short time, which is ideal for mass culture.

Rotifers vary in size depending on strain and culture conditions. Adult sizes range from 123 to 315 microns. This can allow strains to be cultured for size (Yufera, 1982; Snell and Carrillo, 1984). Rotifers can be grown in a range of salinities. Our studies indicate 18 ppt is the optimum for the strain we use. Reproduction in rotifers is salinity-dependent, therefore, the salinity used depends on the strain of rotifer used. Nutrients for the culture are varied and the most common are: single-celled algae, such as *Tetraselmis*, *Chlorella*, *Isochrysis*, etc., bakers yeast, emulsified oil enrichment and artificial diets. There are advantages and disadvantages to each culture media.

Stock rotifer cultures are used as starter cultures to initiate production in larger culture containers. These should be maintained in a separate area from mass culture tanks to prevent contamination. These can be maintained in 1- to 2-liter flasks on algae at 24° to 25°C and a light cycle of 12-light/12-dark. This allows for better water quality and less maintenance. The culture should be restarted periodically (at least every month; more often depending on environmental factors).

Stock cultures can be used to start an inoculant culture, depending on the number of rotifers required.

Cuture Methods

The following is a description of several culture methods used at The University of Texas, Marine Science Institute, Mariculture Program, Port Aransas, Texas, but does not necessarily represent the most advanced methods.

Rotifers are mass cultured in either 1.8-meter diameter, round flat-bottomed tanks that hold up to 1,800 liters of water or 190 liter conical tanks. The tanks are sterilized with 2.5 ppm bleach for 12 to 24 hours, then rinsed and cleaned. Tanks are filled with filtered sea water in which the salinity has been adjusted to 18 ppt with dechlorinated fresh water. The temperature is maintained at 24° to 26°C in the culture room. The round tanks have continuous light supplied from light banks with 40-watt fluorescent bulbs over the tanks. The conical tanks have no additional light other than room ceiling lights. All of the tanks are aerated.

Method Using Baker's Yeast and Emulsified Oil

Set up round tanks and conical tanks as described above. On day one, yeast is fed at 0.6 to 0.8 g/L, emulsified oil at 1.0 ml/10L and rotifers inoculated at

10 to 40/ml after being rinsed three times through a 60 micron filtering cloth, with fresh 18 ppm adjusted filtered sea water, this is to rid culture of most contaminants. From day two yeast is fed at 1.5 g/10^6 rotifers and emulsified oil at 3 ml/10^6 rotifers daily until rotifer density reaches 50/ml. Then yeast is added at 1-1.3 g/10^6 rotifers and emulsified oil at 2 to 3 ml/10^6 rotifers daily until density reaches 100/ml. Then yeast is fed at 0.6 to 1.0 g/10^6 rotifers and emulsified oil at 2 ml/10^6 rotifers until density reaches 150 to 200/ml at which time harvesting may begin (Figure 1).

Harvest

Drain 20 to 25 cloth to collect rotifers. Refill tanks with filtered sea water adjusted to a salinity of 18 ppt. Add yeast at 0.6 to 0.8 g/10^6 rotifers and emulsified oil at 2 to 3 ml/10^6 rotifers. Repeat daily unti rotifer population declines. To count rotifers, take a subsample of the culture and count the number of rotifers in at least three one milliliter samples. This gives the average number of rotifers per milliliter. When this is multiplied by the volume of the rotifer tank, it gives you the number of rotifers in the tank. If the rotifer population is very high, the sample will need to be diluted before counting.

Precautions and Limitations

- Ciliates can be a major problem, especially when using yeast and emulsified oil as a food source. Even if ciliates are not numerous they still can cause a decrease in rotifer density, as they will compete with rotifers for food. If ciliate numbers are high they must be filtered out using a 60-micron filtering cloth.
- The yeast and emulsified oil that are not ingested by the rotifers can cause water quality problems which may cause a decrease in the rotifer density. Debris can be removed by vacuuming the bottom of the tank.

Method Using Algae (*Isochrisis galbana*), Yeast and Emulsified Oil

Set up 1,800-liter tank as described above and inoculate with a 12-liter carboy of algae and algae media (0.2 ml/L of f/2 media) as shown in Figure 2. Methods of algae culture are discussed in the paper by Treece and Wohlschlag.

On the second and third days, add 0.1 ml/L of F/2 media and when the algae density reaches 132,000 cells/ml, inoculate rotifers at 1-10/ml. When the concentration of algae decreases, begin adding yeast at 50 g/tank and start adding emulsified oil at 1-2 ml/10 L each day.

When rotifer density reaches 100/ml or more, increase the daily amount of yeast to 0.7 to 1.0 g/10^6 rotifers and emulsified oil to 2 to 3 ml/10^6 rotifers. Harvesting rotifers can begin when the density reaches 200 rotifers/ml. This is accomplished by draining 15

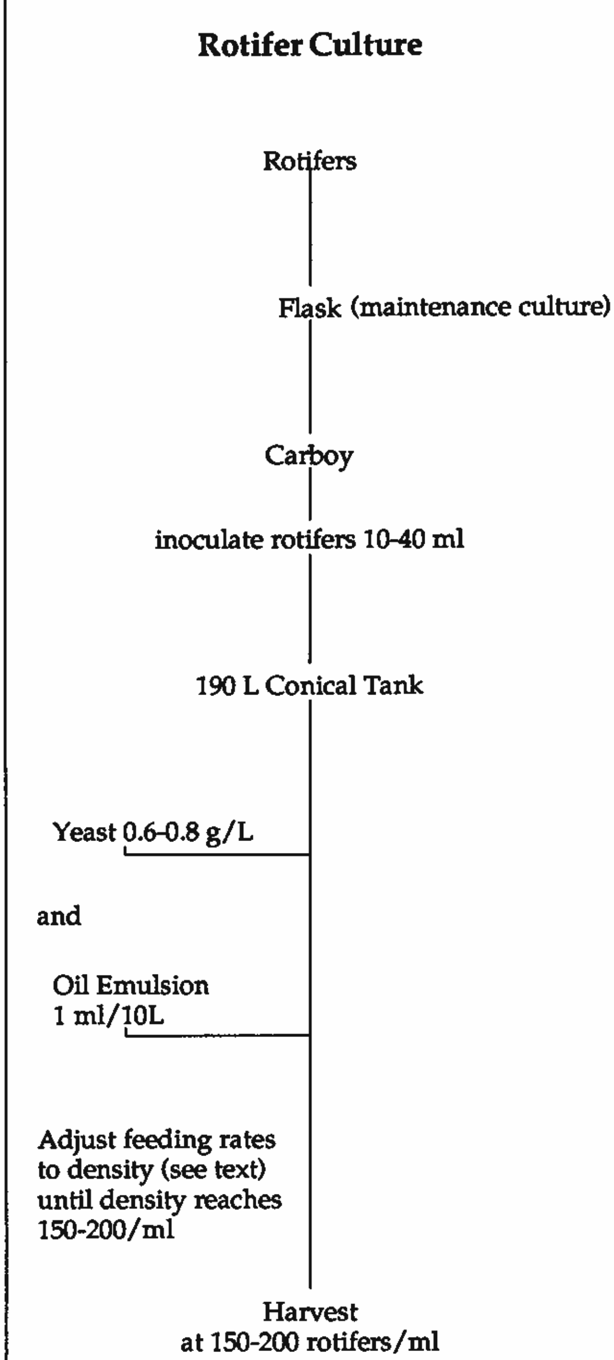


Figure 1. Diagram of rotifer culture using bakers yeast and emulsified fish oil.

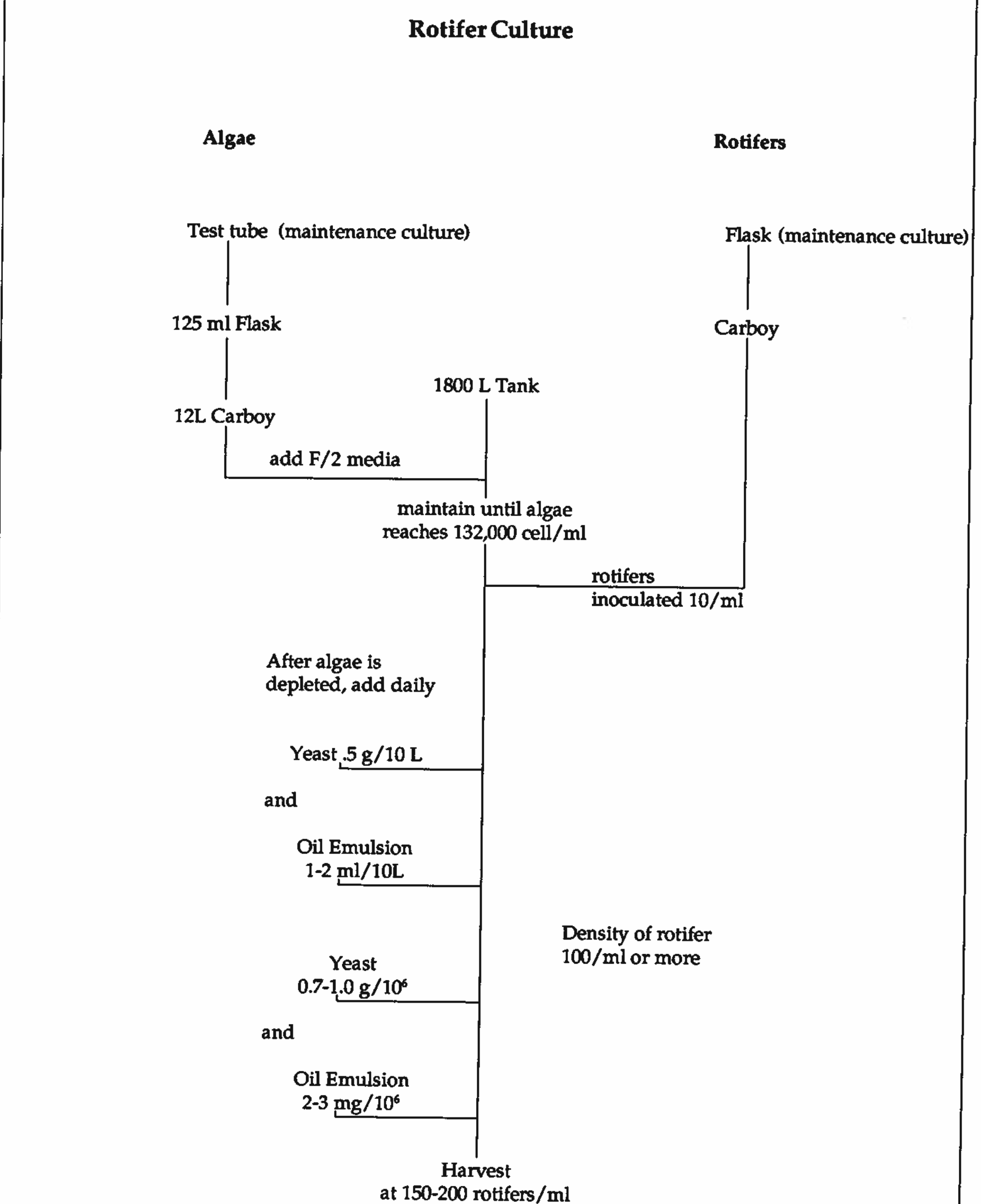


Figure 2. Diagram of rotifer culture method using algae, yeast and emulsified fish oil.

to 25 minutes and collecting rotifers in 60-micron filtering cloth daily. Repeat until the rotifer density drops. This culture method should maintain rotifer densities at 150 to 200/ml for about 30 days.

Precautions and Limitations

- When using algae and algae media,contaminants sometimes appear in tanks. Some kinds of algae – such as blue greens (Cyanobacteria*), – can be harmful to rotifers. The tank must be drained, cleaned and restarted.
- Tanks must have light banks for algae growth.
  *Note: Some blue-green algae can be and is used as food for rotifers (Snell *et al.*, 1983).

Advantages

- Algae can improve water quality.
- It is not solely dependent on algae as food source.
- Improved nutritional value.
- Increased production.

Methods Using Algae as Sole Nutrient Source

When using algae, two methods can be used: algae cultured separately from rotifers or both in the same culturing container. The University of Texas at Austin, Marine Science Institute has used modifications of both methods but has found for our purposes the above methods were more suited for our laboratory.

Algae and Rotifers Cultured Separately, Continuous Culture

Algae is cultured in an 1,800-liter tank using fish emulsion as fertilizer as described by Treece and Wohlschlag. Rotifers are cultured in separate 1800-liter tanks. Both tanks have continuous illumination from light banks over each tank and each is well aerated. Culture room is kept at a temperature of 24+2°C.

All tanks are sterilized with 2.5 ppm bleach for 12 to 24 hours, then cleaned and rinsed. Algae tanks are filled with filtered sea water (through a one-micron filter). The salinity depends on the species of algae and the strain of rotifer to be cultured, but generally is in the range of 16 to 30ppt. The tank is then inoculated with 100 liters of algae stock (130,000 cells/ml). If this amount of algae stock is not available, lower the volume of sea water in the tank. It should take three to four days to reach an algal density of 132,000 cells/ml. If a smaller volume was used, increase by doubling the volume with filtered sea water and fertilizer daily until volume of 1800 liters is reached. This culture can now be used as a nutrient for rotifer culture by draining 50 to 60 percent of the tank volume daily and refilling tank with filtered sea water and adding fertilizer. This should be done daily even if algae is not needed.

The rotifer culture tank is filled with 900 liters of filtered sea water and 900 liters of algal culture water. Rotifers are added at least 1/ml (more if available). When algae have been consumed and culture water becomes clear, rotifers may be harvested. Rotifers are harvested by draining 30 to 50 percent of tank volume and collecting rotifers in a 48-micron filtering cloth. The culture should have a density of rotifers between 100-150/ml. To maintain this rotifer count refill the tank with algal culture water and repeat daily until rotifer population declines (Arnold, *et al.*, 1976).

Precautions and Limitations

- Labor intensive.
- Dependent on one nutrient source.

Advantages

None for our laboratory.

Algae and Rotifer Cultured Together, Batch Culture

Fifty-liter polyethylene bags are used as culture container. They are clamped and attached to support frame. Illumination is supplied by 40 watt fluorescent lamps (on wall light banks). Aeration is supplemented with 1 percent CO_2 to promote algae growth. A valve is attached to the top for aeration and sea water addition and another valve at the bottom of the bag is attached for harvesting rotifers.

Bags are filled with filtered sea water. Algae media is added (f/2) and algae is inoculated at 500 cells/ml. Algae is grown for three days at which time rotifers are inoculated at 10/ml. It should take approximately four days to reach maximum rotifer density. Densities as high as 400 rotifers/ml have been achieved. Rotifers are harvested by draining the entire bag. The bag is then discarded and a new bag is used to restart a new culture (Trotta, 1981; Trotta, 1983).

Precautions and Limitations

- Requires a continuous culture of algae and rotifers (unless using commercially available rotifer cysts) as inoculating cultures.

Advantages

- Technique requires less space for culture area.
- Short time of culture growth period.
- Less care of cultures, one system for both algae and rotifers.

There are several other culture methods (Lubzens, 1987). Each method can be modified to suit the needs of the hatchery.

Feeding Rotifers to Larval Fish

Larval red drum begin feeding about day-three after hatching with the development of their mouth parts (earlier if high temperatures, later if low temperatures). Rotifers are fed to fish at this time at a rate of three to five rotifers/ml until larger foods can be consumed by larval fish (Holt *et al.*, 1981).

To calculate the number of rotifers needed for the larval fish tank, you need the number of rotifers harvested (density of rotifers per ml times volume of culture drained) and the volume of fish tank. If the tank has a volume of 200L you need 600,000 to 1,000,000 rotifers to have three to five rotifers/ml.

Due to the loss of the nutritional value of rotifers over six hours after they have been harvested, it best to feed rotifers to fish at least two times a day, or whenever rotifer density drops below 3/ml (Gatesoupe and Robin, 1982).

Obtaining Stock Cultures and Nutrients

• A culture of the rotifer Brachionus can be obtained from Carolina Biological Supply Company, 2700 York Road, Burlington, N.C. 27215, 800/334-5551. Also, Florida Aqua Farms, Inc., 5532 Old St. Joe Road, Dade City, Fla. 33525.

• Algae culture information can be found in the previous paper by Treece and Wohlschlag.

• Dry bakers yeast can be purchased from wholesale grocery companies in most cities. They come in 1 to 2 pound bags and 20 to 35 pound bags. We found three brands available in Texas. SAF-Instant, distributed by SAF Products Corporation, Peavey Center, 11 Peavy Road, Chaska, Minn. 55318, 612/448-7005 and 800/428-8459. The nearest office to Texas is 200 East Reynolds Road, Lexington, Ky. 40503, 606/273-8529, 800/626-6355. The second brand is Red Star Bakers Instant, distributed by Universal Foods Corporation, Milwaukee, Wis. 53201 and Amelia Street, Dallas, Tex. 75235, 214/631-6376. The third brand is Fleischmann's Active Dry Yeast, distributed by Standard Brands, Inc., New York City, N.Y. 10022, 800/932-7800. The Texas distributor is Nabisco Brands, Inc., 3001 LBJ Freeway, Dallas, Tex. 214/484-1544. Yeasts can also be purchased from Florida Aqua Farms, Inc.

• The emulsified oil adds essential fatty acids and vitamins not found in yeast. Watanabe *et al.* (1983) and Gatesoupe, and Luquet (1981) showed that fatty acids will improve rotifer growth and/or larval fish growth. This is a mixture of oil (cod liver oil or menhaden oil), raw chicken egg yolk, vitamin E (Tocopherol) and a vitamin mix (AIN Vitamin Mixture 76). The eggs can be purchased in grocery stores. The oil, vitamin and vitamin mixture can be purchased from ICN Nutritional Biochemicals, Customer Services Department, 26201 Miles Road, Cleveland, Ohio 44128, 800/321-6842. Va. 22539.

Emulsified oil is a mixture of sea water, fish oil, egg yolk at a ratio of weight/volume of oil mixture and vitamin E added at 0.1 oil mixture. The mixture is blended for two minutes in a blender and then is stored in a refrigerator up to one week.

Literature Cited

Arnold, C.R., *et al.* 1976. Methods and techniques of spawning and rearing spotted seatrout. (section on production of rotifers). Southeastern Association of Game and Fish Commissioners, Thirtieth Annual Conference.

Gatesoupe, F.J. and Luquet, P., 1981. Practical diet for mass culture of the rotifer, *Brachionus plicatilis*: application to larval rearing of sea bass, *Dicentrarchus labrax*. **Aquaculture**, 22:149-163.

Gatesoupe, F.J. and J.H. Robin. 1981. Commercial single-cell proteins either as sole food source or in formulated diets for intensive and continuous production of rotifers (*Brachionus plicatilis*). **Aquaculture**, 25:1-15.

Gatesoupe, F.J. and J.H. Robin. 1982. The dietary value for sea bass larvae (*Dicentrarchus labrax*) of the rotifer, *Brachionus plicatilis*, fed with or without a laboratory-cultured alga. **Aquaculture**, 27:121- 127.

Hirata, H., S. Yamasaki, T. Kawaguchi and M. Agawa. 1983. Continuous culture of the rotifer, *Brachionus plicatilis*, fed recycled algal diets. **Hydrobiologia** 104:71-75.

Holt, J., R. Godbout and C.R. Arnold. 1981. Effects of temperature and salinity of egg hatching and larval survival of red drum, *Sciaenops ocellata*. **Fishery Bulletin**, Vol. 79, No. 3.

Lubzens, E. 1987. Raising rotifers for use in aquaculture. **Hydrobiologia** 147:245-255.

Scott, A.P. and S.M. Baynes. 1978. Effect of algal diet and temperature on the biochemical composition of the rotifer, *Brachionus plicatilis*. **Aquaculture**, 14:247-260.

Snell, T.W., C.J. Beiberich and R. Fuerst. 1983. The effects of green and blue-green algal diets on the reproductive rate of the rotifer, *Brachionus plicatilis*. **Aquaculture**, 31:21-30.

Snell, T.W. and K. Carrillo. 1984. Body size variations among strains of the rotifer, *Brachionus plicatilis*. **Aquaculture**, 37:359-367.

Treece, G.D. and N.S. Wohlschlag. 1987. Algae culture for a redfish hatchery. In press.

Trotta, P. 1981. A simple and inexpensive system for continuous monoxenic mass culture of marine micro algae. **Aquaculture**, 22:383-387.

Trotta, P. 1983. An indoor solution for mass production of the marine rotifer, *Brachionus plicatilis* Muller fed on the marine microalga *Tetraselmis suecica* Butcher. **Aquacultural Engineering** (2):93-100.

Watanabe, T., T. Tamiya, A. Oka, M. Hirata, C. Kitajima and S. Fujita..1983. Improvement of dietary value of live foods for fish larvae by feeding them on three highly unsaturated fatty acids and fat-soluble vitamins. **Bulletin of the Japanese Society of Scientific Fisheries** 49(3):471-479.

Yufera, M. 1982. Morphometric characterization of a small-sized strain of *Brachionus plicatilis* in culture. **Aquaculture**, 27:55-61.

Raising Food Organisms for Intensive Larval Culture: III. Artemia

Granvil D. Treece
Sea Grant College Program
Texas A&M University
College Station, Texas 77843-4115
and
Nancy Wohlschlag
Division of Life Science
The University of Texas at San Antonio
San Antonio, Texas 78285

Adult *Artemia* (brine shrimp) were first described by Schlosser in the mid-1700s. Long before being scientifically described, brine shrimp had been associated with better salt production in brine pools (Clark and Bowen, 1976). Seal (1933) and Rollefsen (1939) reported the value of freshly hatched *Artemia* nauplii as food for fish fry, and even since, the exploitation of the cysts has gradually increased.

Until the late 1970s, commercial supplies of cysts were available only from the United States and Canada. Salt pools, ponds, lakes or salinas with *Artemia* populations are found worldwide; the distribution of these salinas is not continuous. Certain bodies of salt water lack brine shrimp either because of a failure of natural dispersion or because of periodically unfavorable climatic conditions. However, their distribution is rapidly expanding. Since 1977, successful inoculations have been achieved in areas that previously did not have *Artemia* populations (Brazil, India, the Philippines, Thailand and other countries). To give one example, nauplii out of 250 grams of San Francisco Bay cysts were introduced in a limited number of evaporation ponds on a brine production farm in the Rio Grande de Norte area, Macau, Brazil. The ecological conditions in the Macau salinas, with intake waters coming from a rich mangrove area, turned out to be very favorable for *Artemia* production. The population spread over several thousand hectares and within one year over 15 metric tons of cysts were harvested. For several years, the yields exceeded 30 metric tons of cysts per year (Sorgeloos *et al.*, 1986).

The technical feasibility of *Artemia* production in temporary salt ponds was demonstrated in Southeast Asia by Sorgeloos (1978). Inoculation tests with various geographical strains provided new information on genotypes and phenotypes (Vanhaecke and Sorgeloos, 1979). The application of these inoculation principles helped alleviate a cyst production problem which occurred in the late 1970s. Although cyst production was the primary goal, the exploitation of the adult *Artemia* biomass as a protein source for many aquaculture organisms was also vigorously pursued. By 1979, previous shortages were over and the price of high quality cysts dropped from $US32 per pound to $US16 per pound, or $US35 per kilogram.

Today, there are many geographical strains of *Artemia*. More than 50 have been registered from countries such as Algeria, Argentina, Australia, Brazil, Bulgaria, Canada, China, France, India, Israel, Iran, Iraq, Italy, Japan, Kenya, Mexico, Peru, Puerto Rico, Spain, Tunisia, United States, U.S.S.R., and Venezuela. Numerous commercial harvesters and distributors exist that sell brands of different qualities. The present cost of good quality cysts can range from $12 to $30/lb. ($26 to $66/Kg) and the buyer can expect to have 200,000 to 300,000 nauplii hatch from each gram of cysts.

Life History Aspects and Research on *Artemia*

In nature, *Artemia*, are found only in natural or man-made brine lakes, salterns (also called salinas) and have been observed swimming among precipitated crystals of sodium chloride in saturated brine (Conte et *al.*, 1972). In the laboratory, however, *Artemia* are extremely euryhaline, withstanding salinities from 3 ppt to 300 ppt (Bayly, 1972). It has been suggested that the reason brine shrimp survive only in high salinities in nature is because of the absence of competitors and predators (Morris, 1956).

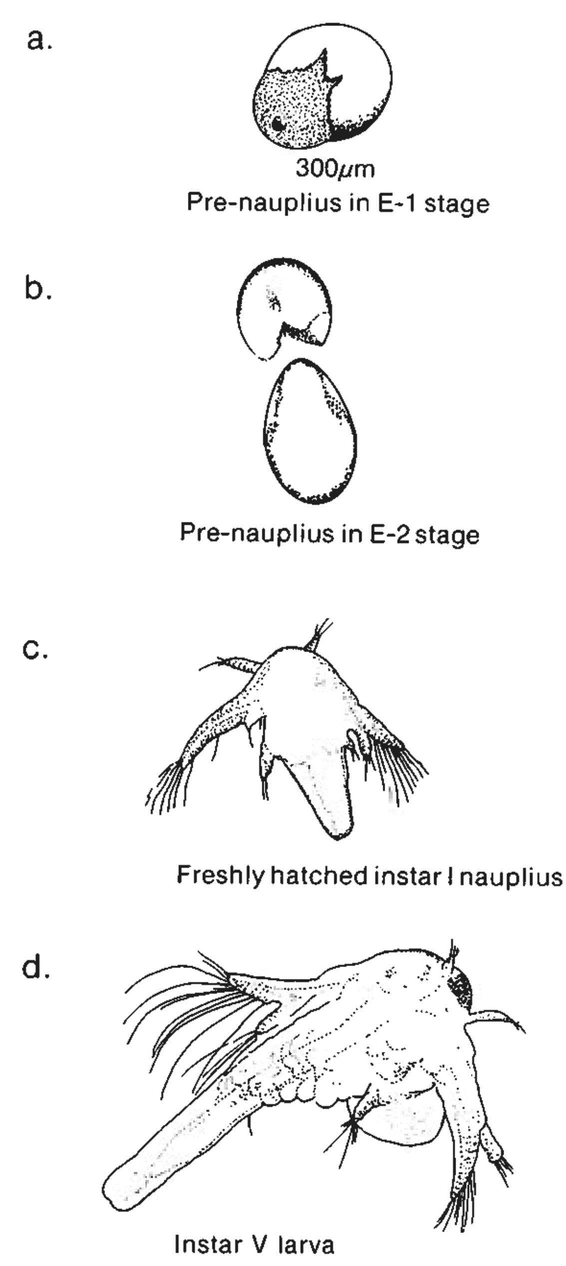
Artemia show good survival in temperatures ranging from 15° to 55°C (McShan *et al.*, 1974). The animal is also quite tolerant of high ammonia levels, remaining active at levels near 80 to 90 ppm (4.4 to 5.0 mg atoms NH_4 - N/liter) (Bossuyt and Sorgeloos, 1978, 1980). Brine shrimp show good growth on a variety of dried, frozen and live microscopic algae, yeast and bacteria; and it has even been grown on organic aggregates formed by the bubbling of water.

Larvae and adults can be reared at extremely high densities, e.g. 3,000/liter, with only a moderate amount of special treatment such as feeding them plenty of algae (Sorgeloos, 1976 and Solomon, 1980). The environmental and nutritional requirements of brine shrimp remain approximately the same throughout their life. Then can achieve sexual maturity two weeks after hatching, and are able to produce 40 larvae per day throughout their six months to one year life span. Food conversion efficiencies are high in comparison to other animals, ranging between 20 to 80 percent.

The value of *Artemia* in aquaculture is due to the unique characteristics of its reproduction, development, and physiology (Sorgeloos and Personne, 1975). Brine shrimp have two modes of reproduction: 1) ovoviviparous, when nauplii hatch in the ovisac of the mother and are born live; 2) oviparous, when embryos at the gastrula stage of development are encased in a hard capsule, or cyst. The dehydrated cyst can be stored for months or years without loss of hatchability (Dutrieu, 1960). The cyst is 200 to 300 micrometers in diameter, depending upon the strain. Its external layer is composed of a hard, dark brown, lipoproteinaceous chorion (Anderson *et al.*, 1970). Osmotic withdrawal of water, dehydration by air, or anoxia causes the encysted embryo to enter little or no sigh of life. The cryptobiotic state of the cyst is essentially complete desiccation, temperatures over 100°C and near absolute zero, high energy radiation and a variety of organic solvents (Clegg, 1974). Yet only water and oxygen are required to initiate the normal development of the embryo. This durable, easily hatched diapause state makes *Artemia* cysts a convenient, constantly accessible source of live animals for the redfish hatchery operator.

After being place in seawater at 28°C, within 24 hours the chorion, or hard coating of the cyst, breaks (See Figure 1a) and the embryo, still surrounded by a transparent hatching membrane, is released (Figure 1b). The embryo can be seen moving within the membrane, and within a few hours, the nauplius breaks free of the hatching membrane and becomes free-swimming (Figure 1c), using specially modified antennae for locomotion and food-filtering (Sorgeloos, 1979). The nauplii can live on yolk and stored reserves for up to five days (Olson, 1979), but its caloric and protein content constantly diminish during this time (Benijts *et al.*, 1976).

The lipid level and fatty acid composition of newly hatched *Artemia* nauplii can be highly variable, depending upon the strain (Schauer *et al.*, 1980; Olney *et al.*, 1980; Seidel *et al.*, 1980, 1982; Soejima *et al.*, 1980; and Leger *et al.*, 1985). The presence of highly unsaturated fatty acids (HUFA) in *Artemia* has been the source of many studies. Most have revealed one thing; the level of HUFA in *Artemia* is directly related



to culture performance in larval fish and crustaceans Leger *et al.*, 1985), 1986a,b). This confirmed the theory by Watanabe *et al.* (1978), that the presence of essential fatty acids is the principal factor for the food value of brine shrimp. Low levels of HUFA resulted in low survival and previously, vary from strain to strain, but also vary within a strain from one harvest to another (Watanabe *et al.*, 1978, 1980; Leger *et al.*, 1986a). It has also been shown in other studies that the type of food consumed by the parent *Artemia* greatly influences the fatty acid profiles in the cysts, and manipulation of the food conditions has thus far been limited to small scale operations such as intensive tank systems (Vos *et al.*, 1984; Lavens *et al.*, 1986; Yates *et al.*, 1987).

The chemical breakdown of one well-known brand of *Artemia* cyst is as follows: on a dry weight basis the cysts contained 28.8 percent crude protein, 10.0 percent crude fiber and 10.0 percent crude fat. The fatty acid profile of the cyst was as follows:

Percent Unsaturated Fatty Acids		Percent Saturated Fatty Acids	
Arachidonic	1.18	Arachidic	5.13
Clupanodonic	0.47	Myristic	2.20
Linolenic	26.42	Palmitic	10.96
Oleic	27.09	Stearic	7.28
Palmitoleic	6.23		

It is important that the *Artemia* nauplii are harvested and fed to the fish larvae in their most energetic form, i.e. as soon as possible seawater at room temperature (mostly in outdoor conditions) results in a continual decrease in the energy content of the nauplii, since they cannot is significantly lower when older, starved nauplii are used (instar II-III stages) versus freshly hatched instar I nauplii (Figure 1c). Problems are not only associated with the decrease in nutritional value (drop dry weight and caloric content) but also with their increasing size (becoming too large for the fish larvae). Other problems associated with feeding older nauplii to fish larvae are: swimming rates (becoming too fast for the fish larvae to catch), and color perception (freshly hatched nauplii are dark orange and are much easier to seen than the starved nauplii, which are transparent (Sorgeloos *et al.*, 1986).

Nutritionally, *Artemia* nauplii seem to meet the necessary requirements of red drum larvae, but as previously mentioned, the nutritional value among different *Artemia* strains is highly variable. To document these variations in strains, an international interdisciplinary study on brine shrimp was initiated in 1978. Recently, several techniques were used to enhance the nutritional value of the "poorer" strains. Even though these techniques are not recommended as part of the standard procedure at this time, they may help overcome future problems with inferior cysts.

The technique for improving the nutritional value of *Artemia* nauplii consists of hatching and separating the nauplii from the debris; then holding the nauplii for up to three days in an enriched medium containing marine algae, encapsulated diets, yeast and/or oil emulsions (see Leger *et al.*, 1986c).

Considerable research has been conducted on *Artemia*. A three-volume publication resulted from an international symposium held in 1980. Dr. Patrick Sorgeloos and his group with the *Artemia* Research Center, State University of Ghent, has contributed much to our knowledge of the brine shrimp. Research directly related to finfish and crustacean aquaculture include; the artificial inoculation of ponds with brine shrimp; optimization of the use of brine shrimp cysts in aquaculture facilities; controlled mass production of *Artemia* adults and cysts; comparative studies of the various geographical strains; and many others.

The following compilation of important parameters is based largely on the work of Sorgeloos and his group. While extensive literature exists on the hatching of brine shrimp (review in Sorgeloos, 1980), the actual procedure of hatching *Artemia* cysts is simple. However, when working with large numbers and high densities of cysts, the following parameters should be considered to assure maximum hatching efficiency.

Hatching and Harvesting Techniques

Critical Parameters for Optimum *Artemia* Hatching

- Keep cysts dry. (Place in desiccator after can is open).
- Temperature: Hold constant during hatching (25° to 30°C). Below 25°C and they will hatch slowly, above 33°C, they cyst metabolism is greatly affected.
- Salinity: 5 ppt. (Use natural seawater and dilute to 5 ppt with dechlorinated tapwater) or use 2 grams of technical grade salt per liter of tapwater. Determine salinity with a refractometer. Research has shown that nauplii have a higher energy content when hatched at a lower salinity (Sorgeloos *et al.*, 1986).
- Oxygen: Oxygen levels above 2 mg/l (2 ppm). Constant aeration is necessary during hydration and hatching; it helps disperse the cysts.
- Cyst Density: 5 grams of cysts per liter of water.
- Light: A bright, continuous light above is necessary during hatching (2,000 lux, 20 cms from the hatching containers).
- Cyst Disinfection: Is highly recommended to assist with improving hatching yields and killing bacteria that are often found on the cysts and could be harmful to red drum larvae.

Precautions and Limitations

- Do not use airstones to aerate cysts because they will create foam.
- Do not put more than the recommended 5 grams of cysts per liter of water (foaming may also be caused by the addition of too many cysts in the hatching container).
- If a sufficient oxygen level cannot be maintained without foam formation or mechanical injury of hatching nauplii, add a few drops of a non-toxic, food-grade antifoaming agent (silicone).
- A buffer may be necessary during hatching to keep the pH above 8.0
- According to Sorgeloos *et al.*, 1986, some strains of *Artemia* nauplii (e.g. Chaplin Lake – Canada, Great Salt Lake – Utah, etc.) are very difficult to separate from debris, the hatched cysts and unhatched cysts. This problem may be overcome by decapsulating. The use of decapsulated cysts eliminates naupliar separation problems, reduces surface bacterial populations and makes it possible for fish larvae to ingest and digest the *Artemia* before they are hatched. One disadvantage is that decapsulated cysts are not buoyant, and will settle out if extra circulation or aeration is not provided. Most hatchery managers prefer to aerate gently and not circulate or exchange water at all during the first week since the larvae are quite fragile.

Recommended Procedures for Hatching *Artemia*

- Obtain good quality *Artemia* cysts from a reputable company that offers a hatch guarantee.
- Set up hatching containers as follows:
  a. Determine the amount of *Artemia* required according to the size and demand of the hatchery.
  b. Size your hatching containers accordingly. Sizing is site-specific, so the important parameters of the hatching procedure will be presented here, and the numbers and sizes of containers will be left to each individual hatchery. Any funnel-shaped, transparent container will do for hatching.
  c. A typical *Artemia* hatching stand is illustrated in Figure 2. Hatching containers consist of clear plastic sediment settling cones. The cones can be purchased open-ended with a plastic valve inserted at the small end for draining, or the cones can be purchased without the opening and a siphon can be used to remove hatched *Artemia* after they settle to the bottom. Aeration to the cones is provided by connecting a glass pipet to an air line and allowing the weight of the glass pipet to hold the air supply in place, at the bottom of the cone. A fluorescent tube is placed near the hatching containers.

  Conical shaped, clear plastic bags also work well as hatching containers, but the seawater has a tendency to corrode clips and metal ring stands used to support these bags. The wooden Imhoff cone support or stand (Figure 2) is superior to other arrangements which we have tried. These can be "home-made" very easily. For additional ideas on the use of larger hatching containers, see Sorgeloos *et al.*, 1986. Some hatcheries utilize clear plastic, five-gallon drinking water bottles with the bottoms removed. The bottles are inverted and a rubber stopper is firmly placed in the mouth. A hole is drilled in the stopper to allow the placement of a tight-fitting air line, which is later used as a drain tube when harvesting nauplii.
- Cyst Disinfection: Soak cysts for one hour in 20 ppm hypochlorite and tapwater or a 200 ppm mixture for 20 minutes, if in a hurry. For example, this can be done by adding four mls house-hold bleach solution to 10 liters tapwater, and this is enough solution to disinfect one Kg of cysts. Be sure to aerate the disinfection solution so that each cyst is exposed to the disinfectant.
- Pour cysts on to a 120 micrometer sieve and wash them with tapwater.
- Cysts are ready to be placed into the hatching container.
- Start aeration and leave cysts in hatching medium (5 ppt seawater) for 24 hours before taking the first harvest. Remember to keep the temperature 25°to 30°C and turn the light on (placing a 60 watt fluorescent bulb within 20 cm of small hatching containers, such as the one in Figure 2; use two bulbs for a 20-liter or 5-gallon container; use four bulbs for a 75-liter container, etc.)

Recommended Procedures for Harvesting Nauplii

- After 24 hours, remove the aeration and let settle for five to 10 minutes. A distinct separation will noted. Empty cyst shells float to the surface, nauplii have a tendency to concentrate nearer the bottom. There may also be some debris and unhatched cysts at the very bottom. Open the valve on the cone (Figure 2) to drain debris, close the valve when the newly hatched nauplii begin coming out. A graduated beaker can be used to collect the now-concentrated nauplii. The valve is once again opened and the concentrated nauplii are collected. This procedure is repeated a short time later to ensure that all of the newly hatched nauplii are harvested. Floatation of the cyst shells

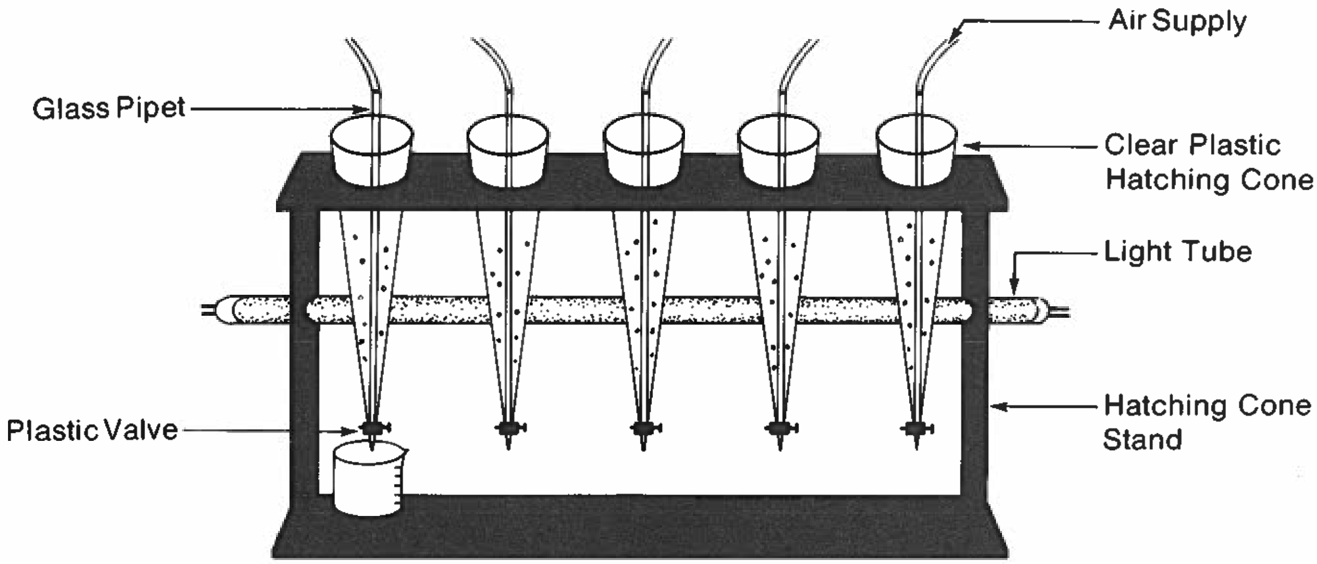


Figure 2. Artemia *hatching cones.*

can also be improved by raising the salinity. The sudden salinity change will not harm the Instar I nauplii.

Cysts from some strains do not all hatch at the same time, and sometimes a number of cysts are still unhatched after 24 hours. If this is the case, place more seawater into the hatching container, aerate, and try the harvesting routine again at 36 hours and finally at 48 hours. Nauplii can also be concentrated with light, since they are phototactic at this state. You may want to utilize this during harvesting, or just turn the overhead light off and let gravity do the concentrating.

Some strains of *Artemia* present more difficulty than others in the separation of nauplii from their old egg cases. With these particular cysts or whenever contamination with empty shells and debris becomes a problem, the use of decapsulated cysts is to be adopted. This is an added step which most hatchery managers try to avoid. The procedure is well documented in the literature (see Sorgeloos *et al.*, 1986 for a more complete discussion of decapsulation techniques).

The decapsulation procedure involves the following steps: 1) hydration of the cysts, 2) treatment in a decapsulation solution, 3) washing and deactivation of chlorine used in the decapsulation and 4) either feeding the eggs directly or will be assumed that decapsulation is not necessary and we will continue with the sequence of procedures).

- In order to prevent contamination of the larval culture tank with glycerol (which is produced by the *Artemia*), hatching metabolites, and excessive bacteria, harvested nauplii are placed on a 125 micrometer sieve and washed with tap water prior to feeding them to the fish larvae.
- Ideally, *Artemia* should be fed immediately after hatching but if this is not possible, freshly hatched nauplii should be stored in the refrigerator at 0° to 4°C in aerated containers. According to Sorgeloos *et al.* (1986) nauplii can be maintained in this state at densities up to 15,000 per ml for up to 48 hours with nauplii viability remaining more than 90 percent.
- After nauplii have been washed and concentrated into a graduated container, they can be counted by mixing the thick nauplii solution and subsampling with a graduated pipet. A count of nauplii can be made by holding the pipet horizontally and counting the number of nauplii seen swimming between two of the graduated hash marks (a known volume, one tenth of a milliliter for example) and extrapolating to give the total number of nauplii harvested. A small volume, automatic pipet can also be used to trap a known volume of concentrated nauplii.

Feeding *Artemia* Nauplii to Red Drum Larvae

Nine to 11 days after hatching, depending on the temperature and the size of the red drum larvae, a gradual transition is made to larger foods (i.e. from rotifers to *Artemia* nauplii). *Artemia* nauplii are maintained in the culture tank at densities between 0.5 to 2.0/ml.

One way to determine the amount of *Artemia* to be added to a rearing tank is by following six easy steps:

1. Number of *Artemia*/ml required = A.
2. Present density in larval rearing tank (#*Artemia*/ml) = B.

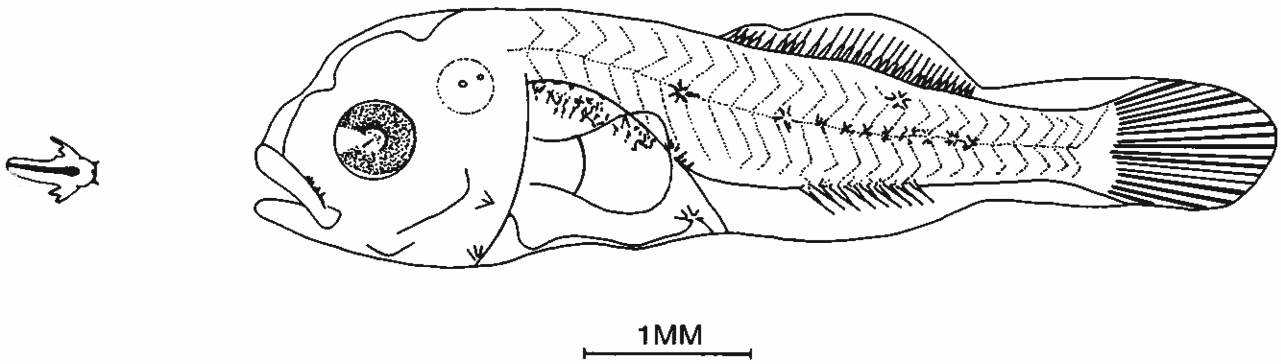


Figure 3. Schematic presentation of respective sizes of predator larval red drum (300 hours post hatch) and freshly hatched Artemia *nauplius (428 micrometers standard length)*

3. A – B = C.
4. C x (Volume of larval rearing tank in ml) - D.
5. Number of *Artemia*/liter in feed container - E.
6. D divided by E, multiplied by 1,000 = #mls of *Artemia to add.*

Artemia are fed to larval fish until they are approximately 15 to 20 days old, when they can be weaned to another less expensive food.

For the red drum larvae, a food organism has to meet certain physical and nutritional requirements. Physically *Artemia* are relatively free of extraneous material and disease producing bacteria (after separation and disinfection techniques described above). The acceptability of *Artemia* by red drum larvae is facilitated by their good perceptibility, catchability and palatability (if fed very soon after they have hatched). However, in some cases *Artemia* (even freshly hatched nauplii) can be difficult to ingest due to their size. The size of a food organism indeed determines whether a larval fish can successfully catch and ingest it. For this reason it is extremely important to continue feeding the smaller food organisms such as rotifers, while introducing the larger food organisms (*Artemia* nauplii) to the fish larvae. The rotifer is generally between 99 to 281 micrometers in length, whereas the brine shrimp nauplius is 428 to 517 micrometers (depending upon the strain). Figure 3 depicts the size of a freshly hatched *Artemia* nauplius relative to a 12 to 13 day post hatch red drum larval fish (Larval sketch was redrawn from Johnson *et al.*, 1977).

Considerable differences in sizes of *Artemia* have been found from strain to strain. Feeding an oversized *Artemia* strain may, therefore, explain poor growth and even mortality due to starvation of the predator fish larvae. Before selecting a strain of brine shrimp for use in a hatchery, it would be wise to ask other hatchery managers for their suggestions and also read the results of the International Studies on *Artemia;* the sections dealing with the culture success obtained with various marine animals fed *Artemia* nauplii from different geographical origins (results for survival and growth), in Leger *et al.*, 1986b, is particularly suggested.

Literature Cited

Anderson, E. *et al.* 1970. The Origin and Structure of the Tertiary Envelope in Thick-Shelled Eggs of the Brine Shrimp, *Artemia*. **J. Ultrastruct. Res.** 32:497-525.

Bayly, I.A.E. 1972. Salinity Tolerance and Osmotic Behavior of Animals in Althalassic and Marine Hypersaline Waters. **Ann. Rev. Ecol. Syst.** 3:233-268.

Benijts, F. *et al.* 1976. Changes in the Biochemical Composition Between the First Larval Stages of the Brine Shrimp, *Artemia salina*. *In:* **Proc. 10th Eur. Symp. Mar. Biol.** 1:1-9.

Bossuyt, E. and P. Sorgeloos. 1978. Culture of *Artemia* Larvae in Air-Water-Life Raceways with Ricebran Suspension. Unpubl. Manuscript.

Bossuyt, E. and P. Sorgeloos. 1980. Technological Aspects of the Batch Culturing of *Artemia* in High Densities. p. 133-152. *In:* **The Brine Shrimp *Artemia*. Vol. 3 Ecology**. Universa Press, Wetteren, Belgium.

Clark, C.S. and S.T. Bowen. 1976. The Genetics of *Artemia salina*. VolI. Reproductive Isolation. **J. Hered.**, 67(6):385-388.

Clegg, J.S. 1974. Biochemical Adaptations Associated with the Embryonic Dormancy of *Artemia salina*. **Trans. Amer. Microsc. Soc.** 93:481-490.

Conte, R.P. *et al.* 1972. Neck Organ of *Artemia salina* Nauplii. **J. Comp. Physiol.** 80:239-246.

Dutrieu, J. 1960. Biochemical and Physiological Observations on the Development of *Artemia salina*. (paper in French) **Arch. Zool. Exp. Gen.** 99:1-34.

Johnson, A.G. *et al.* 1977. Description of Reared Eggs and Young Larvae of the Red Drum, *Sciaenops ocellata*. *In:* **Final Report, FEDERAL AID IN FISHERIES RESTORATION ACT.** Federal Aid Project F-31-R, State-wide Fisheries Research Objective 21: MArine Fish Propagation Study.

Lavens, P. *et al.* 1986. Evaluation of Mono- and Mixed Diets as Food for Intensive Culturing of *Artemia*. *In:* ***Artemia* Research and its Applications.** Vol. 3, Wetteren, Belgium.

Leger, Ph. *et al.* 1985. International Study on *Artemia* XXV. Factors Determining the Nutritional Effectiveness of

Artemia: The Relative Impact of Chlorinated Hydrocarbons and Essential Fatty Acids in San Francisco Bay and San Pablo Bay *Artemia*. **J. Exp. Mar. Biol. Ecol.**, 93:71-82.

Leger, Ph. *et al.* 1986a. The Use and Nutritional Value of *Artemia* as a Food Source. **Oceanogr. Mar. Biol. Ann. Rev.**, 24:521-623.

Leger, Ph. *et al.* 1986b. The Nutritional Value of *Artemia:* A Review. *In*: ***Artemia* Research and Its Applications**. Vol. 3. Universa Press, Wetteren, Belgium.

Leger, Ph. *et al.* 1986c. International Study on *Artemia* XXXV. Techniques to Manipulate the Fatty Acid Profile in *Artemia* Nauplii and the Effect on Its Nutritional Effectiveness for the Marine Crustacean *Mysidopsis bahia*. *In*: ***Artemia* Research and Its Applications.** Vol. 3.

Linnaeus, C. 1758. **Systema Naturae**. Ed.X., Hafniae. 634 pp.

McShan, M. *et al.* 1974. Biological Treatment of Wastewater Using Algae and *Artemia*. **J. Water Pollut. Control Fed.** 46:1742-1750.

Morris, R.W. 1956. Some Aspects of the Problem of Rearing Marine Fishes. **Bull. Inst. Oceanogr.** (Monaco) No. 1082.

Olney, C.E. *et al.* 1980. International Study on *Artemia* VIII. Comparison of the Chlorinated Hydrocarbons and Heavy Metals in Five Different Strains of Newly-Hatched *Artemia* and a Laboratory-Reared Marine Fish. *In*: **The Brine Shrimp *Aretmia*.** Vol. 3. Universa Press, Belgium.

Olson, C.S. 1979. Timing of Developmental Events in *Artemia salina* (Anostraca). **Crustacaena** (Leiden). 36:302-308.

Rollefsen, G. 1939. Artificial Rearing of Fry of Seawater FIsh. Preliminary Communication. Rapp. P.-v. **Reun. Cons. Perm. Int. Explor. Mer.** 109-133.

Schauer, P.S. *et al.* 1980. International Study on *Artemia* IX. Lipid Level, Energy Content and Fatty Acid Composition of the Cysts and Newly Hatched Nauplii from Five Geographical Strains of *Artemia*. p. 365-373. *In*: **The Brine Shrimp *Artemia*** Vol. 3. Ecology, Culturing, Use in Aquaculture. Universa Press. Belgium.

Schlosser (in Kuenen *et al.*) 1938. Historical Notes on *Artemia salina*. **Zool. Med.**, 20:222-230.

Seal, A. 1933. Brine Shrimp (*Artemia*) as a Satisfactory Live Food for Fishes. **Trans. Am. Fish. Soc.**, 63:129-130.

Seidel, C.R. *et al.* 1980. Culture of Atlantic Silversides Fed on Artificial Diets and Brine Shrimp Nauplii. **Bull. Jap. Soc. Scient. Fish.**, 46(2)237-245.

Seidel, C.R. *et al.* 1982. Inter. Study on *Artemia* XXVI. Food Value of Nauplii from Reference *Artemia* Cysts and Four Geographic Collections of *Artemia* for Mud Crab Larvae. **Mar. Ecol. Prog. Ser.** 8(3):309-312.

Soejima, T. *et al.* 1980. International Study on *Artemia* XII. The Carotenoid Compsotion of Eight Geographical Strains of *Artemia* and the Effect of Diet on the Carotenoid Composition of *Artemia*. P. 613-622. *In*: The Brine Shrimp *Artemia*. Vol. 2. Physiology, Biochemistry, Molecular Biology. Universa Press, Belgium.

Soloman, D.W. 1980. Growth Efficiency of the Brine Shrimp *Artemia* Fed a Unialgal Diet... Masters Thesis, The University of Texas at Austin.

Sorgeloos, P. and G. Personne. 1975. Technological Improvements for the Cultivation of Invertebrates as Food for Fishes and Crustaceans. II. Hatching and Culturing of Brine Shrimp, *Artemia salina* L. **Aquaculture.** 6:303-317.

Sorgeloos, P. 1976. The Brine Shrimp *Artemia salina* L.: A Bottle Neck in Mariculture? **FAO Technical Conf. on Aqua.**, Kyoto, Japan.

Sorgeloos, P. 1978. The Culture and Use of Brine Shrimp *Artemia salina* as Food for Hatchery Raised Larval Prawns, Shrimp and Fish in South East Asia. **FAO Report** THA/75/008/78/WP/3, 50 pp.

Sorgeloos, P. 1979. The Life History of the Brine Shrimp *Artemia*. *In*: **Inter. Symposium on the Brine Shrimp *Artemis*.** Abstracts, pp. 1-6. Universa Press, Belgium.

Sorgeloos, P. 1980. The Use of the Brine Shrimp *Artemia* in Aquaculture. p. 25-46. *In* **The Brine Shrimp *Artemia*.** Vol. 3. Ecology, Culturing, Use in Aqua. Universa Press, Belgium.

Sorgeloos, P. *et al.* 1986. Manual for the Culture and Use of Brine Shrimp *Artemia* in Aquaclture. Prepared for the Belgian Administration for Development Cooperation, and FAO.; State University of Ghent. Belgium (*Artemia* Reference Center, Rozier 44, B-9000 Ghent, Belgium).

Vanhaecke, P. and P. Sorgeloos. 1979. International Study on *Artemia* XIII. The Biometrics of *Artemia* Strains of Different Geographical Origin. **Intern. Symposium on the Brine Shrimp *Artemia*.**

Vos, J. *et al.* 1984. Quality Evaluation of Brine Shrimp *Artemia* Cysts Produced in Asian Salt Ponds. **Hydrobiologia**, 108(1):17-23.

Watanabe, T. *et al.* 1978. Nutritional Quality of Brine Shrimp, *Artemia salina*, as a Living Feed from the Viewpoint of Essential Fatty Acids of Fish. **Bull. Jap. Soc. Scient. Fish.**, 44(10):1115-1121.

Watanabe, T. *et al.* 1980. Nutritional Studies in the Seed Production of Fish. IX. Relationship Between Dietary Value of Brine Shrimp *Artemia salina* and Their Content of W^3 Highly Unsaturated Fatty Acids. **Bull. Jap. Soc. Scient. Fish.** 46:35-41.

Yates, M.E. *et al.* 1987. Effect of Long Chain Highly Unsaturated Fatty Acids on Penaeid Larval Nutrition. Presented at the 18th Meeting of the World Aquaculture Society.

Saltwater Pond Fertilization

Robert L. Colura
Texas Parks and Wildlife Department
Perry R. Bass Marine Fisheries Research Station
Star Route Box 385
Palacios, Texas 77465

Production-scale pond culture of red drum fingerlings is a relatively recent development. As such, published information concerning fertilization schedules for red drum fingerling production is limited.

Traditional fertilization procedures for fresh-water hatchery operations have focused on organic fertilizers, such as alafalfa, cottonseed meal, or manure (Bonn *et al.*, 1976; Rappaport *et al.*, 1977). Accordingly, early Texas Parks and Wildlife Department (TPWD) saltwater pond fertilization strategies relied wholly upon organic fertilizers (Colura *et al.*, 1976; Colura and Matlock, 1984).

In 1975, 341 kg/ha cottonseed meal was used for pond production studies with red drum. Half was applied before filling the pond, and the remainder was added in nine equal applications over the following three weeks (Colura *et al.*, 1976). The total application rate was later increased to 568 kg/ha (TPWD, unpublished data).

More recently, combinations of organic and inorganic fertilizers have been used (Porter and Maciorowski, 1984; McCarty *et al.*, 1986). The combined fertilizer regime promotes the diverse autotrophic and heterotrophic microbial community necessary to improve zooplankton abundance, thereby improving fry survival (Geiger 1983a). Currently, the Perry R. Bass Marine Fisheries Research Station and GCCA/CPL Marine Development Center (two TPWD hatcheries) employ both organic and inorganic fertilizers, but different fertilization regimes, due to differing water sources and production goals (Tables 1 and 2).

The described regimes provide an excellent starting point for developing fertilization programs at new hatcheries. However, response to various fertilizers will vary with the time of year, salinity, temperature, residual nutrients in the ponds from previous fertilizer treatments, and ambient nutrient content of the water source (Geiger, 1983a,b; Colura and Matlock, 1984; Colura *et al.*, 1987). Therefore, hatchery managers may need to adjust fertilizer types and application rates to obtain maximum zooplankton production while maintaining adequate water quality for optimum survival and growth of the fish.

Literature

Bonn, E.W., W.M. Bailey, J.D. Bayless, K.E. Erickson and R.E. Stevens (eds.). 1976. Guidelines for striped bass culture. Striped Bass Committee of the Southern Division, **American Fisheries Society**, Bethesda, MD.

Colura, R.L., B.T. Hysmith and R.E. Stevens. 1976. Fingerling production of striped bass (*Morone saxatilis*), spotted seatrout (*Cynoscion nebulosus*), and red drum (*Sciaenops ocellatus*) in saltwater ponds. **Proc. World Maricul. Soc.** 7:79-92.

Colura, R.L. and G.C. Matlock. 1984. Comparison of zooplankton production in brackish water ponds fertilized with cottonseed meal or chicken manure. **Proc. Texas Chapter, American Fisheries Society** 6:68-83.

Colura, R.L., G.C. Matlock and A.F. Maciorowski. 1987. Zooplankton abundance in unstocked mariculture ponds at three salinities. **Prog. Fish Cult.** 49:253-259.

Geiger, J.G. 1983a. Zooplankton production and manipulation in striped bass rearing ponds. **Aquaculture** 35:331-351.

Geiger, J.G. 1983b. A review of pond zooplankton production and fertilization for the culture of larval and fingerling striped bass. **Aquaculture** 35:353-369.

McCarty, C.E., J.G. Geiger, L.N. Sturmer, B.A. Gregg and W.P. Rutledge. 1986. Marine finfish culture in Texas: A model for the future. pp 249-261 *In*: B. Stroud, ed. **The role of fish culture in fishery management. An international symposium on the use of cultured fish in fishery management.**

Porter, C.W. and A.F. Maciorowski. 1984. Spotted seatrout fingerling production in saltwater ponds. **J. World Maricult. Soc.** 15:222-232.

Rappaport, U.S. Sarig and U. Bejerano. 1977. Observations on the use of organic fertilizers in intensive fish farming at the Ginosar station in 1976. **Bamidgeh** 29:57-70.

Table 1. Red drum pond fertilization schedule used in 1988 by the TPWD at the Gulf Coast Conservation Association/Central Power and Light Co. Marine Development Center located near Corpus Christi, Texas. All fertilizer rates are calculated on per hectare basis.

Day	Treatment	Day	Treatment
1	Begin pond filling	22	56 kg cottonseed meal
2	28.4 L liguid urea[a]	27	56 kg cottonseed meal
	11.8 L phosphoric acid[b]	29	56 kg cottonseed meal
4	454.5 kg cottonseed meal	30	28.4 L liquid urea
5	28.4 L liquid urea		11.8 L phosphoric acid
	11.8 L phosphoric acid	34	56 kg cottonseed meal
10	11.8 L phosphoric acid	36	56 kg cottonseed meal
13	56 kg cottonseed meal	40	28.4 L liquid urea
15	56 kg cottonseed meal		11.8 L phosphoric acid
16	stock fish	41	Harvest
20	56 kg cottonseed meal		
21	28.4 L liquid urea		
	11.8 L phospheric acid		

[a]32 percent N
[b] 54 percent P_2O_5

Table 2. Red drum pond fertilization used at the TPWD Perry R. Bass Marine Fisheries Research Station, Palacios, Texas. All fertilizaer rates are calculated per hectare.

Day	Treatment
1	Spread 282 kg cottonseed meal (CSM) on the dry pond bottom. Fill to approximately 100 cm deep
3	Continue filling. Add 9 L phosphoric acid[a] and 4.6 kg urea[b]
7	Spread 31.3 kg CSM
10	Spread 31.3 kg CSM, stock fry
12	Spread 31.3 kg CSM, 3 L phosphoric acid, 4.6 kg urea
15	Spread 31.3 kg CSM
17	Spread 31.3 kg CSM
19	Spread 31.3 kg CSM, 3 L phosphoric acid, 4.6 kg urea
21	Spread 31.3 kg CSM
23	Spread 31.3 kg CSM
24	5.7 kg/ha salmon starter
25	Spread 31.3 kg CSM, 3 L phosphoric acid, 4.6 kg urea

[a] 55 percent P_2O_5
[b] Urea 45 percent N

Zooplankton Composition and Dynamics in Fingerling Red Drum Rearing Ponds

Leslie N. Sturmer
Harbor Branch Oceanographic Insitution Inc.
Oyster Farm Project
St. George Island
P.O. Box 581
Eastpoint, Florida 32328

Red drum larvae, as most marine fish larvae, are planktonic predators. Successful pond production of red drum depends upon the availability of suitable live food organisms of the proper size and in sufficient numbers. This ensures initiation of feeding by larvae or fry and subsequent growth and survival during the 30- to 35-day rearing period. Consequently, red drum fingerling culture requires the management of the lower trophic levels of planktonic food chains in saltwater ponds.

Zooplankton Communities

Although bacteria and protozoans may be important in the diet of larval red drum (particularly for first feeding larvae) zooplankton are their major food source until they reach approximately 1-1/2 inches. Phytoplankton are of importance as they represent the base of the food chain, which culminates in red drum production (Figure 1).

The dominant zooplankton groups in rearing ponds include the Rotifera (rotifers) and a sub-class of the Crustacea, the Copepoda (copepods). Numerous other invertebrates, such as larval stages of barnacles, crabs, mollusks, polychaetes and aquatic insects, may be present at various times and eaten by red drum. However, rotifers and copepods remain the preferred prey(Colura *et al.*, 1976; McCarty *et al.*, 1986).

General Characteristics

Rotifers

Rotifers are distinguished by the presence of an anterior ciliated crown, or corona. When beating, it appears as a rotating wheel (Figure 2). The currents that it sets up direct food particles toward the mouth. The major part of the body, or trunk, is either elongate or saccular. The cuticle of the trunk is frequently thickened to form a conspicuous encasement called the lorica. The foot, or terminal end of the body, is narrower than the trunk region. The corona provides swimming propulsion and crawling along substrates is facilitated by the "foot." Common genera found in saltwater ponds are *Brachionus* and *Keratella*.

Rotifer species vary in size, which is expected of organisms tolerant of many enviromental variations and widely distributed. Typical lorica lengths for *Brachionus plicatilis*,a dominent species, range from 0.1 to 0.3 mm. Rotifer size is considered to be the

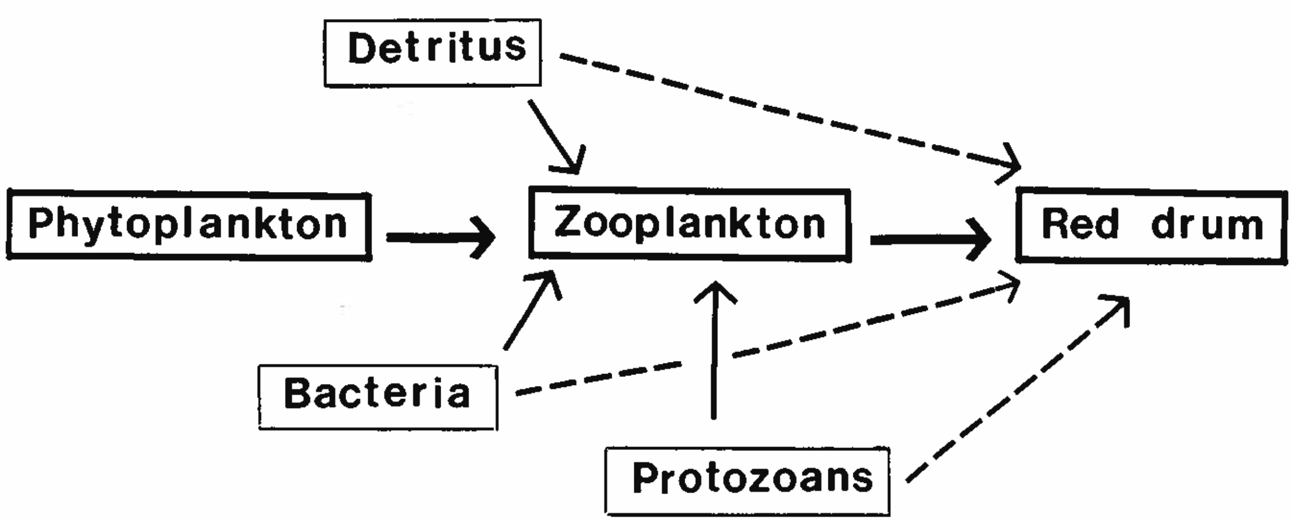


Figure 1. The food chain in a red drum fingerling rearing pond schematically represented.

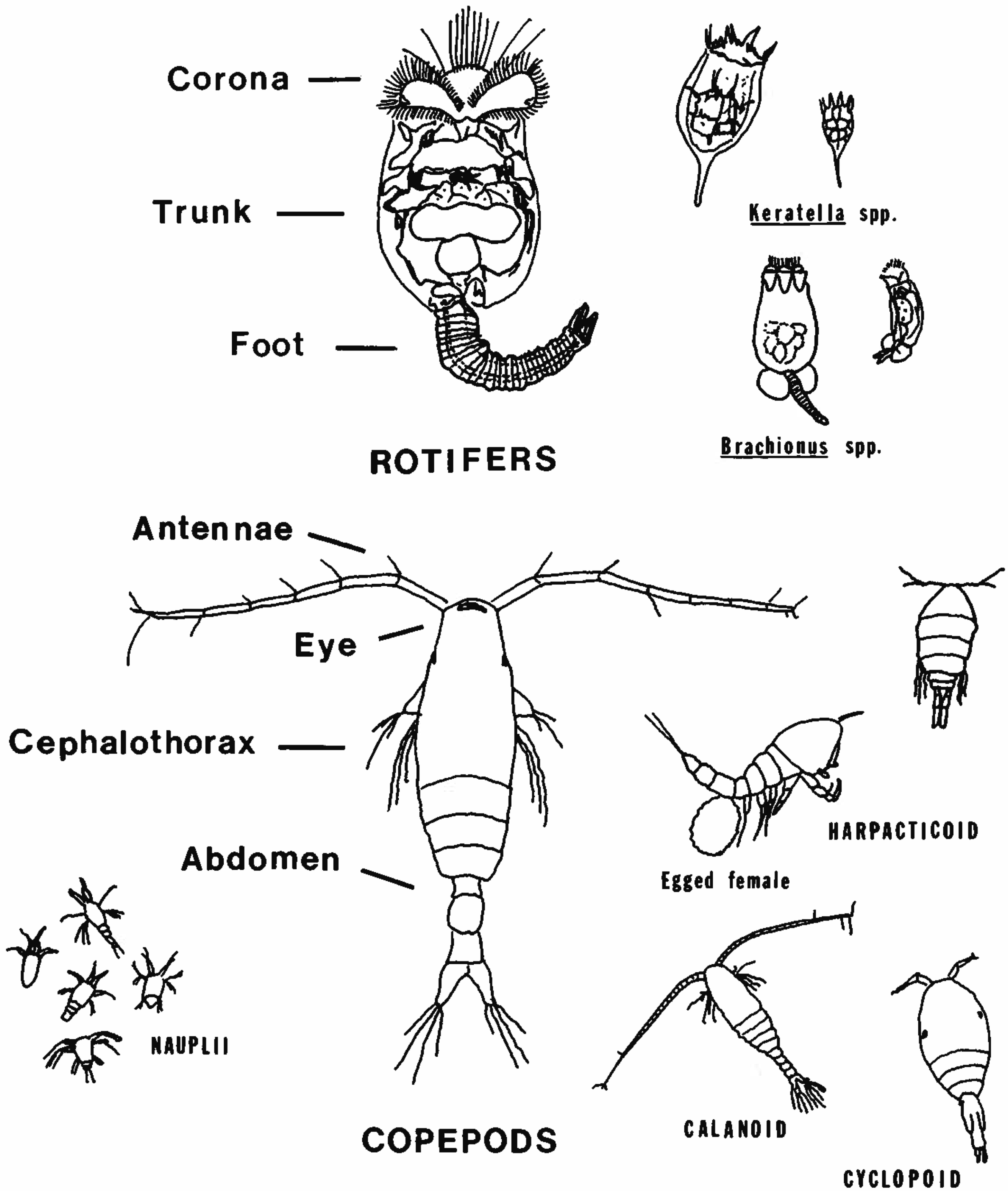


Figure 2. Distinguishing characteristics of rotifers and copepods, the two dominant zooplankton groups in saltwater rearing ponds (not to scale).

critical feature which determines their suitability as a food organism for marine fish larvae.

Copepods

Copepods are characterized by a cylindrical shape with a trunk comprised of 10 segments, consisting of head, thorax and abdomen (Figure 2). The long, first antennae and medial eyespot in the head region are perhaps the most conspicuous features. Movement is generally "jerky" and the various appendages, particularly the antennae, are used in swimming.

Copepods also exhibit a wide range of body sizes, with adults generally varying from 0.5 to 5.0 mm. The various larval stages (six naupliar and six copepodite) provide a variety of smaller forage items. The main suborders of copepods found in red drum rearing ponds include calanoids (*Acartia*, *Calanus* and *Pseudocalanus* spp.), harpacticoids (*Tisbe* and *Tigriopus* spp.), and, to a lesser extent, cyclopoids.

General information on these zooplankton communities can be found in Barnes (1968), Hutchinson (1957), Newell and Newell (1977),and Pennak (1978).

Further information on rotifers can be found in Edmondson (1965) and Walker (1981). Marshall and Orr's (1955) classic monograph is still worth consulting for an introduction to copepods. Useful information is also to be found in Wickstead (1962) and Gauld (1966). The most recent data, however, are not collected under any one title, but are found in the open literature.

Life History

Geiger (1983a,b) provides an excellent review on growth, reproduction and succession patterns of zooplankton communities. He summarizes relationships in freshwater rearing ponds as they relate to the culture of striped bass fingerlings. Cladocerans, or "water fleas," a major zooplankton group in the production of freshwater fishes, do not tolerate salinities above 2 to 3 ppt and generally are not found in saltwater ponds. Nonetheless, similarities in life history patterns are found in both the rotifer and copepod groups, whether they are from a freshwater or marine environment.

Feeding

Rotifers

Most rotifers are non-selective, suspension feeders. Their diet includes various planktonic algae, as well as bacteria, yeasts, and protozoans. *Brachionus plicatilis* filters phytoplankton less than 15 microns in size.

Copepods

Herbivorous copepods are primarily filter feeders and typically feed on particles from 10 to 50 microns. Larger particles, such as the larger diatoms which may exceed 100 microns in diameter, may also be taken, but with a different type of feeding behavior. Copepods not only show a preference for larger particles but also a greater degree of specialization in their feeding habits. When filtering, copepods can also ingest suspended detrital material, the so-called "marine snow".

In a nutrient-rich environment, such as a fertilized rearing pond, food competition should be minimal between these groups. However, as zooplankton populations become abundant following phytoplankton blooms, food shortages may eventually occur. The ability of copepods to capture a wider range of food particles, especially larger items, would allow for an advantage over rotifers during these conditions.

Reproduction

Rotifers

The complex life cycle of rotifers involves both parthenogenic (asexual) reproduction in favorable conditions, and sexual reproduction in conjunction with, or in anticipation of, unfavorable conditions. Fertilized eggs form resting eggs which contain dormant embryos. These hatch on an environmental cue. Studies suggest that high population density and changes in the quality and/or quantity of food are key factors which induce the production of resting eggs. The number of eggs per female also is a function of the favorability of the environment. For example, there is a relationship between the quantity of food

Table 1. Life history parameters of major saltwater zooplankton groups (from Allen, 1976).

Parameter	Temperature (°C)	Rotifer	Copepod
Egg-to-egg generation (days)	20	2-3	13-15
	25	1.2-1.7	7-8
Total offspring per lifespan	20	15-25	250-500
	25	15-25	500-750
Lifespan (days)	20	12	50
	25	5	40
Peak reproduction (days)	20	3.5	24.0
	25	2.0	17.5

and fecundity of rotifers (Pourriot and Snell, 1983). Less clear is the effect of the quality of food on rotifer reproduction.

Rotifers exhibit rapid development, with lifespans of five to 12 days, which compensates for their small brood size, 15-25 per female. Large increases in rotifer populations are facilitated by parthenogenesis and generation times of one to three days. Under optimal conditions, *Brachionus plicatilis* can double its population every two to three days. This life history strategy can add measurably to the zooplankton biomass in rearing ponds.

Copepods

Copepods differ from rotifers in their lack of parthenogenesis. Mating follows sexual maturity and fertilized eggs hatch into the first larval stage. Fecundity data reveal a wide variation, 250 to 750 eggs per female, as the quantity of eggs correlate with body size. Compared to rotifers, copepods have a lower potential for exponential increase as lifespans can range from 40 to 50 days. Consequently, generation times are longer and can range from seven to 12 days.

Population Dynamics

With a basic knowledge of the behavior and life histories of the two major saltwater zooplankton groups, the pond manager can at least understand and, at best, attempt to predict succession patterns in rearing ponds. When a pond is filled in preparation for stocking fry, surface water supplies provide a natural inoculum of zooplankters. Their responses to management practices are dictated by differences in feeding habits and reproductive capacities, as well as by competition and predation. Additionally, the abundance of planktonic populations is modified by a variety of physical and chemical environmental factors, such as temperature, salinity, dissolved oxygen, pH, and the availability of nutrients for the first trophic level (Geiger, 1983b).

Interaction of these two groups with each other influences trends in plankton composition. Rotifers, with their relatively short life cycle and unspecialized feeding habits, rapidly develop large populations under optimal environmental conditions. However, once carrying capacities are reached for a given environment, their numbers may either stabilize or decline. Populations of copepods, with their longer life cycles, greater capacity for selective feeding, and greater ability in predator avoidance, tend to reach their peak abundance following the rotifer peak. (Allen, 1976). Patterns in the timing and magnitude of these dynamics are seasonal as well as biogeographical.

An important factor in determining zooplankton community structure and the objective of nursery pond management is predation by larval and fingerling fishes. Marine fish larvae are "sight-feeders" ingesting individual particles selected from among available food organisms. Food selection is influenced by both food size and fish size (Houde, 1973). Fish predation can regulate mean size and species composition of the zooplankton community (Stenson, 1982). Thus, the primary concern of the pond manager is the development and maintenance of proper-sized food organisms throughout the rearing period.

Another concern in fish production is the period of initial feeding and, consequently, the time of stocking ponds. Increased fingerling survival results when first-feeding fry are stocked into ponds dominated by zooplankters of a size that can be consumed. Rotifers and the first naupliar stages of copepods are within the acceptable size range of initial food items. The density of these food items must be above some threshold level in order to minimize larval mortality. However, most culturists agree that if rotifers are the sole source of food, a higher concentration may be required for growth (Houde and Taniguchi, 1979). In addition, the impact of bacteria and protozans, generally found in large numbers in intensively fertilized ponds, during the initial phase of feeding must be considered.

Succession in Rearing Ponds

Data from several research facilities on the Gulf Coast illustrate the cyclic nature of zooplankton populations and their response to different locations, water sources, seasons, and fish species.

Zooplankton population dynamics and their effects on red drum fingerling production have been investigated at the GCCA-John Wilson Marine Fish Hatchery in Corpus Christi, Tex. (McCarty *et al.*, 1984). In 1983, the first year of production, copepods were the dominant zooplankters (Figure 3). Copepod nauplii provided an adequate forage base for first-feeding fry with densities at stock ranging from 50 organisms/liter in the summer to 135 nauplii/liter in the spring. Nauplii densities gradually increased after the second week in the ponds. The concurrent decline of copepod adults showed a switch in size preference of the fingerling fish generally around the tenth culture day. Populations of rotifers were either low or not detected in all rearing seasons, except in summer, when densities exceeded 250/liter in several ponds. Stomach analyses confirmed that red drum remained zooplanktivorous throughout the culture period.

Zooplankton data from rearing ponds at Texas Parks and Wildlife Department's Marine Fisheries Research Station in Palacios, Tex., illustrated prey response to predation by another sciaenid fish, spotted seatrout (Porter and Maciorowski, 1984). During the first culture week the smaller-sized prey were utilized with densities at stock of 220 rotifers/liter and 105 copepod nauplii/liter (Figure 4). After which,

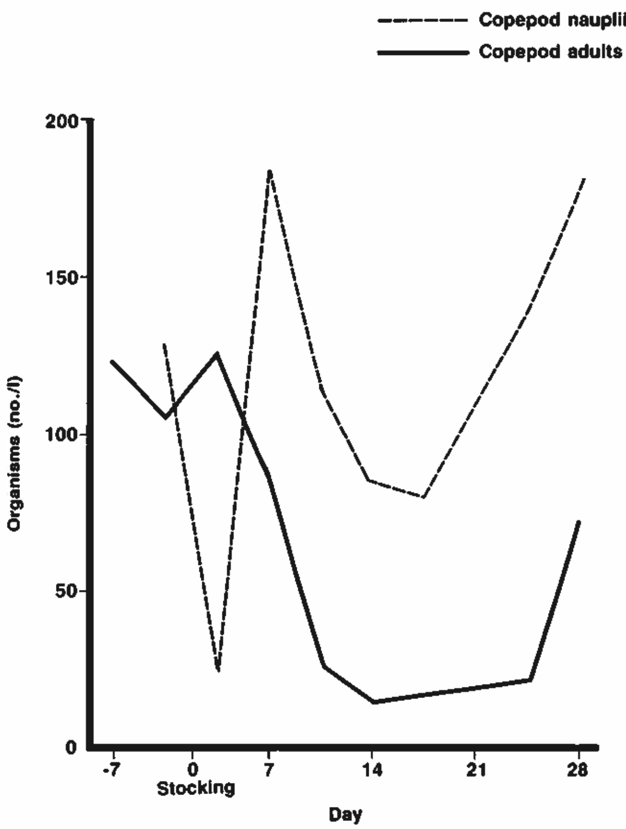


Figure 3. Zooplankton population dynamics in red drum rearing ponds at the GCCA-John Wilson Marine Fish hatchery in Corpus Christi, Tex. (from McCarty et al., 1984).

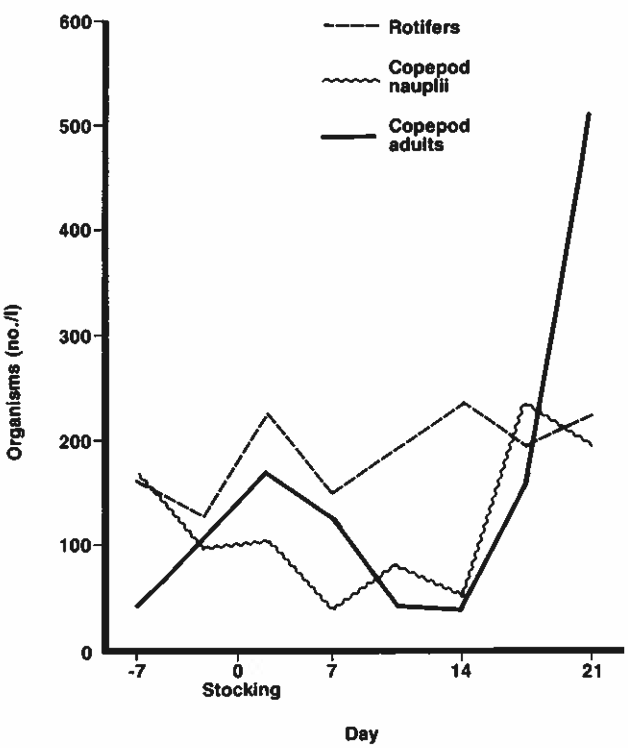


Figure 4. Zooplankton population dynamics in spotted seatrout rearing ponds at Texas Parks and Wildlife Department's Marine Fisheries Research Station in Palacios, Tex. (from Porter and Maciorowski, 1984).

populations responded by expanding. Trout larvae continued to reduce adult copepod populations through the second culture week, at which time a change in food preference was indicated by increased zooplankton densities and confirmed by an increased occurrence of insects (corixids) in the stomach contents. In contrast to red drum, trout larvae have a larger mouth gape at the on-set of feeding and prefer benthic or nektonic organisms at a smaller size (Colura *et al.*, 1976).

A similar pattern was noted in spotted seatrout ponds at the Claude Peteet Mariculture Center in Gulf Shores, Ala. (Minton, unpublished data). Initial levels of rotifers were extremely high in these ponds, ranging from 1,500 to 8,500 organisms per liter at stock (Figure 5). Trout larvae reduced rotifer populations by the third culture day, as well as the copepod nauplii in the ponds. The small peak of copepod adults (400/l) during the first culture week indicated these populations had not yet been utilized. By the tenth day, however, these larger forage organisms were eliminated. Plankton samples during the remaining rearing period reflected interspecific competition between the two zooplankton groups, with populations going through several cycles prior to termination.

Management of Zooplankton Production

Management of rearing ponds for the desired type and abundance of zooplankton during the critical stock and post-stock weeks may require one or more of the following practices: fertilization, liming, chemical treatment and inoculation. As Boyd (1979) cautioned, it is unreasonable to assume that a single management strategy would be effective for all conditions. Ponds differ greatly in morphometry, hydrology, soil chemistry, and water quality, so their responses to a given program will vary greatly. However, guidelines for establishing a suitable management program for saltwater ponds may be generated or modified from those previously reported (Porter and Maciorowsky, 1984; McCarty *et al.*, 1986). Geiger (1983a) summarized recommendations for zooplankton management in freshwater ponds.

Fertilization

The ability of water to support zooplankton depends on many factors, but the most important is the availabliity of nutrients to stimulate growth and reproduction of the food chain components. Addition of fertilizers to ponds is justified by providing these nutrients in either two forms – inorganic or organic.

Phytoplankton, the base of the marine planktonic food chain, require inorganic nutrients, carbon dioxide, water and sunlight to produce their own food. The key nutrient in regulating phytoplankton or autotrophic production in freshwater is phosphorus.

In brackish or marine systems, nitrogen is generally considered to be the limiting nutrient(Smith, 1984). This suggests that fertilization of saltwater ponds to increase algal abundance will, in general, require a fertilizer with relatively more nitrogen (Daniels et *al*., 1987).

Liquid fertilizers are recommended over the use of granular or powdered fertilizers because of their faster solubilization and more uniform distribution of nutrients in the water column (Davidson and Boyd, 1981).

Organic fertilizers consist of various animal and plant wastes and meals. The heterotrophic production of microorganisms is stimulated by the decomposition of these fertilizers. The rate at which organic matter will decompose when added to water is a function of its carbon to nitrogen ratio. For this reason, many pond managers recommend the use of cottonseed meal, chicken manure, or alfalfa meal as organic sources with low C:N ratios. Additionally, the presence of inorganic nitrogen increases the rate of decomposition. Geiger (1983a) also recommended that the organic component should be of a fine particle size to allow for faster colonization by bacteria and protozoans, as well as increased decompositon.

An optimal fertilization regime combines inorganic fertilizers to directly stimulate phytoplankton production and organic fertilizers to directly encourage growth of microorganisms, which, in turn, feed the zooplankton. Unfortunately, simple methods for precisely determining fertilizer requirements of individual ponds are not available. However, the timing and amounts of fertilizer listed in Tables 1 and 2 of the previous paper should be sufficient as a starting point. Refinements can later be made on a site-specific basis.

Liming

Many freshwater ponds with waters less than 20 mg/l total alkalinity should be limed to insure a good response to inorganic fertilization (Boyd *et al*., 1978). Liming also increases hardness, pH and phosphorus and carbon availability. This results in better phytoplankton growth, increased zooplankton production, and, ultimately, improved fish production.

Seawater is alkaline, exhibits a strong buffering capacity, and has pH values generally in the range of 7.8 to 8.2. Consequently, liming is usually not considered in saltwater pond preparation. A very important exception is when the pond is located in an area with so-called acid-sulfate soils. Liming would improve the productivity of such sites (Boyd, 1979).

Chemical Treatments

Several studies have been published on regulating zooplankton quantity and composition through the use of chemical applications. Very low concentrations of organic phosphorus acid-esters were used to kill cladocerans, but did not affect rotifers, allowing the latter to prevail in treated ponds (Tamas, 1979). The effect of the pesticide trichlorfon, commercially available as Masoten or Dylox, temporarily caused a reduction in the number of copepods while phytoplankton and rotifers were unaffected (Snow *et al*., 1980). Competitive and predacious aquatic insects and fairy shrimp were also suppressed, improving chances of striped bass fry survival in freshwater rearing ponds. While these chemical applications appear promising, it must be cautioned that at present, none of the above compounds are registered for use in food fish culture. This restricts further development of their use for zooplankton manipulation in red drum rearing ponds.

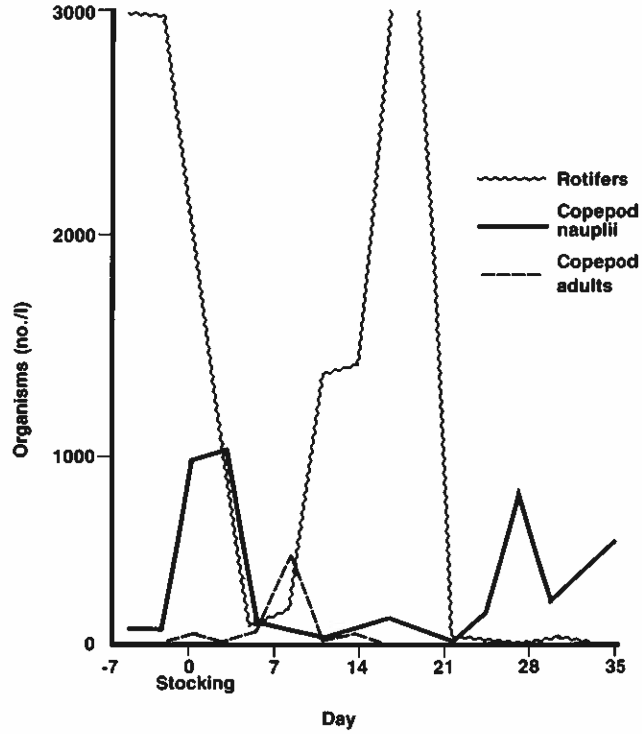


Figure 5. Zooplankton population dynamics in spotted seatrout rearing ponds at the Claude Peteet Mariculture Center in Gulf Shores, Ala. (Minton, unpublished data).

Of the compounds approved by the U.S. Environmental Protection Agency, the herbicide simizine is effective in selective control of blue- green algae. This is an important management tool, particularly in summer, when the mat-forming algae predominates. By allowing unicellular algal species an opportunity to bloom, this important trophic level for zooplankton development is not disrupted. It must be cautioned that due to the prolonged persistance of this algicide, a decrease in overall phytoplankton abundance may result and pond preparation may require a longer time period (Tucker and Boyd, 1978).

Inoculation

Dependence upon the water supply to introduce desired zooplankters when filling ponds is not always reliable, especially if the source is poor in nutrients or from a saltwater well. Inoculation of ponds with selected species at fertilization may provide greater predictability in directing the development of the zooplankton community. In addition, high post-stocking zooplankton densities are difficult to maintain after fish are introduced without re-seeding the pond. Desirable forage organisms may be made available by collecting them from preferred natural habitats or by cultivating them in separate, smaller ponds under intensive conditions. Zooplankton can also be mass-cultured using laboratory techniques developed for a number of invertebrates (Kinne, 1976; Lawrence, 1981).

Red drum rearing ponds were inoculated with mass-cultured rotifers in an attempt to increase the number of suitable-sized prey organisms at time of stocking (Sturmer *et al.*, 1985). Rotifers reared in outdoor holding troughs were fed a combination of baker's and Torula yeasts (Fontaine and Revera, 1980). Figure 6 illustrates the response of the rotifer inoculate during the pre-stocking period. Rotifer populations declined following a late spring cold front before completion of fry stocking. Rapid decreases in water temperatures have been shown to induce a switch from parthenogenic to sexual reproduction in rotifers (Gilbert, 1974). However, of the stocked ponds in which rotifers were in log-phase reproduction, red drum survival and daily production were greater than corresponding non-inoculated ponds. These data suggest that by manipulating the zooplankton community, sufficient food of an adequate size can be made available prior to the on-set of fry feeding.

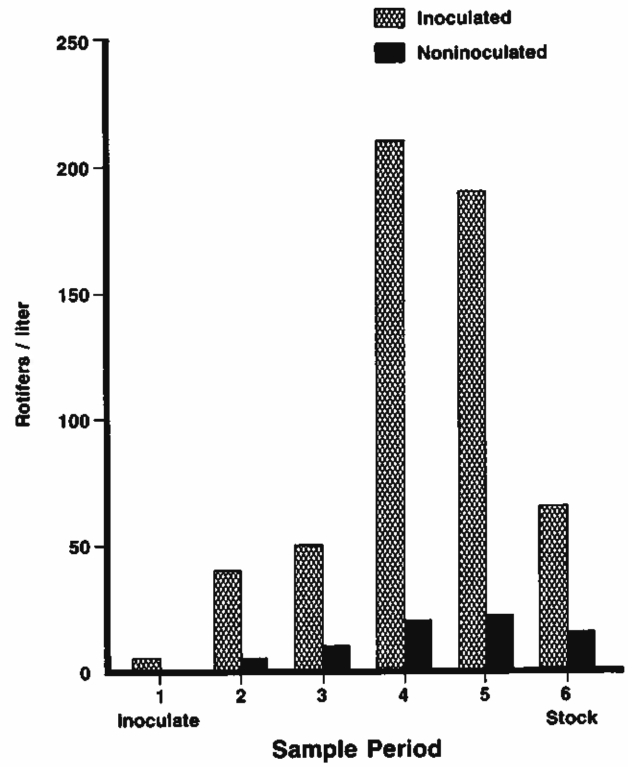


Figure 6. Response of rotifers in inoculated and non-inoculated ponds prior to stocking with red drum fry (Sturmer et al., 1985).

Zooplankton Sampling and Monitoring

Zooplankton sampling and monitoring of red drum rearing ponds can give the pond manager information by which he can make important management decisions both before and during the culture period. Knowledge of zooplankton populations aid in determining which ponds to stock as fry become available or when fertilizer applications should be made. Finally, this information is useful in determining when to harvest and in predicting harvest success. Although time-consuming, these procedures are justified as they can reduce the amount of "trial and error" associated with pond management.

Sample Gear/Methodology

The abundance of zooplankters is usually too low for accurate direct enumeration, even in fertilized ponds. As a consequence, organisms must be concentrated before counted. An integral piece of gear required for collecting and concentrating zooplankters is the plankton net. One of the most widely used devices for obtaining quantitative and qualitative samples is the Wisconsin-style plankton net obtained from Wildco Supply Company, Saginaw, Mich. (Phone: 517/799-8100). This sampler is available with either 80-micron or 153-micron Nitex netting. The smaller mesh size is recommended so rotifers and copepod nauplii are retained in the sample. A lower cost sampler (Student plankton net) is also available from Wildco Co. However, applications of this net are somewhat limited.

Sampling methods, standardized for environmental monitoring, involve pulling a plankton net through a body of water and concentrating the collected organisms. These procedures are summarized by the American Public Health Association (APHA *et al.*, 1975) and are satisfactory for large, deep bodies of water, such as lakes, reservoirs, or oceans. The types of pulls or tows described are vertical, horizontal and oblique. Of the three, the oblique tow in which the sampler is lowered to some pre-determined depth and then raised at a constant speed for a known distance is best adapted for pond monitoring. The distance the net is towed depends on the size of the pond but, in most cases, 25 to 100 feet is necessary for an adequate sample. For ease, the bridle of the net can be tied to a short pole that can be held from the pond bank. The volume (ft^3) of water filtered through the net is estimated by the formula $V = (3.14)r^2d$, where r is the radius of the net opening in feet and d is the distance in feet the net is towed. This value can then be converted to gallons ($1\ ft^3 = 7.48$ gal) or liters ($1\ ft^3 = 28.32$ l).

An alternative method, and one better suited for sampling small, shallow rearing ponds, is one in which a known volume of water is collected and then filtered through the plankton net to concentrate organisms. This is facilitated by the use of portable, flexible-impeller pumps, operated on 12-volt DC or 110-volt AC. Little Giant Pump Company, Oklahoma City, Okla. (Phone: 405/947-2511) manufactures several units (Models PPS-12 or PPS-110) with pumping rates of 20-30 l/min. Advantages of the pumping method over towing include expediency of sample time and more accuracy in determining the volume of water filtered. In addition, there is less tendency for the plankton net to become clogged with algae, detritus and mud or torn by rocks and barnacles. Geiger (1983a) in comparing the two methods found no differences in types or numbers of zooplankters. He also confirmed there was no avoidance by larger species. The only disadvantage is the initial cost of the pump ($75 to $95).

A zooplankton sampling apparatus incorporating the use of a pump was devised by Farquhar and Geiger (1984). This device consisted of a boom with a counter-weight and attached pump, mounted on a truck (Figure 7). A detailed account of construction and sampling procedures are included in the paper. Zooplankton samples were collected with the truck positioned near the pond and the boom, with an intake structure at the terminal end, extended over the sample station. By slowly raising and lowering the intake from pond bottom to surface, the entire water column was sampled. The pump discharge (vinyl tubing fitted with garden hose connectors) directed the water sample through the plankton net, suspended in a calibrated bucket. Following their techniques, two people could efficiently collect 10 samples per hour, including moving from pond to pond and sample preservation (McCarty *et al.*, 1986).

The number of samples collected per pond and the volume of water filtered per sample are related to pond size. Two samples, each 20 liters, gave an adequate assessment of zooplankton populations in 2.0-acre hatchery ponds (McCarty *et al.*, 1986). Pond sample sites were located at the shallow and deep ends on opposite corners. This strategy was devised to minimize zooplankton population variance due to depth or wind interactions. In smaller ponds, less than 0.5-acre, one sample site located at the harvest basin or deep end would suffice. If a larger sample is desired, the volume of water pumped and filtered can be increased.

The timing of sample collection is also of importance. Taking samples twice weekly allows the pond manager to determine if populations are increasing or decreasing during the pre- or post-stock weeks. To further reduce variability in the sampling regime, samples should be collected at a designated time and day.

Sample Handling/Evaluation

After sampling, a concentrated volume of zooplankters is available for evaluation. This concentrate, dependent on the amount of detrital matter or

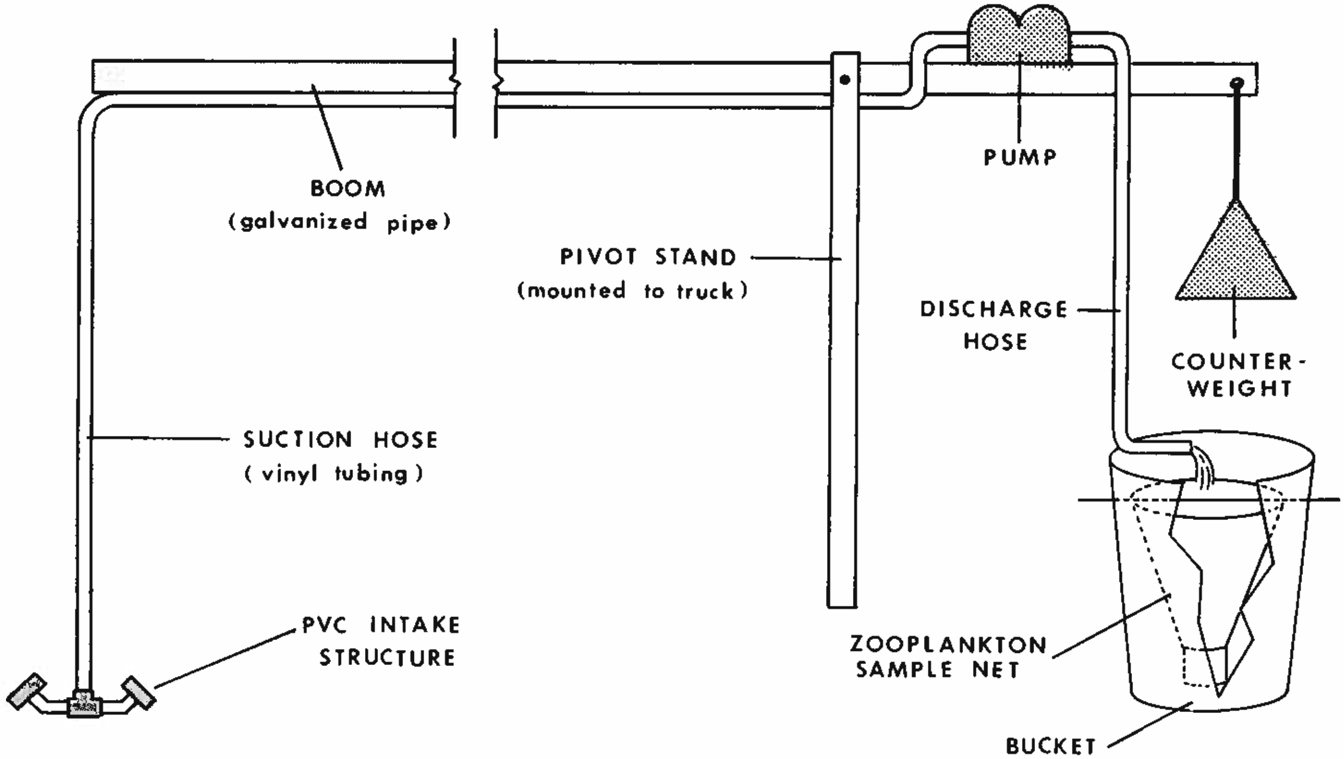


Figure 7. Portable zooplankton sampling apparatus for monitoring plankton populations in fingerling rearing ponds (modified from Farquhar and Geiger, 1984).

zooplankton densities, is usually 100 milliliters. Care must be taken to rinse the net thoroughly with filtered seawater to remove all organisms. The sample is then poured into labeled jars and preserved for later analysis. Several fixatives are recommended by APHA *et al.* (1975) and include Lugol's solution, 70 percent ethanol, or 5 percent buffered formalin. The latter is preferred and prepared by adding 2 grams of borax to every 98 milliliters of formalin (37 to 40). By adding sucrose (40 g/l) to the fixative and chilling, distortion of the organisms and loss of eggs from gravid females are prevented. In detritus-laden samples, a 0.04 percent rose bengal stain will help to differentiate zooplankters from plant matter.

The method of microscopic examination described by either the Environmental Protection Agency (1973) or APHA *et al.* (1975) may be employed for both qualitative and quantitative analysis of zooplankton. The following equipment is required: a binocular microscope with 40 to 100 X magnification; a Whipple ocular micrometer grid; a counting chamber, preferably a Sedgwick-Rafter (S-R) cell; and, a 1-ml large bore pipet or medicine dropper. The pipet is used to transfer a 1-ml aliquot of well-mixed sample into the counting chamber (Figure 8). By directing the sample diagonally across the bottom of the cell, the cover slip will rotate into place as the cell is filled. Before counting, allow the sample to stand for several minutes to permit settling. For larger organisms, such as copepod adults, the entire chamber should be counted. When smaller organisms predominate, such as rotifers or copepod nauplii, a strip method can be used. When making a strip count, two to four strips (width of Whipple grid) should be examined the length of the S-R cell, depending on the density of organisms. As the area of the S-R counting cell (50 mm long x 20 mm wide x 1 mm deep) and Whipple grid (calibrated with stage micrometer) are known, the number of organisms counted can be expanded to number of organisms per milliliter of concentrated sample. The following formula can be used:

$$\text{Number per milliliter of sample} = \frac{C \times 1000\ \text{mm}^3}{L \times D \times W \times S}$$

C = total number of organisms counted
L = length of strip (S-R cell) mm
D = depth of strip (S-R cell), mm
W = width of strip (Whipple grid), mm
S = number of strips counted

The final results should be expressed as total organisms or number of rotifers, copepod adults, and copepod nauplii per liter of pond volume. This can be calculated by taking into account the volume of the concentrated sample, the volume of pond water filtered, and the number of zooplankters counted in the Sedgwick-Rafter cell.

Samples can also be evaluated qualitatively to assess the reproductive state of the zooplankters. The number of eggs per rotifer or copepod female can be used as indicators of upcoming population peaks.

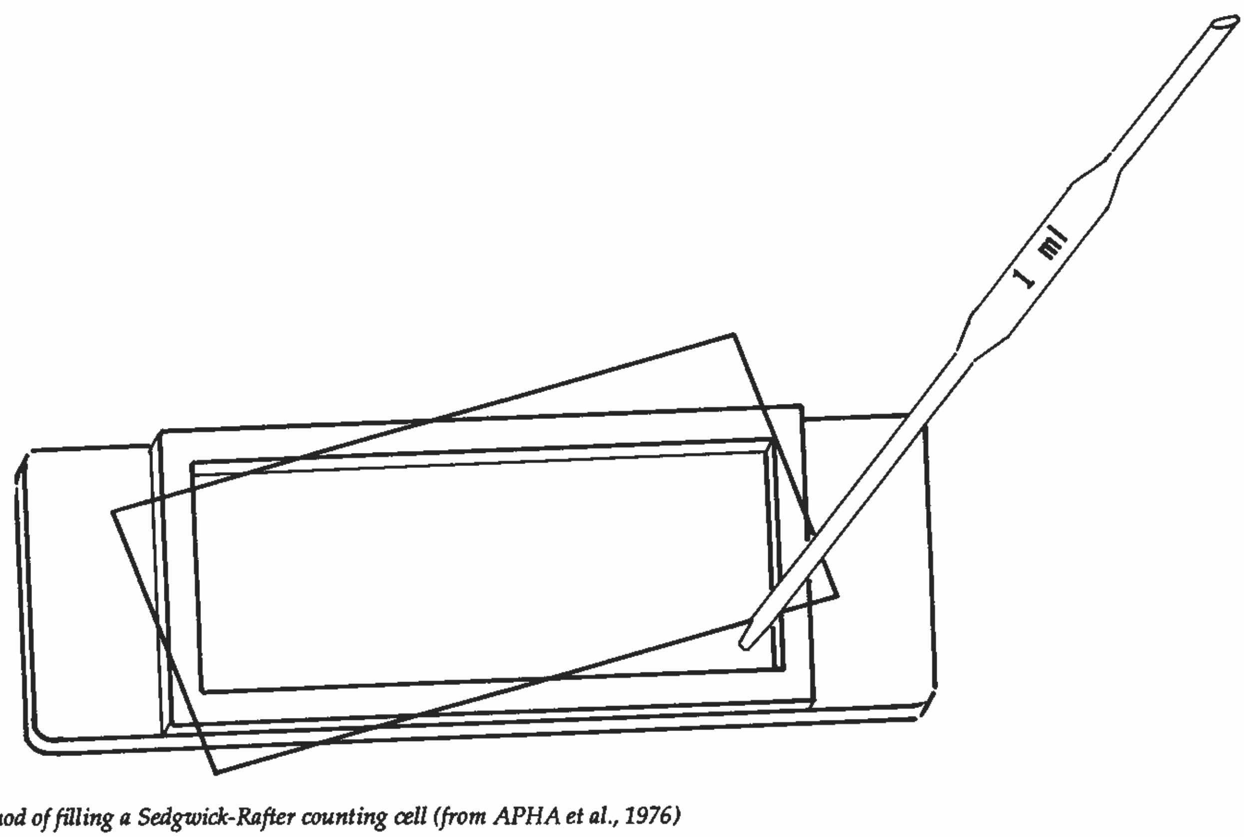


Figure 8. Method of filling a Sedgwick-Rafter counting cell (from APHA et al., 1976)

Appearance of rotifer resting eggs, indicating a switch from parthenogenetic reproduction, is associated with adverse environmental conditions.

Identification of the predominate zooplankters to family or genus, if possible, can be conducted to understand zooplankton community structure or succession patterns in ponds further. Taxonomic references recommended are Davis (1955), Edmondson *et al.* (1959), Hyman (1951), and Pennak (1978). Local keys or references are recommended for further speciation.

Precautions and Limitations

- As cautioned previously, the variation associated with pond culture is great. Pond management recommendations should be developed specifically for each location with several alternatives proposed. Zooplankton responses in composition and abundance to selected management decisions can vary not only from site to site but also from pond to pond.
- In spite of the acknowledged benefits of zooplankton monitoring to the pond manager, the expenditure of time involved in sampling and processing of samples is costly.
- There is also a need to further identify measurements of the production process involving critical links in the food chain. Conceptual and mathematical models of production dynamics that have been developed for marine ecosystems should be modified and applied to the special case of the pond system. This would facilitate effective management of commercial rearing ponds.

Acknowledgements

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References

Allan, J.D., 1976. Life history patterns in zooplankton. **Am. Nat.**, 110:165-180.

APHA (American Public Health Association), American Water Works Association, and Water Pollution Control Federation, 1975. Standard Methods for the Examination of Waters and Wastewater, 14th ed. American Public Health Association, Washington, D.C., 1193 pp.

Barnes, R.D., 1968. **Invertebrate Zoology**. W.B. Saunders Co., Philadelphia, Pa., 743 pp.

Boyd, C.E., 1979. Water Quality in Warmwater Fish Ponds. Auburn University Agricultural Experiment Station, Auburn, Al., 359 pp.

Boyd, C.E., J.W. Preacher and L. Justice. 1978. Hardness, alkalinity, pH and pond fertilization. **Proc. Ann. Conf. S.E. Assoc. Fish & Wildl. Agencies**, 32:605-611.

Colura, R.L., B.T. Hysmith and R.E. Stevens. 1976. Fingerling production of striped bass (*Morone saxatilis*), spotted seatrout (*Cynoscion nebulosus*), and red drum (*Sciaenops ocellatus*) in saltwater ponds. **Proc. World Maricul. Soc.**, 7:79-92.

Daniels, H.V., C.E. Boyd and R.V. Minton. 1987. Effects of three nitrogen and phosphorus liquid fertiizers on water quality in brackishwater ponds. Presented at 18th annual meeting, World. Maricul. Soc., Jan. 18-23, Guayaquil, Ecuador.

Davidson, R.E.and C.E Boyd. 1981. Phytoplankton response to liquid fertilizers. **Prog. Fish-Cult.**, 43(3):126-129.

Davis, C., 1955. The Marine and Freshwater Plankton. Mich. State Univ. Press, East Lansing, Mich.

Edmondson, W.T., 1965. Reproductive rate of planktonic rotifers as related to food and temperature in nature. **Ecol. Monogr.**, 35:61-111.

Edmondson, W.T., H.B. Ward. and G.C. Whipple. (ed.) 1959. **Fresh-water Biology**. John Wiley and Sons, New York, N.Y., 1248 pp.

Environmental Protection Agency, 1973. Biological Field and Laboratory Methods for Measuring the Quality of Surface Waters and Effluents. EPA 670/4-73-001, Office of Research and Development, U.S. Environmental Protection Agency, Cincinnati, Ohio.

Farquhar, B.W. and J.G. Geiger. 1984. Portable zooplankton sampling apparatus for hatchery ponds. **Progressive Fish- Culturist**, 46: 209-211.

Fontaine, C.T. and D.B. Revera. 1980. The mass culture of rotifer, *Brachionus plicatilis*, for use as foodstuff in aquaculture. **Proc. World Maricul. Soc.**, 11:211-218.

Gauld, D.T. 1966. The swimming and feeding of planktonic copepods. *In:* **Some Contemporary Studies in Marine Science**, H. Barnes (ed.), Allen & Unwin, London, pp. 313-334.

Geiger, J.G., 1983a. Zooplankton production and manipulation in striped bass rearing ponds. **Aquaculture**, 35:331-351.

Geiger, J.G., 1983b. A review of pond zooplankton production and fertilization for the culture of larval and fingerling striped bass. **Aquaculture**, 35:353-369.

Geiger, J.G., W.P. Rutledge and H.J. Warren. 1986. Improved pond management strategies for the culture of striped bass and hybrid striped bass: a two-year case study. Presented at 17th annual meeting, **World Maricul. Soc.**, Jan 19-23, Reno, Nevada.

Gilbert, J.J., 1974. Dormancy in rotifers. **Transactions American Microscopical Society**, 93:490-513.

Houde, E.D, 1973. Some recent advances and unsolved problems in the culture of marine fish larvae. **Proc. World Maricul. Soc.**, 3:83-112.

Houde, E.D. and A.K. Taniguchi. 1979. Laboratory culture of marine fish larvae and their role in marine environmental research. *In:* F.S. Jacoff (ed.), **Advances in Marine Environmental Research.** Proceedings of a symposium: Rep. No. EPA-600/9-79-035:76-205. Environ. Res. Lab., U.S. Environmental Protection Agency, Narrangansett, R.I.

Hutchinson, G.E., 1957. **A Treatise on Limnology: Vol. I Geography, Physics, and Chemistry**. John Wiley and Sons, N.Y., 1015 pp.

Hyman, L.H. 1951. **The Invertebrates: Acanthocephala, Aschelminthes and Ectoprocta, Vol. III**. McGraw-Hill, New York, N.Y., 572 pp.

Lawrence, S.G. (ed.), 1981. Manual for the Culture of Selected Freshwater Invertebrates. **Can. Spec. Publ. Fish. Aquat. Sci.**, No. 54, 169 pp.

Kinne, O., 1976. **Marine Ecology. Vol. III. Cultivation, Part 1.** Wiley-Interscience Publication, New York, N.Y., pp. 212-216.

McCarty, C.E., L.N. Sturmer and W.P. Rutledge. 1984. Hatchery production of red drum fingerlings in Texas. Presented at 15th annual meeting, **World Maricult. Soc.**, March 18-22, Vancouver, B.C.

McCarty, C.E., J.G. Geiger, L.N. Sturmer, B.A. Gregg, and W.P. Rutledge. 1986. Marine finfish culture in Texas: a model for the future. *In* Stroud, R.R. (ed.), **Fish Culture in Fisheries Management. American Fisheries Society, Proc. of a Symposium on the Role of Fish Culture in Fisheries Management at Lake Ozark, Mo.**, March 31-April 3, 1985, pp. 249-262.

Marshall, S.M. and A.P. Orr. 1955. **The Biology of a Marine Copepod, *Calanus finmarchicus* (Gunnerus)**, Oliver & Boyd, Edinburgh, 188 pp.

Newell, G.E. and R.C. Newell. 1977. **Marine Plankton-a Practical Guide.** Hutchinson & Co., London, 244 pp.

Pennak, R.W., 1978. **Fresh-water Invertebrates of the United States.** John Wiley and Sons, New York, N.Y., 803 pp.

Porter, C.W. and A.F. Maciorowski. 1984. Spotted seatrout fingerling production in saltwater ponds. **Proc. World Maricul. Soc.**, 9:311-332.

Pourriot, R. and T.W. Snell. 1983. Resting eggs in rotifers. **Hydrobiologia**, 104:213-224.

Smith, S.V., 1984. Phosphorus versus nitrogen limitation in the marine environment. **Limnology and Oceanography**, 29(6):1149-1160.

Snow, J.R., T.A. Al-Ahmad and J.E. Parsons. 1980. Rotifers as a production diet for striped bass fingerlings. **Proc. Ann. Conf. S.E. Assoc. Fish & Wildl. Agencies**, 34:280-291.

Stenson, J.A., 1982. Fish impact on rotifer community structure. **Hydrobiologia**, 87:57-64.

Sturmer, L.N., C.E. McCarty, J.G. Geiger and W.P. Rutledge. 1985. Pond management strategies for enhanced pond production of red drum (*Sciaenops ocellatus*) fingerlings. Presented at 16th annual meeting, **World Maricul. Soc., Jan. 13-17**, Orlando, Fla.

Tamas, G., 1979. Rearing of common carp fry and mass cultivation of its food organisms in ponds. *In*: E. Styczynska-Jurewicz, T. Backiel, E. Jaspers and G. Persoone. (ed.), **Cultivation of Fish Fry and its Live Food, European Mariculture Society**, Sp. Publ. No. 4: 199-209.

Tucker, C.S. and C.E. Boyd. 1978. Consequences of periodic applications of copper sulfate and simizine for phytoplankton control in catfish ponds. **Trans. Am. Fish. Soc.**, 107(2):316-320.

Walker, K. 1981. A synopsis of ecological information on the saline lake rotifer *Brachionus plicatilis* Muller. **Hydrobiologia**, 81: 159-167.

Wickstead J.H. 1962. Food and feeding in pelagic copepods. **Proc. Zool. Soc. London**, 139:545-555.

Redfish Fingerling Harvest

Richard Fernandez
Texas Parks and Wildlife Department
John Wilson Marine Fish Hatchery
4300 Waldron Road
Corpus Christi, Texas 78418

Abstract

Redfish fingerlings are hardy and tend to be less susceptible to stress than many other species. Generally, larger redfish fingerlings adapt better to stress than smaller ones.

Fingerlings at the John Wilson Marine Fish Hatchery are harvested approximately 30 days after stocking. Prior to harvest, samples are collected at the harvest box on a weekly basis. Several factors are considered prior to harvest: pond zooplankton levels; length; weight; and conditions of fish. Optimum harvest length is 30 to 35 mm (1 1/2 inches) or about 1,200 to 1,300/lb.

Catch basins, or kettles, are highly recommended for the collection of redfish fingerlings. Basins and kettles differ in design but all should have a water source at, or within, the basin to ensure good water quality and prevent low dissolved oxygen problems at time of harvest.

When draining ponds, it is essential to drain slowly at night to prevent fish from being impinged on the screen. Drain valves are shut during the night prior to harvest and water sprayed into ponds to ensure that dissolved oxygen levels remain high. Ponds are then started down about 4 a.m. and harvested before 10 a.m., thus avoiding the midday rise in water temperature, which can be stressful. During harvest, fish are dipped from the kettle and put into pre-weighed buckets filled with water, then placed into hauling trailers. At random intervals during harvest (smaller fish enter the catch basin before larger ones do), five samples of 100 fish are collected. Samples are weighed and the number of fish per pound are estimated.

Transportation and Acclimation of Red Drum Fingerlings

Joseph P. McCraren*
Office of Extension and Publications
Research and Development
U.S. Fish and Wildlife Service
National Fisheries Research Center
Leetown Box 700
Kearneysville, W.V. 25430

A number of questions remain to be answered before large-scale red drum production can become a reality. Among these are fingerling transport requirements and acclimation to freshwater. This paper will attempt to summarize the current state of knowledge on these particular subjects.

Much of the information in the initial section of this paper is taken from a relatively comprehensive publication on fish transportation by McCraren (1978) and McCraren and Millard (1978). That information has been updated with data from a manuscript by Carmichael and Tomasso (1988). Two additional recommended publications on the general subject of fish transportation are by Berka (1986) and Johnson (1979).

Fish Transportation

History

The first recorded attempts to transport and culture fish are from China. The Romans are also known to have cultured and transported fish. Published tales of distribution in more contemporary times must surely rival those of the Chinese and Romans in terms of ingenuity and resourcefulness. For example, there was Dr. Livingston Stone's transportation of 35,000 shad fry in milk cans by railway from the east to west coast before the turn of the century. Water in the cans was changed every two hours, if water was available. This ambitious venture resulted in the establishment of an extensive shad fishery in the Sacramento River and other Pacific coastal streams.

In the early 1880s it was understood that to haul live fish in closed containers (in this case, wooden kegs) it was necessary to "provide sufficient air for reoxygenating the water, maintain a suitable water temperature, keep the water free from slime and impurities, and prevent excessive heating or shaking to protect the fish from injury, since fish with broken fins and scales did not keep well." The importance of adding ice to the water was also recognized, since at a lower temperature water not only absorbs more oxygen, but the quantity of oxygen used by fish is reduced. Further, that "prior to transporting, only perfectly sound fish should be selected, fish should be confined for some time prior to transporting, and a few days before the journey no food should be given so that the water did not become impure with vomit and excrement."

Additional contributions of historical interest include the transoceanic shipment of smallmouth bass from Maryland to South Africa by freighter and later (in 1939) by air after the species became established. Fuqua and Topel transported channel catfish eggs and fry in Fearnow pails by sedan delivery truck from Iowa to Montana, a tedious, but nonetheless successful trip. Leach described the hauling of brook and rainbow trout fry in 10-gallon milk cans in railroad baggage cars. The principal method in the late 1930s, however, involved the use of railroad cars especially built for fish transportation and equipped with pumps that forced air through the cans.

An outstanding early contribution, primarily due to its forthrightness and farsightedness, was a letter from Roger Burrows to the Editor of the **Progressive Fish-Culturist** (1937). Burrows stressed the importance of fish arriving at stocking points in optimum condition, and that, to accomplish this, obsolete equipment such as truck-borne pails and barrels should be replaced by tank trucks with circulation systems, ice compartments, and aeration devices. During the ensuing years, the truck envisioned by Burrows was developed. Transportation methods have improved dramatically within the last 30 to 40 years. Larger and better distribution tanks have been designed specifically to haul fish more efficiently. A better understanding of the physiology of fishes and how they respond to certain environmental changes has also contributed greatly to improvements in the transportation of live fish.

*Presently, Executive Director, U. S. Trout Farmers Association, P.O. Box 220, Harpers Ferry, W.Va. 25425.

Modes of transportation

Irrespective of species or purpose, fish transportation plays an important role in fisheries management. The objective of those charged with this function is to transport as many fish as possible in as little water as possible, with as little loss as possible, and as economically as possible.

Fish are transported in a variety of ways, ranging from the use of plastic containers shipped via postal service to costly diesel truck- trailer units.

Traditionally, the truck has been the principal means of conveyance for most species. Schultz described an extraordinary use of trucks, which involved the transportation of anesthetized and iced walleyes without water. This procedure is an obvious exception to the rule, as most federal and state hatcheries use 1.5-ton vehicles and water. A number of 1- and 2-ton units are also reportedly in use. The cruising speed of 1.5-ton vehicles generally coincides with established state speed limits. For short trips and small numbers of fish, most hatcheries use 0.5- and 0.75-ton pickup trucks.

Most large trucks now in use have four-speed transmissions and are equipped with two-speed axles. Stake-bed trucks are widely used in some areas. Most hatcheries affix tanks directly to the truck's frame.

An increasingly popular innovation in fish transportation is the use of gooseneck trailer-borne tanks and pickup trucks. These units are appealing from both the standpoint of cost and versatility. For example, the trailer/pickup truck combination probably costs one-quarter of what a conventional tank truck would cost. Another decided advantage of trailers is that trucks can be incorporated into routine station duty when they are not used for fish distribution. Dual wheel pickup trucks with gooseneck units are used by the Texas Parks and Wildlife Department to transport 1 to 2 in.redfish fingerlings from hatcheries to bays and inland lakes (Chamberlain, 1986).

Tank Construction

Size, Shape, and Materials

Most commercial tanks are rectangular. Vehicles typically carry one or two tanks. One compartment tanks are most common but some may have two, three, or four. The volume of most compartments ranges from 100 to 200 gallons, with some exceeding 300 gallons.

Most tanks are constructed of fiberglass or aluminum, but many kinds of materials have been used in the past: plywood, redwood, stainless steel, glass, galvanized iron, sheet metal and others.

Aluminum has several appealing qualities. It is light-weight, corrosion resistant, is easily mass produced, and has excellent cryogenic properties. To avoid problems with various water qualities, it is recommended that an alloy range of 3003H14-6061T6 be adhered to. Aluminum elliptical tanks used at Federal hatcheries in the Southwest are constructed of 3/16-inch material. Most marine fish facilities, however, use fiberglass tanks.

Insulation

Most tanks constructed in recent years are insulated. Styrofoam, fiberglass, urethane foam, and cork board are most commonly employed. Urethane foam appears to be most popular at the present time. Insulation is used as a filler between rigid tank walls or to cover circulation lines. It virtually eliminates the need for icing during short trips and simplifies temperature manipulation over long distances.

The key parameter for comparing the relative effectiveness of insulating materials is the thermal conductivity (K). Lower K values imply better insulating ability. The following is a list of insulating materials and their respective K values:

Brick	5.00	Expanded vermiculite	1.60
Oak	1.18	Pine	0.74
Balsa wood	0.58	Cork	0.29
Styrofoam 33	0.28	Fiberglass	0.25
Kapok	0.24	Urethane	0.18

Circulation/aeration

The purpose for circulation within a transportation tank is to supply well-aerated water of optimum quality to all parts of the tank as rapidly as possible. Success is related to tank shape, location and efficiency of aeration devices and circulation pattern.

The exchange of gas between water and the atmosphere is important in fish transportation. Transport water must contain adequate oxygen; pH levels must remain within a tolerable range; and undesirable quantities of dissolved ammonia and carbon dioxide must be suppressed. The solution to this complex problem has been through application of the oxygenation principle. That is, a potentially lethal situation is circumvented by maintaining high dissolved oxygen tensions in transportation water through aeration.

Reports from years past concerning aeration are instructive. One individual used a storage tank filled with compressed air to aerate tanks and cans of fish transported on fisheries cars. Another method is still in use today. This approach is based on the liberation of oxygen through perforated rubber tubing.

Currently, most transportation units are aerated with gaseous, bottled oxygen. The oxygen is commonly liberated within tanks via carbon stones or rods but Micropore tubing, Bioweave and Porex are also used. Agitators are the second most popular means of aeration and many haulers use agitators in combination with oxygen, providing both aeration and mixing of water. Twelve-volt direct-current agitators, powered by the truck's battery and alternator, are most commonly used by fish haulers.

The Fresh-flo device, a popular aerator , is commercially available in a variety of sizes. (Use of trade names does not imply endorsement of commercial products.) The system depends on centrifugal force created by a high speed motor-driven impellor that pulls water into a system of vanes, producing the turbulence needed to mix water with air while concurrently removing noxious gases, such as carbon dioxide. This aerator has been used for years with highly satisfactory results.

Hauling units used by the Texas Parks and Wildlife Department to transport redfish fingerlings were described by Hammerschmidt (1986) as three chambered, wood and fiberglass tanks, 3.0 M long x 1.2 M wide x 0.8 M high, (9.87 ft. x 3.94 ft. x 2.62 ft.) with each chamber having a liquid capacity of approximately 1950 L (515.2 gal.). Colura (personal communication) indicated that the Palacios hatchery uses bottled oxygen liberated through carbonstones or Bioweave for aeration purposes and mentioned that Fresh-Flo aerators are no longer used as they are corroded by saltwater (i.e., maintenance problems), and small redfish fingerlings tend to be drawn up against them. It should be noted that aerators may cause excessive foaming when used in seawater.

Water Quality Maintenance and Monitoring

Oxygen

As was mentioned previously, the approach for providing fish with ample oxygen and supressing lethal concentrations of ammonia and other toxic metabolic products has been through application of the oxygenation principle. Most drivers simply attempt to maintain a level of no less than 5 ppm dissolved oxygen during transport. As an example of the interaction and importance of oxygen and temperature on fish we can examine the work of Moss and Scott (1961). Their data were obtained by subjecting several species of fish (bluegill sunfish, largemouth bass, and channel catfish) to sudden or gradual changes in water temperature, ranging from 25° to 35°C (77° to 95°F). All three species required more oxygen when they were subjected to sudden changes in temperature.

Some transport units are equipped with oxygen monitoring equipment. Some use ampere gauges installed in truck cabs to indicate the functional status of agitators. A rheostat in the truck cab enables the driver to control the speed of Fresh-flo aerators. Some truckers carry portable oxygen kits/meters for use when they suspect problems while enroute to stocking sites. At temperatures below 20°C (68°F), Colura (personal communication) cautioned that 1- to 2-inch red drum fingerlings lose equilibrium and rise to the surface during transport under oxygen supersaturated conditions. When the oxygen is turned off and levels return to normal, the fish settle down.

Temperature

Temperature of transport water has generally been controlled by insulation and the judicious use of ice. The obvious disadvantages associated with using ice are inconvenience, its temporary stabilizing effects on temperature, its diluting effect on salinity, and its unavailability in remote areas.

However, most individuals continue to use ice or insulation, or both, for temperature control. Most persons transporting fish apparently check water temperatures with pocket thermometers while enroute; electric thermometers are little used. Most pondfish are transported at water temperatures of 65° to 70°F. The preferred temperature for red drum is 70°F.

Metabolic Waste Products

The effects of metabolic waste products and related substances on pondfish during transport have received little attention. This is rather surprising, since most fish culturists are aware that excretory products in conjunction with accumulations of regurgitated food, mucous, and loose fish scales, all tend to reduce water quality over a period of time.

Ammonia, for instance, is the principal nitrogenous end product excreted by fish. Toxicity is directly related to the amount of ammonia present in its unionized form, NH_3. It is known that increased pH and temperature, reduced oxygen and carbon dioxide at salinities above or below blood isotonicity (isotonic – no osmotic difference; one solution having the same osmotic pressure as another), increase the proportion of NH_3 to NH_4+ resulting in increased toxicity. For example, as concentrations of ammonia and related excretory products increase in water, rainbow trout lose the ability to use oxygen efficiently. More specifically, as the ammonia level reaches 1 ppm, the oxygen content of the blood decreases to one-seventh normal and the carbon dioxide content increases about 15 percent. The result of this combination of circumstances is death by suffocation.

Work by Holt and Arnold (1983) on the effects of ammonia (and nitrite) on growth and survival of red drum eggs and larvae showed that concentrations as low as 0.3 ppm un-ionized ammonia significantly reduced survival of newly-hatched larvae in the first two weeks. However, concentrations twice as high were tolerated by 3-week-old post-larvae. Their data suggest that un-ionized ammonia may be a potential hazard in red drum culture systems and, perhaps, transport units. The most commonly used method for ammonia suppression is that of reducing water temperature. Low temperatures not only reduce ammonia production, but oxygen consumption as well. Significant amounts of ammonia cannot be removed from water by aeration.

Carbon dioxide may be an important limiting factor in fish transportation. A product of fish and bacterial respiration, the gas tends to acidify transport water. Increased carbon dioxide, in combination with low pH, reduces the affinity of fish blood for oxygen (the "Bohr" effect). Fish may die under these conditions, even though oxygen levels are seemingly adequate. It was reported that rainbow trout tolerate carbon dioxide at levels less than 15 ppm in the presence of reasonable oxygen concentrations and temperatures, but become distressed when carbon dioxide levels approach 25 ppm.

Chemical Additives

Therapeutants

Bacterial numbers are known to increase in closed transport systems. Their effect is that of competing directly with the fish for oxygen as well as by excreting toxic metabolic wastes. Acriflavine is commonly used as a transport bactericide at a rate of 1 to 2 ppm. Furacin at 5 to 15 ppm is also used, as is Terramycin, although to a lesser extent. Although no treatment levels are given, Chamberlain (1986) noted that red drum were hauled in water containing acriflavine or furacin. Fisheries personnel must be aware that only Sulfamerazine, Terramycin, Romet-30, and salt (sodium chloride) are currently registered as therapeutants for use on food fish and that constraints exist and vary between each, irrespective of their approved status (Schnick *et al.*, 1986).

Common salt (sodium chloride) has been used by fish culturists for years as a disease prophylaxis, and a general therapeutant. For example, when moving channel catfish or largemouth bass broodfish to spawning ponds, some aquaculturists will briefly "bathe" each fish in a strong salt solution (50-100 ppt) in a tub before releasing it in the pond. Reasons for doing so are often general in nature, i.e., overwintering parasites and bacteria will be reduced or eliminated, excess mucous will be reduced, gills will be "cleaned," or "the saltbath just makes 'em feel good."

When striped bass culture became increasingly widespread, the use of salt as a handling and transport medium increased as well, probably due to the species' anadromous (fish that leave the sea and migrate up freshwater rivers to spawn) character. When queried as to why they hauled stripers in a 1 percent (10 ppt) salt solution culturists typically replied, "because they haul and handle better" in a saline solution.

Almost 30 years ago, Norris *et al.* (1960) discussed the role of osmosis in helping fish feel better in a saline solution. (Osmosis is the tendency of a solvent (i.e., water) to pass through a semi-permeable membrane, such as the wall of a living cell, into a solution of higher concentration, so as to equalize concentrations on both sides of the membrane.)

These authors pointed out that saltwater and freshwater species alike are constantly expending energy to maintain an osmotic balance between themselves and their environment. Thus, more oxygen is consumed than would be used if the fish were in a solution that was isotonic to their blood. Although little experimental work had been done on the transportation of fish in saline solutions, they cited a study in which a saltwater species was handled and transported in varying concentrations of seawater. The fish appeared to tolerate handling and transport much better in isotonic solutions. Since the concentration of salts in most fish blood (both freshwater and saltwater forms) is 1 to 1.2 percent (10-12 ppt), we might conclude that one reason striped bass were found to tolerate handling and transport better in a 1 percent salt solution was that the solution was essentially isotonic. As Robertson *et al.* (1987) stated in a discussion regarding salinity and its positive effects on stress, "salt lessens the osmotic gradient between the transport medium and fish blood and helps alleviate the adverse effects of osmotic imbalance."

As will be seen in the discussion on stress, work on the use of salt and combinations of salt and other compounds has increased dramatically in recent years as knowledge of their value has progressed. More culturists are using saline solutions for transport than in past years but still to a limited degree, according to Carmichael and Tomasso's (1987) survey.

Anesthetics

Benefits derived from the use of anesthetics are primarily based on reducing the metabolic activity of fish (which in turn results in less oxygen consumption, less carbon dioxide production, and less excretion of nitrogenous wastes) and in reducing the effects of external stimuli on fish. Anesthetics used at lower rates have a sedating or calming effect on fish, often enhancing survivability of some species during transport or handling. For instance, anesthetics are commonly used during handling of striped bass broodstock to alleviate stress. Anesthetics have been shown to reduce the stress response in red drum when used prior to handling (Robertson *et al.*, 1987).

Work by Carmichael *et al.* (1984) with largemouth bass demonstrated that the use of anesthetics before, during and following transport increased both short and long-term survival. The use of anesthetics in combination with salt have also been shown to reduce stress during transport. Chamberlain (1986) noted that sedating levels of 0.25 to 2.0 ppm quinaldine or 5.0 to 20.0 ppm MS-222 have been used for hauling red drum fingerlings. However, as with saline solutions, the use of anesthetics continues to be limited. Where MS-222 is concerned, a contributing factor may be that a 21-day withdrawal period (after use/before harvest) is associated with its use on food fish.

Anti-foaming Agents

The formation of scum and foam on the surface of transport water may result from drug use or excessive mucous associated with large numbers of fish and long distances. Irrespective of cause, excessive foaming interferes with the observation of fish during transit and may inhibit gas exchange. To combat this problem, many hatcheries use a 10 percent solution of Dow Corning's antifoam AF emulsion at the rate of 1 oz. (or 25 cc)/100 gallons of water. For maximum effectiveness, the chemical should be added before drugs are used or fish are loaded. Antifoam is a nontoxic and useful substance.

Hauling Loss

Wedemeyer (1970) states that stress is "the sum of all the physiological responses by which an animal tries to maintain or re-establish a normal metabolism in the face of a physical or chemical force." Stress that exceeds a fish's ability to adjust will be lethal. Less severe stress may predispose fish to infectious disease or to physiological disorders.

Although there are differences, many stress-induced metabolic alterations known for higher vertebrates are also recognized in fish. Changes in plasma glucose and stress hormones such as corticosteroids have been found to occur in stressed fish as well as in higher animals such as man. Studies of the effects of typical hatchery manipulations (crowding, handling, hauling) on fish have shown that fish do indeed become stressed, and that losses may occur immediately or be delayed. However, there are practices that may be employed to negate or reduce loss due to stress.

In 1972, Wedemeyer investigated the effects of handling stress on juvenile coho salmon and steelhead trout in both soft water and in water to which sodium chloride (salt) and calcium sulfate had been added. He found that significant osmoregulatory and metabolic dysfunction occurred and persisted for approximately 24 hours after fish in soft water were handled. Stress was partially or completely eliminated for fish held in water containing 0.3 percent (3 ppt) salt and with a calcium ion level of 75 to 120 ppm. In 1974, Wedemeyer and Wood recommended that fish be handled and hauled in water containing at least 0.1 to 0.3 percent (1 to 3 ppt) salt and that calcium chloride be added (where necessary) to increase total hardness (as calcium carbonate) to at least 50 ppm.

McCraren (1974) describes a large-scale production program involving advanced largemouth bass fingerlings (8 to 10 inches T. L.) reared at three federal hatcheries which were marked and transported in conjunction with two state agencies. Reports from the southeast suggested that these fish (grown-out on formulated feed) were delicate in nature. Short-term handling/hauling tests aptly showed that attention to details and minimal handling would be necessary to avoid losses. Successful stocking of the fish (some 35,000 bass weighing approximately 12,300 lbs.) was accomplished by: removing most of the fish from ponds by seining, using Autocranes and partially-filled containers of water to transfer fish from seines to trailer-borne tanks at the pond, terminating feeding 48 hours prior to harvest, keeping fish in water (a "water cushion") whenever possible, avoiding graders made of wire or other abrasive materials and grading only when necessary, providing a rest period in the holding house prior to shipment, utilizing a distribution unit that had quick-release gates and pipes to reduce handling, hauling in water containing 0.5 ppt) salt and 5.0 ppm Furacin, maintaining a low temperature of 55°F enroute, providing ample aeration and liberating bottled oxygen through carbon stones, and lastly, using a loading rate (2.2 lbs/gal.) predetermined along with other practices prior to transport. Ten distribution trips of up to 13 hours duration were required to complete the program.

Carmichael (1984) monitored the transport of 5 lots of advanced largemouth bass fingerlings from Texas to Nevada and Arizona (30 hours or longer in duration). Three of the five lots were successfully moved. It was found that weight per unit volume (loading) is less for smaller bass (5.0 inches) than it is for fish about 8.0 inches in length. Plasma corticosteroid and plasma glucose levels remained high for 24 hours following transport. It took nearly 63 hours for plasma chloride to return to normal. The bass required approximately 64 hours or more to recover from trips of this duration. Hauling water was 22°C (71.6°F) at loading, cooled to 17°C (62.6°F), contained 0.5 percent NaCL (5 ppt) for approximately 16 hours, and was exchanged for hardwater in New Mexico (2,346 ppm bicarbonate) for the balance of the hauling(s). Carmichael *et al.* (1983) also contributed to our understanding of handling/hauling stress on smallmouth bass, and has developed an integrated, physiologically-based protocol for handling/hauling largemouth bass that has application for other species.

For those who hadn't experienced the occasional personal trauma if working with striped bass in the 1960s, publication of the Bonn *et al.* (1976) "guidelines" for same warned of the species' tendency to "shock" during handling and harvest. A thorough and detailed "how to" account of harvesting, handling, and transporting striped bass is by Turner (1984). This includes: avoidance of high temperatures during harvest, using glass V-traps for harvest, placing fish immediately in oxygenated water containing 1 to 2 percent salt (10 to 20 ppt), then into holding tanks containing a 1 percent (10 ppt) solution of salt, providing fish a rest period, hauling in a medium of 1 5 ml antifoam. The fish are kept in saline water at every possible point in the harvest-handling-hauling scenario.

Hauling densities vary but the fish/water ratio never exceeds 60 g/l(0.5 lb./gal.). On long trips, the density is reduced to 30 g/l(0.25lb./gal.). Agitators, oxygen, insulation and quick-release valves and stocking pipes are used. If necessary, tempering is accomplished at the stocking site by pumping water from the site to be stocked into the hauling tank with a 2 h.p. gasoline-engine pump. Tempering requres 1 to 2 hours for a 1400 L (369.9 gal.) tank.

Parker (1984) reports results of a study wherein an investigator found that the addition of 10 g/L (ppt) of salt and 25 ppm MS-222 in hauling water prevented the loss of chloride ions from hybrid striped bass. Based on the increase in corticosteriods, it was determined that the fish were stressed by normal netting/handling procedures. Survival 72 hours after hauling was 100 percent for fish exposed to salt and anesthetic and 75 percent for those treated solely with the anesthetic.

Caldwell and Tomasso (1984) evaluated stress induced in red drum fingerlings (0.2 to 0.8 g) by handling, hauling and net confinement. The trend in stress indicators (plasma glucose and plasma chloride) followed the same general pattern for both stocking procedures and confinement. Overall loss after nine hours of confinement was 53 percent. Physiological response of the species to these stressforms were less than those observed by the authors in other species.

Robertson *et al.* (1987) provide not only an excellent review of stress in fish culture, also but the results of work designed to measure the stress response of red drum to typical hatchery transport. The authors also comment on methods to reduce biochemical stress responses.

For their purposes the authors focused on measuring the more reliable stress indicators in fish, i.e., plasma cortisol (neuroendocrine), plasma glucose (metabolic) and osmolality/electrolytes (osmoregulatory disturbances).

In the handling portion of the study, red drum were transferred from raceways to a small livebox, confined there for 30 minutes and then returned to the raceways. Based on plasma cortisol and glucose measurements, the fish reacted dramatically to the procedure, although similarly with respect to the reaction observed in other fish. The cortisol response lasted less than three hours, whereas the glucose response lasted for six hours. Both plasma cortisol and glucose returned to baseline values after one day and levels stabilized after six days. In the transportation work, red drum were hauled for five hours in 32 ppt seawater containing a sedating level of MS-222 (5 ppm). After one hour all parameters were significantly elevated. Plasma glucose and cortisol levels were similar to or either higher or lower than those for other species. These parameters remained elevated, as they often do throughout transport, whereas plasma osmolality recovered during the same period. Delayed mortality has been attributed to electrolyte imbalance in fish. However, in this work, the fish were hauled in 32 ppt seawater, a situation wherein the fish would be expected to lose water to the hauling environment and gain electrolytes. All parameters returned to resting levels one day after transport. No mortality occurred during or following transportation in this study.

To alleviate the effects of stress associated with cultural practices, the authors advise against exposing fish to multiple stressors, but support the usefulness of hauling fish in low salinity water, the use of sedating doses of anesthetics, and the use of combinations of salt and anesthetics during transport.

Harvest and Pre-hauling Treatment of Fish

Proper techniques for harvesting and holding pondfish before shipment are well documented. Most losses are directly related to improper handling that may result in outright mortality or assume a lingering nature, such as fungus infection. Lewis and Bender (1960) described how a latent bacterial infection among golden shiners assumed acute proportions after harvest-associated stress. Undoubtedly, many improperly handled fish are stocked each year that eventually die unnoticed.

Most pondfish hatcheries consist of numerous earthen ponds that are normally harvested through a combination of seining and draining or trapping. During summer, this procedure is often complicated by high water temperatures. When this occurs, water levels are generally lowered to a point where removal of fish can be accomplished rapidly during the morning, when the water and air are cooler.

When ponds contain large poundages of channel catfish, for example, a substantial portion may be removed by seining before the pond is lowered. The rest can then be safely removed from collection basins. Largemouth and striped bass fingerlings are safely harvested with glass V-traps while ponds are being lowered. The traps are positioned in front of outlet screens and a stream of fresh water is introduced near the trap to attract fish. Fingerlings are periodically removed from the traps with a dip net as the pond is drained, transferred to tubs or a small pick-up truck holding tank and moved to the holding house. Consideration should be given to holding and transporting freshwater fish in a saline solution.

V-traps have been used at the Palacios, Tex., hatchery to harvest red drum fingerlings but results have been mixed. More work with them is being considered, according to Colura (personal communication). Currently, 1 to 1.5 inch red drum fingerlings are harvested 30 to 40 days after stocking from ponds with collection basins. The fingerlings are dip-netted from the basin as the last of the water is drained from the pond.

For small poundages of fish, the use of a dip net and tub tends to reduce physical damage. Large mesh nets are understandably avoided. At most stations, a small fine-meshed net is used, particularly when scaled fish are involved.

Considerable thought should be given to the design of transport units used to move fish from pondside to holding house. If existing holding-house facilities permit their use, tanks with a quick-release gate valve and a length of polyvinyl chloride pipe will allow fish to be unloaded directly and rapidly into holding tanks, thereby eliminating additional handling.

Holding-houses should be designed with the aforementioned approach in mind. This can be accomplished in at least one of two ways: enough space should be allowed for trucks to drive through holding houses, or unloading ports should be incorporated in holding house walls that allow fish to be unloaded into tanks from outside the building. Hogan (1938) presented several sound suggestions for holding house design. Some of his suggestions were incorporated into a holding house at the Dexter, N.M., National Fish Hatchery.

Pondfish are generally provided with a rest period of several hours before they are graded and weighed. The use of properly sized grading devices constructed of non-abrasive materials is essential.

Boxes with metal bars in the bottom or vertical metal bar graders are usually used for grading. Several commercial models are available.

To reduce fouling of transport water with fecal material and regurgitated feed, culturists do not feed pondfish before transport. It is also believed that starvation tends to make fish more tolerant of temperature fluctuations and handling. The starvation period is generally 48 hours, but during cool weather, a 72-hour period is warranted. The starvation period may be reduced by cessation of feeding one or two days before harvest.

Chemical treatments should be applied during the holding period, no later than 12 hours before shipment. Treatments should not be applied before fish have voided their stomach contents. Fish receiving tank treatments should be observed continually to avoid undue stress or loss.

Loading Methods

The same consideration should be given fish when they are loaded onto a transport unit as when they are harvested. The initial shock of handling and introduction into a transport unit may induce strong physiological stress. Caillouet (1967) demonstrated the build up of high concentrations of lactic acid by fish during periods of high activity. Such a stress creates an oxygen debt which is alleviated very slowly, and may last through a hauling period. Thus, loading stress may also contribute to post-stocking mortality.

Whenever possible, fish should be conditioned (tempered) before they are loaded into hauling tanks. Usually, however, this is not practical, and fish are tempered after they are loaded on distribution units. This is a satisfactory method, but the reduction in water temperature must not be too rapid. Proper tempering requires 20 minutes at the very least, for every 10°F change in temperature. The addition of ice is the most common method of reducing and holding water temperatures within transport units. One-half pound of ice per gallon of water reduces water temperatures by about 10°F. Most pondfish are transported at 65° to 70° F. Fish can be hauled at lower temperatures, but it is believed that they are unnecessarily stressed when retempered to a higher temperature at the delivery point. At temperatures lower then 60°F, some species tend to become listless. For trucks with recirculation units the affected fish may gravitate towards, and block, intake screens.

Transporting fish at relatively low temperatures is beneficial in many respects. It reduces metabolism rates, which lowers oxygen consumption and the production of organic wastes. High oxygen levels are more easily maintained in cooler water. It should be remembered that fish "shock" as easily when moved from cold to warmer water as when moved from warm to cooler water. This may be an especially important consideration when fish are unloaded at stocking or delivery sites.

Where the red drum is concerned, a hauling temperature near 70°F (21°C) is preferred to suppress metabolism. Hauling in freshwater is not recommended. If fish are to be stocked in freshwater, saline hauling water may be diluted to 10 ppt and upon arrival at the stocking site, receiving water is pumped into the tank for one to four hours to acclimate the fish before release (Chamberlain, 1986).

Hauling Criteria

Loading rates for pondfishes vary widely between hatcheries. Maximum loading capacities of individual transportation units have not been determined for the most part. An examination of pondfish and salmonid loading rates, in terms of weight per unit volume of water, implies that units distributing salmonid species (i.e., rainbow trout) are generally the more sophisticated and advanced in design. However, these data are misleading when we realize that most pond-reared species are shipped as small fingerlings and in relatively small quantities. Further, warmwater hatcheries rear a variety of species and stocking policies and inherent variables, such as growing season and water quality, often differ significantly between hatcheries.

There tends to be confusion concerning the reporting of loading densities. In many instances, displacement of water by fish is ignored. Generally, densities are reported as pounds of fish per gallon of water, whereas other reports are in terms of pounds of water required for each pound of fish. McCraren and Jones (1978) suggested an approach for standardized reporting.

Transportation Guidelines

In general, fewer pounds of small fish than of large fish can be transported per gallon of water. The risk of loss increases as time spent en route increases. Consequently, lengthy distribution trips demand care on the driver's part and mechanically proven equipment. The transportation unit should be capable of holding metabolites to a minimum, maintaining at least 5.0 ppm dissolved oxygen en route, and maintaining temperatures at the desired level. To provide a basis for comparison between transport of red drum and other species, the following includes data relative to largemouth bass, bluegills and striped bass.

Largemouth Bass and Bluegills

According to Carmichael and Tomasso (1987), largemouth bass (1000 to 2.5 fish/lb.) are generally transported at 70° to 79° F and at a hauling density of 0.5 lb/gal. or less. The average trip is five hours in duration or less. Most haulers use acriflavine at 2 ppm. Anethestics are rarely used. However, salt is commonly used at 5 ppt. About 1/4 of the haulers use an anti-foaming agent.

Bluegills ranging in size from 2,500 to 25 fish/lb. are generally transported at temperatures of 60° to 68° F. The most commonly reported hauling density is 0.5 lb/gal. or less. Most trips are of five hours duration or less and acriflavine is commonly used as a transport antibiotic. Anesthetics are not used, but salt is used 65 percent of the time at 3 to 5 ppt. Antifoam is used about one-quarter of the time. From the aforementioned paper, it was also found that for striped bass fry, fingerlings, and broodfish (weighing 15 to 30 lbs. each) most are commonly hauled at temperatures of 70° to 75° F. The hauling density most employed is 0.5 lb/gal. or less. Most trips last four hours or less and antibiotics (furacin or acriflavine) are generally used. Anesthetics see limited use but sodium chloride at 2.5 to 5 ppt is used most of the time and some individuals use calcium chloride in combination with salt. Antifoam is normally used as well.

The use of salt is much more widespread than it used to be, probably due to a better understanding of the osmoregulatory process in fish. Note that calcium chloride is at least used by some to further alleviate stress when transporting the more sensitive striped bass, an anadromous species found to adapt to a freshwater environment.

Red Drum

Guidelines are not well-developed for this species and, in some instances, reflect what has been learned to date for the striped bass. This is understandable as the species has only recently been cultured (Colura *et al.*, 1976). Hauling densities, for example, are similar to those used for striped bass –1/3 lb./gal. for hauls less than eight hours and not more than 1/4 lb./gal. for hauls exceeding eight hours (1- to 2-inch fingerlings). As was referenced previously, antibiotics (acriflavine and furacin) are often used as are anesthetics such as MS-222. Interestingly, a study by Robertson *et al.* (1985) that evaluated stress from transport of larger red drum (5.12 to 8.27 inch S.L.) demonstrated that the fish could be transported in water of high (32 ppt) and low (4 ppt) salinities with little or no loss but that no apparent benefits were derived from the use of MS-222 (5 ppm to 5.5 hour haul; 25 ppm - 2.5 hour haul).

Colura (personal communication) stated that for shipments from one saline environment to another temperature was the principal consideration.

In 1986, Carmichael and Tomasso (personal communication) monitored shipments of red drum fingerlings to the John Wilson hatchery. The fingerlings were approximately 1 inch in length, were from three different ponds and, depending upon which pond they were harvested from, averaged 1400/lb., 1800/lb., 2800/lb., respectively. Transport time was 5 1/2 hours and the fish were held in separate tanks upon arrival at John Wilson and oberved for 10 days. The fish were hauled in 33 ppt (pondwater) at a density of 0.1 lbs./gal. Hauling and hatchery temperatures (80.6° to 84.2° F) and salinities (33 to 41 ppt) were similar. Harvest procedures were essentially the same for all three lots of fingerlings. However, at the end of the observation period (and in the absence of replicates) mortalities ranged from 12 to 54 lot, the 2800/lb. lot sustained a 30 the slightest loss (12 percent). Conclusion: size was not as important as initial condition.

Hammerschmidt and Saul (1984) and Hammerschmidt (1986) investigated the initial survival of red drum fingerlings stocked in Texas bays, as part of the state's massive enhancement program, for the period 1983-85. Overall mean survival, based on 24 hour. observation of caged fingerlings at the stocking sites, was 89 percent (1983) and 86 percent (1984).

In 1984, differences in water temperature between hauling trailer and release site ranged from 32° to 41° F. Salinity values were higher (36 to 49 ppt) in the hauling trailer than at the release site (26 to 36 ppt). Acclimation time ranged from 0.2 to 2.4 hours.

For both years, the authors concluded that harvesting, transport and stocking procedures were adequate. However, there were significant differences noted in survival between stocking dates, suggesting that the condition of fingerlings at stocking was important. In 1983 mean survival ranged from 62.0 to 98.7 percent and in 1984 from 34.0 to 100.0 percent. Survival in 1984 was also apparently linked to initial stocking densities in ponds, i.e., ponds stocked at higher densities produced smaller fingerlings that were stressed more easily during harvest and transport. Additionally, the authors felt that survival may have been influenced by varying acclimation periods and variation in bay holding conditions. Please note the difference in conclusions (size vs. condition) between this work and the aforementioned by Carmichael and Tomasso.

Unloading Methods

Stocking pondfish is often a complicated process. Pond-reared species may be stocked over a large geographical area. This entails making numerous stops at prearranged locations and removing small quantities of various species and sizes of fish by dip net. For large bodies of water, however, modern distribution units employ a quick-release valve and gate which allows fish to be discharged from tanks within seconds. When tank design or stocking commitments dictate that dip nets be used to remove fish from tanks, the same care should be given the fish as when they are harvested or loaded.

Meyer *et al.* (1983) recommended tempering before stocking if the receiving water differed from the hauling temperature by more than 5° F. If water temperatures are known to differ extensively, drivers may allow the temperature to rise gradually during the last few hours on a long trip or gasoline powered pumps may be used at the stocking site to gradually exchange water in the transport tank. To avoid problems, it is suggested that the temperature of the water at the receiving site be known in advance to ensure that proper precautions may be taken.

Acclimation

Red drum are euryhaline (able to exist in waters with wide variations in salt content) and found in waters with salinities ranging from 0 to 50 ppt. The optimum range is reported to be 30 to 35 ppt. There is a direct relationship between size and salinity, small fish being more commonly found at lower salinities and large fish at higher salinities (Perret *et al.*, 1980). It is this wide tolerance to salinity and the fact that the species can survive and grow in freshwater that has led catfish farmers to consider them as an alternative species. They are also being looked at by saltwater shrimp producers and for rearing in coastal Louisiana saltwater impoundments. Consequently, acclimation of the species to environmental conditions outside of its optimum environmental range(s) continues to be of interest to those who wish to culture, transport, stock, etc.

The species is found over a wide range of salinities but it cannot survive in low ionic strength freshwater. Red drum must maintain their body fluids at an osmotic strength equivalent to about 10 ppt salinity. If they are held in water of less than 10 ppt, loss of body salts by diffusion to the external water (environment) will occur. They can compensate to some degree by absorbing ions from the water but if the rate of loss exceeds replacement death will result (Chamberlain, 1986).

Laboratory studies by Miranda and Sonski (1985) suggest that mortality of red drum in freshwater is related to levels of individual ions rather than total ionic concentration, in particular sodium and chloride. Survival of fingerlings (1.93- and 3.9-inch av. T.L.) was greater than 80percent at chloride levels above 130 ppm. The larger fingerlings tended to be more tolerant than the smaller fish, requiring approximately 100 ppm chloride for survival.

In a preliminary evaluation of survival and growth of juvenile red drum in fresh and salt water, Crocker *et al.* (1981) held larval and juvenile red drum in dechlorinated and freshwater for 96 hours. Survival for larvae (23 days old, 0.24 inches SL) was 5 percent, for postlarvae (34 and 47 days old, 0.64 to 0.78 inches SL) was 70 percent, and for juveniles (57 days old, 2.24 inches SL) was 95 percent. Survival in control salinities of 10 ppt was 90 percent or more. These results indicate that tolerance to the dilute freshwater was size dependent. These workers also compared the growth of 2-inch fingerlings in saltwater (35 ppt) and freshwater (195 ppm chloride, 44 ppm calcium). After 30 days, survival for each group was 93 percent; saltwater fish were significantly larger. It was concluded that the fast growth and high survival of red drum greater than 0.79 inch SL was indicative of its potential as a sport or commercial species in either fresh or saltwater aquaculture. In a later study, Crocker *et al.* (1983) found that juveniles (1.57-1.93 inch SL) tolerated abrupt transfer from seawater (28 ppt) to freshwater (less than 1 ppt), prompting the authors to state that red drum are highly efficient osmoregulators, capable of adapting to a wide range of environmental salinities.

Dolman (1983) investigated the tolerance of red drum fingerlings to freshwater as part of a freshwater stocking project. In the first of three laboratory experiments, red drum fingerlings (1.85 inch TL) were tempered from saltwater (25 ppt) for two, four or eight hours to synthetic seawater of 100 to 1,100 ppm total dissolved solids (TDS). It was found that the fingerlings would not survive in water with a TDS equal to 100 ppm but did so at higher levels. Further, tempering for four hours or more enhanced survival. In the second trial, 1.97 inch (TL) fingerlings were tempered from 25 ppt seawater to freshwater (TDS = 340 ppm) containing various concentrations of sodium chloride for two, four or eight hours. Survival was labelled adequate above 125 ppm chloride and tempering period had no effect on survival. The author suggested that use of a simple chloride ion determination kit could be used in the field to test the adequacy of natural waters for stocking. From the final phase of testing, it was determined that larger fingerlings (3.9 inches) were more tolerant of low concentrations of NaCl than smaller fish (2.05 inches) thereby increasing the possibilities of using the fish in a greater number of freshwater reservoirs. However, production of larger fingerlings involves supplemental feeding which increases overall cost. A brief discussion followed describing the successful introduction of fingerling red drum into Lake V. Braunig (freshwater) and fisherman acceptance (90 percent) of the species.

Colura (personal communication) states that successful introduction of fingerlings is dependent upon the receiving freshwater having a chloride ion content of 150 ppm or more. Also, that since fingerlings are transported in saltwater, half of the water may be changed immediately, but when the salinity reaches 10 ppt tempering will take no less than two to three hours. The larger the fish, the easier it is to acclimate them to freshwater. Some individuals apparently feel that a longer acclimation period is required. Temperature of hauling and receiving water is also important. If stocking in saltwater, only temperature is a consideration.

In addition to being euryhaline, red drum are also eurythermal (can live in a wide range of temperatures). From the Perret *et al.* (1980) review we find that the species has been collected from Florida rivers at temperatures ranging from 35.6° to 84.2° F. Young drum (0.3 inches) have been taken at 68.9° to 87.8° F, whereas juveniles (1.69 to 4.33 inches) have been collected at 56.8° to 83.8° F. Highest catches were reportedly at temperatures of 68.0° to 77.0° F.

Miranda and Sonski (1985) studied the survival of red drum fingerlings in freshwater in terms of tolerance to low temperature. For fingerlings (6.22 inches average Total Length) held in tap water with a concentration of 150 ppm chloride, lower lethal temperatures ranged from 37.4° to 33.4° F when temperature was reduced. Fish did not feed between 9° to 5° C (48.2° to 41° F). The authors concluded that lethal temperatures did not differ greatly from those reported in saltwater, suggesting that salinity may not effect temperature tolerance to a great degree. Mortality in freshwater is probable when temperatures reach 4° C (39.2° F) or less for several days. Feeding is likely to be affected in freshwater that drops to 9° to 7° C (48.2° to 22.6° F) for extended periods of time. Consequently, despite the importance of chloride concentrations in freshwater, temperature range would also be expected to limit successful freshwater introductions of red drum.

Relating the work of Texas A & M University researchers, Chamberlain (1986) reported the following with respect to studies involving low temperature tolerance of 1–to 2-inch fingerlings in relation to combinations of salinity and calcium concentrations: greatest low temperature tolerance combination was 5 ppt salinity and 100 ppm calcium; cold tolerance was better at 5 and 10 ppt (near the isomotic blood concentration); than at 0, 20, or 35 ppt salinity. Cold-tolerance at 100 and 400 ppm calcium levels was greater than at 10 ppm calcium.

Since it appears at this point in time that survival of red drum in freshwater is linked to a chloride content of at least 130 ppm, and that a calcium hardness of at least 100 ppm enhances tolerance to low temperature and salinity, is it logical or feasible to consider large-scale chemical alterations of a less-than-suitable environment? This is being done in the catfish industry, steelhead trout hatcheries in the northwest alter water sources, and it has been attempted on a pilot-scale with red drum (Chamberlain, 1986).

Precautions and Limitations

It must be remembered that the following information is principally based on laboratory work and the Texas stocking program experience. Therefore, most of these data relate to 1- to 2-inch red drum fingerlings. Since a food fish industry encompasses all life stages of a species, it is apparent that much remains to be learned and shared.

Transportation

In Texas hauling units, Fresh-flo aerators are not recommended as they tend to pull 1- to 2-inch fingerlings up against them. Corrosion due to saline waters also causes aerator maintenance problems. Texas reports loss of equilibrium in 1- to 2-inch fingerlings during transport at temperatures below 68° F in an oxygen supersaturated environment.

Know your chemicals; know the difference between levels of anesthesia.

Although the red drum apparently tolerates stress as well as other species, easy does it is a cardinal rule during all phases of harvest through unloading. Don't multiply stress! Keep fish in water at every step possible, don't overload dip nets, seines and tubs. Harvest early when temperatures are cool. Consider glass V-trap as a means of pond harvest – it's less stressful. Use correct mesh sizes and material; use graders of proper size and construction (non-abrasive materials). Crew and equipment should always be readied well in advance of actual operations. Consider holding house and tank designs, valves, pipes to reduce stress to fish. Starve fish prior to shipment (48 hours); don't treat fish less than 12 hours prior to shipment. Observe a minimum oxygen requirement of 5 ppm. When tempering, fish can tolerate a 5° F change every 20 minutes.

Hauling red drum in freshwater (low in chloride) is not recommended. If hauled in saline water to a freshwater stocking site, hauling water may be diluted to 10 ppt upon arrival and then exchanged with site water for one to four hours. If hauling from saline site to saline stocking point, only temperature is a consideration.

Large red drum (5 to 8 inches) haul well at from 4 ppt to 32 ppt salinity. Anesthesia was not found to enhance their survival under these conditions.

Hauling densities for 1- to 2-inch fingerlings, based on striped bass work, seem appropriate for the purpose of hatchery transfer, bay and lake stockings in Texas.

Small fingerlings and their condition at harvest both appear to contribute to mortality on occasion. Answers are needed. There also appears to be some question regarding acclimation period. Is four hours long enough? Know your receiving water temperature and chemistry (chlorides and hardness) before hauling to alleviate problems.

Acclimation

Red drum are adaptable to a broad range of salinities, but 30 to 35 ppt is optimum. Larger drum apparently tolerate greater salinities. The species cannot survive in low ionic strength freshwater. They must maintain body fluids at an osmotic strength equivalent to 10 ppt salinity. They can compensate to a degree by absorbing ions from the water. Sodium and chloride are two particularly important ions. Survival in excess of 80 percent has been observed for 2- to 4-inch fingerlings in water with chloride levels above 130 ppm. Four-inch fingerlings are more tolerant than 2- inch fish, requiring about 100 ppm chloride.

Larger fish (2-inch) are more tolerant to dilute seawater than smaller fish (1/4-inch). Survival for 2-inch fingerlings in 35 ppt salinity was 93 percent in freshwater with 195 pm chloride and 44 ppm calcium, it was also 93 percent. Better growth is associated with a saltwater environment.

With a minimum of four hours tempering, 2-inch fingerlings tolerated transfer from 25 ppt salinity to water with a total dissolved solids content of 100 ppm or more. Tempering was not required for 2-inch red drum transferred from 25 ppt to freshwater containing at least 125 ppm chloride. Natural waters with a chloride content of 150 ppm or more should be adequate for red drum fingerling survival.

Four-inch fingerlings are more tolerant to low sodium chloride content than are smaller fish (2-inch). Fingerlings greater than 2 inches in size acclimate more easily to freshwater. Temperature is the major consideration if hauling in saltwater and stocking in a saline environment.

For 6-inch fingerlings in freshwater containing 150 ppm chloride, lower lethal temperature ranges from 37.4° to 33.4°F. Feeding ceases at 48.2° to 41°F. (This is similar to that observed in saltwater.)

Mortality in freshwater is probable when temperature is 39.2°F or less on successive days.

Feeding (growth, survival) is likely to be affected in freshwater that drops from 48.2° to 44.6°F for an extended period. Temperature then, as well as the chloride ion, is important to freshwater survival (stockings, production).

For 1- to 2-inch fingerlings, lowest temperature tolerated was at 5 ppt salinity and 100 ppm calcium. Cold tolerance was better at 5 to 10 ppt salinity (isotonic) than at higher salinities. Calcium levels of 100 ppm or more apparently enhances tolerance to low T° and low salinity.

Altering environments with additions of chloride and calcium warrants additional investigation.

Recommended Procedures

Transportation

- Gooseneck trailer-borne tanks and pick-up trucks. Less cost; greater versatility.
- Rectangular tanks, fiberglass with urethane foam lining, 300- to 500-gallon capacity, quick release gate valves, lengths of PVC pipe for unloading.
- Aeration provided by bottled oxygen, liberated through carbon stones or Bioweave.
- Maintain temperature of at least 70°F and dissolved oxygen content of 5 ppm. Periodically check fish while enroute. Be prepared for the unusual. Carry back-up materials.
- Use an approved bactericide according to use pattern.
- Haul in saline solution of at least 10-12 ppt.
- Consider use of sedating levels of MS-222 at 5.0 to 20.0 ppm.
- Add 1 oz/200 gal. antifoam before drugs and fish are loaded.
- Be sure that hauling temperature and oxygen level is optimum before fish are loaded and that additives are thoroughly in solution.
- Due to our current lack of knowledge, conduct short-term pilot tests prior to committing large numbers of fish to un-tried equipment and practices. This is extremely important.
- It will take several days for fish to recover from stress associated with routine cultural practices. Carefully plan your operations.
- Harvest in the morning when temperatures are coolest, which makes it easier on fish and crew. Have equipment readied and at pond site.
- Consider use of the glass V-traps. Make sure seine/dipnet mesh size and material is correct.
- Don't overload dipnets or seines.
- Move fish to oxygenated saline water in hauling tank in saline water (tubs, etc.).
- Provide at least an overnight rest period in holding house to alleviate stress and to allow fish to void fecal material. Forty-eight hours would be better.
- If possible, unload fish via PVC pipe into holding house tanks.
- Grade only if necessary and with non-abrasive graders.

- If treating fish for any reason, do so no later than 12 hours before shipping.
- If temperature differences are a consideration, temper at rate of 5° F/20 minutes.
- Do not haul red drum in freshwater that does not contain at least 10 to 12 ppt salt.
- Haul 1- to 2-inch fingerlings at 1/3 lb./gal. for trips less than eight hours in duration and no more than 1/4 lb./gal. for trips in excess of eight hours. Remember, fewer pounds of small fish can be hauled per unit of water than large fish.
- If shipping from saline to saline, only temperature is a concern.
- Fish in poor condition prior to hauling will probably not haul well, nor survive well. It is also likely that larger fish will haul/survive better.
- Know the temperature and chemistry (salinity, chloride, etc.) of your receiving water.
- Temper if receiving water varies from hauling water by more than 5° F. Carry a 2 h.p. pump to simplify water exchange. Tempering on the basis of temperature and/or salinity could take up to four hours.

Acclimation

- Red drum are found in waters ranging from 0-50 ppt salinity. Optimum range is 30 to 35 ppt.
- Small fish are more commonly found in lower salinities. Converse is true for larger red drum.
- Cannot survive in low ionic strength freshwater. Need at least 10 ppt salinity.
- Sodium and chloride are particularly important in freshwater. Rule-of-thumb: chloride level in freshwater should be at least 130 to 150 ppm.
- Growth and survival of 2-inch fingerlings in 35 ppt saltwater and freshwater containing 195 ppm chloride and 44 ppm calcium was comparable (93 percent).
- Juveniles (1 1/2 to 2 inches) tolerated abrupt transfer from 28 ppt seawater to freshwater (less than 1 ppt).
- Growth and survival are better in a saline environment.
- Tempering for four hours or more from saltwater (25 ppt) to synthetic seawater containing 100 ppm total dissolved solids or more enhanced survival.
- Tempering from 25 ppt seawater has no effect if freshwater contains above 125 ppm chloride.
- Consider the use of a field kit to test freshwater for chloride ion adequacy.
- Four-inch fingerlings are more tolerant of low salt (NaCl) concentrations than are 2-inch fish.
- If fingerlings are transported in saltwater, to freshwater, half of the water may be changed immediately until 10 ppt is reached, then temper for two to four hours.
- Remember temperature acclimation in all cases (saline-saline, saline-freshwater, etc.).
- Salinity probably doesn't effect temperature tolerance significantly.
- Lower lethal temperatures for 6-inch fish in 150 ppm chloride (freshwater) ranged from 37.4° to 33.4° F.
- Mortality in freshwater is probable when temperatures reach 39.2°F or less for several days.
- Despite the importance of chloride concentrations in freshwater, temperature will limit growth and survival, as well.
- Tolerance to low temperatures for 1- to 2-inch fingerlings was found to be greatest at 5 to 10 ppt salinity and 100 ppm or 400 ppm calcium.
- Altering freshwater environments with additions of chloride and/or calcium to enhance growth and survival is provoking but warrants further investigation.

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Literature

Berka R. 1986. The transport of live fish: a review. EIFAC (European Inland Fisheries Advisory Commission) Technical Paper 48. 52 pp.

Bonn, E.W., W.M. Bailey, J.D. Bayless, K.E. Erickson and R.E. Stevens, editors. 1976. Guidelines for striped bass culture. Striped Bass Committee of the Southern Division, **American Fisheries Society.** 103 pp.

Caillouet, C.W. 1967. Hyperactivity, blood lactic acid and mortality in channel catfish. Agric. and Home Econ. Exp. Sta. Iowa State Univ., **Res. Bull. 551,** 17pp.

Caldwell, C.A. and J.R. Tomasso. 1985. Stocking and handling-induced stress in red drum fingerlings. **Proceedings Annual Conference Southeastern Association of Fish and Wildlife Agencies,** 39:238-247. Texas. [Personal communication to Joseph P. McCraren, Extension Fisheries Specialist, U.S. Fish and Wildlife Service, Leetown, West

Virginia.] April 1987.

Carmichael, G.J. 1984. Long distance truck transport of intensively reared largemouth bass. **Progressive Fish-Culturist** 46(2):111-115.

Carmichael, G.J. and J.R. Tomasso. 1988. Survey of fish transportation equipment and techniques. 20 pp.Progressive Fish Culturist 50:155-159.

Carmichael, G.J., G.A. Wedemeyer, J.P. McCraren and J.L Millard. 1983. Physiological effects of handling and hauling stress on smallmouth bass. **Progressive Fish-Culturist** 45(2):111-113.

Chamberlain, G. 1986. Red drum aquaculture status. Texas A&M University System, Texas Agricultural Extension Service, **Coastal Aquaculture** 3(3):1-20.

Colura, R.L., Biologist, Texas Parks and Wildlife Department, Palacios, Tex. [Personal communication to Joseph P. McCraren, Extension Fisheries Specialist, U.S. Fish and Wildlife Service, Leetown, West Virginia.] April 1987.

Colura, R.L., B.T. Hysmith and R.E. Stevens. 1976. Fingerling production of striped bass (*Morone saxatilis*), spotted seatrout (*Cynoscion nebulosus*) and red drum (*Sciaenops ocellatus*), in saltwater ponds. **Proceedings of the Annual Meeting World Mariculture Society** 7:79-92.

Crocker, P.A., C.R. Arnold, J.A. DeBoer and G.J. Holt. 1983. Blood osmolality shift in juvenile red drum, *Sciaenops ocellatus* L. exposed to fresh water. **Journal of Fish Biology** 23(3):315-319.

Crocker, P.A., C.R. Arnold, J.A. DeBoer and J.D. Holt. 1981. Preliminary evaluation of survival and growth of juvenile red drum (*Sciaenops ocellatus*) in fresh and salt water. **Journal of the World Mariculture Society** 12(1):122-134.

Dolman, W.B. 1983. Red drum freshwater stocking evaluation. Texas Parks and Wildlife Department, Federal Aid Project F-31-R-9. Performance Report, December 31, 1983. 4 pp.

Hammerschmidt, P.C. 1986. Initial survival of red drum fingerlings stocked in Texas bays during 1984-1985. **Texas Parks and Wildlife Department Management Data Series** 106. 14 pp.

Hammerschmidt, P.C. and G.E. Saul. 1985. Initial survival of red drum fingerlings stocked in Texas bays 1983. pp. 13-28 in **Proceedings of the Texas Chapter, American Fisheries Society, 7.** University of Texas Marine Science Institute, Port Aransas, Texas.

Holt, G.J. and C.R. Arnold. 1983. Effects of ammonia and nitrite on growth and survival of red drum eggs and larvae. **Transactions of the American Fisheries Society** 112(2B):314-318.

Hogan, J. 1938. Improved shipping facilities recommended. **Prog. Fish- Cult.** 40:41-42.

Johnson, S.K. 1979. Transport of live fish. Texas A&M University System, Texas Agricultural Extension Service, Fish Disease Diagnostic Laboratory FDDL-F14. 13 pp.

Lewis, W.M. and M. Bendey. 1960. Heavy mortality of Golden Shiners during harvest due to a bacterium of the Genus Aeromonas. **Prog. Fish-Cult.** 22(1):11-14.

McCraren, J.P. 1978. History. pp. 1-7 *In* **Manual of fish culture, Section G: fish transportation**. U.S. Fish and Wildlife Service, Washington, D.C.

McCraren, J.P. 1974. Hatchery production of advanced largemouth bass fingerlings. **Proceedings Annual Conference of Western Association of Game and Fish Commissioners** 54:260-270.

McCraren, J.P. and J.L. Millard. 1978. Transportation of warmwater fishes. pp. 43-88 *In* **Manual of fish culture, Section G: fish transportation.** U.S. Fish and Wildlife Service, Washington, D.C.

McCraren, J.P. and R.W. Jones. 1978. Suggested approach to comparing and reporting loading densities for fish transport units. **Progressive Fish-Culturist** 40(4):169.

Meyer, F.P., K.E. Sneed and P.T. Eschmeyer (eds.) 1973. **Second Report to the Fish Farmers.** U.S. Fish and Wildlife Service, Res. Pub. #113, 123 pp.

Miranda, L.E. and A.J. Sonski. 1985. Survival of red drum fingerlings in fresh water: salinity and thermal minima. **Proceedings of the Annual Conference Southeastern Association of Fish and Wildlife Agencies** 39:228-237. (In press.)

Moss, D.D. and D.C. Scott. 1961. Dissolved oxygen requirements for three species of fish. Trans. Am. Fish. Soc. 90(4):377-393.

Norris, K.S., F. Brocato, F. Calandrino and W.N. McFarland. 1960. A survey of fish transportation methods and equipment. Calif. Fish Game 46(1):6-33.

Parker, N.C. 1984. Culture requirements for striped bass. pp. 29-43 *In* J.P. McCraren, ed. **The aquaculture of striped bass: a proceedings.** UM-SG-MAP-84-01. University of Maryland Sea Grant, College Park, Maryland.

Perret, W.S., J.E. Weaver, R.O. Williams, P.L. Johansen, T.D. McIlwain, R.C. Raulerson and W.M. Tatum. 1980. Fishery profiles of red drum and spotted seatrout. Gulf States Marine Fisheries Commission, Ocean Springs, Mississippi.

Robertson, L., P. Thomas and C. Arnold. 1985. Physiological evaluation of transportation stress in cultured juvenile red drum. p. 2 in **Proceedings of the Texas Chapter, American Fisheries Society, 7.** University of Texas Marine Science Institute, Port Aransas, Texas. (Abstract.)

Robertson, L., P. Thomas, C.R. Arnold and J.M. Trant. 1987. Plasma cortisol and secondary stress responses of red drum to handling, transport, rearing density and a disease outbreak. **Prog. Fish-Cult.** 49(1):1-12.

Schnick, R.A., F.P. Meyer and D.L. Gray. 1986. A guide to approved chemicals in fish production and fishery resource management. University of Arkansas Cooperative Extension Service, Little Rock, Arkansas and U.S. Fish and Wildlife Service, LaCrosse, Wisconsin. 24 pp.

Turner, C.J. 1984. Striped bass culture at Marion Fish Hatchery. pp. 59-85. *In* J.P. McCraren, ed. **The aquaculture of striped bass: a proceedings.** UM-SG-MAP-84-01. University of Maryland Sea Grant, College Park, Maryland.

Wedemeyer, G. 1972. Some physiological consequences of handling stress in the juvenile coho salmon (*Oncorhynchus kisutch*) and steelhead trout (*Salmo gairdneri*). **Journal Fisheries Research Board of Canada** 29(12):1780-1783.

Wedemeyer, G. 1970. The role of stress in the disease resistance of fishes. pp. 30-35 *In* S.F. Snieszko, ed. A symposium on diseases of fishes and shellfishes. **American Fisheries Society Special Publication No. 5.**

Wedemeyer, G.A. and J.W. Wood. 1974. Stress as a predisposing factor in fish diseases. **U.S. Fish and Wildlife Service Fish Disease Leaflet** 38. 8 pp.

Environmental Requirements of Red Drum

William H. Neill
Department of Wildlife and Fisheries Sciences
Texas Agricultural Experiment Station and
Texas A&M University
College Station, Texas 77843

All organisms thrive only under specific sets of environmental conditions. For euryhaline fishes, meaning those such as red drum (*Sciaenops ocellatus*) that can tolerate substantial variation in salinity, important factors of the physico-chemical environment include salinity, temperature and dissolved oxygen. When fish live at high densities, as they must in intensive aquaculture, metabolic wastes such as ammonia and its derivatives assume importance.

Environmental requirements may change over the life-span of the organism. Red drum and other marine sciaenids begin life in seawater, then passively drift or actively migrate into the brackish waters of bays and estuaries where they pass their larval and juvenile stages. As the fish grow toward adulthood, they migrate seaward and, in the process, encounter progressively increased salinity. In long-lived species such as red drum, the general seaward migration may involve a series of annual migrations, with fish moving into deeper, more saline water in winter (and perhaps also during the warmest weather of summer) and returning to fresher parts of the estuary during spring and fall. Presumably, these features of life history reflect age/size-dependent shifts in tolerable and optimal values of environmental variables.

Physiological Roles of Important Environmental Factors

A fish may be considered a moving, growing and reproducing physiological "engine" that uses oxygen (and nutrients from food) and produces wastes such as ammonia. Temperature sets the pace, or metabolic rate, of the engine. In water with non-optimal levels of dissolved solids (in aggregate, salinity), the engine must do extra work to maintain a proper internal environment with respect to water and ions (osmoregulation); such metabolic loading reduces the engine's capacity for other work, such as growing.

Dissolved Oxygen

Oxidation of food materials or stored energy reserves requires oxygen that generally must be acquired via the gills from the water. The fish's respiratory and circulatory systems are designed to function efficiently over a range of dissolved oxygen concentrations, from air saturation down to a critical oxygen concentration, that, in effect, marks the lower limit for prolonged existence. The critical oxygen concentration is increased by elevated temperature, activity, and metabolic loading caused by environmental stressors such as ammonia and non-optimum concentrations of ions.

Temperature

Rates of all chemical reactions, including those that comprise metabolism, vary with temperature. Ordinary fishes like red drum are poikilotherms; that is, heat moves so readily between the water and the fish that its internal tissues are typically at nearly the same temperature as the water– except when water temperature is changing rapidly, on time-scales of seconds or minutes. Although thermal lag occasioned by such rapid changes may play an important role in the fish's ability to detect and orient to temperature gradients, we may conclude that rates of metabolic work inside the fish generally are set by average temperature of water surrounding the fish.

A resting fish's metabolic demand for oxygen generally increases exponentially with temperature, doubling or trebling for each 10°C rise (i.e., $2 < Q10 < 3$). However, the physiological capacity to supply this oxygen rises to a maximum at some intermediate temperature and then declines precipitously as temperature increases farther. The difference between obligatory demand for oxygen and capacity to supply it is called metabolic scope. Metabolic scope reaches a maximum at a species-specific optimum temperature, which is somewhat lower than the temperature permitting maximum supply. Under otherwise favorable conditions, the optimum temperature for many warm-water fishes is 24° to 30°C. Because environmental stressors can elevate obligatory demand for oxygen and reduce physiological capacity to supply it, these stressors can reduce metabolic scope at all temperatures and shift (in general, reduce) the optimum temperature for maximizing the surplus metabolic capacity that, in combination with adequate food, leads to growth.

Beyond lower and upper extremes of temperature, fish cannot grow and they can live only for a limited time. In general, the logarithim of survival (=resistance) time at such lethal temperatures is linearly related to temperature. If the fish can resist heat death at 32°C for 1,000 minutes and at 34°C for 100 minutes, it will die at 36°C in only 10 minutes. The specific relationship depends on the fish species (and perhaps size/age), general health, past thermal experience, and levels of non-thermal factors such as dissolved oxygen and salinity.

Salinity

Salinity is a term used to quantify the aggregate of solids dissolved and mostly ionized in water. Principal contributors to salinity of both seawater and the blood plasma of fish are Na^+, Cl^-, SO_4^-, Mg^{++}, Ca^{++}, and K^+; however, both the ratios and the total concentrations of these ions differ markedly between seawater and the blood plasma of fish. Moreover, euryhaline fish generally are able to maintain near-constant concentrations of ions in their blood plasma despite considerable variation in environmental salinity; thus, such fish as red drum are good osmoregulators. But, osmoregulation requires metabolic work, and the amount of this work must increase as environmental salinity diverges from the optimum. The optimum environmental salinity is not necessarily that which is equal (isosmotic) to blood salinity.

Hardness

Another consideration ancillary to salinity, but central to osmoregulation, involves water "hardness" conferred by Ca^{++} and Mg^{++}. Seawater is hard; it contains about 400 ppm Ca^{++} and 1,200 ppm Mg^{++}. Very hard freshwater also may contain 400 ppm Ca^{++} but usually less than 200 ppm Mg^{++}. Hardness ions are important in that they limit permeability of cell membranes to other ions and to water. Fish generally must do more osmoregulatory work in soft water than in hard water.

Ammonia

No discussion of potentially important environmental factors would be complete without some consideration of ammonia and pH. Ammonia is the chief end-product of protein catabolism (break-down) in fish and, in warm water with pH above 8, a substantial proportion occurs in the highly toxic un-ionized form (see table 2.9 in Boyd, 1979). Un-ionized ammonia in the water interferes with oxygen uptake and ammonia excretion at the gills. In unpolluted natural waters, ammonia seldom reaches concentrations high enough to be toxic to fish, but, in intensive fish culture, ammonia produced by the fish themselves and through decomposition processes can reach lethally high levels, particularly in alkaline waters.

Ammonia in water can be converted by bacteria to nitrite and then to nitrate. In recirculating aquaculture systems, most of this conversion is accomplished in a biofilter that provides surfaces on which the bacteria live and over which the water is circulated. In natural waters and in aquaculture ponds, oxidation of ammonia to nitrate must be accomplished by the microflora that live on the substrate and on particles suspended in the water.

Ammonia's intermediate oxidation product, nitrite, is also toxic to fish. However, problems involving nitrite toxicity are rare in waters that contain concentrations of chloride above 100 ppm.

Adaptations of Fish to Environmental Variation

In the previous section, we saw that fish have characteristic physiological responses to important components of the physico-chemical environment, and that the response to one component typically depends on levels of other components, giving rise to complex physiological interactions. Having established this, it is important to realize that the environment varies from time to time and from place to place, and that fish cope with such variation both by physiological and behavioral means.

Fish, like other organisms, have evolved an array of behaviors that allow them to exploit spatial variation of environment; behaviors that tend to optimize environment we call habitat selection or enviroregulation. Thus, fish move toward the surface when dissolved oxygen is deficient and into deeper water when a passing cold front drops water temperatures. Such responses are obvious. What may be less obvious is that fish continuously enviroregulate, taking advantage of environmental gradients and patchiness that may be inconspicuous to the casual observer. The benefit to the fish is that it keeps itself in those parts of the habitat that are environmentally best. The disadvantage to the fish farmer is that the fish aggregate at high densities in only a fraction of the habitat, leaving the remainder under-used. From this, it follows that fish farmers should strive to maintain all of the habitat (pond, raceway, etc.) at environmental values near optimum for the species being cultured.

Variations in habitat quality over time cannot be exploited through enviroregulatory behavior. In dealing with temporal variation, fish must rely on physiological mechanisms of adaptation that are termed acclimation or acclimatization. In essence, exposure to a change in the level of some environmental factor results in physiological adjustments that enable the fish to better perform at the new level of the factor and shift the fish's lethal limits in the direction of the change. For example, transfer of a 20°C acclimated fish to 30°C eventually will result in increased metabolic scope at 30°C and in greater heat tolerance (but lowered cold tolerance).

Fish make acclimatory adjustments to temperature, dissolved oxygen, salinity, ammonia, and many other physico-chemical factors. But these adjustments take time – from several hours to several weeks. The rates of acclimatory processes seem to have evolved to keep pace with normal rates of seasonal change in environment.

Red Drum Specifics

Now that the general pattern of fish-environment relations has been established, it is time to focus on red drum. At the outset, it must be admitted that specific environmental requirements of red drum are not well known.

Salinity, Water Hardness, and Osmoregulation

In their natural habitat, adult and large subadult red drum most often are found in diluted/concentrated seawater of 20 to 40 ppt and rarely above 50 ppt; however, juvenile red drum can be found in the freshest parts of the bays. Red drum eggs and newly hatched larvae apparently require salinities above 25 ppt; however, juveniles larger than about 4 cm (standard length) tolerate direct transfer from seawater to freshwater.

Experiments suggest that juvenile red drum can survive and grow in water with less than 1 ppt dissolved solids, provided that sufficient hardness (>100 ppm Ca^{++}) chloride (>150 ppm Cl^{-}) are present. Salinities between 5 and 10 ppt seem optimum for growth of fish 1-10 cm in standard length. At larger sizes, the optimum salinity for growth probably increases; however, the evidence for this is only circumstantial: Dr. D.E. Wohlschlag and associates have found that standard metabolism of 15-19 cm (standard length) red drum is minimum in seawater diluted to about 20 ppt (although the blood plasma is isosmotic with seawater diluted to about 11 ppt). Similar experiments with subadult to adult spotted seatrout (*Cynoscion nebulosus*) show that they, too, do least osmotic work at salinities near 20 ppt.

Red drum stocked in various inland waters, with non-seawater-like mixtures of ions ranging from near zero to 15 ppt in aggregate, have survived and, in some cases, grown to adult size. Still, I must caution that most of our inferences about salinity requirements of red drum are based on observations or experiments in diluted/concentrated seawater or in artificial media made with seawater-like proportions of the various salts.

Temperature, Thermal Tolerance, and Behavioral Thermoregulation

Among the more serious obstacles to culture of red drum is their relative intolerance to cold, which makes outdoor overwintering of this species problematic in most of Texas and the other Gulf states. We believe that osmoregulatory work is the common denominator of an important interaction among salinity, water hardness and cold stress. Our experiments with juvenile (1 to 4 cm standard length) red drum suggest that cold tolerance is greatest in 5 to 10 ppt water with high hardness (>100 ppm Ca^{++}). Under these conditions, juvenile red drum can survive a very gradual decline in temperature to values as low as 8° to 10°C (ultimate lower lethal temperature); reduction of hardness to 10 ppm Ca^{++} and either reductions or increases in salinity raise ultimate lower lethal temperatures into the teens. Still higher temperatures can be fatal if warm-acclimated red drum are subjected to an abrupt decline in temperature that persists longer than several hours. But the same fish can survive a plunge to near-freezing temperatures, if the plunge is of brief duration. Thus, red drum have been observed alive at temperatures as low as 2°C in Texas and Florida estuaries.

So how much cold does it take to kill a red drum? The answer to this question cannot be given in degrees or even in degree-days. The answer must await development of a dose-model that can take into account the precise nature of the temperature decline, previous thermal history of the fish, salinity, water hardness and perhaps even photoperiod. And before the model must come more experimentation.

We know less about the red drum's heat tolerance than about its cold tolerance. Red drum have been observed alive at temperatures up to at least 33°C in estuaries.

The optimum temperature for hatching of red drum eggs and larval survival is 25°C (at 30 ppt salinity); presumably, this temperature is near the overall optimum for the species. Very limited data suggest that red drum prefer temperatures between 22° and 25°C. Thermal optima and preferenda undoubtedly are dependent on salinity, for such dependency is characteristic of euryhaline fishes.

Dissolved Oxygen Requirements

Limited observations in our laboratory suggest that the critical oxygen concentration for juvenile red drum is about 2 ppm – in 5 to 10 ppt artificial seawater at 24° C. One would expect the critical oxygen concentration to be least at the optimum salinity and less at low temperatures than at high temperatures.

Ammonia Toxicity

Un-ionized ammonia concentrations above 0.3 ppm adversely affect survival of three-week-old red drum fry; presumably, older fish are more tolerant. However, concentrations of un-ionized ammonia greater than 0.1 ppm can depress growth in some fishes.

Precautions and Limitations

What we do not know about the environmental requirements of red drum by far outweighs what we do know. We know that red drum tolerate broad variation in salinity. We don't know how performance varies with variation in the components of salinity, or how salinity requirements change with size/age. We know that red drum are relatively cold-intolerant, but we don't know enough to predict the consequences of a particular sequence of low-temperature exposure. We know that red drum need oxygen, but not exactly how much, as a function of salinity, temperature, size/age of fish, etc. Given our general ignorance and the probable complexity of interactions between red drum and their environment, about all we can do is make educated guesses that are designed to err on the safe side of expensive mistakes. That is the spirit in which the following recommendations are made.

Environmental Management for Red Drum Culture: Strategy and Tactics

Choice of Site

From an environmental perspective, the ideal site for culture of red drum has access to both seawater and hard (>100 ppm Ca^{++}) freshwater, average minimum January temperatures above 10°C (and/or naturally or industrially heated water), and soils and topography suitable for pond construction. Since these restrictions would limit the choice of U.S. sites to narrow strips of coast in south Texas and southern Florida (or where heated effluents are available), it is appropriate to consider management strategies and tactics that might relax the restrictions.

Winter Temperature

In thermally marginal areas, some protection from cold-kill during winter might be afforded by constructing elongate, extra-deep (8 to 10 feet) ponds that are oriented with the long axis NE-SW (or perpendicular to prevailing cold-weather winds) and protected by windbreaks on the northwest side. The additional costs of such construction and difficulties of harvesting deep ponds could be prohibitive.

If the average minimum January temperature at the site is much below 10°C, the grower must decide whether or not he will try to keep red drum year-round. If the decision is "yes," he must overwinter the fish indoors or in a greenhouse where temperature can be maintained continuously above 10°C and abrupt declines in temperature prevented. If salinity/hardness guidelines (see below) cannot be followed, the minimum temperature should be raised to 15°C.

Would-be producers of red drum who cannot meet the low-temperature requirement must be satisfied with a production strategy that does not involve overwintering fish.

Salinity and Hardness of Water

Unless the would-be producer of red drum is willing to treat his water chemically, sites with only soft (<100 ppm Ca++) freshwater are inappropriate for red drum production. Limited culture of red drum may be feasible without water treatment if either saline water or hard freshwater is available. If, as I suspect, red drum do require increasing salinity as they grow, sites with only hard freshwater or brackish (<5 ppt) water may be limited to production of fingerlings unless the grower is willing to invest in considerable quantities of salt (see below). At present, it would seem prudent for growers to avoid the reproductive phase of red drum culture at any but coastal sites with access to natural seawater of high quality (although we have spawned red drum in artificial seawater at our experimental hatchery in Bryan, Tex.).

Only ordinary and relatively inexpensive chemicals are needed to increase salinity and hardness of water. Contaminant-free rock salt (NaCl) can be used to elevate salinity. Because salt does not react appreciably with other constituents of the water or bottom muds, the amount needed to raise salinity is easily computed: 4,480 pounds of NaCl raises the Cl^- concentration of 1 acre-foot of water by 1 ppt (1,000 ppm). Deficiencies of water hardness (and pH) can be corrected by addition of agricultural lime ($CaCO_3$). Although Boyd (1978) provides guidelines for liming ponds, complexities of chemical interactions between water and soils probably justify professional analysis on a site-by-site basis.

Finally, I urge that the source water for any prospective red drum- culture enterprise be bioassayed (after appropriate chemical treatment) with red drum before resources are committed. This precaution is especially important in the case of saline groundwaters, which are highly variable in their constituents.

Dissolved Oxygen and Metabolites

Conventional tactics would seem appropriate for red drum: Avoid overstocking and overfeeding; aerate whenever dissolved oxygen at a depth of 50 cm (20 inches) drops below 3.0 ppm. Expect production to suffer whenever dissolved oxygen is below 5 ppm or un-ionized ammonia exceeds 0.1 ppm at a depth of 50 cm.

Literature Cited

Boyd, C.E. 1979. **Water Quality in Warmwater Fish Ponds.** Auburn University Agricultural Experiment Station, Auburn University, Ala. 359 p.

Stickney, R.R. 1979. **Principles of Warmwater Aquaculture.** J. Wiley and Sons, Inc., New York, NY. 375 p.

Nutrition and Feeding of Red Drum

Edwin H. Robinson
Mississippi State University
Delta Branch Experiment Station
Stoneville, Mississippi 38776

The nutrition of terrestrial animals has been intensively studied for many years, whereas research concerning the nutrition of fish is in its infancy. Because of the lack of nutritional information, the composition of the first prepared fish feeds was based largely on the proximate composition of natural foods consumed by the fish or the nutrient requirements of other simple-stomached animals. Feeds formulated on these bases were adequate at low stocking densities because of the contribution of natural foods. However, as stocking rates increased, the need for more precise nutritional information became apparent.

Fish nutrition researchers are faced with the unique problem of working with aquatic animals, but by adaptation of research methods common in terrestrial animal research and by development of new methods, some 40 chemicals have been shown necessary for the normal metabolic function of fish. Qualitatively, these requirements are similar to those of land animals; i.e., amino acids, fats, carbohydrates, vitamins, minerals, oxygen and water are required for normal growth and development.

Most of the nutritional research with fish has been conducted with the salmonids and with catfish; little work has been done concerning the nutritional needs of red drum. However, using feeding habits of red drum in nature, nutritional data from studies with other fish, and available nutritional data from red drum studies (summarized below), feeds can be formulated that are adequate for rearing red drum in captivity.

Post-larval red drum of all sizes feed primarily on small crabs, but shrimp and small fish are also consumed. Growth in nature is rapid, with fish reaching about one pound in the first year, about three pounds the second year, and six to eight pounds the third year. Based on feeding habits and growth rates in nature, red drum require a feed that contains liberal levels of high-quality protein. Further, the carnivorous nature of red drum indicates that some animal protein is needed in the diet to provide critical nutrients and to increase palatability. (This has been true of other carnivorous fish studied thus far.)

Though some nutritional requirements may be estimated based on feeding habits, such observations provide little insight into the requirements for micronutrients such as amino acids, vitamins and minerals. Until more nutritional information is available, formulations for red drum feeds will have to be based, in part, on data derived from nutritional studies with other fish.

Nutrition and Feeding Studies

Summary

Nutrition and feeding studies with red drum have been conducted at various universities and state agencies. Most research has been directed toward early life stages and little research has dealt with the grow-out phase of red drum production. Even though pond grow-out data are needed, more basic nutritional research underlying grow-out is essential in order to develop red drum feeds. Although knowledge of the quantitative requirement for each nutrient is desirable, a feed specific for red drum can be formulated if quantitative requirements for protein and energy, lysine and methionine, fat, and phosphorus are known. Once the initial diet is formulated, refinements can be made as data become available.

Early work at The University of Texas Marine Research Laboratory demonstrated that juvenile red drum required approximately 50 percent dietary protein for optimum growth in salt water. More recent research conducted at the Aquacultural Research Center at Texas A&M University concentrated on the nutritional requirements of red drum reared under controlled conditions in closed recirculating systems at 6 ppt salinity. The major contributions of this research are as follows: 1) juvenile red drum diets should contain 35 to 45 percent high-quality protein and 3.5 to 4.0 kilocalories of energy per gram of diet; 2) five to six percent fat was adequate for good weight gain, whereas high levels of fat (12 percent and above) depressed weight gain; 3) approximately 0.86 percent dietary phosphorus was required for proper bone mineralization; 4) the dietary lysine requirement was between 4.6 and 5.7 percent of the dietary protein; and 5) some animal protein was required in the diet. Additional work at Texas A&M has demonstrated that tilapia and crawfish were good forage species for red drum grown at low densities.

Information concerning the feeding of food-size red drum under pond culture conditions is limited. Weight gain in various saltwater and freshwater ponds indicate potential to reach a marketable size within one or two years. A pond grow-out study conducted in Alabama at the Claude Peteet Mariculture Center resulted in the production of from 946 to 2,041 pounds per acre of 0.5 to 0.75 pound red drum in 10 months. Fingerling red drum were stocked from 1,700 to 6,700 per acre. Weight gain was slow because of cold temperatures and mechanical problems.

Studies conducted in South Carolina at the Waddell Mariculture Center have shown that red drum can be raised from fingerlings to over three pounds in 22 months on pelleted trout feed (38 percent protein). Production was 2,786 pounds per acre with a survival of 95 percent. Several other feeding studies have been conducted but under less controlled conditions. The results from these studies are quite variable, but generally indicate that red drum can be grown to a harvestable size in salt water and certain fresh waters using commercial feeds. Feed conversion ratios (the amount of dry feed it takes to produce one pound of fresh, whole fish) from 1.5 to greater than six have been reported. It is likely that conversions of 1.8 to 2.0 can be routinely achieved under culture conditions.

Precautions and limitations

Since feed cost represents approximately one-half of production costs in aquaculture, careful consideration should be given to the feed used. Bear in mind that although red drum can be produced using trout and catfish feeds, a feed formulated specifically for red drum should be more efficient. Since there is limited information available concerning the actual nutritional requirements for red drum, any feed recommended for grow-out of red drum is a "best guess" based on the data currently available. As with any other feed, initial formulations for red drum feeds will require refinement. As you read the following section be aware that much of the discussion is based on experiences with other warmwater fish and is not necessarily specific for the red drum. However, the information should be applicable to the culture of red drum.

Procedures

Feed selection

Proper feed selection is important from both a nutritional and economical view. A feed should supply the necessary nutrients in a form that is readily consumed by the fish and at an economical price. Feed quality and type are of primary importance when selecting a feed.

Feed quality is primarily determined by the ingredients that are mixed to make the feed. The culturist should be aware that a mixture of ingredients is necessary to provide a balance of nutrients because no single ingredient is adequate as the total nutritive source. Thus, a high quality feed should contain a blend of animal and plant products as well as supplemental vitamins and minerals.

Brand Name

32% Floating Fish Food

GUARANTEED ANALYSIS

Crude Protein	not less than	32.00%
Crude Fat	not less than	4.00%
Crude Fiber	not more than	5.50%
Moisture	not more than	12.00%

INGREDIENTS

Composed of Soybean Meal, Ground Yellow Corn, Fish Meal, Rice Bran, Animal Fat, Dicalcium Phosphate, Vitamin A Acetate (Stability Improved), D-Activated Animal Sterol (Source of Vitamin D3) in Gelatine Sugar Starch Beadlet, Vitamin E Supplement, Vitamin B12 Supplement, Riboflavin Supplement, Niacin, D-Calcium Pantothenate, Folic Acid, Menadione Sodium Bisulfite, Complex (Source of Vitamin K Activity), Pyridoxine Hydrochloride, Thiamin Mononitrate, Choline Chioride, d-Biotin, Manganous Oxide, Zinc Oxide, Iron Carbonate, Copper Oxide, Calcium Iodate, Cobalt Carbonate, and Coated Ascorbic Acid Granules (Vitamin C).

Manufacturer's Address

Figure 1. Example of a feed tag from a bag of catfish feed. (Brand name and address omitted.)

Most feeds manufactured in the United States for use in fish culture are of relatively high quality. Feed formulations are generally proprietary and the exact composition is not released to the culturist. However, the feed tag (see example in Figure 1) provides information concerning the composition of the feed as well as certain nutritional information. The culturist can use this information to assist in judging the nutritional quality of the feed. For example, using the example given in Figure 1, it can be seen that the feed contains a high level of protein and is low in fiber; this generally signals a high quality feed. Also, it can be seen that the feed contains soybean meal (a high-quality plant protein) and fish meal (a high-quality animal protein). If questions arise concerning feed composition that cannot be determined from the feed tag, ask a representative of the feed manufacturer. Although the representative may not reveal actual amounts of a particular ingredient, they should be willing to reveal nutrient sources which will be beneficial in judging feed quality.

Feed quality is also dependent on processing. The culturist has little control over processing, but certain characteristics of the feed will indicate whether it has

Table 1. Suggestions for feeding red drum[1].

Fish size (inches)	Feed type[2]	Protein level (percent)	Feeding rate[3] (percent)
1 – 3	#2 crumble	45	5
3 – 6	#3 crumble or 3/32 to 1/8 inch sinking or floating pellet	45	5
6 – 8	3/32 to 1/4 inch sinking or floating pellet	35–45	4
8 – harvest	3/32 to 1/4 inch sinking or floating pellet	32–38	3

[1]Modified from data provided by Texas Parks and Wildlife Department and Texas A&M University
[2]Recommended feed for small fish (1 or 2 inches) is a feed containing high levels of animal protein, such as a salmon feed. Other feeds containing high levels of animal protein can be used. Larger fish (6 inches or more) may be fed a pelleted feed that contains lower amounts of fish meal; that is, a feed formulated for redfish, a trout feed, or a high-quality catfish feed.
[3]Daily feeding rate based on percentage of body weight of standing crop. Fish may be fed to satiation if so desired. If satiation feeding is used, comply with cautions given in the feeding section of this paper. These rates are based on temperatures that are in the optimum range for red drum growth.

been properly processed. For example, there should be a minimum of feed dust ("fines") and the feed should not crumble easily from simply applying pressure with the hand. Also, if a floating feed is used the feed should float. Sinking feeds should have adequate water stability in order to provide the fish an opportunity to consume the pellet before the pellet dissolves in the water. Culturists can observe these characteristics and make some judgement as to the quality of the feed.

In certain situations, it is not economical to select the highest quality (generally more expensive) feed. For example, if the culturist wishes to feed fish stocked at low densities where natural foods are available, lower quality feeds which need not provide all essential nutrients may be adequate. These feeds are called "supplemental feeds" because they supplement the natural food available in the pond. On the other hand, if the culturist wants to get maximum production in heavily stocked ponds or to feed fish confined in cages or raceways, a high quality feed that provides all the essential nutrients (complete feed) should be used. Most commercial fish feeds are complete feeds. Basically, there is no real need to pay for an expensive feed if one is not needed; select the quality of feed that fits your particular management scheme.

Feed type is largely dependent on fish size and management practices. Fry require a feed of small particle size (flour or meal), while fingerlings require a larger particle size (various crumbles or pellets) and advanced fingerlings to marketable fish require a still larger size (large pellets) feed. Selection of a feed that can easily be consumed should give the best performance. Recommendations of feed type are given in Table 1.

Most of the feed used in fish production will be fed to fish ranging in size from advanced fingerling to marketable-size. Pelleted feeds, either floating (extruded) or sinking (hard pellet), are used. Most culturists, at least in catfish farming, prefer the floating feed because of its management value; that is, they can observe the fish feeding and may be able to detect diseases or other problems. Other advantages of the floating feed include better water stability and no loss of pellets in bottom muds. However, floating feeds cost 10 to 20 percent more to manufacture than sinking feeds.

Although sinking feeds are less expensive, their use requires a higher level of management because of the possibility of losing pellets in the bottom muds and water stability of the pellets is relatively short. Production of catfish using sinking feeds is as good as when using floating feeds, particularly if the manager is experienced. Sinking feeds may be advantagous during winter feeding and when feeding medicated feeds.

There is no clear evidence of whether red drum production is better on sinking or floating feeds. The fish will train to feed at the surface on floating feeds and will take sinking feeds as they fall through the water column. There is some evidence to indicate that a slow-sink pellet might be good for feeding red drum in ponds.

Red drum feeds are likely to be fairly expensive because of the need for liberal amounts of animal protein and total protein in the diet. An example of a feed that could be used for growing advanced fingerling red drum to marketable size is given in Table 2. The formulation given is for a floating feed. The feed could be made to sink with minor adjustments in the

Table 2. Example of a feed for grow-out of red drum from approximately 6 inches to harvest[1].

Ingredient	Percentage (as fed)
Soybean meal (48 percent)	50
Menhaden fish meal	12
Shrimp-head meal	5
Ground corn	28.9
Vitamin premix[2]	0.1
Mineral premix[2]	0.1
Fish oil[3]	1.5
Dicalcium phosphate	2.4

[1]Based on available data on red drum nutrition. Feed formulation is for floating feed but a sinking feed can be made with slight modifications in the formulation or in processing methods.

[2]Based on catfish premixes. Amount added will depend on premix manufacturer.

[3]Spray on finished pellet.

formulation and in the processing method. As previously stated, feeds used to produce trout and catfish can be used to grow red drum. Trout feeds are generally higher in protein and contain more animal protein than a typical catfish feed. A catfish feed that contains 10 to 12 percent fish meal would probably be adequate for grow-out of advanced fingerlings. There is some evidence that feeds containing shrimp meal may increase palatability.

Feed storage

Fish feeds are usually packaged in multi-wall bags or have plastic liners to retard moisture uptake and help protect flavor, aroma, and color.

Nevertheless, both bagged and bulk feeds should be stored in a cool, dry area, since climate is the most important factor determining the effectiveness of a storage area for feed. Care should be taken to provide air circulation space and rodent control in the storage area. Storage time will vary depending upon environmental conditions; however, as a rule of thumb, 60 to 90 days is normally the maximum safe storage time for a fish feed. If feed is to be fed that has been stored for a considerable time, examine it for mold and staleness before feeding. Moldy feeds tend to become lumpy and discolored (bluish-green) and are relatively easily recognized. Do not feed a moldy feed.

Techniques for feeding

Small redfish fingerlings (one inch or so) may be trained to accept artificial feeds within a few days by initially offering a salmon starter feed (or other high-quality feed) mixed with frozen brine shrimp. The fish should be offered the mixture several times a day. Gradually decrease the amount of brine shrimp in the mixture until the fish are consuming adequate amounts of the feed. Training is most easily observed when fish are trained in tanks prior to being stocked into ponds. However, fish can be trained to feed on artificial feeds in ponds by offering the feed as a supplement to the natural food. Feed should be offered at a rate of approximately five percent of body weight divided into at least two feedings per day. It is important to increase the particle size as a fish grows as well as to change the feeding rate. Follow directions given on the feed tag or those suggested in Table 1.

Feeds for advanced fingerlings to marketable-size fish can be either floating or sinking pellets. If a floating feed is used the feed should be placed into the pond from an upwind position. It is probably best to offer sinking feeds at the same place each day. Some catfish culturists have utilized a mixture of 85 percent sinking and 15 percent floating feeds. This strategy gives the culturist an opportunity to observe the fish while reducing feed cost. Slow sinking feeds, which allow the fish to consume the feed as it falls through the water column, may offer advantages for feeding red drum.

It is recommended that feeds be offered in the morning only after the dissolved oxygen levels have started to increase and no later than the middle of the afternoon, to allow digestion to occur during periods of high dissolved oxygen concentrations. Fish of all sizes do not consume and assimilate feed efficiently when the oxygen levels are low.

Feeding rates depend on standing crops, water quality and water temperature. In the single stocking-single harvest program, in which fish of similar size are stocked and grown to a harvestable size, feeding an average of three percent of standing crop is probably adequate. Other culturists may choose to feed to satiation. Regardless of feeding technique, the culturist must be careful, at high standing crops, to offer only as much feed as can be utilized without adversely affecting water quality. Feeding rates recommended for red drum are given in Table 1; these rates are based on a limited amount of data simply because few red drum have been grown from fingerling to harvestable size in ponds.

Culturists who use cages, net pens, and raceways often offer the amount of feed that will be consumed within 10 to 15 minutes. They may feed more than one time per day and benefit from considerably faster growth rate and better feed conversion. Fish reared in ponds also benefit from multiple feedings; however, logistically it is often impractical to feed fish in ponds more than one time a day, particularly on large farms.

Recognition and Control of Diseases Common to Grow-out Aquaculture of Red Drum

S.K. Johnson
Extension Fish Disease Specialist
Department of Wildlife and Fisheries Sciences
Texas A&M University
College Station, Texas 77843

There is much to be learned about red drum diseases and their management. The development of red drum aquaculture is in its early stages. This is particularly true of the focus of this section—the grow-out phase. This paper discusses red drum disease problems in some order of priority and suggests methods for their recognition and management. Selection of diseases for presentation was largely based on experience with disease in the mariculture of warm-water fish, and a more limited experience with red drum grow-out.

Amyloodinium ocellatum (Brown, 1931)

Species Associations

There are a number of parastic dinoflagellates. Those invading fish were taxonomically reviewed by Lom (1981) who disclosed that all true ectoparasitic dinoflagellates of fish were first described as members of the genus *Oodinium*. This taxon includes parasites on invertebrates. Figure 1 depicts the general structure of the parasitic stage of the fish infesting genera. The genus *Crepidoodinium* has one species, *C. cyprinodontum* (Lawler 1967), which is associated with cyprinodontid fishes in brackish water environments. *Piscinoodinium* spp. are associated with a variety of freshwater fishes and the monotypic *Amyloodinium* with a variety of marine fish species. Lom (1981) reports on two other monotypic genera—*Oodinioides* (Reichenbach-Klinke, 1970), which has questionable status as a true dinoflagellate, and *Ichthyodinium* (Hollande et Cachon, 1953), which is an internal parasite of the fry of clupeoid fishes.

Distribution

Amyloodinium ocellatum is a widely distributed fish ectoparasite (Paperna, 1983; Becker, 1977; Lawler, 1980). It is not host specific and a large number of teleosts are infested (Lawler, 1979). There is one report of a severe infection of an elasmobranch (Lawler, 1980).

Parasitism in mariculture is quite common, par-

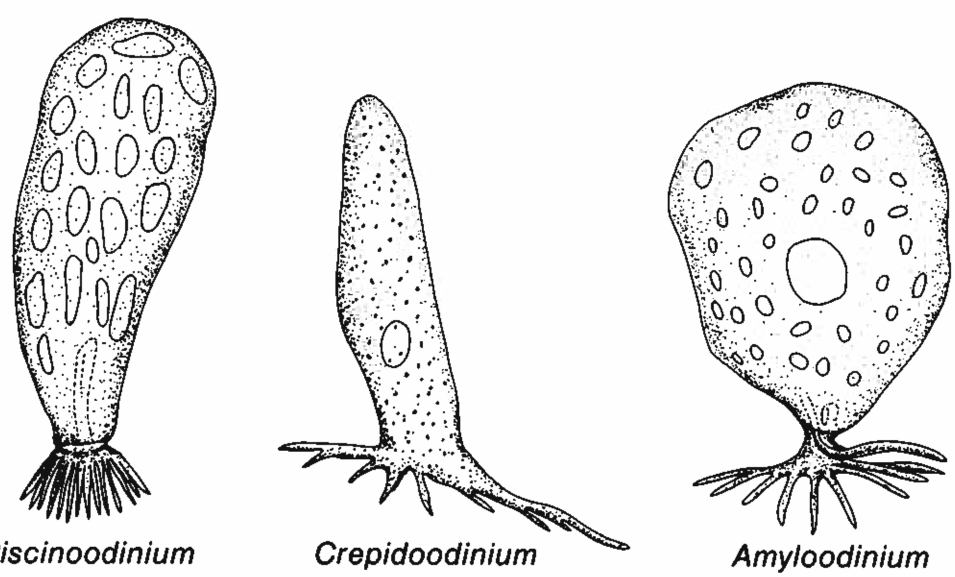


Figure 1. Comparison of dinoflagellates commonly parasitizing fishes. Piscinoodinium *is a genus that infects a variety of freshwater fishes and is sometimes encountered in freshwater aquaria.* Crepidoodinium *is a parasite of cyprinodontid fishes.* Amyloodinium *is the only dinoflagellate parasite expected on red drum.*

ticularly when dense fish populations are maintained. Infestation can be especially serious during the larva-to-fingerling state. Red drum infestations have been common in experimental units in Texas where fingerlings have been used in nutritional and other studies.

Life History

In general, the life cycle of the parasite begins when an infective swarmer (dinospore) encounters a fish host. The swarmer attaches to the host and develops into the parasitic stage (trophont). Then the parasitic stage detaches and transforms into a reproductive stage (tomont) by forming a capsule. The tomont will undergo division to produce as many as 256 dinospores. The time required to complete a cycle at optimal conditions is less than 12 days. This means that, within a short period, a single infective unit could, under optimal conditions, produce tens of thousands of dinospores in the second generation and millions in the third. Knowledge of this reproductive cycle and of *A. ocellatum's* tolerance to temperature and salinity was recently advanced by Paperna (1984a). He found that trophont length at normal detachment varied with temperature, ranging from 85 to 100 microns at 19°to 24°C, to more than 100 microns at lower temperatures (16° C). Attachment normally lasted two to five days at 19° to 24° C. However, smaller individuals that detached after 24 hours at optimal temperatures did divide and quickly produce two dinospores. At 20° C and 20 to 40 ppt, Paperna shows the time period from detachment to production of infective swarmers to be three to six days. This conforms to results by Johnson (1984) and Nigrelli (1936). Bower *et al.* (1987) found dinospores infective for at least six days.

Pathology

Pathological studies have been reported by Lom and Lawler (1973) and by Paperna (1980). *A. ocellatum* causes tissue hyperplasia, fusion of gill lamellae and other degenerative effects commonly produced by many fish ectoparasites.

Control

The biological characteristcics of *A. ocellatum* favor its explosive development in intensive culture situations. It is difficult to prevent infestations in aquaculture because, as Paperna (1984b) has shown, the salinity and temperature ranges suitable for the parasite are extremely broad. The reports of Lawler (1977) and Johnson (1984) support this conclusion. Kingsford (1975) and Lawler (1977b) advocate the use of brief freshwater dips, but agree that eradication is not possible. Trimble (1979) reported that infested red drum recovered after repeated water replacement in a culture pond. Once the parasite has infested a dense fish population, it is difficult to control by selective toxicity. Chemicals for *Amyloodinium* control are listed in Hoffman and Meyer (1974) and in Lawler (1977a). Copper sulfate, formalin, and copper sulfate plus citric acid have been found useful.

Formalin was used by Paperna (1984b). He found in vitro that exposures to 25 to 200 mg/L formalin caused trophonts to transform into tomonts within one to three hours. He demonstrated that complete dislodgement of trophonts was effected by 150 to 200 mg/L formalin in six hours and by 100 ppm in nine hours. The fish were reinfected, however, after dislodged tomonts produced a new generation of dinospores.

The use of copper sulfate for dinoflagellate infestations was initiated by Dempster (1955) in public

Table 1. Control of *Amyloodinium ocellatum* with three chemicals.

Test Chemical	Concentration mg/1 (ppm)	Result 24 and 48 hours after treatment
Cutrine (Chelated Copper)	1.0*	Lightly infested
Cutrine	2.0, 6.0	Uninfested
Copper sulfate ($CuSO_4 5H_2O$)	1.0*	Infested
Copper Sulfate	2.0	Lightly infested
Copper Sulfate	6.0+	-
Benzalkonium Chloride	0.1	Infested
Benzalkonium Chloride	1.0	Lightly infested
Benzalkonium Chloride	2.0+, 4.0+	-
Benzalkonium Chloride	0.1 followed 3 and 5 days later by 0.1	Lightly infested
Benzalkonium Chloride	1.0 followed 2 days later by 1.0	Uninfested

*Application level for plant control
+Notably toxic to fish
-Too toxic for practical application

aquaria and used subsequently for a number of years. One of the problems with the copper produced from copper sulfate (cupric sulfate) is that the copper ion is rather reactive and disappears from the water column in a short time. Aquarists were advised to monitor the level of copper ion so that an appropriate level of copper could be maintained as a parasiticide (Kingsford, 1975).

Other copper compounds followed the use of copper sulfate, chief among them being copper citrate or mixtures of copper sulfate and citric acid (Dempster, 1972). The citrated copper supposedly caused copper to stay in solution longer, but this was not confirmed by Keith (1980). Paperna (1984b) found that copper sulfate (cupric sulfate) at therapeutic levels did not interrupt tomont division but did kill dinospores. Best results were obtained by continuous application of 0.75 mg/1 of copper sulfate for seven days.

Johnson (1984) used infested sciaenids to evaluate the effectiveness of several herbicides, diluted seawater and a disinfectant. Control was achieved with copper sulfate, benzalkonium chloride and a chelated copper compound (Table l).

Copper sulfate gave only partial control at levels (single dose of 2.0 mg/1 in static water for test duration) non-toxic to the fishes. After initial tests revealed that benzalkonium chloride was partially effective in single dosing, a second dosing method was attempted. The initial dose was 1 ppm. A second dose two days later resulted in apparent parasite elimination. Chelated copper compound (Cutrine) gave excellent results at 2.0 and 6.0 ppm; the parasites were eliminated within 24 hours of application. Several hatchery and experimental applications of chelated copper algacides (Cutrine, Applied Biochemists, Waukesha, Wisconsin, USA; Copper Control Argent Chemical Laboratories, Redmond, Washington, USA) have subsequently provided excellent control (Johnson, unpublished). In Alabama mariculture ponds,a chelated compound controlled but did not eliminate the parasite. Elimination was achieved, however, in tank units (John Hawke, Louisiana State University, personal communication). To this author's knowledge, benzalkonium chloride has not been evaluated in the field because of the apparent advantage of chelated copper compounds.

Chelated copper compounds produce rapid results without a premature loss of the therapeutant or the risk of a narrow safety range. There have been claims of success in using chelated copper in aquaculture, but these have not been experimentally verified. It would be advantageous to develop a single dose treatment rather than the continuous application approach. Since many hatcheries operate by recirculation, a single dose treatment would limit metallic input and should better preserve system quality.

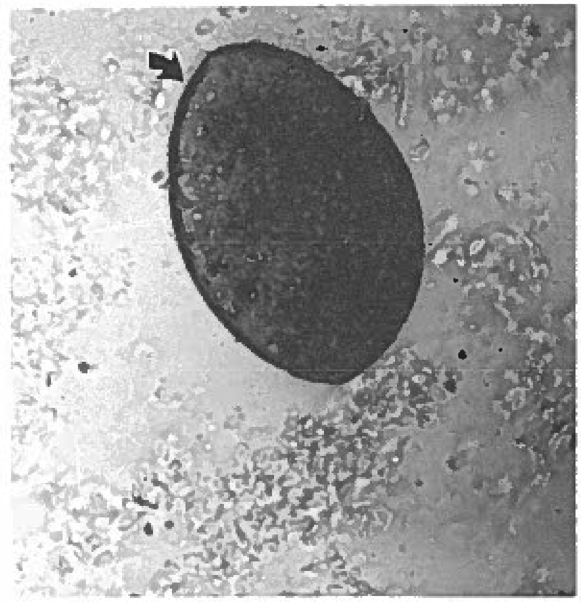


Figure 2. Typical shape of a detached mature parasitic stage of Cryptocaryon. *It is a somewhat indistinctive ciliate which does not have an obvious and characteristic nucleus as does* Ichthyophthirius.

Cryptocaryon irritans Brown 1951

Distribution and Species Associations

The ciliated parasite *Crptocaryon irritans* (hereafter denoted *Cryptocaryon*) is apparently distributed throughout the world on a wide range of marine fish hosts. The parasite is very common in the ornamental fish trade, where it affects many species, and occasionally it is reported in the literature from public aquaria. The red drum has not escaped its effects (Overstreet, 1983 citing Texas location; J. Hawke, personal communication, a Louisiana location).

Corliss (1977) placed *Cryptocaryon* in the family Ichthyophthiriidae Kent along with *Ichthyophthirius multifilis*, its close relative and the cause of "Ich" disease of freshwater fishes. Sikama (1937) first reported the parasite as a marine form of its freshwater relative and later (Sikama, 1961) named it *I. marinus*. By that time, however, Brown in 1951 had already officially named the species *Cryptocaryon irritans*.

The parasite's appearance when removed from its fish host is much the same as *Ichthyophthirius* (Figure 2). An important difference in the two parasites is that trophonts of *Cryptocaryon* do not have the tubular, U-shaped macronucleus characteristic of *Ichthyophthirius*. The macronucleus consists instead of a less distinct set of four spherical bodies linked into a U-shape. This macronucleus is usually obscured by granules or ingested cellular debris and is apparent only with staining. The parasite's body shape is more oval and the buccal cavity more pronounced than *Ichthyophthirius*. *Cryptocaryon* attains a size of up to about 450 x 350 microns and is marine, whereas

Ichthyophthirius may reach twice this size and is a freshwater animal.

Life Cycle

The life cycle of the parasite could be said to start when a free-swimming infective stage (tomite) invades the epithelium of the fish host. Both skin and gills are parasitized. *Cryptocaryon* increases greatly in size within the host tissue during the parasitic or feeding stage (trophont). After three to seven days the trophont leaves the host and transforms into an encysted stage (tomont). Many tomites develop within the tomont until they are released as infective individuals. According to Nigrelli and Ruggieri (1966), 200 or more tomites may be produced depending on the size of the tomont. The estimated the life span of the tomite is 24 to 48 hours. An average duration of the feeding stage apparently has not been determined, but should be several days. The transformation to the encysted or tomont stage takes place in a few hours, but the time from tomont development to tomite release may take four to seven days at 30°C and nine to 12 days at 20°C(Cheung *et al.*, 1979).

Manifestations and Pathology

A pathological summary of cryptocaryoniasis is given by Lom (1984). Factors that reduce resistance cause outbreaks. Infected fish show respiratory distress, restlessness and occasional scratching against surfaces. Macroscopic white to gray pustules form where trophonts burrow under the epidermis. The epithelium becomes undermined and will slough in time. Gills are particularly susceptable to damage.

Control

Ornamental fish handlers have traditionally used 0.1mg/l malachite green. Huff and Burns (1981) concluded that hypersalinity of 45 percent or more would remove excess mucus and better expose *Cryptocaryon* to chemical treatments. Several workers have recommended copper or copper plus formalin treatments in fish tanks (Blaisiola, 1976; Colorni, 1987). Cheung *et al.* (1979), Colorni (1989, 1987) and Herwig (1978) reported that reduced salinity procedures control the parasite. These and other treatment methods for small volumes are given in Table 2.

Chemicals listed in Table 2 would be too costly for pond application. Malachite green is economical, but because of its carcinogenic properties it is not approved for aquaculture use in this country. Copper sulfate (1 mg/l) is economical and has fully controlled *Cryptocaryon* in a Louisiana experimental hatchery tank (J. Hawke, personal communication). The use of chelated copper compounds apparently has not been evaluated in pond systems. *Cryptocaryon* is difficult to control because of its site of infestation. Skin and mucus protect the trophont from chemicals applied temporarily to the water, and thus sustain the life cycle. Exposure to treatment concentrations must therefore be maintained until the parasite exits the host and becomes susceptible.

Bacterial Infections

Bacterial diseases are common in all marine fish culture, and are associated with conditions stressful to fish. Handling, crowding and inferior water quality particularly contribute to the occurrence of bacterial diseases. In comparison to more established marine aquaculture fishes, knowledge of bacterial diseases of red drum is lacking mainly because the culture of this fish is in its infancy.

Kinds of Infectious Bacteria and Distribution

• **Gram negative rods** — Of the various bacteria affecting marine fishes, *Vibrio* is encountered most often. Seven species of *Vibrio* have been described as pathogens of marine fish: *V. anguillarum, V. ordalii V. alginolyticus, V. carchariae ,V. cholerae,V. damsela* and *V. anguillarum* has perhaps been isolated most often from marine fish, and*V. alginolyticus* and*V. parahaemolyticus* also occur frequently. *Aeromonas* spp. commonly infect at lower salinities.

Vibrio and *Aeromonas* consist of rod-shaped, motile, Gram-negative, oxidase-positive, and anaerogenic fermentators of glucose. *Aeromonas* is not sensitive to pteridine compound 0/129, but *Vibrio* is. Laboratory differentiation of the various species is conducted by

Table 2. Some treatments for control of cryptocaryoniasis.

Treatment	Application	Control Achieved	Reference
Reduced salinity	50 percent seawater or more	Yes, in 6 days	Cheung *et al.*, 1979
	10 g/l for 3 h 4 times at 3-day intervals	Yes, in 10 days	Colorni, 1987
Physical	Switch tanks for 4 times at 3-day intervals, dry and clean tanks between uses	Yes, in 10 days	Colorni, 1987
	Move fish to open	Yes	Colorni, 1987

using long-established bacteriological procedures to compare characters (Conroy, 1984; Buchanan and Gibbons, 1974).

These Gram-negative rods can produce either localized or systemic infections. Fish with localized infections are characterized by having frayed fins and surface lesions. Organ swellings, exudate formation in the body cavity and hemorrhaging are manifestations of fishes with systemic infections.

• **Eubacterium** — This bacterial group includes gram positive rods that form into long, unbranched filaments. It occurs in red drum and other marine fish (Henley and Lewis, 1976). It grows in anaerobic environments and require specialized techniques for isolation from brain tissue.

• **Streptococcus** — This group is identified by coccus (spherical or oval) shapes occurring in chains and exhibiting Gram positive staining. Infections typically cause inflammation and hemorrhaging in affected tissues. Notably affected are tissues of the opercular area and intestine. Although not recorded from red drum, *Streptococcus* is considered a problem in marine fish culture in the Gulf region (Rasheed and Plumb, 1984). The group is included here as a possible problem in red drum culture.

Because of their history in other marine fishes, certain other bacterial groups should be considered during isolation procedures. Included would be acid-fast bacteria and myxobacteria.

Dynamics of Development

Bacteria usually affect fishes that are weakened and have reduced resistance. However, if exposed to excessive numbers of virulent bacteria, even fish in good condition can become diseased. Epizootics usually occur when weakened fish harbor infections that increase the numbers of pathogenic bacteria in the water. In those circumstances, the bacteria become both agents of demise and predisposing stressors (Defigueiredo and Plumb, 1977). Marine fish drink water to maintain osmoregulatory balance. This likely enhances the chance for infection through the digestive tract when fish experience stressful conditions. Most *Vibrio* species require salt water to live, but *V. anguillarum* is more euryhaline and has been cultured routinely in media without added salt (Kuo *et al.*, 1976). Obviously, water heavily laden with nutrients will favor the development of excess bacteria including the pathogenic species. This is especially true where decomposing food material of animal origin is present.

Control Methods

General management procedures should include keeping the water as clean as possible, but as culture systems age and food is added, bacterial numbers increase. Some hatchery managers conduct regular bacterial counts and correct with cleaning procedures or increased water exchange. Antibacterials are often added to hatchery water as both a preventative and corrective, but the cost of this procedure is too great to use in ponds.

Instead, antibacterials are administered as a food additive. This practice may be effective unless the infection is widespread in the fish population, in which case oral administration of antibacterials is of little benefit because heavily infected fish refuse to feed. In these cases control methods must focus on killing bacteria in the water column. To accomplish this, oxidizing chemicals such as potassium permanganate have been used with some success (Phelps *et al.*, 1977; Jee and Plumb 1981).

It is important to establish that diseased fish are indeed being killed by bacteria. This may be presumptively accomplished by isolating bacteria from suspect fish. The isolation process makes use of traditional broad spectrum media such as trypticase soy agar (TSA) or brain heart infusion agar (BHI) incorporating small amounts of NaCl (i.e. 3 percent). Thiosulphate citrate bile salts agar (TCBS) also works well for *Vibrio*. Some isolation media are more selective or specific. Examples for routine use are Cytophaga Agar for Myxobacteria and/or agar containing blood for *Streptococcus*. Medium containing blood has too short a shelf life for most field stations. Azide Dextrose Broth or Dextrose Agar dosed with 200 mg/l sodium azide will isolate Gram positive bacteria, which can be further characterized as to genus with a simple catalase test.

Other Disease Conditions

Cold Extremes and Fungal Parasitism

There are many reports of red drum and other coastal fishes dying in large numbers when temperatures reached the teens or when winds of cold fronts drove water temperatures quickly downward. Fungi are sometimes associated with such mortalities (Figure 3).

Red drum become susceptible to fungal disease when their mobility is impaired and they are unable to leave shallow, low salinity waters suddenly chilled by severe winter cold fronts. The fungi develop opportunistically upon fishes weakened by extremes or abrupt changes in temperature. *Saprolegnia* or closely related fungal forms are most common.

Worm and Crustacean Parasites

Red drum host a variety of worm parasites. Perhaps most notorious are the larval tapeworms (spaghetti worms) that invade the edible, muscular areas. These and other worm parasites are not considered a major threat to fish health, but certainly they affect marketability. Examples of worm parasites of red drum are depicted in Figures 4 and 5.

Perhaps 20 crustacean parasites (Overstreet, 1983)

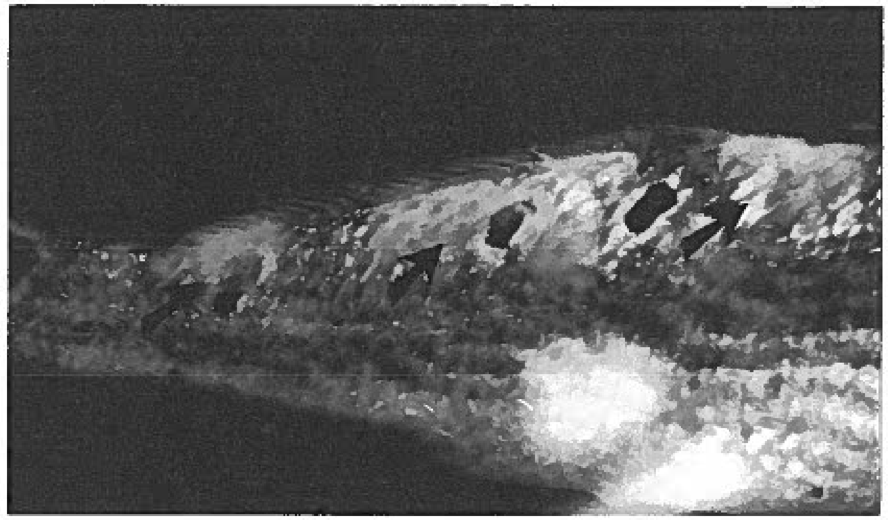


Figure 3. An infestation of red drum with several patches of fungus

will affect the red drum. These parasites (copepods, isopods and branchiurans) produce pathological effects as they affix to external body parts. One group of copepods, the caligoids, attach to hosts as a chalimus larval stage early in their life cycle. Chalimus larvae are often seen infesting younger fish. Fortunately, crustaceans can be controlled with chemicals. Crustacean genera known to parasitize red drum are depicted in Figure 6.

Lymphocystis

Lymphocystis disease affects both freshwater and salt water fishes. It is caused by a virus which invades the cells of the skin tissue and causes a cellular size increase of as much as 10,000 fold. The skin will have a granular or wart-like appearance (Figure 7). Affected tissue will slough off in time and the fish will regain a normal appearance. The main impact upon red drum culture would be product marketability. Although there are no reports of this disease in red drum, the frequency of its occurrence in other drums suggests that it could become a problem.

Trichodinids

Trichodinid protozoa are very common on freshwater and marine fishes, and the red drum is no exception (Trimble, 1979). It is probable that heavy infections will occasionally occur. These external parasites can be controlled by applying formalin as a chemotherapeutic according to established procedures. Trichodinids are depicted in Figure 8.

Toxic Algae

In 1986 there was a devastating red tide along the Texas coast that killed an estimated 22.2 million fish. Water became heavily concentrated with the principal causative agent of Gulf red tide, a toxin-producing dinoflagellate alga named *Ptychodiscus brevis*. This and other toxin-producing algae can affect the culture of red drum.

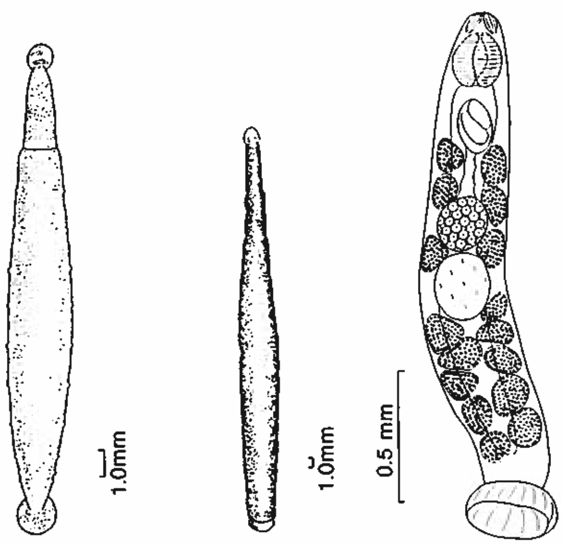


Figure 5. Leeches and a trematode. Calliobdella *and* Mazobdella *have been listed by Overstreet (1983) as leeches that affix in the mouth and branchial cavity of red drum.* Udonella *is a monogenetic trematode that normally attaches to copepods that parasitize fishes and has been noted on red drum also. Overstreet (1983) lists* Neoheterobothrium, Diplectanum, *and* Cynoscionicola *as monogenic trematodes attaching to the gills of other marine drums. Perhaps as more parasitic studies occur some of these will be found to also parasitize red drum. [Figures redrawn from:* Calliobdella *and* Mazobdella *(Sawyer* et al., *1975);* Udonella *(Schell 1985).]*

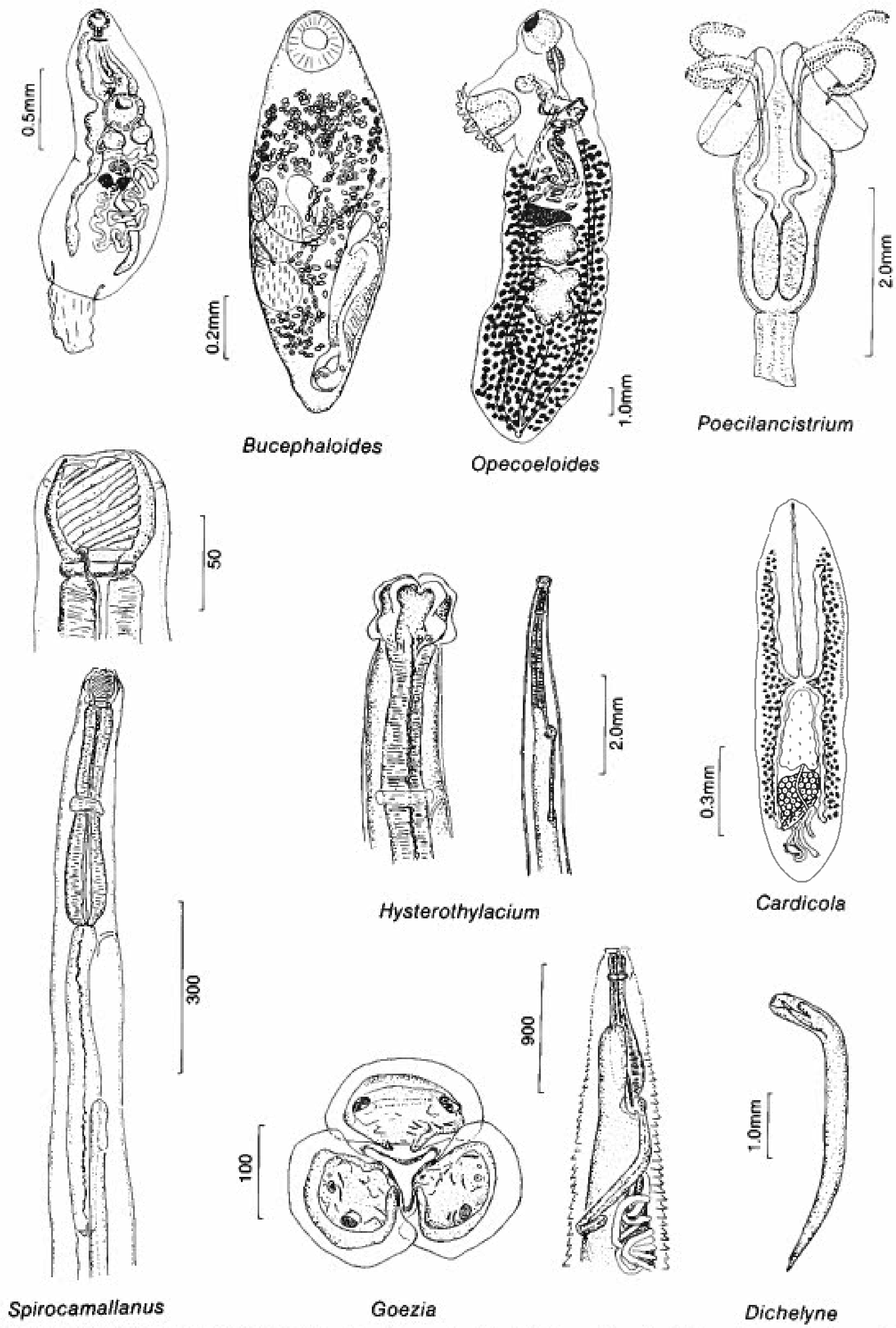


Figure 4. Some reported or probable internal parasites of red drum. Lecithochirium, Bucephaloides, Opecoeloides *and* Cardicola *are internal trematodes. The first three are found in the digestive tract whereas the latter is found in the heart or major blood vessels. Another trematode* (Stomachola) *is sometimes found imbedded in tissues of drums and is large (up to 1/2 inch) and conspicuous (pink).* Poecilancistrium *is a tapeworm that is found as an immature stage in the red drum. It is one of the "spaghetti worms" that commonly infests flesh of drums. The figure shows only the head or scolex.* Spirocamallanus, Hysterothylacium, Goezia, *and* Dichelyne *are nematodes. Except for the latter, views of the head end (anterior) are shown. In the case of* Goezia, *a top view of the head end is shown. [Figures redrawn from:* Lecithochirium *(Manter, 1947);* Bucephaloides *(Riggin and Sparks, 1962);* Opecoeloides *(Sogandares-Bernal and Hutton, 1959);* Cardicola *(Schell, 1985);* Poecilancistrium *(Thatcher, 1960);* Spirocamallanus *(Fusco and Overstreet, 1978);* Hysterothylacium *(Deardorff and Overstreet, 1981);* Goezia *(Deardorff and Overstreet, 1980);* Dichelyne *(Chandler, 1935).]*

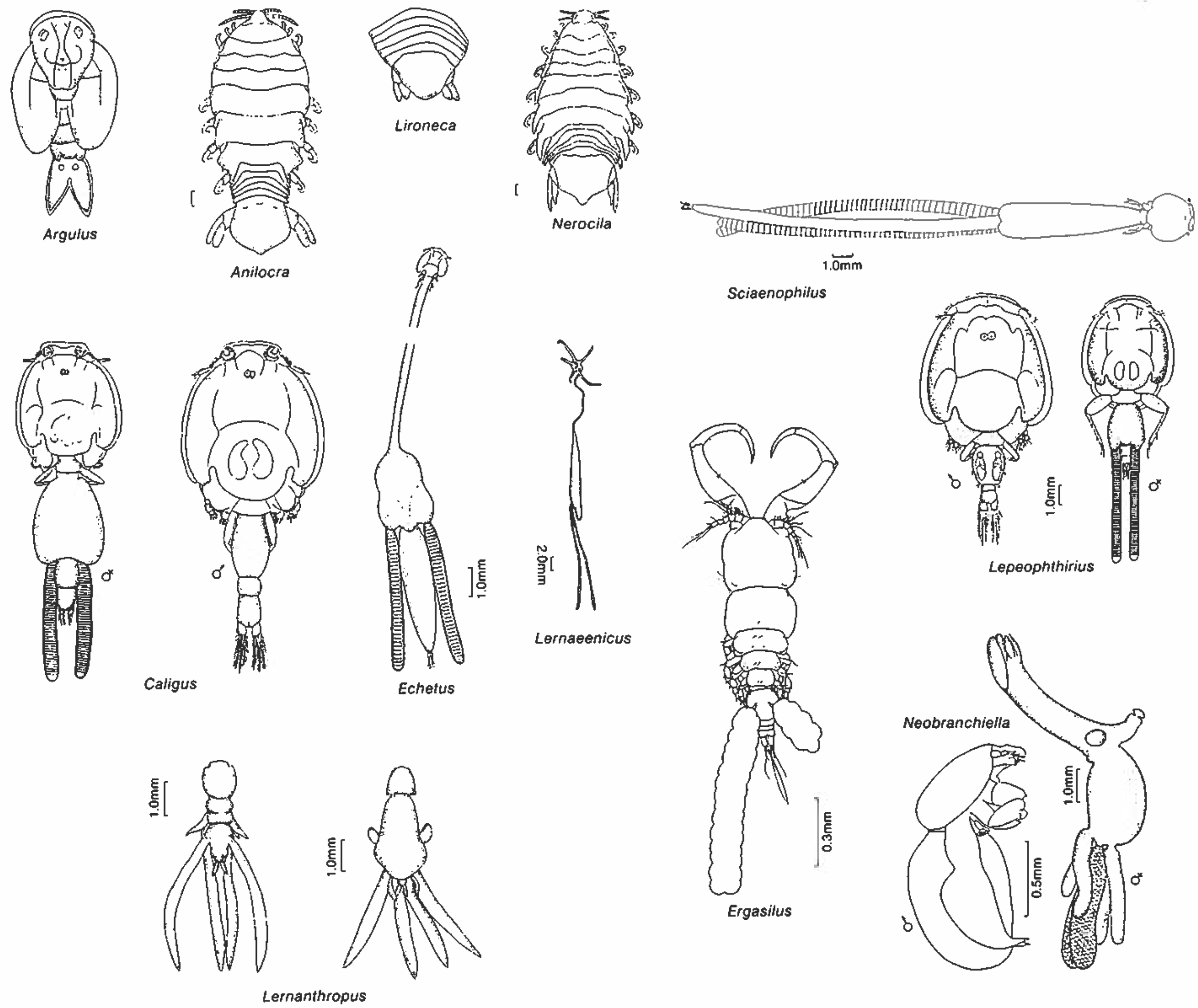


Figure 6. Reported or probable crustacean parasites of red drum Argulus *belongs to the crustacean group, Branchiura. The species reported from red drum is* Argulus *bicolor. More study will probably discover other parasites of this group on red drum. These parasites affix to the skin and surfaces of the mouth cavity.*

Anilocra, Nerocila, *and* Lironeca *are isopod parasites of red drum. The first two are found on the skin and the latter in the gill cavity of the host. Only the posterior portion of* Lironeca *is shown in the figure.* Cymothoa, *another isopod genus sofar reported from drums other than red drum, if found within the mouth cavity.*

Caligus, Echetus, Lernaeenicus, Lernanthropus, Sciaenophilus, Lepeophthirius, Ergasilus, *and* Neobranchiella *are copepod parasites. Most will be found attached to the gills or within the gill or mouth cavity.* Lernaeenicus *will attach by embedding the anterior or head portion into the skin on the body surface.* Caligus *may sometimes be found on the body surface as the parasitic adult.Several of these copepods produce larval stages which infest the skin of an array of fishes. The more common* Caligus *and* Lepeophthirius *produce a larvae with a stage known as chalimus. This larvae will anchor to the host skin by a filament.* Caligus *and* Lepeophthirius *are very similar copepod groups. They may be easily distinguished by the presence in* Caligus *of lunules, which are obvious circular structures lying anterior between the antennae. Ergasilus has not been reported from the red drum; but, because it is a common gill parasite and has been found on other drum species, it is figured as a probable copepod parasite of red drum.*

[Figures redrawn from: Argulus *and* Lernanthropus *(Bere, 1936);* Anilocra, Nerocila *and* Lironeca *(Schultz, 1969);* Caligus, Echetus, Lernaeenicus, Lepeophthirius, Neobranchiella *and* Sciaenophilus *(Yamaguti, 1963);* Ergasilus *(Roberts, 1969).]*

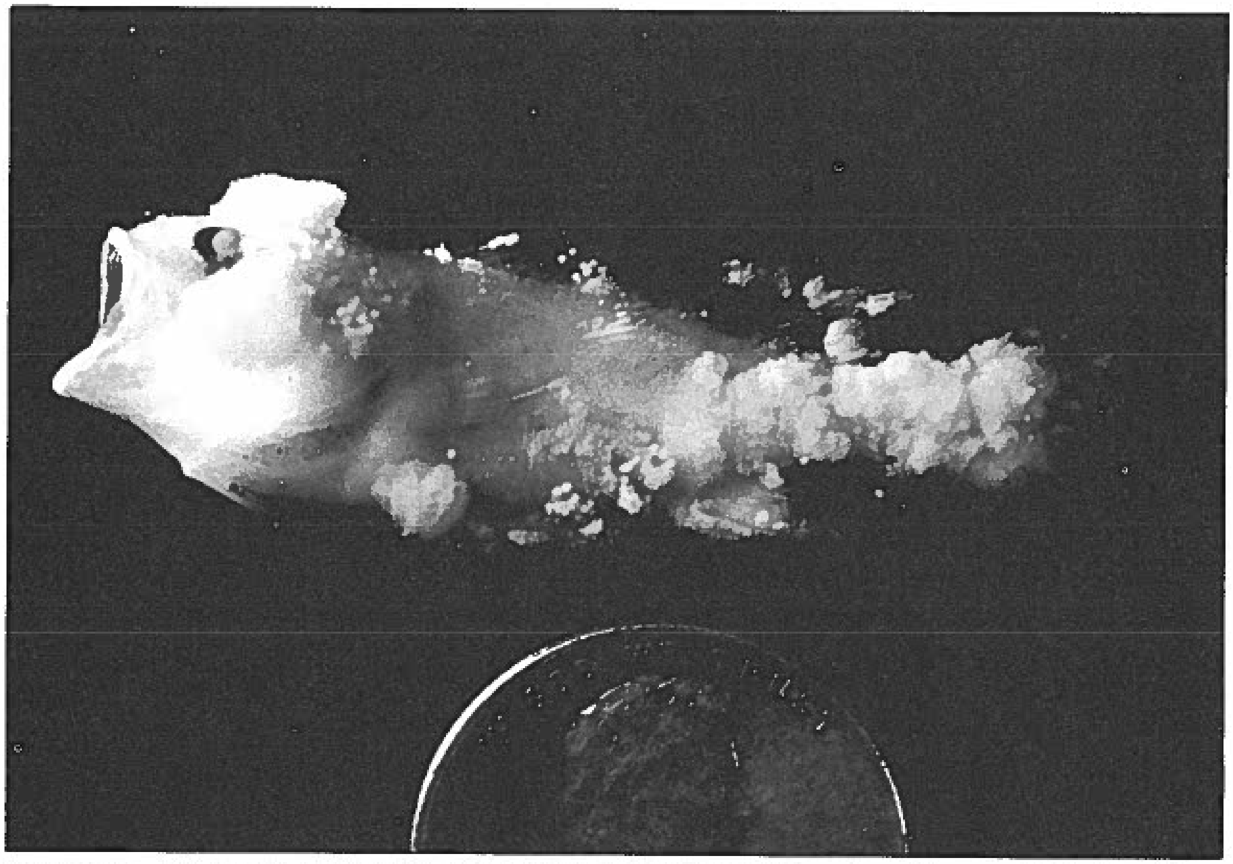
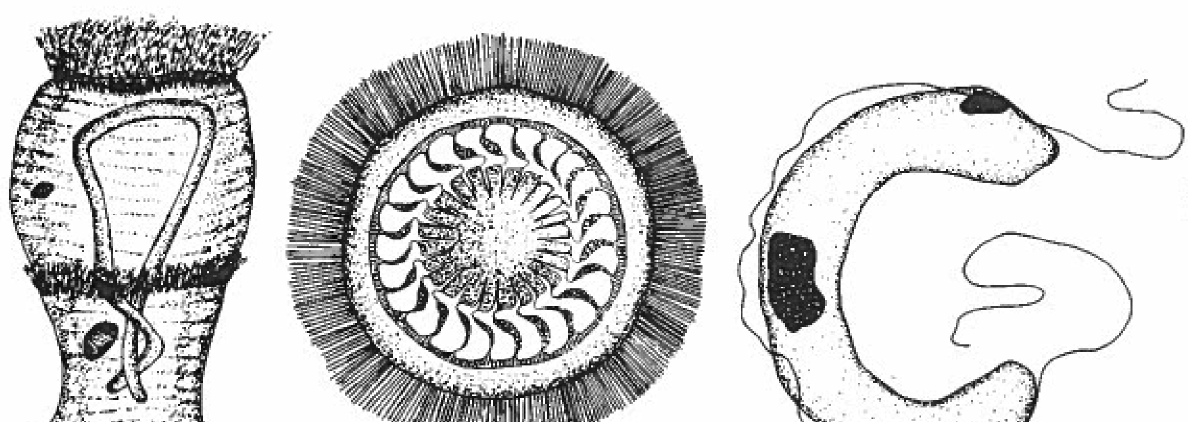


Figure 7. Lymphocystis disease of a drum (star drum). Although lymphocystis disease has not been associated with red drum aquaculture, the prevalence of this viral disease in fingerling drums from wild capture suggests that it could become a potential problem.



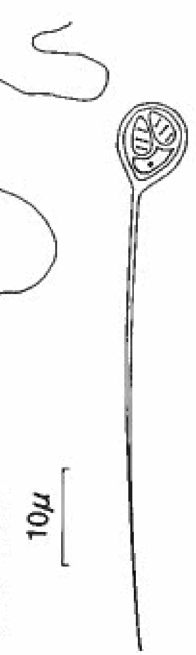


Figure 8. Protozoa expected to occur on or in red drum. Ambiphrya *and* Trichodina *occur on gills and body surfaces of their hosts.* Cryptobia *occurs on the gill surfaces. Henneguya is an internal protozoan parasite with a complex life cycle that has spores form within a capsule in the host tissues. The figure depicts one of many spores that will be found within the capsule.* Tripartiella, *another trichodinid found on the red drum, is very similar in appearance to* Trichodina *and is distinguished by minor features such as extent of ciliary rows about the oral opening. These protozoa should not become a problem in red drum aquaculture unless present in exceptionally large numbers on the fish. [Figures redrawn from:* Ambiphrya, Trichodina *and* Cryptobia *(Wellborn and Rogers, 1966);* Henneguya *(Joy, 1972).*

The normal occurrence of the alga is considered to be 1 individual/ml; the killing threshold is 250/ml. The 1986 red tide reached 100,000/ml in some areas (Martin, 1987). Monitoring water for algae both at the source and in culture ponds appears to be a good management tactic. This would enable the manager to change the water source if it becomes contaminated. Algal development in culture water can be controlled by algacides.

Gonyaulax monilata is another toxin-producing dinoflagellate of the Gulf Coast. Fish kills from this alga are not as common as from *P. brevis*, but cell concentrations capable of producing toxicity are similar (Siever, 1969). *P. brevis* and *G. monilata* are depicted in Figure 9.

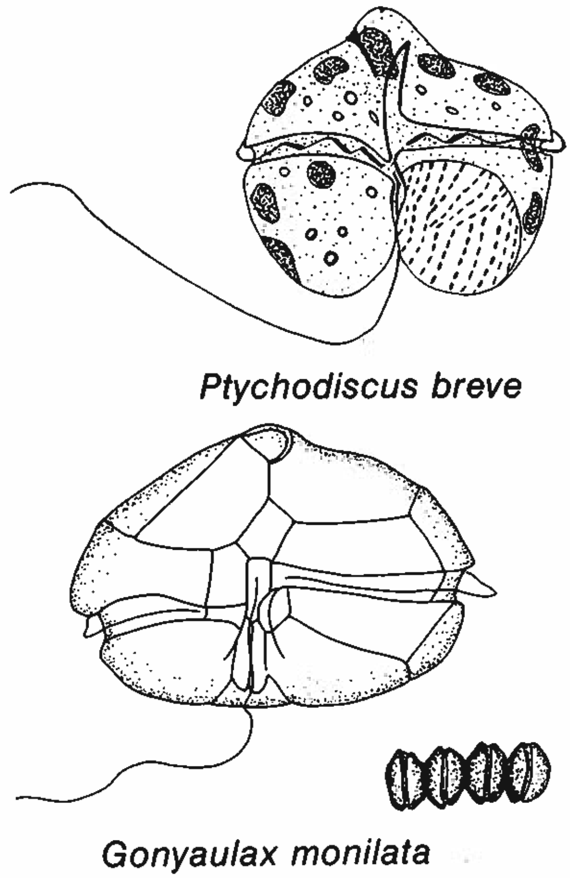


Figure 9. Ptychodiscus brevis *and* Gonyaulax monilata, *two known toxin-producing algae of the Gulf.* Gonyaulax monilata *will occur free or in groupings as shown. [Figures redrawn from:* Ptychodiscus brevis *(Steidinger and Joyce, 1973);* Gonyaulax monilata *(Howell, 1953).*

Recommended Procedures

Collecting And Immobilizing Specimens

Collect moribund (sick), not dead fishes for examination, being careful to keep the fish cool after removal from the culture pond. This may be accomplished by placing the specimen in a plastic bag with a cup of pond water. If the fish is not to be examined within a few minutes, it should be placed on ice.

Even moribund fish will flip about and make examination difficult. Smaller fishes can be subdued with anesthetic in the holding water (MS-222 or quinaldine). Mixing anesthetic with a small amount of water and spraying it onto the gills with a syringe will immobilize larger animals.

Recognition Methods

Parasites may be detected visually. Smaller forms will require the use of a microscope and may be observed by preparing slide wet mounts.

Algae also can be detected with a microscope.

Bacterial and fungal invasion usually can be determined by observing gross manifestations, but it is often necessary to isolate and culture bacteria.

When isolation onto agar produces numerous colonies of the same bacterial species it is a clear indication of infection. Bacteria so isolated may be subjected to a simple antibacterial sensitivity (susceptibility) test to help establish the best method for therapy. Field determination of species is usually not necessary. Assignment to a general bacterial group is easily accomplished by noting the cell structure, by staining or by other simple bacterial techniques. Some bacterial forms (i.e. myxobacteria) can be distinguished from wet mounts using 400X magnification. Treatments will be similar for most types of infection.

Examination For Parasites And Algae

Equipment And Materials

Compound microscope
Dissecting microscope
Glass slides and cover skips
Dissecting kit
Counting slides
Dropper bottle
Small glass dishes (petri dishes)

Methods For Parasites

Protozoa and other small, external parasites may be observed by making wet mounts. The aim of the procedure is to establish and estimate the occurrence of the parasites on skin and gills. For skin examination, some disease specialists suggest scraping a small spatula or scalpel across the epithelium and then removing the scraping into a drop of water prior to applying a coverslip. A # 1 1/2 or thicker coverslip may be used for the same scraping procedure, thus eliminating the use of a dissecting tool. Gills are examined by clipping several filament tips with dissecting scissors and depositing them into a water drop on a slide. Applying a cover-slip completes the wet mount. Wet mount examination is best conducted using 100X magnification. Copepods, isopods and other larger parasites are seen with careful visual examination. Smaller forms of these groups are best detected with a dissecting microscope. Some, such as *Ergasilus*, are typically gill parasites. To examine them, remove a gill arch complete with filaments and place it into a small dish of water. Internal parasitic worms are exposed by dissection. Some will be in the muscular flesh, whereas others are found in and about the organs. Adult tapeworms inhabit the intestine. Once isolated, worms are best observed by placing them in a small dish of water and using a dissecting microscope.

Methods For Algae

Algae can be identified and quantified using various slide apparatuses designed for this purpose. Hemacytometers, Sedgwick-Rafter cells, and specially designed counting slides are available. A Sedgwick-Rafter funnel may be used to concentrate phytoplankton. An inverted microscope is useful for identifying and quantifying of algae. Dinoflagellates are motile and it is helpful to kill them for study. Strong fixative solutions should be avoided because they can disrupt the integrity of the delicate cells.

Examination Methods For Bacteria

Equipment and materials

Flame source (alcohol lamp, propane torch, cigarette lighter)
Compound microscope
Scalpel
90 percent alcohol solution
Cotton
Inoculating loop
Petri dishes
Bacterial isolation media (see below)
Hot plate
Sensitivity discs
Sterilizing unit (autoclave, pressure cooker)
Flask with screw-top lids
Gram stain kit with methanol for fixation
Acid-fast bacteria stain kit Media preparation.

Media preparation

Bacteriological media containing agar are available from various commercial sources. The standard preparation uses powdered materials. An appropriate amount of powder is added to water and the mix is heated to or near boiling to completely dissolve the powder. The mix may then be made sterile by placing it in an autoclave or pressure cooker so as to attain 121 °C for 15 minutes at 15 pounds of pressure. If screw-top flasks are used, then part of the media can be set aside in a refrigerator for future use. In such cases, the flasks containing solidified media may be heated to dissolution in a water bath and poured directly into sterile petri dishes. Plastic petri dishes are commercially available. If media is sterilized in glass petri dishes the autoclave or pressure cooker should be allowed to cool down slowly, so that pressure will be lowered to the point that the material will not boil out. Warm sterile media may be poured directly into sterile petri dishes and allowed to cool until the media hardens. Lids should be tilted slightly so that steam will vent; this prevents condensation of excess water over the media.

- Brain heart infusion agar:
  dehydrated BHI agar ....26 gm
  distilled water ....500 ml
  Heat to boiling to dissolve the medium completely.
  Sterilize.
  Plates: Autoclave. Pour sterilized media into sterile petri dishes (1/3 full). Let stand for 10 minutes with lids off center so as to make an outlet for steam.
  Cover and let stand until media hardens.
  Invert and identify the media; and refrigerate.
- Brain heart infusion agar with salt:
  Add NaCl to the above medium to make 3 percent salt medium. Perpare as indicated for BHI.
- Tryptic soy agar:
  dehydrated T S agar ....20 gm
  distilled water ....500 ml
  Heat to boiling to dissolve the medium completely.
  Sterilize. Prepare as for BHI. For redfish work make up 35 salt medium.
- Azied dextrose broth and dextrose agar with sodium azide (for gram positive bacteria). Azide dextrose broth:
  beef extract..4.5 gm
  tryptone ....15.0 gm
  dextrose ....7.5 gm
  sodium chloride ....7.5 gm
  sodium azide ....0.2 gm
  distilled water .... 1 liter
  Azide dextrose broth is available from Difco Laboratories. *Streptococcus* can be separated from *Staphylococcus* and *Micrococcus* with the catalase test. For dextrose agar add 0.2 gm sodium azide/liter medium.
- Cytophaga agar (Anacker and Ordal, 1958):
  Tryptone ....0.25 gm
  Yeast extract ....0.25 gm
  Sodium acetate 0.1 gm
  Agar ....4.5 gm
  Distilled water ....500 ml
  Adjust pH to 7.2 – 7.4
- Bacteriological media are available from:
  BBL Division of Bioquest, Box 243 Cockeysville, Md. 21030 and Difco Laboratories, Detroit, Mich.

Isolation

It is essential that the aseptic technique be used. Bacteria of all sorts are present on the surface of fish and dissecting equipment. The kidney is a good location for bacterial isolation from blood. A red drum's kidney is located within the body cavity along and beneath the backbone. It can be accessed by cutting directly into the body cavity slightly below the lateral line. Once the body cavity is opened, a loop can be inserted into the kidney and a sample removed for spreading on agar medium. Tools for the procedure may be flamed prior to use and the skin surface around the point of incision disinfected by alcohol swabbing. Surface lesions may also be swabbed and dried and a sample obtained with a loop thrust into the lesion. The intent is to avoid contamination by extraneous bacteria.

Culture

Bacteria should be incubated at water temperatures comparable to those at which fish were cultured or at room temperature. It is important to enclose the plates to prevent contamination by insects in laboratories not fully equipped for microbiology. Bacterial colonies of infected fish will be numerous and appear similar in color and shape. A typical colony is selected for staining or streaking on another plate for susceptibility testing.

Staining procedure: Gram stain.

A. Preparation of solutions:

1. Modified Hucker's Crystal Violet

Solution A
ethyl alcohol 95 percent ....20 ml
crystal violet (certified) ....2 g

Solution B
ammonium oxalate ....0.8 g
distilled water ....80.0 ml

Mix solutions A and B

2. Iodine
iodine ....1 g
potassium iodine ....2 g
distilled water ....300 ml

3. Decolorizer
ethyl alcohol ....95 ml
acetone ....5 ml

4. Counterstain
safraine 0.85 percent ....6 g
ethyl alcohol ....20 ml
distilled water ....200 ml

B. Procedure:

1. Touch loop to colony and mix into a drop of water on slide.
2. Smear suspension over slide surface.
3. Air dry smear about 15 minutes.
4. Flood with methyl alcohol for 2 minutes.
5. Rinse with distilled water.
6. Flood smear with crystal violet solution and let stand 1 minute.
7. Wash smear briefly with tap water and drain off excess water.
8. Flood smear with iodine solution
9. Wash with tap water and decolorize until solvent flows colorless from the slide.
10. Wash briefly with tap water.
11. Counterstain with safranine for 20 seconds.
12. Wash briefly with tap water, blot dry and examine.

Result: Gram positive organisms are blue; gram negative are red.

Comment: A modified version of this staining procedure, using paper strips, is available from the Gugol Stain Co. 43-50 11 St., Long Island City, N.Y. 11101.

Staining procedure: Acid fast bacteria.

Gugol Stain Co. (see above) has a simple stain kit for acid fast bacteria that uses color impregnated paper strips. It can be used with smears prepared in the manner described above. Result: Acid fast bacteria stain red, others green or blue.

Susceptability testing procedure.

1. From an isolated bacteria colony, remove a loop of bacteria and place on BHI plate.
2. Using a sterile swab (cotton on wooden stick) or the loop, distribute the bacteriato all parts of the media surface.
3. If media surface is not particularly moist then addition of several ml of sterile water will facilitate distribution of bacteria.
4. Add sensivity disks to the plate, spacing so that overlaps of diffusion will not cause confusion when reading.
5. Bacteria will not grow on the area of an antibacterial disk that affects them. Areas of no growth or limited growth determine the susceptibility of the bacteria.
6. Determine whether the microorganism is very sensitive, moderately sensitive, slightly sensitive or resistant.
7. If the sensitivity disks are not marked it will be necessary to mark their names on the plate surface just under the disks.
8. Inhibition by vibriostat. Make a 5 percent solution of 2, 4-D amino 6-, 7-disopropyl pterdine phospate (known as 0/129) in distilled water or chloroform. Dip blank antibiotic sensitivity disks in solution and allow preparation to dry. Use as for prepared antibiotic disks by the method described above under susceptibility testing. Inhibition of growth by this compound is characteristic of most vibrios. O/129 is available from: Gallad Schlesinger Chemical Mfg. Corp. 854 Mineola Ave., Carle Place, Long Island, NY 11514, and BDR Chemicals Ltd., Poole, England.

Table 3. **Amount of antibiotic in grams added to 1 pound of feed.**

Percent of body weight fed per day	Dosage in grams of antibiotic per 100 pounds of fish per day 1.0	2.0	2.5	3.0	3.5	4.0	4.5	5.0	6.0	8.0	10.0
1%	1.00	2.00	2.50	3.00	3.50	4.00	4.50	5.00	6.00	8.00	10.00
2%	0.50	1.00	1.25	1.50	1.75	2.00	2.25	2.50	3.00	4.00	5.00
3%	0.33	0.67	0.83	1.00	1.14	1.33	1.50	1.66	2.00	2.66	3.33

Control

The method of grow-out affects the potential for occurrence of a particular disease, and, to some extent, the management options. For example, the application of selective toxins can be a difficult matter in cage culture. The rearing of red drum in water of very low salinity hampers development of marine disease agents but opens possibilities for infection/infestation by more freshwater forms

Chemical use has its limitations. Oral administration of antibacterial agents can be difficult with infected fish when infirmity suppresses appetite and lowers intake of the medicine. It would be beneficial to establish a safe procedure for bacterial control in the water column of grow-out ponds. The use of potassium permanganate for this purpose has not been established in mariculture because seawater enhances the danger of manganese dioxide precipitation on gills. Frequent use of copper compounds may increase the environmental load of copper and thereby adversely affect the pond system's productive base. **The legality and public health aspects of using certain chemicals also must be considered (Schnick, 1986). At present no antibiotics are cleared in the U.S. for use in red drum aquaculture.** Special permission could probably be obtained for use of antibiotics used in aquaculture of other fish species.

Control Methods For Pond Grow-Out

Oral Administration of Antibiotics

Treating fishes orally with antibiotics has become a common practice in the fish culture industry. Oral administration of antibiotics has been successful in correcting light kills caused by bacterial invasion.

In certain areas, commercially prepared fish feeds with antibiotic additives are available. This type of product is convenient and in many cases more economical than feeds with self-applied antibiotics. A farmer may decide, however, that the "do it yourself" approach would be the better choice for his particular needs.

Unrestrained use of antibiotics as a precautionary measure may greatly reduce the expected margin of profit. There is no evidence that shows that feeding antibiotics will enhance the growth of fishes as it does for other cultured animals.

Many antibiotics are available in technical grades such as poultry or veterinary-type formulations. The drugs should be purchased in this form, since they are much less expensive than the highly-purified products made for human use. Several antibiotics are marketed as "water soluable concentrates." The products consist of more than 20 percent active ingredient. These are preferred to those of weaker strength because there is less bulk of inert ingredients to hamper mixing. Weights can be determined with reasonable accuracy by simple proportioning of package contents.

Equipment and materials

Pelleted feed

Antibacterial (water soluble powder)

Vegetable oil

Container for mixing

Method

• **Preparing coated feeds.** Thoroughly mix the correct amount of powdered antibiotic in oil. Fold the oil-antibiotic mixture into the daily pellet allotment. Stir gently until the pellets are uniformly coated. Cod liver oil may be more readily accepted by red drum than vegetable oil. Enough oil should be used to provide a thin film of coating around the pelleted fish food. A cement mixer can be used to mix large feed volumes.

• **Dosage.** Recommended dosages of antibiotics usually are expressed as weight of drug per weight of fish per day. For example, they are expressed as grams per 100 pounds of fish per day or milligrams per kilogram of fish per day. When figuring the dosage of antibiotic, only the weight of the active ingredient (the antibiotic) of the purchased formulation should be considered. The weight of fishes can be estimated by sampling fishes to get an estimated average length and then comparing the length to a length-weight table.

Cultivated fish might be expected to run slightly heavier in weight. Table 3 gives the amount of antibiotic in grams that is to be added to 1 pound of feed at various dosage rates. From Table 3 and a length/weight determination, one may establish the amount of antibiotic that will be used per day during treatment.

• **Example.** A pond contains an estimated 10,000

Table 4. Cost of drugs required to treat 1,000 pounds of fish for one day*

Rate of dosage (grams per 100 pounds of fish per day)	Cost per gram of drug used: 0.5 cents	1 cents	2 cents	3 cents	4 cents	5 cents	10 cents	20 cents
0.5	$0.025	$0.05	$0.10	$0.15	$0.20	$0.25	$0.50	$1.00
1.0	.05	.10	.20	.30	.40	.50	1.00	2.00
2.0	.10	.20	.40	.60	.80	1.00	2.00	4.00
3.0	.15	.30	.60	.90	1.20	1.50	3.00	6.00
4.0	.20	.40	.80	1.20	1.60	2.00	4.00	8.00
5.0	.25	.50	1.00	1.50	2.00	2.50	5.00	10.00
6.0	.30	.60	1.20	1.80	2.40	3.00	6.00	12.00
7.0	.35	.70	1.40	2.10	2.80	3.50	7.00	14.00
8.0	.40	.80	1.60	2.40	3.20	4.00	8.00	16.00
10.0	.50	1.00	2.00	3.00	4.00	5.00	10.00	20.00
		30 cents	40 cents	50 cents	60 cents	70 cents	80 cents	90 cents
0.5		1.50	2.00	2.50	3.00	3.50	4.00	4.50
1.0		3.00	4.00	5.00	6.00	7.00	8.00	9.00
2.0		6.00	8.00	10.00	12.00	14.00	16.00	18.00
3.0		9.00	12.00	15.00	18.00	21.00	24.00	27.00
4.0		1.200	16.00	20.00	24.00	28.00	32.00	36.00
5.0		15.00	20.00	25.00	30.00	35.00	40.00	45.00
6.0		18.00	24.00	30.00	36.00	42.00	48.00	54.00
7.0		21.00	28.00	35.00	42.00	49.00	56.00	63.00
8.0		24.00	32.00	40.00	48.00	56.00	64.00	72.00
10.0		30.00	40.00	50.00	60.00	70.00	80.00	90.00

* Modified from Piper and Wolf (1959) **Prog. Fish Cult.** 21 (3):135-137.

red drum fingerlings that average 5 inches in length. The feeding rate of these fishes is 3 percent of body weight per day. The dosage of antibiotic has been suggested to be 4.5 grams of antibiotic per 100 pounds of fish per day. How much antibiotic should be used?

First, determine how many pounds of fish are present: Because there is limited knowledge base for red drum length-weight relationships, a reliable tabular summation cannot be provided at this time. Therefore, the producer must determine poundage by obtaining a population sample from the pond, weighing and computing an average weight, and multiplying that average weight by the number of fish he believes to be in the production pond.

For the purposes of this example, let's assume that a group of 5-inch finglerlings average 16.1 grams in weight, so 10,000 would represent 16.1 x 10,000 = 161,000 grams. In pounds this would be 161,000 grams divided by 454 grams/pound = 355 pounds of fish. Fish fed at 3 percent of the body weight would be fed 355 x .03 = 10.65 pounds. From Table 3 we find that at a 3 percent feeding rate and a dosage rate of 4.5 grams per 100 pounds of fish per day the number of grams of antibiotic should be added to 1 pound of feed is 1.50. Now to answer the question of how much antibiotic should be added to the feed that is fed each day: 10.65 pounds of feed x 1.5 grams/pound of feed = 16 grams. The 16 grams is the weight of the active ingredient that is required, but the weight of the total formulation that is to be added must be derived. A 20 percent active ingredient preparation would be added to the feed in an amount of 80 grams.

Cost

The producer should determine whether or not the cost of antibiotic treatment is economical. Table 4 provides information based on a fish crop of 1,000 pounds and will aid in the derivation of drug cost.

- **Example.** How much will it cost to treat 4,000 pounds of fish for 10 days when the cost of the drug is 2 cents per gram and the suggested dosage is 4.0 grams per 100 pounds of fish per day?

Consulting Table 4, we find that at a dosage rate of 4.0 grams per 100 pounds of fish per day and at a cost of 2 cents per gram, it will cost 80 cents to treat 1,000 pounds of fish for one day. But there are 4,000 pounds of fish to be treated for a period of 10 days.

Calculating: 80 cents x 10 x 4 = $32.00 (cost of treatment).

Conversions:

454 grams = 1 pound = 0.454 kilogram

28.35 grams = 1 ounce = 0.0625 pound

1 gram = 1000 milligrams = 0.001 kilogram.

Treatment Of Water In Pond Grow-Out

Several types of fish parasites can be successfully controlled by introducing a selective toxin into the pond water. The parasite will succumb to the chemical sooner or at a lower level than will the fish. Undesirable algae and water-borne bacteria also can be controlled by this method. The goal of such treatment is to obtain an even distribution of the chemical throughout the pond. Areas that are too weak in concentration may allow parasites to survive the treatment, while areas with excessive concentration (hot spots) may be lethal to the fishes.

Determining pond volume

Check the engineering design papers for your pond. If pond volume is not stated, estimate the average depth using soundings obtained with a marked pole or line. A surface acre is 43,560 square feet which is about 209 x 209 feet. Everyone should know the length of their normal stride. Mine is 2.5 feet. Once the dimensions of the pond are determined, volume may be determined in cubic feet or acre feet. An acre foot is 1 acre of 1 foot depth. A mg/liter or part per million in a cubic foot is 0.028 grams and in an acre foot is 2.7 pounds.

Metric measure is somewhat simpler. A hectare is 10,000 square meters or 100 x 100 meters. A cubic meter equals 1,000 liters and for practical purposes a liter of water weighs 1 kilogram (=1000 grams = 1000 cc = 1,000,000 mg). One gram (gm) in a cubic meter equals one part per million or 1 mg/liter.

Application

• **Small Ponds.** Shallow ponds that are approximately 1/10 acre in size, or larger ponds that are long with width of 50 to 80 feet, can be treated from the bank. An effective method is to dissolve or dilute the chemical in a quantity of liquid sufficient for broadcasting over the surface of the whole pond. This liquid may then be carried around the bank and broadcast with a long handled water dipper or similar device that allows the liquid to be cast a distance of 30 feet. The chemical should be spread evenly over the entire surface. Portable chemical sprayers may also be used to apply the chemical. Volatile chemicals such as formalin are best applied by pouring the fluid slowly into the water while wading. If the pond is deep, a boat may be used to accomplish the task. Small ponds more than six feet deep should be stirred with a pump or outboard motor to insure an even distribution of chemical. Shallow ponds normally have enough circulation to disperse the chemical to the bottom. In ponds with deep and shallow areas, surface application should be appropriately greater over the deeper water.

• **Large Ponds.** Chemical application from a boat equipped with an outboard motor is a convenient means for treating larger ponds. The chemical is poured slowly into the propeller wake at a rate that will assure complete coverage of the pond. A heavier application should be made in deeper areas than in shallow ones to allow equilibrium to be reached as quickly as possible. The chemical should be applied as a liquid diluted with water because this method allows more even distribution than powders. It is a simple matter to prepare the amount of solution needed for a given application. A 35-gallon drum outfitted with a faucet and hose is an inexpensive and efficient apparatus that will do a good job. The faucet allows regulation of the liquid to a constant flow. With the correct flow set, the boat follows a zig-zag course across the pond that provides complete coverage.

Disinfection Of Ponds

An often overlooked method of disease control (one that is not harmful to the consumer) is early harvest. It can be a good option if fish are of harvestable size. When a harvest or cleanout is undertaken, a disinfection often follows. Disinfection methods can eliminate or reduce harmful pathogens and parasites or break cycles of parasites by removing intermediate hosts. Pond disinfection is difficult because some microbes can remain protected in the deeper layers of bottom muds. Dehydration is effective if water entry can be curtailed and there is a good chance of prolonged dry weather. Chemicals involve more cost. Chemicals applied to shallow water or wet mud surfaces are most economical. Quick lime (calcium oxide) at 380 gm/m2 (Hoffman, 1976) is sometimes added to wet mud by broadcasting (Finlay, 1978). Or, 1 percent sodium hydroxide plus 0.1 percent teepol (a detergent) can be applied to bottoms at a rate of 1/2 gallon/m2. Chlorine can be maintained in the water column at suitable concentrations (10mg/l) for several days to disinfect the water.

Control Methods For Cage Culture

Culture of red drum in cages contained in a closed system or pond can be approached by the methods described above for pond grow out. Cages suspended in open waters present more of a challenge. Oral administration of antibacterials is possible, but use of selective toxins becomes impractical where treatment must be of long duration. Short term application of selective toxins involves enveloping a cage with a polyethylene bag and providing aeration to prevent asphyxiation. Parasitic isopods can be eliminated by introducing 500 mg/liter formalin for several minutes (E. Williams, Univ. Puerto Rico, Mayaguez, personal communication), but they will return once the chemical is no longer present.

Disease Control In Raceway Grow-Out

Most of the disease organisms mentioned for pond culture occur also in raceway grow-out. Similar too are the methods used for their control. Disinfection can be more beneficial in raceways because man-made materials such as walls and bottoms allow better exposure of pathogens to disinfectants. Smaller volumes of water in raceway systems make possible the use of more costly chemicals.

Raceway Disinfection

Chlorination is often used for static treatments of systems and as a wall wash. Exposure times and concentrations are as follows: 200 mg/l for 30 minutes; 100 mg/l for 1 hour; and 10 mg/l for 24 hours. Wall washes with chlorine compounds can be very noxious. Such efforts, therefore, should be limited to small jobs with adequate ventilation. Chlorine is available in liquid form as household bleach (5.25 percent sodium hypochlorite) or in powdered form for swimming pools (calcium hypochlorite, 50 to 70 percent available chlorine). Other chemical disinfectants include an 80 percent active product of benzalkonium chloride which is sometimes used for a 1-day exposure at a concentration of 1200 mg/l. Various iodophores also can be helpful. Solutions of 50 mg/l active ingredient are used for disinfecting of water and 250 mg/l for disinfecting of equipment. Isopropyl and ethyl alcohol have been used to disinfect standing water in tanks, as has hydrochloric acid. Formalin has been used for disinfection as a 1 percent solution. The benefits of cleaning and dehydration should always be considered in raceway disinfection.

Treating Raceways Containing Fishes

Chemicals can be added to the water of recirculating systems or to raceways with flow shut off. Two approaches are usually utilized. One is the exposure of fishes and pathogens to a relatively strong dosage of chemical for a few minutes to a few hours. The other is a longer exposure to chemicals (especially antibacterials) that show low toxicity to the fishes. Continous addition of chemicals has not gained widespread adoption but has been described by Post (1983).

Examples Of Calculating Concentrations For Fish Vats (Tanks)

Example 1. A fish vat is 10-feet long and 4-feet wide with a water depth of 3 feet. How much chemical should be added to attain a concentration of 1 part per million (= 1 mg/l)? First calculate the weight of the water in the tank:

Water weight = L x W x D x 62.4
10 x 4 x 3 x 62.4 = 7,488 pounds
L = length, W = width, D = depth, 62.4 lbs. per cubic foot

Next calculate the weight of chemical needed by comparing the known ratio and the unknown ratio:

1 mg/l = 1/1,000,000
1/1,000,000 = (N) pounds/7,488
By cross multiplication the following is obtained:
1,000,000 x (N) = 1 x 7,488
(N) = 1 x 7,488/1,000,000 = 0.007 pounds
one pound = 454 grams or 16 ounces
0.007 pound in grams is 0.007 x 454 = 3.2 grams
in ounces 0.007 x 16 = 0.112 ounce

Volume may also be determined in cubic meters. A cubic meter consists of 1,000 liters and weighs 1,000 kilograms. One gram in a cubic meter is thus equal to one part per million or 1 mg/l.

Example 2. (Where chemical is already in mixed form) Water soluble antibiotic is 10 grams active material in a 6.4 ounce (181 grams) package. In the problem above, 3.2 grams were needed for a 1 ppm concentration in the vat. The amount needed may be calculated by comparing the known ratio the unknown ratio:

3.2 grams/(N) oz. = 10 grams/6.4 oz.
(N) = 3.2 x 6.4/10 = 2.05 ounces = amount needed

Dosage concentrations also can be determined using conversion factors. In this case the volume or weight of the vat water is determined and then compared to an appropriate conversion factor.

Some useful conversions are:

1 part per million (1 mg/l) =	0.0038 grams per gallon
	0.028 grams per cubic foot
	0.13 ounce per 1,000 gal.
	1 ounce per 1,000 cubic ft
	1 gram per cubic meter

Application

The calculated dosage is first weighed. The amount determined in the first example above (3.2 grams or 0.11 ounce) is very small and a weight balance or scale for such amounts usually is not available in the field. It is possible in such cases to use a less sensitive weighing device to weigh an amount 10 times that required. The weighed sample is then added to a volume measuring device such as a measuring pitcher. In the case of the small amount mentioned above, 1.1 ounces could be added to eight ounces of water in a pitcher. Water is then added to the 10 ounce mark. A 1-ounce dosage of this solution will result in the desired 0.11 ounce concentration in the fish vat. One type of balance that is both readily available and inexpensive is the type used in the loading of hunting ammunition.

Chemical should be evenly distributed in the water. Mixing the chemical in water prior to dosing is usually helpful, and extra amounts may be prepared in advance. If water flow is to be reduced during treatment, then precautions should be taken to assure adequate aeration to support oxygen levels.

Use of Diagnostic Laboratory

The Texas Agricultural Extension Service operates a fish disease diagnostic laboratory on the Texas A&M University campus. The laboratory is in its 16th year of service to Texas fish farmers.

References

Anacker, R.L. and E.J. Ordal. 1959. Studies on the myxobacterium *Chondrococcus columnaris*. I. Serological typing. **J. Bacteriol.** 78:25-32.

Becker, C.D. 1977. Flagellate parasites of fish. pp. 357-416, *In:* **Parasitic Protozoa, Vol. I, Taxonomy, Kinetoplastids and Flagellates of Fish,** J.P. Kreier,(ed.) Academic Press, NY.

Bere, R. 1936. Parasitic copepods from Gulf of Mexico fish. **Amer. Mid. Nat.** 17:577-625.

Blaisiola, G.C. 1976. A review of whitespot, *Cryptocaryon irritans*. **The Marine Aquarist** 7:5-13.

Bower, C.E., D.T. Turner and R.C. Biever. 1987. A standardized method of propagating the marine fish parasite, *Amyloodinium ocellatum*. **J. Parasitology,** 73:85-88.

Brown, E.M. 1951. A new parasite protozoan the causal organism of a white spot disease in marine fish-*Cryptocaryon irritans* gen. and sp. n. **Agenda and Abstracts of the Scientific Meetings of the Zoological Society of London,** 1950, No. 11:1-2.

Buchanan, R.E. and N.E. Gibbons (eds.). 1974. **Bergey's manual of determinative bacteriology, 8th edition.** Williams & Wilkins Company, Baltimore, MD. 1268 pp.

Chandler, A.C. 1935. Parasites of fishes of Galveston Bay. **Proc. U.S. Nat. Museum** 83:123-157, plates 6-12.

Cheung, P.J., R.F. Nigrelli and G.D. Ruggieri. 1979. Studies on cryptocaryoniasis in marine fish: effect of temperature and salinity on the reproductive cycle of *Cryptocaryon irritans* Brown 1951. **J. Fish Dis.** 2:93-97.

Colorni, A. 1985. Aspects of the biology of *Cryptocaryon irritans,* and hyposalinity as a control measure in cultured gilthead sea bream *Sparus aurata*. **Dis. Aquat. Org.,** 1:19-22.

Colorni, A. 1987. Biology of *Cryptocaryon irritans* and strategies for its control. **Aquaculture** 67:236-237.

Conroy, D.A. 1984. Diseases caused by microorganisms: agents, bacteria. pp. 48-88 *In:* O. Kinne,(ed.) **Disease of Marine Animals Vol. IV, Part 1,** Introduction Pisces. Biologische Anstalt Helgoland, Hamburg. 541pp.

Corliss, J.O. 1977. Annotated assignment of families and genera to the orders and classes currently comprising the Corlissian scheme of higher classification for the Phylum Ciliophora. **Trans. Amer. Micros. Soc.** 96:104-140.

Deardorff, T.L. and R.M. Overstreet. 1980. Taxonomy and biology of North American species of *Goezia* (Nematoda: Anasakidae) from fishes, including three new species. **Proc. Helminthol. Soc. Wash.** 47:192- 217.

Deardorff, T.L. and R.M. Overstreet. 1981. Review of *Hysterothylacium* and *Iheringascaris* (both previously = *Thynnascaris* (Nematoda: Anasakidae) from the Northern Gulf of Mexico. **Proc. Biol. Soc. Wash.** 93:1035-1079.

Defigueiredo J. and J.A. Plumb. 1977. Virulence of different isolates of *Aeromonas hydrophilia* in channel catfish. **Aquaculture** 11:349-354.

Dempster, R.P. 1955. The use of copper sulfate as a cure for fish diseases caused by parasitic dinoflagellates of the genus Oodinium. **Zoologica** (NY), 40:133-138.

Dempster, R.P and W.H. Shipman. 1969. The use of copper sulfate as a medicament for aquarium fishes and as an algacide in marine mammal water systems. **Occasional Papers Calif. Acad. Sci.,** No. 71, 6 p.

Dempster, R.P. 1972. A description of the use of copper sulfate as a cure of gill disease in marine tropical fish tanks. **Anchor** 6:450-452.

Finlay J. 1978. Disinfectants in fish farming. **G.B. Ministry Agric. Fish. Food, Fishery Notes,** No. 59., Lowestoft, England, 7 p.

Fusco, A.C. and R.M. Overstreet. 1978. *Spirocamallanus cricotus* sp. n. and *S. halitrophus* sp. n. (Nematoda: Camallanidae) from fishes in the Northern Gulf of Mexico. **J. Parasitol.** 64:239-244.

Henley, M.W. and D.L. Lewis. 1976. Anaerobic bacteria associated with epizootic in grey mullet (*Mugil cephalus*) and redfish (*Sciaenops ocellata*) along the Texas Gulf coast. **J. Wildl. Dis.** 12:448-453.

Hoffman. 1976. Whirling disease of trout. **U.S. Fish Wildl. Serv. Fish Dis. Leafl.** 47 Washington D.C. 10 p.

Hoffman, G.L. and F.P. Meyer. 1974. **Parasites of freshwater fishes. A review of their control and treatment.** TFH Publications, Inc. Neptune City, NJ.

Herwig N. 1978. Notes on the treatment of *Cryptocaryon*. **Drum and Croaker** 18:6-12.

Huff, J.A. and C.D. Burns. 1981. Hypersaline and chemical control of Cryptocaryon irritans in red snapper, *Lutjanus campechanus,* monoculture. **Aquaculture** 22:181-184.

Jee, L.K. and J. Plumb. 1981. Effects of organic load on potassium permanganate as a treatment for *Flexibacter columnaris*. **Trans. Am. Fish. Soc.** 110:86-89.

Johnson, S.K. 1984. Evaluation of several chemicals for control of *Amyloodinium ocellatum,* a parasite of marine fishes. **Texas A&M University Fish Disease Diagnostic Laboratory Publication,** No. FDDL-M5. 4 p..

Joy, J.E. 1972. A new species of Henneguya (Myxosporidae: Myxobolidae) from the black drum, *Pogonais cromis,* in Clear Lake, Texas. **J. Protozool.** 19:126-128.

Keith, R.E. 1980. Disease prevention and control. **Aquarium.** January 1980. p. 20.

Kingsford, E. 1975. **Treatment of exotic marine fish diseases.** Palmetto Publishing Co. St. Petersburg, Florida. 92 pp.

Kuo, S-C, H-Y Chung and G.H. Kou. 1976. *Vibrio anguillarum* isolated from a Vibrio disease of freshwater cultured ayu, *Plecoglossus altivelis*. **J. Fish Soc.** Taiwan 4:21-24.

Lawler, A.R. 1977a. The parasitic dinofagellate *Amyloodinium ocellatum* in marine aquaria. **Drum and Croaker** 17(2):17-20.

Lawler, A.R. 1977b. Dinoflagellate (*Amyloodinium*) infestation of pompano. pp. 257-264 *In:* C.J. Sinderman, (ed.) **Disease Diagnosis and Control in North American Marine Aquaculture.** Elsevier Scientific Publishing Co., New York.

Lawler, A.R. 1979. North American fishes reported as hosts of *Amyloodinium ocellatum* (Brown, 1931). **Drum and Croaker** 19(1):8-14.

Lawler, A.R. 1980. Studies on *Amyloodinium ocellatum* (Dinoflagellata) in Mississippi Sound: natural and experimental hosts. **Gulf Res. Repts.** 6:403-413.

Lom, J. and A.R. Lawler. 1973. An ultrastructural study of the mode of attachment in dinoflagellates invading gills of Cyprinodontidae. **Protistologica** 9:293-309.

Lom, J. 1981. Fish invading dinoflagellates: a synopsis of existing and newly proposed genera. **Folia Parasitologia** (Praha) 28:3-11.

Lom, J. 1984. Diseases caused by protistans. pp. 114-168 *In:* O. Kinne (ed.) **Diseases of Marine Animals, Vol. IV, Part 1, Introduction Pisces.** Biologische Anstalt Helgoland, Hamburg, 541pp.

Manter, H.W. 1947. The digenetic trematodes of marine fishes of Tortugas, Florida. **Amer. Mid. Nat.** 38:257-416.

Martin, N. 1987. Bitter Harvest, **Texas Shores** 19:21-23.

Morin, L.G. 1980. Disease preventative and control. **Aquarium**, December 1980. p. 20.

Nigrelli, R.F. 1936. The morphology cytology and life-history of *Oodinium ocellatum* Brown, a dinoflagellate parasitic on marine fishes. **Zoologica**, 21:129-164.

Nigrelli, R.F. and G.D. Ruggieri. 1966. Enzootics in the New York aquarium caused by *Cryptocaryon irritans*Brown, 1951 (=Ichthyophthirius marinus Sikama, 1961), a histophagous ciliate in the skin, eyes and gills of marine fishes. **Zoologica** 51:97-102.

Overstreet, R.M. 1983. Aspects of the biology of the red drum, *Sciaenops ocellatus* in Mississippi. Gulf Research Reports, Supplement 1, 45- 68.

Paperna, I. 1980. *Amyloodinium ocellatum* (Brown 1931) (Dinoflagellida) infestation in cultured marine fish at Eilat, Red Sea: epizootiology and pathology. **J. Fish Disease,** 3:363-372.

Paperna, I. 1983. Review of disease of cultured warm-water marine fish. **Rapp. P.-v. Reun. Cons. Int. Explor. Mer.** 182:44-48

Paperna, I. 1984a. Reproduction cycle and tolerance to temperature and salinity of *Amyloodinium ocellatum* (Brown 1931) Dinoflagellida. **Ann. Parasitol. Hum. Comp.** 59:7-30.

Paperna, I. 1984b. Chemical control of *Amyloodinium ocellatum* (Brown 1931) (Dinoflagellida) infections: In vitro tests and treatment trials with infected fishes. **Aquaculture** 38:1-18.

Phelps, R., J.A. Plumb and C.W. Harris. 1977. Control of external bacterial infection of bluegills with potassium permanganate. **The Progressive Fish Culturist** 39:142-143.

Rasheed, V. and J.A. Plumb. 1984. Pathogenicity of a non-haemolytic Group B *Streptococcus* sp. in Gulf killifish (*Fundulus grandis* Baird and Girard). **Aquaculture,** 37:97-105.

Riggin, G.T., Jr. and A.K. Sparks. 1962. A new Gastrostome, *Bucephaloides megacirrus*, from the redfish, (*Sciaenops ocellata*). **Proc. Helminthol. Soc. Wash.** 29:27-29.

Roberts, L.S. 1969. *Ergasilus arthrosis* n. sp. (Copepoda: Cyclopoida) and the taxonomic status of *Ergasilus versicolor* Wilson, 1911, *Ergasilus elegans*, Wilson, 1916, and *Ergasilus celestis* Mueller, 1936, from North American fishes. **J. Fish. Res. Brd. Can.** 26:997-1011.

Sawyer, R T. and A.R. Lawler. 1975. Marine leeches of the eastern United States and the Gulf of Mexico with a key to the species. **J. Nat. Hist.** 9:633-667.

Schell, S.C. 1985. **Handbook of trematodes of North America north of Mexico.** University Press of Idaho, Moscow, 263 pages.

Schnick, R.A., F.P. Meyer and D.F. Walsh .1986. Status of fishery chemicals in 1985. **The Progressive Fish-Culturist** 48:1-17.

Schultz, G.A. 1969. **How to know the marine isopod crustaceans.** W.C. Brown Co., Dubuque, Iowa. 359 pp.

Sievers, A.M. 1969. Comparative toxicity of *Gonyaulax monilata* and *Gymnodinium breve* to annelids, crustaceans, molluscs and a fish. **J. Protozool.** 16:401-404.

Sikama, Y. 1937. Preliminary report on the white spot disease in marine fish. **Suisan-Gakuka:** 7:149-160 (In Japanese).

Sikama, Y. 1961. On a new species of *Icthyophthirius* found in marine fishes. **Sciences Reports of the Yokosuka City, Museum** 6:66-70.

Sogandares-Bernal, F. and R.F. Hutton. 1959. Studies on helminth parasites from the coast of Florida. IV. Digenetic trematodes of marine fishes of Tampa, Boca Ciega Bays, and the Gulf of Mexico. **Quart. Journ. Fla. Acad. Sci.** 21:259-273.

Steidinger, K.A. and E.A. Joyce. 1973. Florida red tides. **Fla. Dept. Nat. Res. Marine Research Laboratory Educational Series No. 17,** 26 pp.

Thatcher, V.E. 1960. Studies on the Cestoda of elasmobranch fishes of the Gulf of Mexico. Part I. **Louisiana Acad. Sci.** 23:65-74.

Trimble, W.C. 1979. Yield trials for red drum in brackish-water ponds, 1976-1979. **Proc. annu. Conf. Southeast. Assoc. Fish & Wildl. Agencies,** 33:432-441.

Wellborn, T.L., Jr. and W.A. Rogers. 1966. A key to the common parasitic protozoans of North American Fishes. **Zoology-Entomology Department Series, Fisheries No. 4.,** Auburn University Agr. Experiment Station, 17 pages.

Yamaguti, S. 1963. Parasitic copepoda and Branchiura of fishes. **Interscience,** N.Y., 1104 pp.

Site Selection for Redfish Culture

Russell J. Miget
Marine Fisheries Specialist
Texas Marine Advisory Service
Rt. 2 Box 589
Corpus Christi, Tx 78410

Site selection for redfish aquaculture can be somewhat confusing as this particular fish is euryhaline; i.e., as an adult it lives in both fresh and salt water. Although this physiological attribute potentially extends the range of redfish grow out operations, to date redfish have not been commercially produced in freshwater impoundments. Adding to the confusion, culture systems include low-density stocking in large impoundments (extensive); semi-intensive production in managed ponds; and intensive culture in greenhouse-covered fiberglass raceways with recirculating water systems. Each of these systems has different land and water requirements.

As redfish regularly migrate between waters of greatly varying salinity (they grow up in brackish bays, then move offshore to true oceanic waters to spawn), much attention has been given to developing culture systems that mimic this aspect of their natural cycle.

A typical semi-intensive system includes a building to house spawning tanks and hatching cones, saltwater ponds to produce fingerlings, and grow out ponds (which are usually saltwater but which may utilize certain fresh waters with adequate chlorides and hardness).

Such a system would most probably be constructed in a coastal area (or over a saline aquifer), since it requires more than one million gallons of saltwater to fill each acre of fingerling ponds to a depth of four feet. Optimal conditions for fingerling production in fertilized ponds is 25 to 30 ppt (parts-per-thousand) salinity (although they have been reported to survive in 10 to 40 ppt), and 28° to 30°C (82° to 86°F). Larval redfish placed in waters cooler than 20°C (68°F) become inactive and may starve to death.

After 30 to 40 days, one-inch fingerlings can be trained to take prepared feeds for further growout. At this time, they can also be acclimated to fresh water, provided the water has at least 130 ppm chloride ions and 100 ppm calcium hardness. Waters deficient in these ions can be supplemented with agricultural-grade salt and limestone.

Although redfish can tolerate a wide range of salinity, they cannot tolerate cold weather. This limitation may well be the determining factor in selecting a site for redfish growout. Since commercial redfish ponds typically contain only 4 to 5 feet of water, strong winds and low temperatures that accompany cold fronts can cause pond temperatures to drop to critical levels in a matter of hours.

Experimental data indicate that young fingerlings are more susceptible to cold water than older fish; they begin to die when exposed to 5° to 8°C water for several days. Low temperatures are even more critical if the fingerlings have not been weaned off live food, because they may become too sluggish to catch their prey. Feeding, and therefore growth, of yearlings and adult fish will decline as wintertime temperatures drop below 20°C (68°F). Fish kills will occur when temperatures remain around 3° to 4°C (37° to 39°F) for several days. This is a critical concern as redfish must be overwintered to reach marketable size.

A solution to this potential problem may be found in the intensive grow out system which employs covered raceways. A variation of this approach is to grow 6- to 8-inch fingerlings in warm-water raceways during the winter, transfer them to outdoor ponds in early spring, then harvest market size (2 to 3 lb.) fish in the late fall.

Fingerling red drum have been stocked in recreational freshwater impoundments where they grow to trophey-sized fish in record time while feeding on abundant natural prey. However, very little work has been carried out to determine the commercial potential of raising red drum in fresh water. Limited research data confirm that the added stress of osmoregulation in freshwater makes the fish less tolerant of cold water, as well as less efficient in converting food to body weight. It would therefore be prudent to move cautiously in developing redfish farms in freshwater until more data are available.

Temperature

Coastal areas are more suitable for redfish growout than inland areas because of access to required salinities and the presence of a more moderate climate. Coastal areas are moderated by the "heat-sink" effect of the surrounding water, thereby reducing the risk of a winter kill. Figure 1 shows average daily

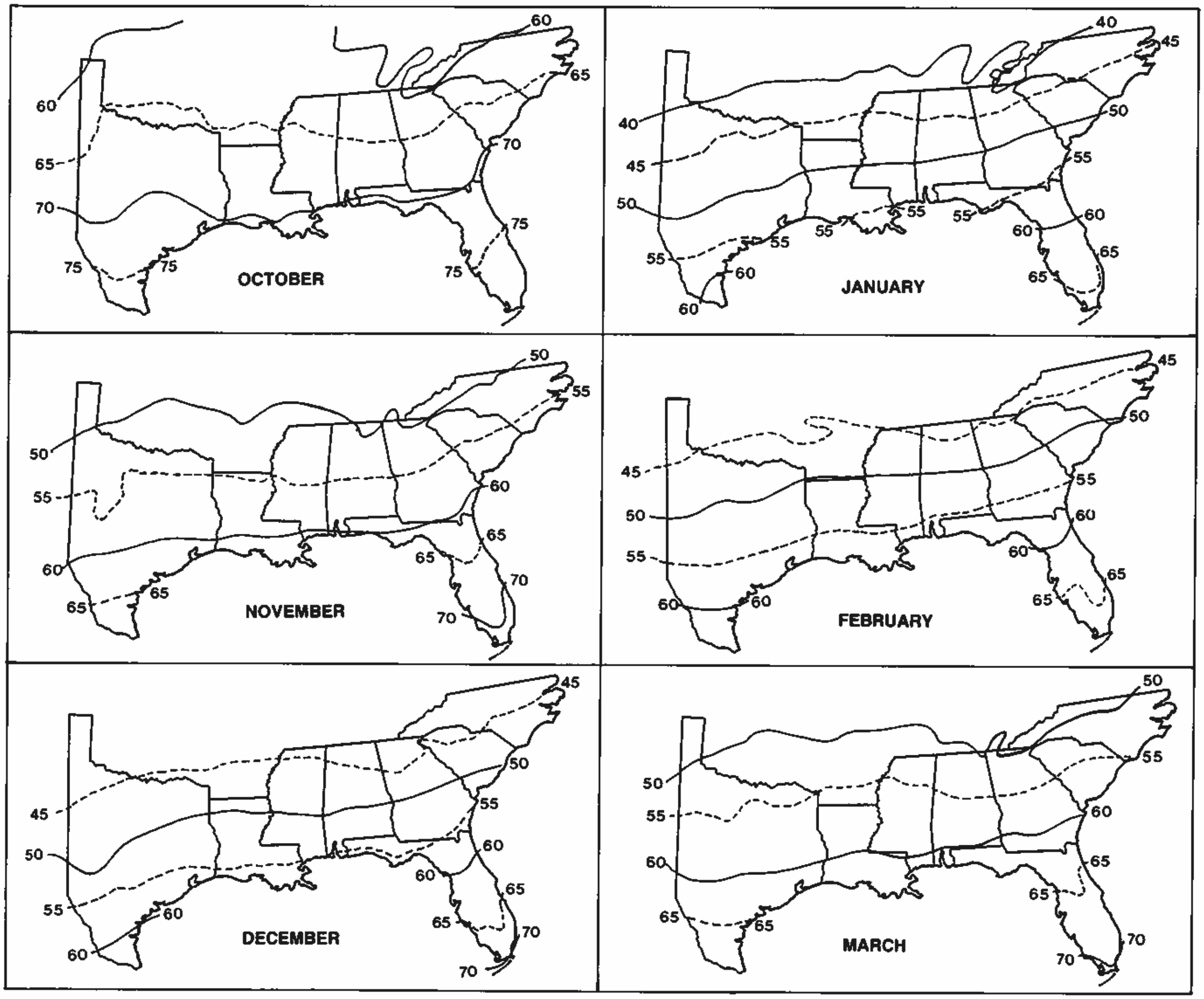


Figure 1. Normal daily average temperature (°F) 1931-1960.

temperature isotherms for the Gulf and South Atlantic states from October through March. Figure 2 shows 30-year normal winter daily maximum, average, and minimum temperature as well as recorded highs and lows. While this 30-year average is a good indicator of the climate, it should be re-emphasized that an unusually severe winter could kill an entire crop of fish in outdoor ponds. Figure 3, therefore, shows the frequency of freezing temperatures along the Gulf and South Atlantic coasts. Table 1 summarizes temperature data as well as freeze frequency at selected cities on the Gulf and South Atlantic coasts.

Isograms of the average number of days between the last spring frost and first fall frost (Fig. 4) more clearly reveal the moderating effect of oceans on coastal climates.

Assuming that a coastal site with access to saltwater (5 to 40 ppt) is most desirable for red drum growout at this time, additional site selection criteria must be evaluated. These are discussed below.

Water Quality

The subject of water quality encompasses a number of physical, chemical and biological variables. Of particular interest to the aquaculturist are: temperature, salinity, pH, alkalinity and dissolved oxygen, as well as potentially harmful pollutants such as heavy metals, domestic and industrial sewage, herbicides and pesticides.

Agencies in each state (listed in "Practical Permitting Suggestions") regularly monitor water quality data at designated sampling sites. They are thus in the position to provide historic data for a number of water-quality parameters which should be considered when choosing a grow out site.

Wetlands, Elevation and the Water Table

There is minimal topographic relief around most Gulf coast and South Atlantic bays and estuaries. Many areas below the 5-foot elevation line have been

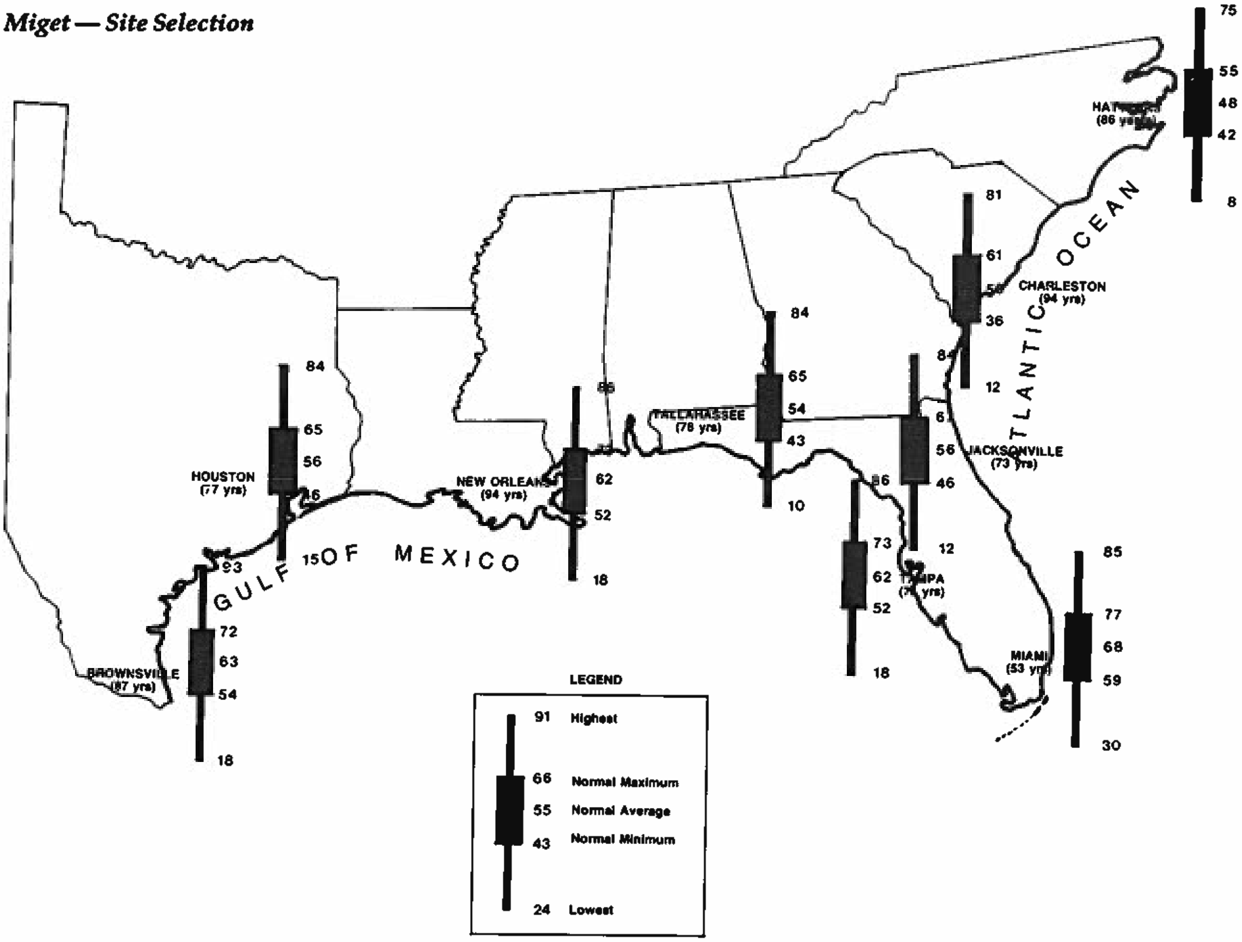


Figure 2. Normal daily maximum, average and extreme temperatures-December.

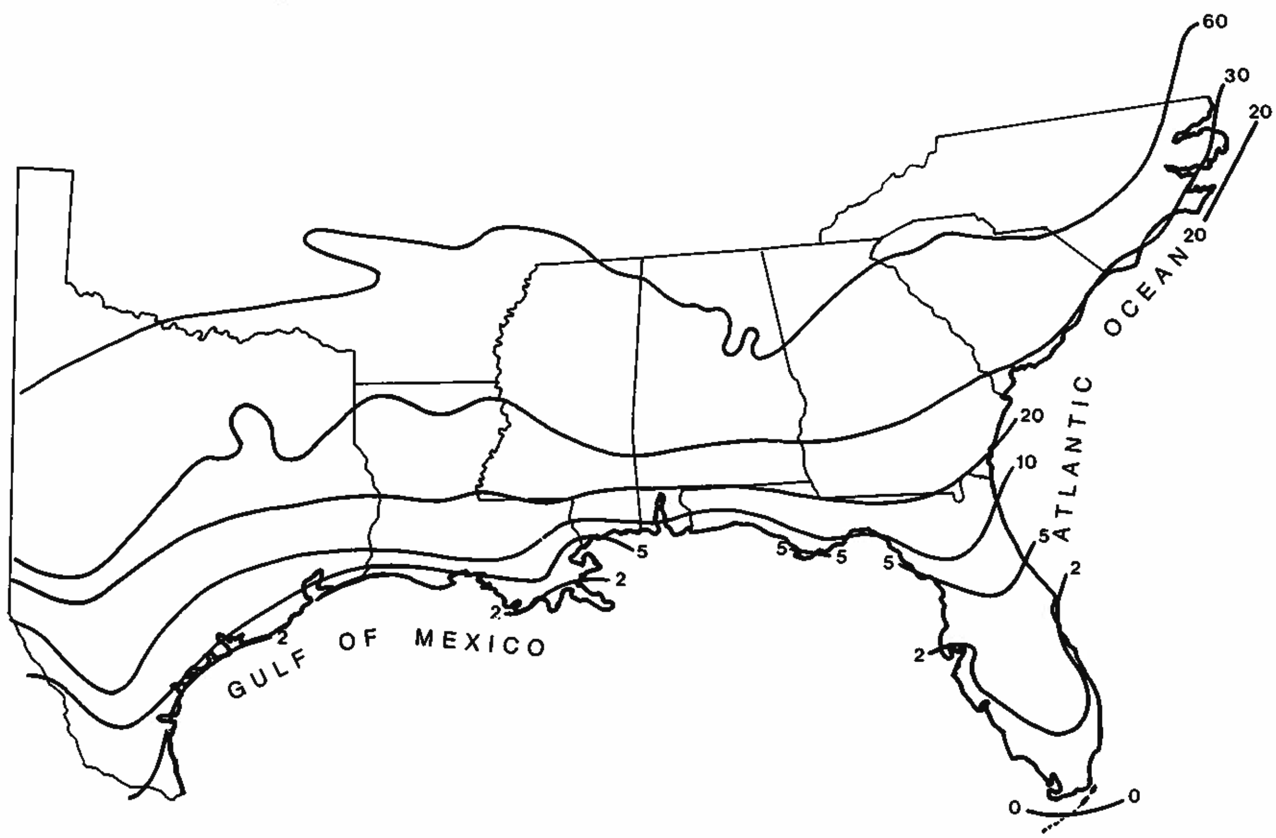


Figure 3. Mean annual number of days minimum temperature 32 °F and below.

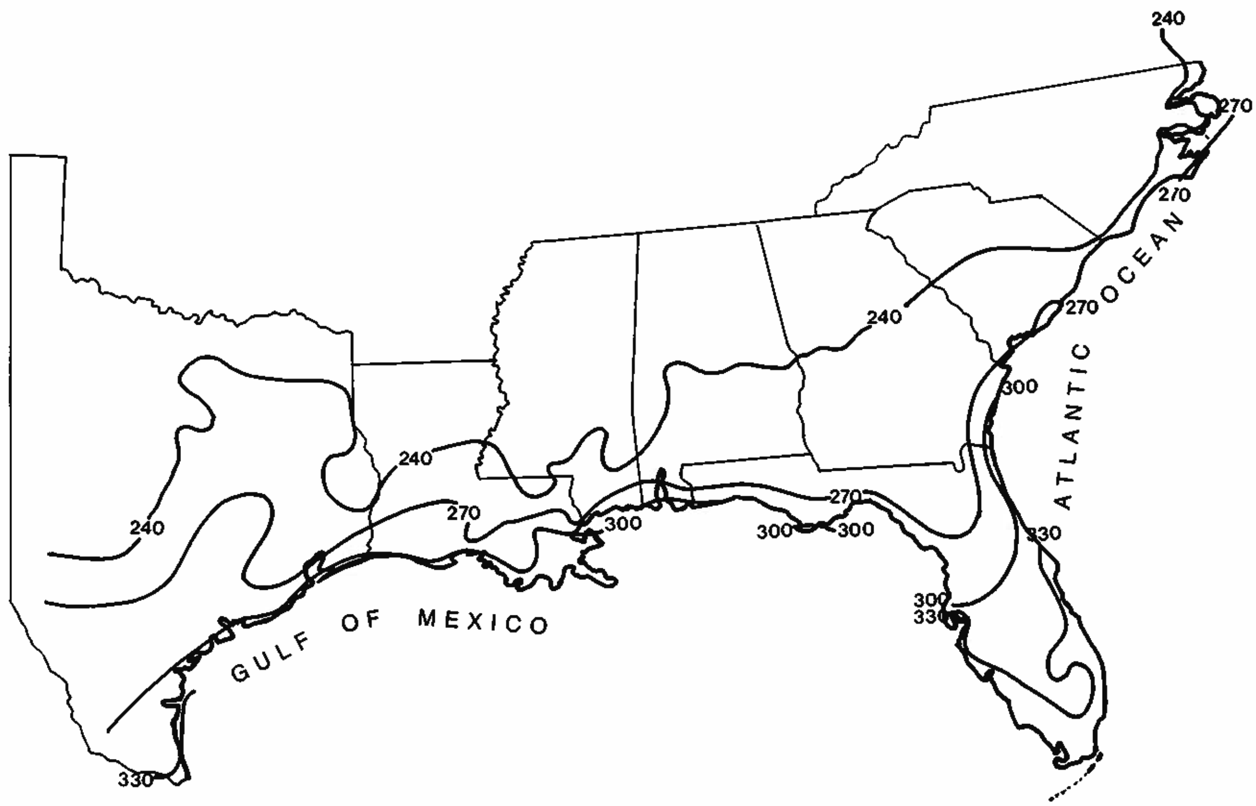


Figure 4. Mean length of freeze-free period (days).

designated as coastal wetlands. Many of these areas are suitable for aquaculture development, but are subject to rigid federal and state legislative restrictions.

Coastal wetlands represent the transition between aquatic and terrestrial ecosystems. A dominant feature of these areas is that soil is at least periodically saturated with, or covered by, water. As highly productive natural systems, they serve as a nursery for commercially important organisms such as shrimp, crabs and many species of finfish.

For reasons such as economic development, wetlands alteration in the United States is generally prohibited. An important exception of interest to the aquaculturist is the instance in which some wetland habitat is unavoidably altered as part of a larger project. (An example might be when an intake canal draws water directly from a bay.) In such cases, an agreement known as mitigation may be entered into with the appropriate permitting agencies. The basic idea is that the developer will agree to make up for the unavoidable loss of habitat caused by his operation by creating a similar one nearby.

As a general precaution, do not buy coastal property below the 5-foot Mean Sea Level (MSL) elevation prior to a wetlands survey conducted by a U.S. Army Corps of Engineer's biologist. The Corps is authorized by federal law to make a determination as to the presence, location and extent of wetlands based on elevation, soil saturation and vegetation. The results of such an on-site wetlands inspection are legally binding when presented in writing. However, it is important to note that as the coastal property changes due to erosion, subsidence or man-made activities, so changes the extent of the Corps jurisdiction over such areas.

Finally, it should also be emphasized that the Corps of Engineers permit only authorizes "an activity" and does not convey ownership or property rights necessary for the activity. Therefore, in the aforementioned example of an intake canal, the aquaculturist must seek appropriate approval (e.g. permit, easement or lease) from the state if the canal extends onto state-owned submerged lands.

Geological Survey Quadrangle maps are a good source of information about coastal property. Maps include detailed elevation contours (5-foot intervals), as well as road access to all coastal properties. Such maps will indicate bluffs (above 5-foot elevation) adjacent to the shoreline which would be logical sites to further evaluate.

Soil Characteristics

The next step in site selection involves contacting an agency that can save a project considerable time

Table 1. Temperature Normals (°F) 1950-1980.

Station		Jan	Feb	Mar	Apr	May	Jun	Jul	Aug	Sep	Oct	Nov	Dec	Ann
Texas														
Brownsville	Max	69.7	72.5	77.5	83.2	87.0	90.5	92.6	92.8	89.8	84.4	77.0	71.9	82.4
	Min	50.8	53.0	59.5	66.6	71.3	74.7	75.6	75.4	73.1	66.1	58.3	52.6	64.8
	Mean	60.3	62.8	68.6	74.9	79.2	82.6	84.1	84.1	81.4	75.3	67.7	62.3	73.6
	Freeze Freq	1	*	*	0	0	0	0	0	0	0	0	*	1
Corpus Christi	Max	66.5	69.9	76.1	82.1	86.7	91.2	94.2	94.1	90.1	83.9	75.1	69.3	81.6
	Min	46.1	48.7	55.7	63.9	69.5	74.1	75.6	75.8	72.8	64.1	54.9	48.8	62.5
	Mean	56.3	59.3	65.9	73.0	78.1	82.7	74.9	85.0	81.5	74.0	65.0	59.1	72.1
	Freeze Freq	3	1	*	0	0	0	0	0	0	0	*	1	5
Palacios	Max	62.5	65.2	71.5	77.7	83.3	88.4	90.6	90.8	87.9	81.5	71.9	65.6	78.1
	Min	43.9	46.5	53.1	61.9	68.7	74.8	77.2	76.1	71.2	61.4	52.1	46.2	61.1
	Mean	53.2	55.9	62.3	69.8	76.0	81.6	83.9	83.5	79.6	71.4	62.0	55.9	69.6
	Freeze Freq	2	1	*	0	0	0	0	0	0	0	*	1	4
Galveston	Max	59.2	60.9	66.4	73.3	79.8	85.1	87.3	87.5	84.6	77.6	68.3	62.3	74.4
	Min	47.9	50.2	56.5	64.9	71.6	77.2	79.1	78.8	75.4	67.7	57.6	51.2	64.8
	Freeze Freq	2	1	*	0	0	0	0	0	0	0	*	1	4
Louisiana														
Hackberry	Max	59.4	62.6	68.7	75.9	82.4	88.0	90.0	89.8	86.7	79.3	69.6	62.9	76.3
	Min	42.3	45.2	51.9	61.0	67.7	73.2	74.2	73.9	70.8	60.4	50.9	44.8	59.7
	Mean	50.8	53.9	60.3	68.5	75.1	80.6	82.1	81.8	78.8	69.9	60.3	53.9	68.0
	Freeze Freq	6	2	1	*	0	0	0	0	0	0	1	4	14
Houma	Max	64.4	66.7	72.7	79.5	85.1	89.9	90.8	90.6	87.6	80.9	72.4	66.8	79.0
	Min	44.0	45.7	52.4	59.6	65.4	70.4	72.6	72.2	69.3	58.0	50.3	45.4	58.8
	Freeze Freq	5	3	1	0	0	0	0	0	0	0	2	4	15
Mississippi														
Bay St. Louis	Max	58.7	61.8	67.8	75.7	82.6	88.5	90.0	89.8	86.3	78.7	68.2	61.5	75.8
	Min	41.2	43.5	50.2	59.8	66.3	72.0	73.9	73.4	69.8	58.1	49.2	42.8	58.4
	Mean	50.0	52.7	59.0	67.8	74.5	80.3	82.0	81.6	78.1	68.4	58.7	52.2	67.1
	Freeze Freq	6	4	1	*	0	0	0	0	0	*	1	5	17
Alabama														
Fairhope	Max	61.8	64.6	71.0	78.2	84.6	89.6	90.5	90.4	87.2	79.6	70.0	63.8	77.6
	Min	41.5	43.4	49.6	57.0	63.8	69.8	72.3	71.8	68.3	57.2	48.5	43.3	57.2
	Mean	51.6	54.1	60.3	67.6	74.2	79.7	81.4	81.1	77.8	68.4	59.3	53.5	67.4
	Freeze Freq	7	4	1	0	0	0	0	0	0	*	2	5	18
Florida														
Niceville	Max	60.8	63.5	69.6	77.3	84.3	89.5	90.8	90.7	87.7	80.2	70.6	63.8	77.4
	Min	37.7	39.5	46.1	53.7	61.2	67.6	70.7	70.3	66.7	54.1	44.7	39.1	54.3
	Mean	49.3	51.5	57.9	65.5	72.8	78.6	80.8	80.5	77.3	67.2	57.7	51.5	65.9
	Freeze Freq	6	4	1	0	0	0	0	0	0	0	1	3	15
St. Petersburg	Max	70.0	71.1	76.0	81.7	86.7	89.3	90.1	90.0	88.7	83.4	76.8	71.4	81.3
	Min	53.7	54.7	59.8	65.1	70.4	74.5	76.0	75.9	74.8	68.6	61.0	55.3	65.8
	Mean	61.9	62.9	67.9	73.4	78.5	81.9	83.1	83.0	81.7	76.0	68.9	63.4	73.6
	Freeze Freq	0	0	0	0	0	0	0	0	0	0	0	0	0
Homestead	Max	76.5	77.3	81.3	84.3	87.0	88.8	90.2	90.7	89.2	85.6	81.2	77.5	84.1
	Min	53.5	54.0	57.8	61.8	66.1	69.9	71.3	71.7	71.6	67.4	60.6	55.1	63.4
	Freeze Freq	1	0	0	0	0	0	0	0	0	0	0	0	1
Jacksonville	Max	64.6	66.8	73.3	79.7	85.2	88.9	90.7	90.2	86.9	79.7	72.4	66.3	78.7
	Min	41.7	43.3	49.3	55.7	63.0	69.1	71.8	71.8	69.4	59.2	49.2	43.2	57.2
	Mean	53.2	55.1	61.3	67.7	74.1	79.0	81.3	81.0	78.2	69.5	60.8	54.8	68.0
	Freeze Freq	4	3	1	0	0	0	0	0	0	0	1	4	13

Table 1. (continued)

Station		Jan	Feb	Mar	Apr	May	Jun	Jul	Aug	Sep	Oct	Nov	Dec	Ann
Georgia														
Brunswick	Max	64.1	66.3	72.5	79.1	85.2	89.1	91.6	91.0	87.3	79.9	72.4	65.6	78.7
	Min	42.2	43.3	50.0	57.5	64.3	70.2	72.6	72.7	70.1	60.0	50.6	44.3	58.2
	Mean	53.2	54.9	61.3	68.3	74.8	79.7	82.2	81.9	78.7	70.0	61.5	55.0	68.5
	Freeze Freq	7	4	2	0	0	0	0	0	0	0	2	6	21
Savannah	Max	60.3	63.1	69.9	77.8	84.2	88.6	90.8	90.1	85.6	77.8	69.5	62.5	76.7
	Min	37.9	40.0	46.8	54.1	62.3	68.5	71.5	71.4	67.6	55.9	45.5	39.4	55.1
	Mean	49.2	51.6	58.4	66.0	73.3	78.6	81.2	80.8	76.6	66.9	57.5	51.0	65.9
	Freeze Freq	11	8	3	0	0	0	0	0	0	*	4	9	35
South Carolina														
Beaufort	Max	59.8	62.5	69.1	77.1	83.4	87.6	89.9	89.3	84.7	77.3	69.1	61.8	76.0
	Min	38.3	40.0	46.5	54.3	62.6	68.6	71.7	71.4	67.3	56.6	47.0	40.6	55.4
	Mean	49.1	51.3	57.8	65.7	73.0	78.1	80.8	80.4	76.0	67.0	58.1	51.2	65.7
	Freeze Freq	?	?	?	?	?	?	?	?	?	?	?	?	?
Charleston	Max	56.9	58.9	65.1	73.2	80.2	85.0	87.9	87.3	82.9	74.9	66.7	59.6	73.2
	Min	41.4	43.0	49.7	58.3	66.3	72.2	75.2	74.9	70.9	60.4	50.9	44.1	58.9
	Mean	41.2	51.0	57.4	65.8	73.2	78.7	81.6	81.1	76.9	67.7	58.8	51.8	66.1
	Freeze Freq	11	8	3	0	0	0	0	0	0	0	4	10	36
Georgetown	Max	57.8	60.0	66.7	74.9	81.7	86.6	89.2	89.0	84.5	76.5	68.4	60.6	74.7
	Min	36.6	37.7	45.0	53.3	62.0	68.3	71.7	71.2	66.7	55.2	45.5	38.3	54.3
	Mean	47.2	48.9	55.9	64.1	71.8	77.5	80.5	80.1	75.6	65.9	57.0	49.4	64.5
	Freeze Freq	12	4	3	0	0	0	0	0	0	0	4	10	33
North Carolina														
Southport	Max	55.8	57.2	63.4	72.0	79.0	84.3	87.1	87.1	83.3	75.1	66.8	58.8	72.5
	Min	35.5	36.6	43.2	52.2	60.8	67.7	71.9	71.2	66.3	54.3	44.8	37.7	53.5
	Mean	45.7	47.0	53.4	62.1	69.9	76.0	79.5	79.2	74.8	64.7	55.8	48.3	63.0
	Freeze Freq	13	11	3	0	0	0	0	0	0	0	4	10	41
Morehead City	Max	54.5	55.9	62.1	70.5	77.8	83.4	86.5	86.7	83.1	74.8	66.2	57.9	71.6
	Min	36.4	37.0	43.6	52.3	61.2	68.5	72.5	72.4	67.7	56.8	46.5	38.9	54.5
	Mean	45.5	46.5	52.9	61.4	69.5	76.0	79.5	79.6	75.4	65.8	56.4	48.4	63.1
	Freeze Freq	14	11	5	0	0	0	0	0	0	0	4	11	45
Elizabeth City	Max	50.5	52.6	59.8	69.9	76.8	83.6	87.3	86.4	81.2	71.4	62.6	53.9	69.7
	Min	32.2	33.1	40.0	48.6	57.5	65.1	69.5	69.1	63.5	52.1	42.2	34.7	50.6
	Mean	41.4	42.9	49.9	59.3	67.2	74.4	78.4	77.8	72.4	61.8	52.4	44.3	60.2
	Freeze Freq	16	12	7	0	0	0	0	0	0	0	5	14	54

*Less than once in two years.

and money: The U.S.D.A. Soil Conservation Service (S.C.S.). Each county S.C.S. office has General Soils Maps that will show the overall soil composition of the proposed aquaculture site. On the back of the map is a chart describing the suitability of the area of interest for pond construction.

More detailed soil composition descriptions are given in the county soil profile books, (i.e. soil type contours superimposed over aerial photographs), which are also available at S.C.S. offices. Soil chemists will discuss the suitability of various soils for pond construction as well as visit the actual site to take core samples and analyze them for clay content. The following soil characteristics are necessary for the excavated part of the pond to retain water:

- 25 percent or more clay to at least 3-foot depth;
- no bedrock or cemented layers within 3 feet of the surface;
- no water table within 3 feet of the surface.

If the site has been farmed previously, a thorough check for herbicide and pesticide residues should be carried out. This includes taking soil samples for analysis from two areas in particular, low areas where runoff accumulates, and any areas which were used to store pesticides or to transfer them to spraying equipment. An accurate history of the farm will help locate sample areas.

S.C.S. engineers can advise on avoiding properties subject to flooding. They also can help produce accurate pond layout and design plans which take advantage of the topography, thus minimizing earth moving costs.

Additional Site Selection Criteria

Other criteria which should be considered for each particular site include the following:

- Permitting/Legal
- Tides and currents in water supply
  - Tidal range
  - Storm surge
  - Salt wedge in river
- Accessibility
  - Roads
  - Docks
  - Rail Service
  - Security
- Utilities (availability/cost)
  - Electrical power
  - Diesel fuel for generators
  - Fresh water (municipal or on-site wells)
- Supplies and Equipment
  - Feed
  - Fertilizer
  - Ice
  - Earthmoving machinery
- Labor (availability/cost)
  - Skilled
  - Unskilled
- Processing and marketing
- Land costs

Practical Permitting Suggestions

William R. Younger
Texas Marine Advisory Service
County Courthouse, Room 326
Bay City, Texas 77414
and
Joe Moseley and James A. Shiner
Principals, Shiner, Moseley & Assoc.
Engineers & Consultants
Corpus Christi, Texas 78415-1818

There is nothing magical or mysterious about 99.9 percent of all regulatory activity – despite what some consultants will tell (sell) you. A carefully applied combination of thorough preparation, common sense, logical thinking and professional behavior (without any undue rhetoric or posturing) should result in timely processing of an application. During the application process, it's important to bear in mind the fundamental importance of regulatory agencies for our own benefit.

Regulations have been enacted for legitimate purposes

Virtually all restrictions related to development of an aquaculture facility originated from broadly accepted needs—be it water quality protection, wetland preservation, heritage, public health, or others. Unfortunately, most such laws and regulations predate the current boom in mariculture and do not take into consideration its unique circumstances. As a consequence the nondiscriminatory application often results in some strange, and even counterproductive circumstances. This situation makes it doubly critical that an early and cooperative working relation be established with the various regulatory agencies.

Regulatory personnel are reputable, competent, professionals

The myth of a nameless, faceless bureaucrat is just that – a MYTH! The resource agencies are normally staffed with scientists and engineers who are technically competent and generally oriented toward helping get good projects approved. However, they are legally and ethically bound to enforce the applicable laws and regulations. They **cannot** change either the laws or regulations, and except in limited circumstances, they are not empowered to grant variances. Thus, the objective becomes finding a way to make a mariculture project fit within existing requirements – possibly with some innovative interpretations. Lastly, it must be recognized that PEOPLE make decisions about the laws and regulations, and all actions should take this fact into account.

Obtaining and Completing Permits

Request and review permits

The first step in this process is to request copies of the application forms and companion materials from all permitting agencies in order to become acquainted with the required information and the form in which it is to be submitted. A state-by-state list of regulatory agencies is provided in Table 1. Follow this step with informal meetings to gain a full understanding of the requirements and procedures and eventually to lay out preliminary plans to the agencies. It is particularly important to include those agencies whose function is described as Administrative Review in these informal pre-application discussions. The preparation of pre-development drawings, maps, descriptive reports, etc. as documentary support will prove most helpful to the permitters in unofficially sanctioning this proposal and/or pointing out its weaknesses or inadequacies.

This application preview can often curtail costly mistakes before they occur. It creates a cushion that allows re-designs or modifications to be made in order to come up to regulatory standard, or the subsequent selection of an alternative culture site, which may have more suitable development characteristics and/or be less environmentally sensitive. An additional positive aspect of this pre-application dialogue is that it does not necessarily require the services of lawyers, engineers, or other consultants. DO NOT purchase or lease any property, even if it fully meets your established operational criteria, until you are reasonably certain the parcel(s) are permittable. A Corps of Engineers biologist can be requested to make an onsite evaluation of your proposed location and give you a written description of its characteristics. (See site selection paper by R. Miget in this section)

Apply For Ample Needs

Include sufficient excess in the applications to offer protection in the event of an emergency and/or expansion of the operation. This could be especially critical in an emergency situation requiring the additional diversion or discharge of water. It is usually far less difficult and costly to be granted permission for requirements that are beyond the projected demands of your initial operation than it would be to submit additional applications to service unforeseen or underestimated needs.

File complete applications

Follow through with thorough, accurate, and carefully prepared application documents. If for some reason an application cannot be completed, contact the agency to determine if a mutually agreeable option is open to supply their informational needs. Do this prior to submittal of the formal application for review and approval – not after.

Each agency is required by law, regulation or policy to respond to a completed permit within a specified time. Do not hesitate to contact a permitting agency to check on the completeness of your application—or it may be needlessly delayed for lack of a crucial drawing, a signature, or a check mark in an appropriate box.

Permitting Commandments

Thou shalt not treat permitting as an afterthought

This should be a critical element in early project planning and site selection. Some permit requirements will vary from site to site; at some locations, certain potentially critical or vital permits required elsewhere may not even be needed!

If the land is already owned, a careful regulatory assessment may strongly suggest against using it for mariculture purposes. Keep in mind that no site is ever perfect from all regards. Just make sure to avoid anything that poses a fatal flaw.

Thou shalt get thy facts together

Substantive technical data are required for the site, the project, and the resources required. The facts should be presented in a clear, technically acceptable fashion. Applicants should become familiar with all applicable rules and regulations for two reason. Avoid surprises and appear competent to the regulator.

Thou shalt not try to bamboozle the agencies or the public

It is in the applicant's interest to present all needed information in a timely fashion and the proper format. The better the agency understands the facts, the easier it will be for them to help. Incomplete or unclear information will translate directly into delays and more time, which is presumed to be of value to the applicant.

Be "heads up" with the public. Without exception, public concern most frequently arises when there is a perception that facts are being hidden. An applicant is under no obligation to publicly tout a project, although certain public notices will be required during the permitting process. Generally, all information submitted to a regulatory agency is available to the public/media. If pressed for information, provide a concise project summary (one to two pages, modeled after a press release) along with a copy of permit applications. Don't expect a big outpouring of public support to help much; although widespread opposition can hurt.

Thou shalt expect help rather than harassment from agency personnel

Generally, agency personnel are very helpful, unless an applicant does something that compromises such assistance. In early discussions with these professionals, expect information and general guidance, not specific decisions on the spot. A refusal to give a spot judgement is not "uncooperativeness;" the system just doesn't work that way, nor would such action be in the interest of either the public or the applicant. If the applicant goes in with a confrontational/adversarial attitude, then this is the type of relationship that will quickly develop.

Thou shalt neither fear nor overly trust consultants

Competent professional consultants can be invaluable in getting a project permitted quickly and smoothly. Most consultants are very competent in a narrow field; unfortunately, some consultants overstate their abilities. Consultants often have agendas of their own, which they do not perceive to conflict with the owners/applicants goals. While such agendas may not conflict per se, they distract from the most immediate need at hand. Every effort should be made to keep energies focused on the objective. Remember you are the one paying the bill and therefore they should respond to your directions. Don't let them lead you into unwanted conditions

Consultants can be, and often are, expensive. However, if taken in context, their fees are often reasonable. (If six months is saved on a $5 million project, then the interest savings alone is on the order of a quarter of a million dollars.) The key in effectively using professional help is to establish a specific work statement, budget, and timetable, and then stick to it! Beware of any "professionals" that want to handle all dealings with the agencies alone and without the owner present. The owner and regulators should get acquainted. A regulatory proceeding will always go better if there is a "comfort level" between all parties. However, professional consultants call the shots" as far as technical needs, procedural requirements, etc.

Thou shalt allot 150 to 200 percent of the time, energy and resrouces expected

Unforseen developments or diversions inevitably arise; while specific items cannot be predicted, contingencies should be provided. If there is a critical timing problem, share it with the appropriate agencies. But, do not expect preferential or special treatment. After all, each persons' problem is always special and urgent to them! Nevertheless, agencies do have a little latitude.

Major Permit Categories

The specific permits required will vary from project to project and location to location. Nevertheless, there are several general categories of regulatory approvals that normally have to be sought; these are:

Water

- A water use or water rights permit, normally issued by the state.
- Discharge permit for routine pond flow=through and draining; nomrally, both a state permit and a federal NPDES (National Pollution Dishcarge Elimnation System) applications are required.
- Well permits in some states may be required.

Wetlands/Navigable Waters

- Any part of the project, including water intake or discharge, that is in federal wetlands jurisdictional area (generally elevation below +3 feet mean sea level) or in the water will require a Sec. 404/Sec. 10 permit from the U.S. Army Corps of Engineers (US CE).
- Most states also require a wetland or shoreline alteration permit.

Public Lands

- Most states claim title to submerged lands, and a lease/permit may be needed for intake structure/channel, even if the actual production facilities are on nearby privately owned uplands.
- Any mariculture project using open water or submerged lands is likely to require state approval for lands.

Organisms

- There's a lot of uncertainty on this aspect. For example, in some states, redfish are classified as sport fish and their commercial taking is prohibited. Although no special law provides for mariculture operations with redfish, enforcement agencies are apparently "understanding." However, some states such as South Carolina, specifically require a red drum culture permit.
- Other possible permits may include shellfish culture, non-native species, general "fish-farming," etc.

Health/Food Related

- Depending on how far "downstream" an operator goes, there may be a whole array of permits that are normally administered by state health departments.

Be Active in the Political Process

Get involved in the political, governmental, research and educational processes that have the potential to influence (either positively or negatively) the development and growth of your industry. For example, mariculturists in Texas recently formed a task force with the assistance of the Texas Agricultural Extension Service and the Corpus Christi Area Economic Development Corporation to introduce legislation to exempt mariculturists from an inappropriate and time-consuming water use permit. Some coastal states, such as South Carolina, are working towards a vastly improved permitting mechanism with their "one stop shopping" feature for coastal aquaculturists. If aquaculture industry associations exist in your area, join them and work through them to formulate and implement a fair and simplified permitting routine. The point is **don't sit back and wait for someone else to do these things for you** – be part of determining the speed and direction of your business!

Conclusion

As final note, the following simple guidelines are reviewed:

- Start early in the permitting process and allow extra time.
- Complete the applications thoroughly and accurately.
- Work toward cooperation, not confrontation. Remember: The individuals responsible for reviewing permit applications are dedicated, competent professionals – treat them accordingly.

Table 1. Summary listing of state and federal regulatory, review and licensing agencies.

Agency	Permit/License
Alabama	
Alabama State Docks Department Chief Administrative Officer P.O. Box 1588 Mobile, AL 36633 (205) 690-6114	License to construct in wetlands or water bottoms
Office of the District Engineer U.S. Army Corps of Engineers P.O. Box 2288 Mobile, AL 36628 (205) 690-2660	Section 10 Permit Section 404 Permit
Alabama Department of Environmental Management 1751 Congressman W.L. Dickson Drive Montgomery, AL 36130 (205) 271-7700	Water Discharge Permit
Baldwin County Health Department P.O. Box 150 Bay Minette, AL 36109 (205) 937-9561 **or** Mobile County Health Department P.O. Box 2867 Mobile, AL 36652-2867 (205) 690-8158	Septic Tank Permit
Alabama Department of Conservation and Natural Resources Marine Resources Division P.O. Box 189 Dauphin Island, AL 36528 (205) 824-2161	Seafood Dealers License Retail Saltwater Fish Dealer's License Saltwater Net License Commercial Hook and Line License Optional — Freshwater Dealer's License Retail Freshwater Fish Dealer's License
Florida	
County Government Planning and Development Department (Listed locally for each county)	Site Plan Application
Florida Department of Environmental Regulation Division of Industrial Wastes 2600 Blairstone Road Tallahassee, FL 32303 (904) 488-0130	Industrial Wastewater Treatment and Disposal Permit
Florida Department of Natural Resources 3900 Commonwealth Blvd. Tallahassee, FL 32303 (904) 488-2725	Administrative Review to assess impacts
Florida Department of Environmental Regualtion Division of Dredge and Fill 2600 Blairstone Road Tallahassee, FL 32303 (904) 488-0130	Dredge and Fill Permit

Table 1. (continued)

District Engineer Corps of Engineers, Jacksonville District P.O. Box 4970 Jacksonville, FL 32232-0019 (904) 791-2234	Section 10 Permit Section 404 Permit
County and Municipal Governments	County or City Easement Permit Water Use Permit
Private Landowners	Easements for private property
Florida Department of Natural Resources Division of State Lands 3900 Commonwealth Blvd. Tallahassee, FL 32303 (904) 488-2725	Public Easement Permit
Water Management Districts (Local offices or contact Department of Natural Resources)	Consumptive Use Permit Storm Water Runoff Permit Water Retention Device Permit
Florida Department of Natural Resources Division of Marine Resources 3900 Commonwealth Blvd. Tallahassee, FL 32303 (904) 488-2725	Mariculture Permit
U.S. Environmental Protection Agency Regional Office 345 Courtland St. N.E. Atlanta, GA 30365 (404) 347-3004	National Pollution Discharge Elimination System
Game and Freshwater Fish Commission Division of Fisheries 620 South Meridian Tallahassee, FL 32301 (904) 488-2725	Aquaculture Permit
Georgia	
Georgia Department of Natural Resources Coastal Resources Division 1200 Glynn Avenue Brunswick, GA 31523-9990 (912) 264-7218	Construction Permits Permit for Collection of Brood Fish Fish Farming License
Office of the District Engineer U.S. Army Corps of Engineers Attn: Regulatory Branch, OP-F (Mr. Osvald) Savannah, GA 31402-0889 (912) 944-5347	Section 10 Permit Section 404 Permit
Georgia Department of Natural Resources Environmental Protection Division 1070 E. Tower 205 Butler Street, S.E. Atlanta, GA 30334 (404) 656-4887	Water Use or Diversion Permit Water Discharge Permit

Table 1. (continued)

Louisiana	
Louisiana Department of Wildlife and Fisheries Division of Fisheries P.O. Box 98000 Baton Rouge, LA 70898-9000 (504) 765-2328	Domestic Fish Farm Certificate
Louisiana Department of Natural Resources Coastal Management Division P.O. Box 44487 Baton Rouge, LA 70804-44487 (504) 342-7591	Coastal Use Permit
Louisiana Department of Environmental Quality Office of Water Resources P.O. Box 44091 Baton Rouge, LA 70804-4091 (504) 342-1265	Water Discharge Permit
Louisiana Office of Public Works Department of Transportation and Development P.O. Box 94245 Baton Rouge, LA 70804 (504) 379-1234	Well Registration
Mississippi	
U.S. Army Corps of Engineers Attn: Regulatory Function Branch P.O. Box 60267 New Orleans, LA 70160 (604) 862-2255	Section 10 Permit Section 404 Permit
Mississippi Department of Wildlife, Fisheries and Parks Bureau of Marine Resources 2620 Beach Blvd. Biloxi, MS 39531	Development activities in coastal counties
Mississippi Department of Wildlife, Fisheries and Parks Aquaculture Permit Bureau of Fisheries and Wildlife P.O. Box 451 Jackson, MS 39205	Cultivation/marketing Permit Broodstock Collection Permit Wholesale Seafood Dealer's License
Mississippi Department of Natural Resources Bureau of Land and Water Resources P.O. Box 10385 Jackson, MS 39209 (601) 961-5200	Permit to withdraw from a body of water Permit to draw water from well of six inches or greater diameter pipe
Mississippi Department of Natural Resources Bureau of Pollution Control P.O. Box 10385 Jackson, MS 39209 (601) 961-5171	Water Discharge Permit

Table 1. (continued)

Mississippi State Board of Health Shellfish Sanitation Branch P.O. Box 328 Gulfport, MS 39502 (601) 863-1036	Processing/packaging Certification Building Plan Approval
County and Municipal Governments	Construction Permits Business Licenses
North Carolina	
North Carolina Wildlife Resources Commission Division of Boating and Inland Fisheries Archdale Building Raleigh, N.C. 27611 (919) 733-3633	Sales Permit
North Carolina Department of Environment, Health and Natural Resources Division of Environmental Management 512 N. Salisbury Street Raleigh, N.C. 27611 (919) 733-2293	NPDES Permit for Water Discharge CAMA Permits
U.S. Army Corps of Engineers Office of the District Engineer P.O. Box 1890 Wilmington, N.C. 28401-1890 (919) 343-4511	Section 10 Permit Section 404 Permit
County and Municipal Governments	Construction Permits Business Licenses
South Carolina	
South Carolina Coastal Council Summerall Center Suite 802 19 Hagood Street Charleston, S.C. 29403 (803) 792-5808	Critical Area Permit
South Carolina Wildlife and Marine Resources Division of Wildlife and Freshwater Fisheries P.O. Box 12559 Charleston, S.C. 29412 (803) 795-6350	Importation of Prohibited Species Permit
County and Municipal Governments	Zoning Compliance/Business Licenses Construction Permits
South Carolina Department of Health and Environmental Control 2600 Bull Street Columbia, S.C. 29201 (803) 734-5300	NPDES Permit
U.S. Army Corps of Engineers District Engineer, Charleston P.O. Box 919 Federal Building Charleston, S.C. 29402 (803) 724-4330	Section 10 Permit Section 404 Permit

Table 1. (continued)

South Carolina Water Resources Commission P.O. Box 4440 3830 Forest Drive Columbia, S.C. 29240 (803) 758-2514	Groundwater Use Permit Water Use Report
Texas	
U.S. Army Corps of Engineers District Engineer P.O. Box 1229 Galveston, TX 77553 (409) 766-3899 or P.O. Box 2948 Corpus Christi, TX 78403 (512) 888-3355	Section 10 Permit Section 404 Permit
U.S. Environmental Protection Agency Region VI 1445 Ross Avenue, Suite 1200 Dallas, TX 75202 (214) 655-7180	National Pollutant Discharge Elimination System Permit
U.S. Fish and Wildlife Service Corpus Christi, TX 78412 (512) 888-3346 or 17629 El Camino Real, Suite 211 Houston, TX 77058 (713) 229-3681	Construction Project Review Fish and Wildlife Import/Export License Designated Port Exemption Permit
National Marine Fisheries Service 4700 Avenue U Galveston, TX 77551 (409) 766-3699	Construction Project Review
Texas General Land Office Coastal Division 1700 North Congress Avenue Stephen F. Austin Building Austin, TX 78701 (512) 463-5225	Lease/Easement for state-owned lands
Texas Department of Agriculture P.O. Box 12847 Austin, TX 78711 (512) 463-7602	Fish-Farmer's License Fish-Farm Vehicle License Cultured-Fish Processing Plant License Bill of Lading for Certain Vehicles
Texas Parks and Wildlife Department 4200 Smith School Road Austin, TX 78744 (512) 389-4633	Sand, Gravel, Shell and Marl Permit Red Drum and Speckled Seatrout Sourcing Permit Freshwater Commercial Fishing Boat License Saltwater Commercial Fishing Boat License Wholesale Fish Dealer's License Wholesale Fish Truck Dealer's License Retail Fish Dealer's License Retail Fish Truck Dealer's License

Table 1. (continued)

Texas Water Commission P.O. Box 13087, Capitol Station Austin, TX 78711-3087 (512) 463-8238	Section 401 Certification Discharge Permit Reclamation-Engineer Permit Water-Use Permit
Texas Department of Health 1100 West 49th Street Austin, TX 78756-3182 (512) 458-7248	Food Manufacturer Registration
Texas Animal Health Commission (Arranged through local veterinarian)	Certification of Veterinary Inspection
Texas Antiquities Committee P.O. Box 12276, Capitol Station Austin, TX 78711 (512) 463-6098	Administrative Review

Pond Design and Construction

Ross Ulmer
Soil Conservation Service
Suite 1321, Federal Building
100 West Capitol Street
Jackson, Mississippi 39269

Fishpond design and construction techniques for red drum aquaculture are presently in the development stages. Features of several research systems, along with some 25 years of Mississippi experience with catfish pond construction, have been incorporated into the following material. System data included in this paper are generalized; requirements for particular sites may vary from these data.

Planning Considerations

Soils must be an early consideration in developing an effective fishpond operation. Sufficient borings and samples should be taken to determine:

- Is the soil water tight in the pond bottom? Can material be compacted in levees? Will seepage through undetected sand lenses be costly over long periods of time?
- Are pesticides, herbicides or other chemical residues present in the soil from farming, cattle, or forest operations?
- Does the soil have a high ground-water table causing construction difficulties? (Added drainage may also be required to provide adequate pond outlet capacities.)

Adequate water quality and quantity are essential to successful pond culture operations. Several "salient" points are:

- Saline water with 25+ ppt (parts per thousand) salt content is recommended to hatch eggs and grow the fry up to about 1 1/2-2 inch size. A dependable source must be developed considering:
  a. Tidal fluctuations may allow only 12 hours for pumping;
  b. Salt water dilution by fresh water stream outflow into coastal areas. Surface water does not have enough salt content and the acidity (pH) is too low; and
  c. Shoreline turbidity will reduce water quality needed for fish. The pump inlet pipe may need to be extended beyond the turbidity zone.
- Storage reservoirs may be used to provide a dependable water supply. Water flow from the reservoir may be pumped or may be by gravity if there is sufficient elevation.
- Pond water exchange up to 20 percent of water volume per day (180 gpm per surface acre of pond) may be considered for extreme conditions (worst case situation). Pond discharge should not be so great that it would exceed water quality limits after dilution in coastal waters. Normal daily water exchange would be only about 1 percent of the total water volume.
- Water aeration equipment (paddlewheels, spray aerators, bubblers, etc.) may be needed to maintain water quality through critical periods of the day. Oxygen production by water stops at sundown (no more photosynthesis) and oxygen consumption by the fish may lower the dissolved oxygen content below the critical level of 3.0 ppm (parts per million).

Pond system layout is also very important to the efficient operation of the aquaculture system. Several important points are:

- Arrangements of ponds to one another and to the water supply will help utilize common levees, water distribution lines, and drainage channels, and improve pond access for feeding, checking, water quality monitoring, and harvesting operations.
- Location of system near access roads should also be considered due to:
  a. Feed delivery by large trucks.
  b. Harvest truck road requirements.
  c. Daily checking and feeding needs.
  d. Construction and subsequent maintenance activities.
- High ground should be considered in selecting the pond sites to:
  a. Build levee above storm tide elevation.
  b. Provide adequate drainage away from the pond drain outlet.

Other factors to be considered in developing an aquaculture system:

- Do you have adequate financing for land, pond construction, water pump/pipe system, feed, labor, etc.? This may require up to $3500 per acre plus land before the first fish is sold.

- Do you have the level of management required to monitor dissolved oxygen and other water quality levels at all hours of day **and night**? This is a high value crop which can be lost in a matter of hours. Dependable labor and equipment must be available when needed.
- Is there a readily available source of fish food? Catfish food contains about 35 percent protein and represents about 40 percent of production costs. When feeding at the rate of about 200 percent of the fish body weight per year, long hauling distances will increase feed costs and increase production costs.
- Are there readily available markets and processing plants for your fish? Can you harvest your own fish or is a commercial harvesting contractor available?

Design of Fishponds

The designs that follow are based on (a) 80-acre tract of land having five grow-out production ponds of 10 acres, five yearling production ponds of 2.5 acres, and three fingerling production ponds of 0.5 acres for a total of 64.0 acres of water (see Figure 1), and (b) a 170-acre tract of land having six grow-out production ponds of 20 acres, six yearling production ponds of 4.5 acres, and eight fingerling ponds of 0.6 acres for a total of 151.8 acres of water (see Figure 2). The grow-out pond to yearling pond size should have about a 4:1 ratio to prevent stocking densities from exceeding manageable limits (3500-4500 #/ac.)

Levee design should include top widths wide enough to accommodate feeding and harvesting equipment. Widths range from 16 feet to 20 feet on the newer catfish ponds. Some older levees less than 16 feet wide have required repair to maintain access widths. Constant wave action (especially the larger ponds) erodes the levee sides and narrows the top width. Important engineering features include:

- Levee side slopes on the inside (wet side) have been flattened to 4:1 (4-foot horizontal to 1-foot vertical) or 5:1 in an effort to reduce the wave damage. The 4:1 slope along with the wider 18-foot to 20-foot top width appears to be a reasonable trade off of initial cost versus later repair expense. The outside slope (dry side) is 3:1 which provides for mowing safety and other maintenance activities.
- Top widths of exterior levees at 18 feet provide continuing access for feeding and maintenance operations. The interior levee (common to more than one pond) are often built with a 20-foot top width. This provides some extra space for harvest trucks, main travel route safety, vehicle passage, and aerator operation.
- Levees are constructed at a minimum height of 1.0 foot above design pool level for access road stability (dryness), wave protection, and storm rainfall containment without overtopping. Additionally, at least 10 percent of fill height should be added to the levee height to allow for settlement and assure that the 1.0 minimum height is maintained.
- Pool depths should range from 3.5 feet to 5.5 feet. The depth, depending on the site, would be an excavation and levee combination (see Figure 3). The bottom of the pond will be graded to the outlet drain pipe. Bottom grades along both levee directions to the outlet will provide adequate drainage using the following:

Fish Size	Bottom Grades
fingerlings	1.0 percent
yearling	0.3 to 0.5 percent
grow-out	0.1 percent

- A small harvest basin 1.0 foot below the graded pond bottom in the fingerling and yearling ponds should be constructed in the lower corner (at outlet drain) to facilitate harvest without damage to fish. A small 10- x 20-foot concrete basin capable of being flushed out would be best for the fingerling ponds. A larger, earth excavated basin about 50- x 50-foot would be adequate for the yearling ponds.

Yardage Volume

Yardage volume for the typical pond layout on 80 acres of land shown in Figure 1 is 82,750 cubic yards. The yardage calculations were made using AUTO POND, a Soil Conservation Service computer program (W.C. Hamberlin 1985, revised B.J. Massey 1987) which balances cut (excavation) and fill ratio between 1.12 and 1.20. The above yardage requirement is for a levee with 4:1 inside (wet) slope and 3:1 outside (dry) slope having 18 foot top width for exterior levee and 20 foot top width for interior levee (see Figure 3). A buried 12-inch PVC pipe 2,000 feet long with outlet pipe and alfalfa valve shutoff to each pond will provide the required recharge and flush water.

Yardage volume for a similar pond layout having a middle area distribution canal at pipe location (water level elevated 2.5 feet above the pool water level) is 106,200 cubic yards. The canal system would also require 13 inflow structures (15-inch diameter CMP with 30-inch diameter slotted board riser capable of varying the pond inflow). A bridge across this 2,000 foot long canal may be desired to facilitate pond access.

Yardage volume for the typical pond layout on 170 acres of land (Figure 3) is 142,500 cubic yards using the same levee slopes and top widths as the 80 acre land area design. The canal distribution yardage

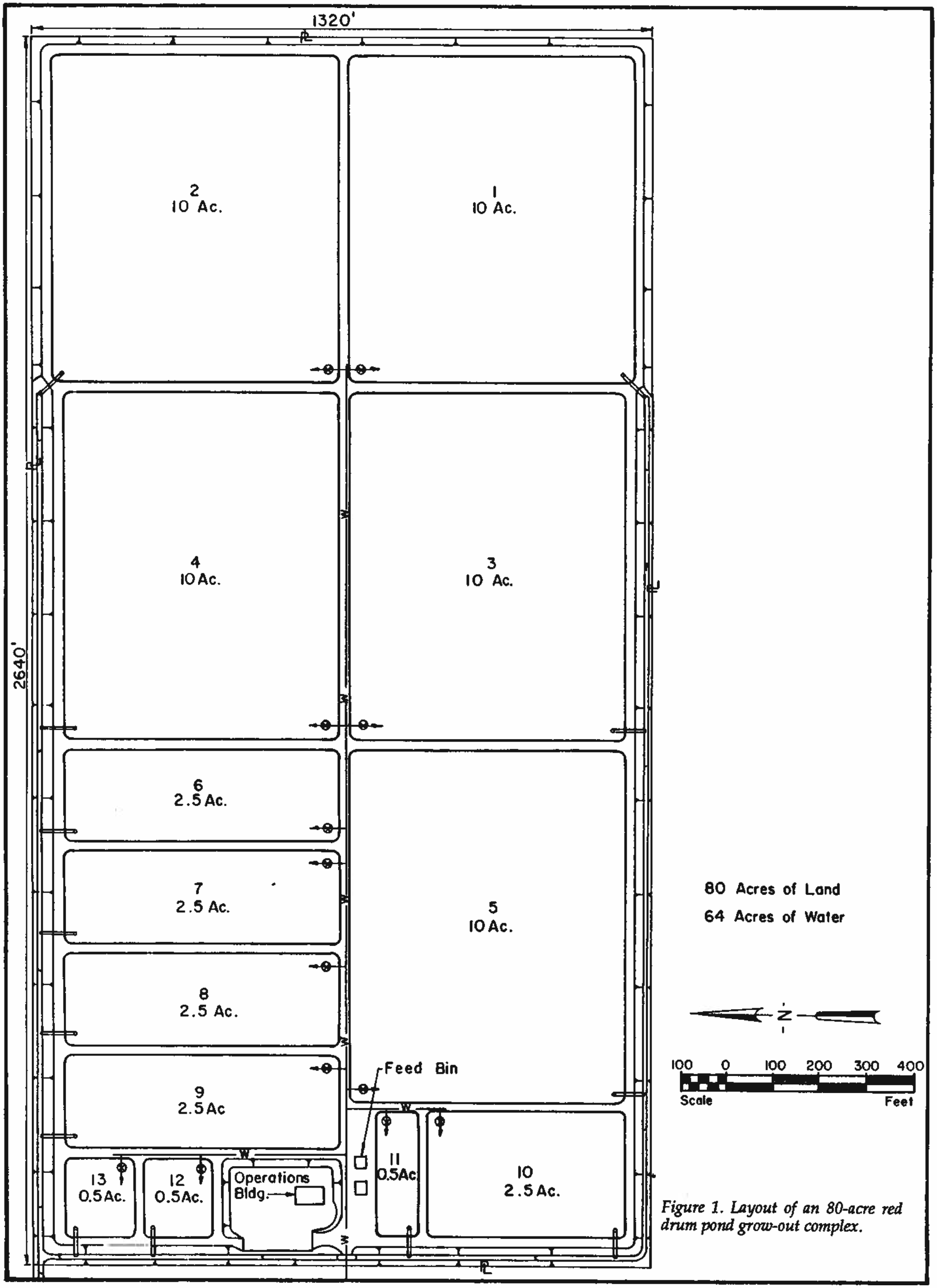
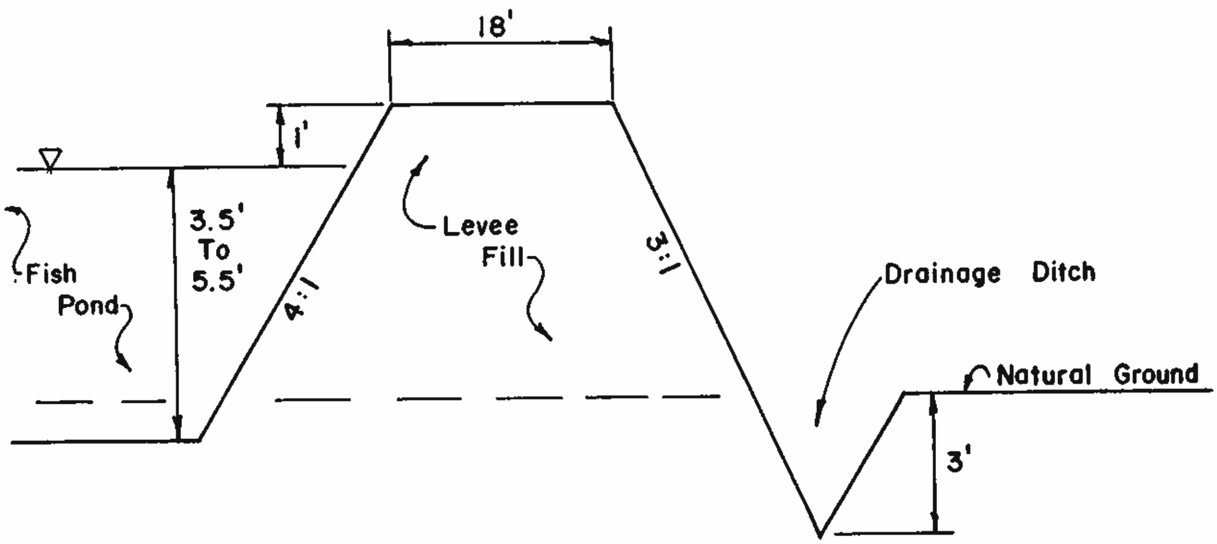
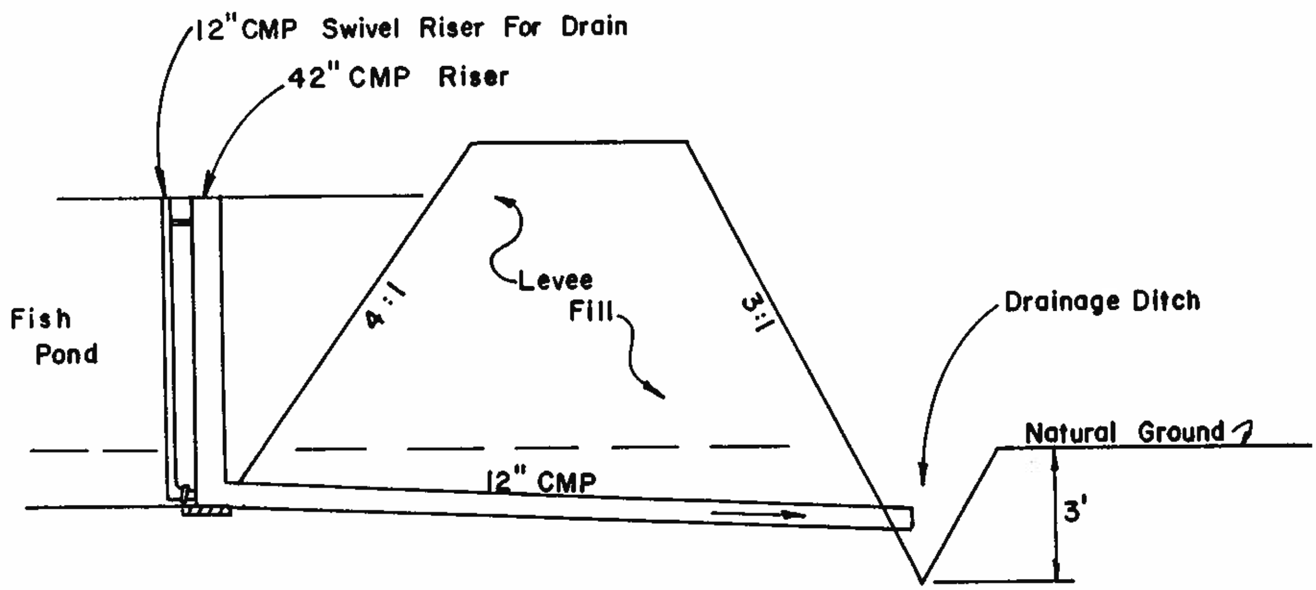


Figure 1. Layout of an 80-acre red drum pond grow-out complex.



TYPICAL OUTSIDE LEVEE SECTION



TYPICAL OUTLET DRAIN PIPE SECTION

Figure 2. Cross-section of pond levee showing recommended dimensions for a red drum grow-out pond.

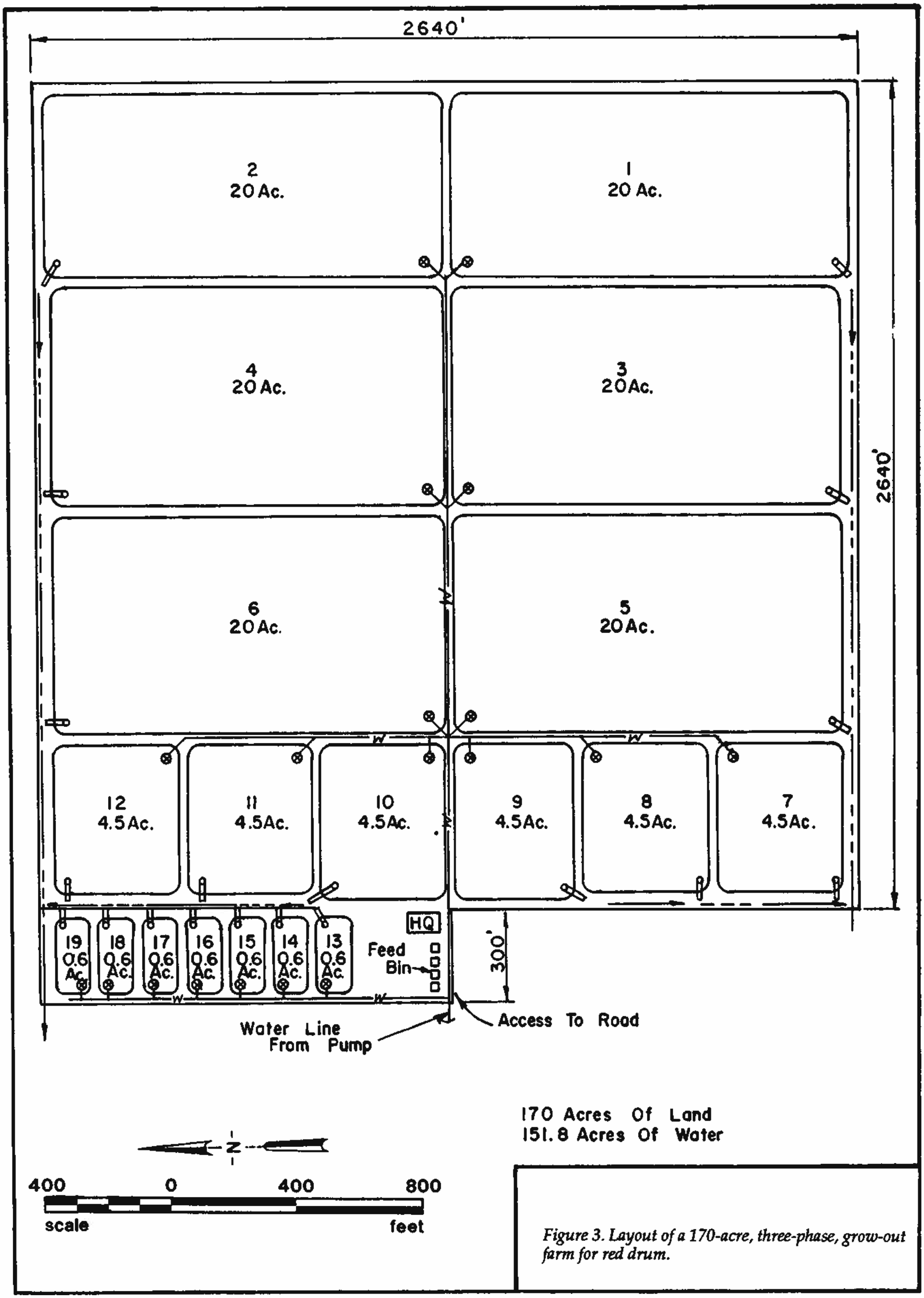


Figure 3. Layout of a 170-acre, three-phase, grow-out farm for red drum.

is estimated at 190,000 cubic yards and would require 19 inflow structures. A 2,000-foot-long 15 inch PVC pipe would be required.

Pump System

The pump system would be located at the western mid-point of the 80 acre and 170 acre tracts. A low lift axial (propeller) pump would respectively provide 1,800 gpm (8-inch diameter, double propeller pump) and 3,600 gpm (10 inch diameter, double propeller pump) with 30 feet of head.

These flows will respectively provide a 20 percent flush of one 10 acre and one 20 acre pond per day or about 2.5 percent flush of all pond areas per day. A single pond flush would be used only in case of emergencies (disease or water quality problems).

Installation of a second pump at the pump house, feeding into the established 2,000-foot PVC pipe would provide a backup for any pump repair and be valuable insurance for fish crop and investment against pump failure.

Drain Design

The outlet drain pipe must be adequate to remove the 20 percent pond flush(1,800 gpm and 3,600 gpm respectively). Design capacity for the drop inlet considered a 4-inch water level increase limit over the pipe riser crest (See Figure 2). A 42-inch pipe riser with a 12-inch outlet will carry the 1,800 gpm. A 48-inch pipe riser with a 15-inch outlet will carry the 3,600 gpm.

The outside perimeter drainage ditch was designed to handle the discharge from all ponds. This capacity would be required when a large storm with 8- to 10-inch rainfall occurs requiring simultaneous discharge.

Drainage ditch capacity under the access road and the PVC pipe will also be required. Twin 24-inch CMP will carry some 23 cfs, adequate for the three 10-acre ponds, the 2.5-acre pond, and the 0.5-acre pond. The drainage ditches were separated for the 170-acre tract of land.

Construction of Ponds

Initial Construction

A farm tractor and scraper(s) have been the most efficient equipment to build fishpond levees due to their speed and adaptability. Laser controlled scrapers add greatly to the accuracy of cuts and fills and speed at which they can be done. The dual wheeled tractors and rubber tired scrapers provide good compaction of the fill material when complete wheel track coverage is made over each layer of fill placed in the levee.

The initial construction operation is to remove all vegetation and topsoil from levee area. This will allow a good bond of fill material with the foundation soil. The topsoil should be placed on the outside levee slopes to encourage vegetative growth.

The levee is left open at the outlet pipe location until all the other levee is completed. This allows free drainage of any storm water during the construction process. Since the pond bottom will be excavated below natural ground line, the outlet channel must also be excavated during the initial construction operation.

Contractors have found it best to start a 300- to 400-foot segment of the levee, finish it to the final elevation and slopes, and then move on to the next segment. This helps maintain levee alignment and shape and make the most efficient use of soil material near the levee.

Drains and Pump

The pond outlet drain pipe (See Figure 3) must be installed in moist soil and have hand compaction around it to assure a water-tight installation. A slide head gate, siphon tube, or swivel riser may be attached to the riser to draw poor quality water off the pond bottom. The PVC pipe from the pump should be installed as the levee is being built. A small, shaped trench under the bottom third of the pipe should be prepared to provide uniform support for the pipe. Hand compacted material on each side and around the rest of the pipe to a distance of 1 foot will provide adequate installation. A minimum of 1 foot of cover over the top of pipe and 1 foot of clearance over drain ditch pipe will be needed for protection. Install pipe off to one side of levee top to reduce potential traffic damage.

The low lift axial pump must be set on a concrete or piling frame in a deepened sump or basin. Water flow to the sump may be through an open channel or underground pipe of sufficient size to match or exceed the pump capacity. Three phase electric power must be brought to the pump for the 30 to 50 hp electric motors.

To assure all-weather access on the levees for feeding, checking, and harvesting, a 4- to 6-inch gravel surface should be applied. Heavy clay soils are very sticky when wet and can allow vehicles to slide into the ponds. Travel on some silty or sandy soils may be possible if vegetated and a day allowed for internal drainage. Successive days of rain may still warrant a gravel surface.

Maintenance

Vegetation maintenance and weed control around the pond are very important as protection against wave damage. Mowing along the levee and pool edge will improve visual checks of the pond, water monitoring access, and harvesting operations. Vegetation may need fertilization at times and care must be taken to prevent contamination of the ponds.

A maintenance and repair schedule should be prepared for the pump installation. The manufacturer will be knowledgeable about lubrication, wear tolerances, and replacement periods. Operation without maintenance considerations will reduce pump efficiency and increase operation costs.

Conclusions

Properly designed production ponds, water supply and distribution systems, and harvest basins are essential elements of successful red drum culture. Experience gained in the design and construction of ponds used in the culture of other aquatic organisms, especially shrimp and catfish, should be adapted to commercial red drum grow-out.

Literature Cited

Ulmer, R.L. 1987. **Planning, design and construction of catfish pond in Mississippi**. Revision of SCS publication by C.B. Tisdale, 1981.

Extensive Aquaculture of Red Drum in the Southern United States

Charles A. Wilson
Coastal Fisheries Institute Center for Wetland Resources
Louisiana State University
Baton Rouge, Louisiana 70803

The levels of aquaculture practice vary in intensity and range from extensive to intensive, depending on the extent of environmental manipulation and control.

Extensive aquaculture, the least expensive of general aquaculture techniques, has been used for over 2,000 years to culture both finfish and crustaceans. Proponents of this non-intensive form of aquaculture use existing or slightly modified impoundments (ponds) to rear aquatic organisms on natural foods available within these systems.

Extensive aquaculture usually involves simple management and harvest of target organisms within an impoundment. It may include slight modifications of the impoundment and low density stocking of a particular species.

Semi-intensive aquaculture is characterized by such increases in energy input as aeration, water control, supplemental feeding, and higher stocking densities; intensive aquaculture involves much closer manipulation and control of the aquatic environment. Intensive aquaculture has greater potential for economic gain; however, it also has higher operating costs and risks. The financial limitations and dedication of the aquaculturist will determine within which part of the continuum his venture will fall.

Extensive culture of redfish offers a low cost, low risk aquaculture alternative for many property owners along the coastal regions of the United States. Based on recent scientific data, redfish can be cultured in salt, brackish, or fresh water, fed natural or prepared diets, and grown to harvest size (4-5 pounds) in about two years.

Caution

Red drum have been proposed as a potential aquaculture candidate for many years. Since the early work of G.R.Lunz and C.Bearden, fisheries biologists with the South Carolina Wildlife and Marine Resources Department, there has been a growing interest in red drum culture. Recent changes in state and federal laws governing the harvest of redfish and the increased demand for red drum have brought about an up-surge in the interest to culture this highly-prized drum.

Due to the high costs associated with intensive aquaculture and the availability of impoundments built in the natural habitat of red drum, many people have expressed interest in using extensive practices to culture red drum. Technologically and biologically it is feasible, however, the economics have yet to be demonstrated. An additional constraint is that the utility of many of the brackish, salt water areas of the United States may be restricted by state coastal zone management regulations. It is very important that the economic feasibility be examined and a proper understanding of the law be gained before attempting coastal aquaculture.

The extensive culture of any organism should be designed to maximize natural productivity in the existing fresh or salt water system while minimizing risk and cost. Like any aquaculture endeavor, it requires an understanding of:

- the biology of the organism to be cultured,
- the biology of potential prey organisms,
- state and federal laws which apply to the venture,
- proper site selection and modification techniques,
- water control,
- acceptable water quality,
- harvest techniques, and
- marketing.

Biology

Although the biology of red drum is covered on other pages of this manual, a brief description of biology as it relates directly to extensive aquaculture is provided below.

Following near-shore or off-shore spawning, the planktonic (floating) eggs and larvae drift into the bays and estuaries where, as juveniles, they spend the first two-to-five years of their lives. Growth rate in the estuaries is known to be rapid, and a fish that hatches in October weighs approximately one pound by the following October (the end of its first year) and can weigh five to six pounds by the end of its second year.

Red drum are opportunistic and feed on the variety of crustaceans and fishes available in the areas they inhabit. Larvae and early juvenile red drum (<15 mm, SL [standard length]) feed primarily on zooplankton. Juveniles (15-75 mm) feed mostly on small bottom invertebrates and the young of other fishes. Fish >75mm have been reported to feed on decapod crustaceans including blue crabs, penaeid and *Palaemonid* shrimp, and other fish. All of these types of prey organisms are available in brackish water and salt water impoundments along the coast of the southern United States. In freshwater, substitute prey items must be made available.

Site Selection and Modification

Selecting a site for the extensive culture of redfish requires consideration of the following factors: laws, accessibility, stability of the impoundment walls (dikes), bottom quality, water quality and quantity, water control, and harvestibility.

Laws

One of the most important criteria in extensive aquaculture is an understanding of the legal constraints and laws that pertain to the area in which the aquaculture venture is planned. Any impoundment modifications attempted should be planned and executed through the cooperation of the Corps of Engineers regional office. In most cases, any modification made in the coastal zone requires a permit that can be obtained through the Corps of Engineers. The Corps will coordinate permit application through the other state and federal agencies involved.

For example, present law in Louisiana prohibits blockage of the free and clear passage of marine organisms (e.g. nets or screens that block a tidal creek), without an approved marsh management plan (Title 56, Section 309 Louisiana Wildlife and Fisheries Code). Therefore, if an aquaculturist wanted to culture redfish in a coastal impoundment of Louisiana that was connected to the open marsh, it would be unwise to stock expensive fingerlings into that impoundment since they could escape. However, a Louisiana impoundment with an approved marsh management plan that calls for weirs (water control structures) to control the water level for the enhancement of vegetation could be used to culture redfish, if the weirs prevented escape and aquaculture was part of the plan. Obviously, different laws apply in different states, and a clear understanding of these state laws is as important as understanding the technology of redfish aquaculture.

Access

The accessibility of an impoundment to be used in aquaculture is important for two conflicting reasons. It is important for the operator to have large vehicle or boat access to the pond in any type of weather. On the other hand, easy access by the operator also means easy access by outsiders, both those with bad intent and the curious. Since theft will always be a potential problem, it would be advantageous to have someone located on site as the crop nears harvest size.

Condition of the Impoundment

During the site selection process, the physical condition of the dikes and water control structures of each impoundment under consideration should be carefully inspected. There are many impoundments in Louisiana, Texas, as well as other areas of the southern United States in which dikes have subsided and are easily breached by extreme high tides. The potential operator should walk the entire length of the dike and make notes of both low areas and areas that might wash out in the near future. Areas susceptible to wash-out are those around water control structures, areas where water is deep on both sides of the dike and areas where the dike has become thin due to erosion. Also included in the operator's inspection should be a note of any alligator or muskrat holes or any holes by other burrowing organisms that may lead to wash out.

Bottom Quality and Depth

Examination of bottom quality, bottom slope, and depth includes consideration of soil type, chemical composition, and general health of the internal vegetation of the impoundment. The operator should collect soil samples from both shallow and deep water areas of the pond and have them analyzed through local county extension agents. The soil samples should be tested for heavy metals, pesticide residue, and other contaminants that may have been reported in the area.

The quality of an impoundment can also be gleaned from its vegetation. It is desirable to have a healthy stand of vegetation in the impoundment to provide habitat for juvenile red drum as well as their prey items. Unsuitable vegetation, characterized by poor marsh grass growth and an abundance of stubble from dead grass, indicates an unhealthy system. Although it is not mandatory, a ratio of approximately 50 percent open water and 50 percent marsh is desirable. However, is more intensive systems, vegetation in the ponds should be avoided.

Depth is an element of bottom quality and an important criteria for the culture of red drum. Most of the coastal impoundments of the southern United States range in depth from 1-3 feet, which is adequate for red drum grow out during most of the year. However, it is advantageous to have a few deep holes in the impoundment for over-wintering fish to escape from drastic temperature changes. Small pockets where the water is 10-12 feet deep are buffered from drastic shifts in water temperature.

It has been reported that red drum cannot tolerate prolonged exposure to water temperatures less than four degrees centigrade. In addition, red drum, like other fish species, are susceptible to rapid changes in the water temperature, which are possible on cold and windy days. Deep water provides insulation from such rapid changes in air temperature. However, the same deep holes are also likely spots for temperature stratification. Such areas are prone to localized fish kill during extremely hot, still weather. Trees or vegetation lines around the ponds also provide some insulation by reducing the wind chilling factor.

Water Quality

Water quality is of primary importance to an aquaculturist and dependent upon many factors. (For a good review of water quality and basic measurement techniques, refer to Boyd's book on water quality.) Before using an impoundment for aquaculture, it is wise to evaluate its water quality.

The red drum is a brackish estuarine organism that has been shown to grow well in both saltwater of varying salinity levels and certain types of freshwater. Most coastal impoundments are located in brackish water areas with salinities ranging from 5 to 20 parts per thousand (ppt) and under natural conditions of favorable water quality. If freshwater aquaculture is planned, it is critical that culture water contains the proper amount of two metals. Texas Department of Parks and Wildlife and researchers at The University of Texas have produced preliminary data showing that chloride levels of 130-ppm and calcium levels of 100-ppm are sufficient to replace the lack of salt in the freshwater systems to be used for redfish culture.

Determination of the origin of the watershed of the impoundment or pond is advised as an indicator of the potential for flooding, pollution, etc. Brackish or freshwater tidal impoundments that are located in drainage basins should be examined and sampled (cast net, seine, traps) for the presence of healthy aquatic organisms as an indicator of water quality. For example, the presence of large, "fat" blue crabs, grass shrimp, and mud minnows usually indicates a healthy impoundment and acceptable water quality. If there is suspicion of runoff of pesticides or other pollutants from nearby agricultural or industrial property, collect a water sample following a heavy spring rain and have it analyzed for likely contaminates.

The color of the water is an indicator of the quality and source of water (depending on the time of year). For example, a distinctive brown (like tea) coloration may indicate that the water source is associated with a swampy area and that the water is high in tannin. Tannin (tannic acid) is leached out of rotting leaves, and excessive amounts can effect water quality. High levels of tannic acid can limit productivity of an impoundment by creating acidic conditions that interfere with plankton growth. Again, the aquaculturist can rely on the organisms present in the impoundment as an indicator of productivity.

Water quality information about an impoundment can also be collected by interviewing people familiar with the area. Questions like:"Has there ever been a fish kill?" or, "Has anyone ever seen crabs crawl out of the water of this impoundment?" (Crabs will climb up the edge of an impoundment during low dissolved oxygen.) give an indication of the frequency of these types of problems. If the common answer is that fish kills occur two to three times every year, particularly following a heavy rain, then the system is not desirable for aquaculture.

The operator may want to take the time to set a few gill nets and/or a few crab traps in the impoundment. The presence of a large number of adult fish and abundant large crabs likely indicate a healthy impoundment. If information about water quality is not available and sampling of the pond indicates that few organisms are present, then the operator should refrain for a year and study the system. Periodic visits to the impoundment during the course of the year should be planned, in order to look for any changes or indicators mentioned above.

Water control

An important criterion in maintaining good water quality is to have sufficient water exchange between the impounded area and the water source. Proper exchange is necessary to maintain adequate dissolved oxygen and nutrient exchange to culture the desired species, in this case, red drum. The operator should have water control structures in place that:

a) provide a cross flow across the pond using at least two water control structures and setting the controls so that the water does not enter and exit through the same pipe and
b) collectively provide average water exchange rates of 1- 5 percent per day over a 7-day period.

There are various types of water control structures used in the southern United States, and they are made of either wood and/or aluminum (Figure.1). Wooden water control structures can be classified as either fixed crest or Egyptian-style. Fixed crest wiers are essentially a wood dam with a fixed height that allows water to go in and out of an impoundment, and are built to prevent drainage beyond a "fixed" level. A recent modification of the fixed crest weir by Barton Rodgers of Louisiana State University is the "slotted weir" (Rodgers *et al.*1987). The slotted weir is designed to allow organisms to move into and out of the system through a vertical slot yet restrict the flow of water to assure that a minimum water level is achieved.

Egyptian-style water control structures are rectangular boxes that run the width of the dike. They have flap gates at each end that can be raised or lowered depending on the desired movement of the water. In South Carolina, a wooden riser-board assembly has replaced the inner flap gate of the Egyptian-style water control structures to control the water level in the impoundment.

Aluminum water control structures are usually made of corregated aluminum pipe. The riser board assembly on the inside of the pond has 6-8 inch wide boards (2"-x-6" or 2"-x-8") that can be stacked on top of one another to maintain the desired water level. On the outside of the pond, a flap gate is positioned over the culvert intake and is used to stop the ingress of water from outside.

The aluminum culvert is the more desired type of water control structure because it is easy to maintain and operate. New aluminum alloys now used to construct the corrugated pipe are very strong and are reported to last many years. Various modifications of the riser board assembly are available, such as the addition of a series of slots for screens (Figure 1).

Harvest

An additional criterion in site selection depends on intended harvest technique. It is much easier to operate an impoundment that drains completely. Along the Atlantic coast of the southern United States, tidal amplitude allows total draining of a pond for harvest. An impoundment in this region should drain well, and any remaining deep holes should be free of trees or other debris that would restrict the use of a net.

Tidal amplitude in the Gulf region is not sufficient for total harvest by draining; therefore, the fish should be harvested by gill net or trawl. The impoundments in this region should have areas where nets can be used for harvest. Capture by gill net combined with other methods (seining) allows harvest of a specific size class and, therefore, more effective management of the population within the impoundment by allowing partial harvest at optimal times and harvest of specific sizes. However, net harvesting is labor intensive and often incomplete.

Pond Preparation

Preparation and/or modification of a pond depends upon the willingness and financial capabilities of the operator. Modifications using heavy equipment are generally expensive and so force the operator into a more semi-intensive operation to increase yield.

Minor modifications can be undertaken using manual labor. Major dike or water control structure work requires the use of heavy equipment, e.g., a drag line. Drag line operators may be available locally and should have a general knowledge of dike work, its limitations, and its costs. The local U.S.D.A. Soil Conservation Service office is a federal agency which is available to assist in pond design and modifications.

Preparing a pond for stocking involves a) treatment of the soil and b) elimination of potential predators. The principal extensive aquaculture techniques for pond preparation discussed below originated in the Philippines and South America and can be used in the United States.

Soils

One should test potential acidity of the soil before final site solution, new construction, or disturbing the bottom of an existing pond. Acidity or potential for acidity is dependent upon sediment composition. When exposed to air, metals in the soil, like iron react with other soil components (e.g. sulfides) to create acid when water is introduced.

Even if the pond soil is not acidic upon measurement it may still have the potential to become acidic. To determine this, remove about 10 g of soil and dry it for a week in the sun. Mix the soil sample with 100 ml of water from the source to be used for growout. Measure the pH; if the pH is low (below 4.5) the pond should never be dried. Pond drying is practiced in areas where the soil organic content is high. Several months prior to loading and stocking, the impoundment should be drained as much as possible. The extent of draining will depend on the tidal amplitude and depth of the pond; it is advantageous to expose as much of the bottom as possible.

The primary reason for bottom exposure is to improve the soil chemistry by oxidizing the soil and removing as much of the hydrogen sulfide as possible. Hydrogen sulfide, a by-product of anaerobic respiration of sulphur bacteria, can reach very high levels in the bottom of impoundments. Low oxygen, promoted by temperature stratification, provides conditions for such unwanted bacterial activity. By exposing these areas to air and sunlight, the bottom becomes oxidized and this process decreases the sulfide level in the soil. Well-aerated, partially oxidized soil makes the bottom better suited for colonization by desired benthic organisms, a critical part of the food chain in an impoundment. Exposing and drying the open flats and bottom areas of an impoundment helps stimulate growth of vegetation.

A general rule for drying time is stated in terms of the size and depth of the cracks that form in the bottom: once the bottom cracks penetrate one to two inches, the pond is sufficiently dry. It is a good practice to fill and drain the pond at least once after drying to remove some of the oxidized material formed on the bottom.

There is a risk in drying ponds built in areas with acid sulfate orcationic clay soils. Iron in acid soils reacts with sulfides to produce iron sulfate. Iron sulfate will react, in turn, when the pond is filled, to produce sulfuric acid. This is a common problem in coastal impoundments in North and South Carolina, and it is referred to as formation of "cat clays." This

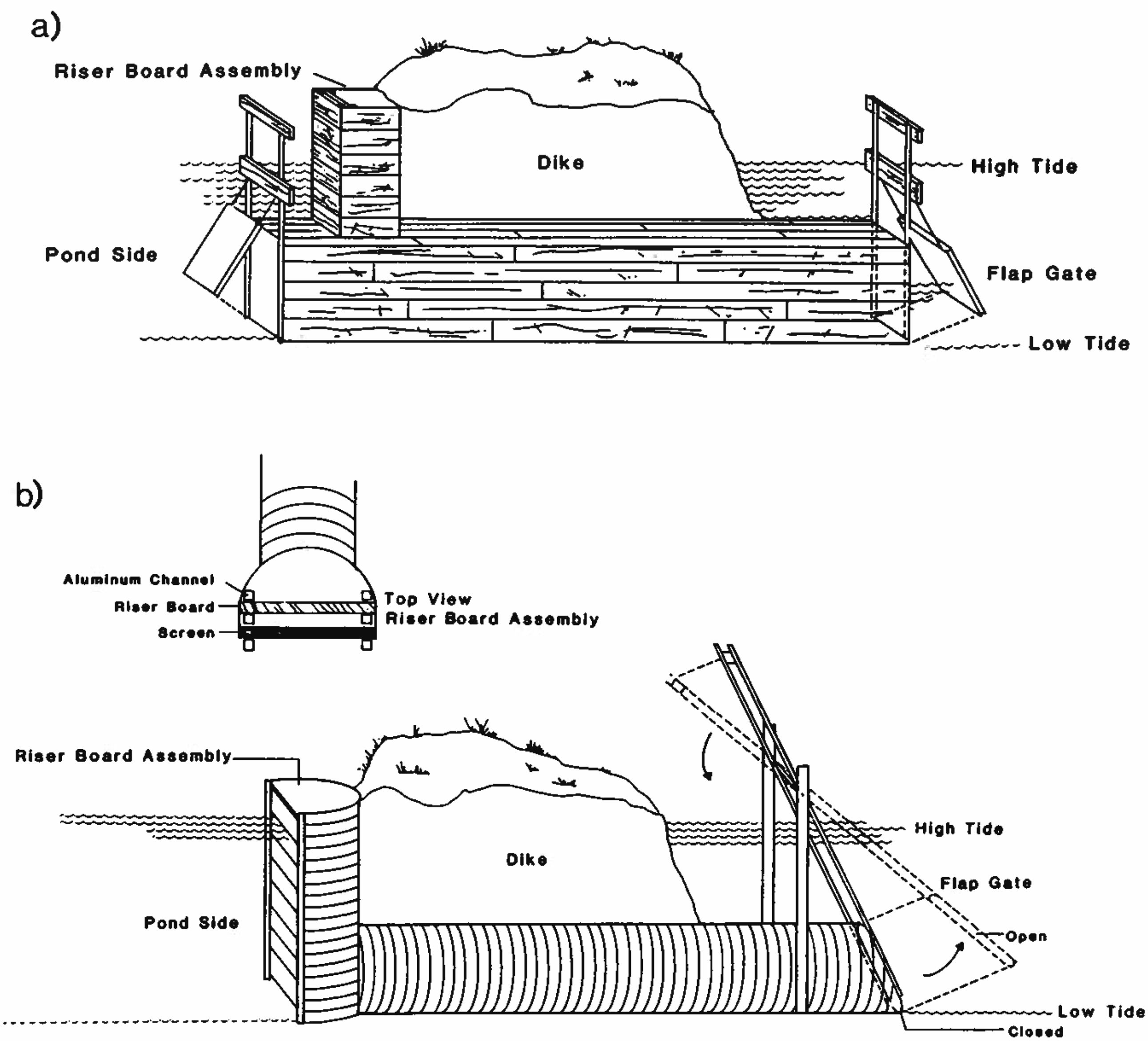


Figure 1. Two types of "Flap Gate" water control structures in Louisiana and South Carolina: a) wooden; b) aluminum.

can cause severe acidic conditions in the soil and greatly reduce the productivity of the impoundment once it is filled. In the event this is detected via pH testing of the water, the pond should go through several (4-6) rinsings and be tested again until the acidic condition, measured by pH, disappears.

Predator Control

After preparation of the pond bottom, the undesired predators that remain should be eliminated, as much as possible. This can be accomplished by:

a. pond drying,
b. gill netting and trapping,
c. treating the water with a fish toxin.

Since red drum feed on a variety of crustaceans and fishes, only the larger predators/competitors in the impoundment should be eliminated before stocking or introducing juvenile red drum. However, the financial success of extensive aquaculture is dependent upon the amount of time and money invested, and the extent of predator control should reflect this.

Predator elimination can be accomplished by the use of gill nets to thin-out the number of fish present in the impoundment. It is recommended that gill nets of mesh sizes (bar) of one, two, four, six, and eight inches be available if predators of these size ranges are known to exist in the impoundment. For example, many of the Gulf of Mexico impoundments contain a large number of garfish which are voracious predators. Gar are typically harvested using six to eight inch gill nets. Adult red drum, speckled trout, tarpon, and ladyfish should also be harvested, to eliminate their potential impact on stocked redfish. Most of these large fish can be caught using mesh sizes of two and/or four inches, depending on the size of the fish present in the impoundment. One inch gill nets can be used to eliminate potential predators and competitors of the juvenile red drum to be stocked, such as small croaker, spot, ladyfish, tarpon, black drum, sheepshead, pinfish, and other common inhabitants of coastal marshes. Usually extensive aquaculture ventures are highly visible to the public and sometimes controversial. Therefore, all gill nets should be operated within the law and the proper licenses possessed by the operators.

The operator may choose not to use mechanical predator control because it is expensive to purchase some of the gear and time consuming to set the nets in a fashion that would be effective. An alternative way of controlling fish predators in an impoundment is the use of an icthyocide (fish toxin). Rotenone is one of the most common chemicals used to control fish. It is used in its original root form (derris root) in many third world countries for the control and/or harvest of fish. It is easiest to use in the commercially available liquid forms (see Appendix 1 for sources and techniques). Crabs are also potential predators and can be controlled with a regular trapping program. Be sure to contact local regulatory agencies about the laws affecting the use of rotenone or other predator control devices.

Filling

Following pond preparation, the pond should be filled prior to stocking. Depending on the laws that are in effect in the state of operation, screens can be placed across the path of the water entering into the impoundment to eliminate undesired organisms. Screen sizes should be approximately 1/2-3/4 inches to allow the ingress of desired feed organisms, such as juvenile crabs, other crustaceans and small fish. Following filling, the pond should remain stagnate for five days prior to stocking to allow the system to stabilize and develop a good stand of plankton.

Stocking

Stocking a pond for extensive culture can be accomplished either by allowing the ingress of wild fingerlings or by collecting juvenile red drum in January and February (as the late stages of larvae and early stages of juveniles migrate in to the estuaries). Red drum can also be acquired from various hatcheries and stocked at low densities. It is recommended that the operator identify a local (or regional) hatchery for stocking fingerlings since the acquisition of wild fingerlings from the area will probably lead to conflict with local fishing groups. The use of red drum fingerlings from other regions (e.g. Gulf of Mexico fish in South Carolina) is not recommended due to the existing uncertainty of stock difference between red drum from different regions. Methods of producing fingerlings are presented in other parts of this manual.

The stocking density used in an extensive system will depend on the amount of predator control that has been used, the availability of natural food in the system, and anticipated grow-out period. It is generally accepted that a natural impoundment can support 200 to 300 pounds per acre of marine organisms (crabs, shrimp, and fish). If predator control has been effective, and the bulk of large predators have been removed, then the majority of this 200 to 300 pounds per acre could be redfish and stocking densities adjusted appropriately. For example, if predator control has been fairly thorough, an expected harvest of 300 pounds per acre of redfish is reasonable. If the target size redfish is 3 pounds, then the fish should be stocked at 100 to 150 redfish per acre. Without predator control, the biomass of other species will be higher, and stocking densities of 50 to 100 fish per acre are recommended.

Depending on the salinity and ionic content of the water in which the redfish are to be grown, fish can be stocked at different sizes. For example, if the impoundment has relatively high salinity water in (22 to 25 parts per thousand), then 5-10 days old larvae could be stocked directly into the impoundment. This is advantageous because larval redfish should cost considerably less than juveniles. Larvae would have a chance of survival because extensive pond preparation practices induce a rich zooplankton bloom, enhancing prey items for red drum larvae. However, survival is likely to be low (<2 percent) due to predators that are abundant in this type of system. Survival can be optimized by using good predator control, well screened water, and if the system meets the criteria described in the manual section on Pond Preparation of fingerlings by Colura.

Stocking red drum into lower salinity (1-15 parts per thousand) and/or freshwater impoundments requires the acclimation and stocking of juveniles (30-50 days old). Acclaimation is the process by which the fish acclimate as the ionic content, salinity, and temperature of the water are slowly changed. It is usually advisable to change the salinity and temperature of the water in which fingerlings are transported at a rate of 1 part per thousand and 1 °C, respectively, per hour by slowly adding pond water.

Growout and Pond Monitoring

Extensive aquaculture involves fairly large impoundment that the operator usually takes little or no control over. In most extensive systems, there is very little that can be done during grow out to change the course of events. However, through the process of pond monitoring, the operator can learn more about the system, including some of the warning signs that exist in impoundments, and the identification of problems once they have occurred. There is nothing worse than having a fish kill and not being able to identify the cause. It is difficult or often infeasible to correct, however, problems such as low dissolved oxygen kills, or disease within large extensive ponds. A general rule of thumb is if things look hopeless, and there are no other alternatives, then harvest and get what you can.

In a large extensive aquaculture system, it is best to set the water control structures so there is a maximum water exchange rate at all times. (However, if larvae are stocked, it is wise to hold the water stagnant for 2-3 weeks, depending on water quality.) The operator should establish and commit himself to a schedule for visiting and monitoring the pond periodically during the growout period. It should include both daytime and nighttime visits and visits at various stages of the tide.

The following equipment for monitoring water quality would be useful in interpreting causes of biological responses such as rapid growth, stress behavior, and mortality within the impoundment:

- dissolved oxygen meter ($800)
- thermometer ($10)
- water quality test kit ($100) (e.g pH, sulfides, ammonia)
- Secchi disk

The operator could easily track several variables while periodically visiting the pond. Data which are kept over a two- or three-year period can be used to make judgements about the system's ability to support a higher density of redfish, whether to reduce stocking density, the need for supplemental feed to increase production, or whether to stock at a different time of the year. It also provides valuable insight into future site selection. Unfortunately, water quality information is useful primarily for long term decision making, because short term management actions have little influence on immediate conditions within an extensive impoundment.

The following discussion presents various physical and chemical variables that can be measured by casual visits to the pond, and includes the equipment required and the value of what is measured.

Oxygen

Oxygen is required for respiration of virtually all organisms within the pond (with the exception of anaerobic bacteria in the sediment).The level of dissolved oxygen is a balance between photosynthetic activity of submerged plants, atmospheric diffusion, and oxygen consumption by organisms and sediments within the pond. For a thorough explanation of oxygen dynamics, refer to Boyd (1979).

Typically, a die-off caused by below level of dissolved oxygen die- off involves the following. Oxygen in coastal impoundments will usually vary from a high of 7 to 8 parts per million (ppm) in the daytime, during active photosynthetic activity, to a low of 2 to 3 ppm at nighttime due to organism respiration. The cycle is fairly predictable under normal conditions and has been measured in many coastal impoundments and ponds along the southern United States. The system may be thrown out of balance if oxygen production or consumption are altered by cloudy weather, a sudden increase in organisms, or lack of water exchange. For example, several cloudy days may reduce the photosynthetic activity of plant life or phytoplankton within the pond, thereby reducing the oxygen levels to early morning lows of 0-2 ppm. Once dissolved oxygen goes below 2 ppm, organisms (including fish) become stressed, and some level of mortality may result. In severe cases, the plankton community can also collapse.

A collapse of phytoplankton can be detected by a sudden clearing of the water, which had previously been turbid or "dirty." Following the crash (die-off), the free swimming and suspended organisms settle out of the water column, and begin to decay. The decay process results in an increase in bacterial activity. The large numbers of bacteria consume most of the oxygen. Two to three days after a die off, fish may be seen swimming on the surface of the pond in an erratic fashion, apparently trying to gasp for air, and crabs might be observed crawling out of the water onto the bank, particularly in the early morning hours. This can go on for several days and is a good indicator that a fish kill is eminent. It also demonstrates that organisms are capable of surviving for short periods of time during low dissolved oxygen. Typically, large fish die first (i.e. they are most sensitive to low D.O.), then large crustaceans and small fish; finally, grass shrimp and mud minnows are the last to go.

In irregular shaped systems, localized D.O. kills may occur but do not necessarily indicate the whole system is lost. In some cases this problem will spread like a disease to other parts of the system as the dead organisms fuel the reductions of D.O. in adjacent waters.

Oxygen is measured using a dissolved oxygen meter. There are several simple-to-use models of dissolved oxygen meters available for approximately $800.00. Although purchase of a dissolved oxygen meter is recommended, it is not necessary that the extensive aquaculturist have one for it to be a successful venture. However, after tracking dissolved oxygen levels for several years and by associating D.O. fluctuations with loss or near loss events, the aquaculturist will learn to associate the color of the water, the time of day, and the habits of organisms to oxygen problems that may be on the horizon.

Turbidity

Turbidity of the water in an impoundment is a good indicator of either high wind activities suspending flocculent material up from the bottom or, more importantly, an indicator of the abundance of zooplankton and plankton in the water column.

Turbidity is typically measured with a Secchi disk,which can be purchased from a variety of scientific supply houses. However, I prefer to use a homemade Secchi disk made from the bottom of a Clorox bottle tied to a piece of string with a weight on the bottom. With a waterproof marker, place a big cross on the upper surface of the plate. Using a centimeter ruler, make marks on the string attached to the plate at 5 centimeter intervals out to 50 centimeters. Lower the Secchi disk into the water and make note of the depth at which the Secchi disk disappears from sight.

The depth at which the disk disappears out of sight expressed in centimeters, is a non-linear function of the density of organisms and detritus in the water. An abundant zooplankton population may be indicated by a brown coloration against the white background of the Secchi disk. High densities of phytoplankton are indicated by a green or golden-brown, depending upon species and physiological state.

Secchi disk readings in a healthy pond range from 25-35 centimeters in the summer time. In the winter time when biological activity is reduced, Secchi disk readings of 50-60 centimeters may be common. If the Secchi disk reading becomes less than 15 centimeters, the density of organisms is dangerously high, and the system is out of balance. If low secchi readings are detected, the system is either unproductive or overstocked.

Temperature

Temperature is important for several reasons and can be measured with a simple household thermometer. For example, prior to impoundment use, the operator should determine the maximum summertime temperatures and minimum winter time temperatures to determine if red drum will survive throughout the planned grow out period. Temperature should be recorded as frequently as possible so it can be correlated with resultant growth rates. Temperature stratification is another prudent concern by midsummer; if the upper portion of the water column is 3 to 4 degrees (C) warmer than the bottom, then a physical boundary exists through which little oxygen will pass. Therefore, oxygen at the surface could be high; yet, because the deeper water is cooler, oxygen exchange may be restricted, and low oxygen conditions on the bottom may thus result. This can lead to a fish kill similar to the previous description.

Growth rate

The most important biological data to collect during extensive culture are the growth rates of the fish. Reduction or cessation of growth rates indicates that the system may be over stocked,(i.e. there are too many fish for the available food) or that water quality problems exist. Fish can be sampled by cast net, trawl, hook and line, or gill net. Comprehensive sampling of wandering species, like reddrum, is very difficult. The operator will learn to associate catch rates with certain densities through experience. The operator should try to catch 15-20 fish each month from representative areas within the impoundment. The length and weight of these fish should be measured and the fish returned to the system. This is best accomplished with hook and line sampling. If the rate of growth is significantly lower than that depicted in Figure 1 of the following paper by Hopkins, then the operator should reduce the density of the fish or add supplemental feeding to increase growth rates.

Harvest

As the end of grow out nears, the operator should work up a harvest schedule. Gill nets will probably be the harvest technique of choice. Gill nets should be purchased and/or repaired if necessary. Fish boxes should be purchased for packing the redfish as they are harvested, and plans should be made with a local fish house for the storage and/or sale of the redfish.

It is wise to follow the price structure of the market as there may bea better time of year to harvest the fish than that which is most convenient for the operator. The potential for increased price should be considered along with the increased risk associated with possibly exposing the fish to extremely low temperatures or otherwise poor water quality.

In impoundments gill nets are more effective if used during the night and in highly turbid water. Since red drum are a schooling species, schools of red drum should be located in the impoundment, and, once identified, these areas should be fished with gill nets until the operator feels that harvest is nearly complete.

If harvest is planned for mid to late summer, gill nets should be fished frequently because the warm water will quickly cause a reduction in fish quality. However, if harvest is scheduled for winter, when water temperatures are cool, the net can be left out longer. Careful and common sense fish handling practices should be used at all times to assure top dollar for the product. The fish should be carefully removed from the gill net to avoid injury. Following removal from the gill net, the fish should be laid in a standard fish box on a bed of ice and packed in a way acceptable to the buyer.

Some buyers prefer a headed, gutted product. Other buyers will accept a whole fish ungutted. The operator should determine ahead of time whether additional processing, such as gutting, heading, scaling, or filleting, is economically advantageous since these steps require time, space, and usually more money.

Summary

The culture of red drum offers a new and exciting alternative for the use of coastal impoundments and

some fresh water ponds in the United States. The growing popularity of red drum along with an increase in its price have further increased the aquaculturist's interest in culturing redfish.

It is important to understand that extensive aquaculture is in essence a backyard, low management operation that will produce only a small number of red drum per acre, yet can be very labor intensive. However, it can provide income from existing underutilized coastal impoundments.

It is critical that anyone interested in using coastal impoundments for aquaculture be certain that every aspect of the intended operation is permitted by law. The operator should contact the appropriate government agencies concerning coastal zone management laws. Constraints on land use, net use, introduction of exotic species, water column use, water bottom ownership, etc. are regulated by governing agencies.

Extensive aquaculturists can expect to make mistakes due to the unpredictable nature of extensive culture in impoundments. However, a good operator will learn from his mistakes. In addition, an extensive aquaculturist should not depend upon extensive culture as his only source of income.

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Appendix

Rotenone

Rotenone is a common icthyocide used at final concentrations ranging from 0.1 to 5 parts per million to kill fish in both fresh and salt water to kill fish. Rotenone interferes with the movement of oxygen across the gills of the fish, in affect causing the fish to suffocate; it is not a poison. Rotenone comes in both powdered and liquid forms, and is available from a variety of companies in the United States, such as:

Pennick Corporation, Lindhurst, N.J.

Prentice Drug and Chemical Company, Inc., New York, N.Y.

Tifa Limited, Millington, N.J.

Fairfield American Corporation, N.J.

Wool Faulk Chemical Works, Inc., Fort Valley, Ga.

The liquid form of rotenone is available in concentrations of 2+ and 5 percent concentrations and can be diluted with water for application. The recommended dosage of 5 percent rotenone is apporximately one gallon per three acre feet of water (one acre-foot is one acre of water one foot deep). Higher dosages are required for high salinity water. The powdered form of rotenone is relatively difficult to use because it does not easily dissolve in water, hence avoid using the powdered form if possible.

Cost of treatment is reduced if as much water as possible is drained from the impoundment. It is particularly critical to treat the deep portions where predatory fish congregate when the water level is lowered.

The liquid form of rotenone can be distributed as a half and half mixture with water in a garden sprayer. It can be sprayed on the surface of the water and/or mixed in the propeller wash of an engine or electric motor. Since it is supplied in an oil base, rotenone will tend to float unless the water is agitated by wind, fish, or mechanical agitation.

It is very important that the fish killed with rotenone not be eaten. Although slight exposure to rotenone by man is not lethal, it does cause a numbing of the lips and moist membranes of the inner part of the nose and mouth if the fumes or spray are inhaled.

Depending on the state of operation, a permit may be required for the use of rotenone, so it is wise to inquire about application guidelines from the supplier or state agency in your area.

Extreme care should be used when distributing retonone so as not to introduce it into the open marsh or riverine system. The treatment should be totally contained in the impoundment. Following treatment with rotenone, all water exchange should be stopped and the treated water held in the impoundment for at least five days. Rotenone is convenient to use because it will photo-oxidize after two to three days in the water (depending on the concentration). Rotenone can also be neutralized using potassium permanganate at a dosage of 1 mg per liter for each .05 mg per liter of rotenone (1 mg per liter = 1 ppm). The effect of potassium permanganate is immediate in eliminating the effect of rotenone.

Caution! Both rotenone and potassium permanganate are highly irritating and possibly toxic. Proper clothing should be worn when using them as a treatment.

References

Allan, P.F. 1950. Ecological bases for land use planning in Gulf coast marshlands. **J.Soil and Water Conserv.** 5:57-60.

Arnold, C.R., J.D. Williams, A. Johnson, W.H. Bailey and J.L. Laswell. 1977. Laboratory spawning and larval rearing of red drum and southern flounder. **Proc. Annu. Conf. Southeast. Assn Fish Widl. Agen.** 31:437-440.

Baldwin, W.P. 1968. Impoundments for waterfowl on South Atlantic and Gulf coast marshes. In: J.D. Newson, ed. **Proc. Marsh and Est. Mgt. Symp**, LSU: 127-133.

Bass, R.J., and J.W. Avault, Jr. 1975. Food habits, length-weight relationship, condition factor, and growth of juvenile red drum, *Scianeops ocellata*, in **Louisiana Trans. Amer. Fish Soc.** 104:35-45.

Baughman, J.L. 1947. The loss of fish due to freeze. Tex. Parks Wildl. Dept., **Texas Game and Fish Magazine**. March 1947. 3p.

Bearden, Charles M. 1967. salt-water Impoundments for Game Fish in South Carolina. **Prog. Fish. Cult.** (29):123-127.

Bok, A.H. 1984. Extensive culture of two mullet species in freshwater impundments in the eastern Cape. **S. Afr. J. Zool.** 19:31-36.

Boothby, R.N., and J.W. Avault, Jr. 1971. Food habits, length-weight relationship and condition factor of the red drum (*Scianeops ocellata*) in southeastern Louisiana. **Trans. Amer. Fish. Soc.** 100(2):290-295.

Boyd, C.E. 1979. Water quality in warm water fish ponds. Alabama Agricultural Experiment Station, Auburn University.

Broom, J.G. 1969. Pond culture of shrimp on Grand Terre Island, La., 1962-1968. **Proc. 21st Annu. Session Gulf Caribb. Fish. Inst.** 1968:137-151.

Chabreck, R.H. 1960. Coastal marsh impoundments for ducks in Louisiana. **Proc. Southeaster Assoc. Game and Fish Commissioners.** 14:24-29.

_______1968. Weirs, plugs, and artificial potholes for the management of wildlife in coastal marshes. In: J.D. Newson, ed. **Proc. Marsh Estuary Mgt. Symp.** Div. Cont. Educ., LSU:178-192.

_______and C.M. Hoffpauir. 1965. The rise of weirs in coastal marsh management in **La. Proc. Annu. Conf. Southeast. Assn. Game Fish Comm.** 16:103-112.

Cook, H.L., U. Pongsuwana, L.D. Rajamanickam and R. Khamis. 1984. Experience in establishing vegetation on acid sulphate pond dikes. In: **Malaysia Coast Aquaculture Development**, FAO of the U.N. 215-224 p.

_______and U. Pongsuwana, and S. Wechasite. 1984. Recommendations for construction and management of brackish-water aquaculture ponds in areas with acid sulphate soil. In: **Malaysia Coastal Aquaculture Development**, FAO of the U.N. 243-260 p.

_______and A.A. Rahman, L.D. Rajamanickan and M.Yasin. 1984. Use of screens and boards in water management for tidal ponds. In: **Malaysia Coastal Aquaculture Development**, FAO of the U.N. 183-194 p.

Crocker, P.A., C.R. Arnold, J.A. DeBoer, and J.D. Holt. 1981. Preliminary Evaluation of Survival and Growth of Juvenile Red Drum (*Scianops ocellata*) in Fresh and Salt Water. **J. World Maricult. Soc.** 12(1):122-134.

Davies, W.D. and W.L. Shelton. 1983. Sampling with Toxicants. **Fisheries Techniques.** 199-213 p.

Dawson, U.K., P.D. Harman, D.P. Schult, and J.L. Allen. 1983. Rapid Method for Measuring Rotenone in Water at Piscicidal Concentrations. **Trans. of Am. Fish. Soc.** 112:725-727.

Dean, J.M. 1972. **The Use of Old Rice Fields for Aquaculture.** Technical Report #2, Belle W. Baruch Coastal Research Institute.

de los Santos, C., Jr. 1978. **Modern Aquaculture for the Phillipines.** Yuhum La Defensa Press, Iloilo City, Phillippines. 224 p.

Gedney, R.H., Y.C. Shang, and H.L. Cook. 1983. **Comparative study of tidal and pumped water supply for brackishwater aquaculture ponds in Malaysia.** South China Sea Fish. Dev. and Coord. Programme. 40 p.

Green, A.W., H.R. Osburn, G.C. Matlack, and H.E. Hegen. 1985. Estimated survival rates for immature red drum in Northwest Gulf of Mexico bays. **Fish. Res.** 31:263-277.

Gulf States Marine Fisheries Comm. 1980. Fishery profiles of red drum and spotted seatrout. No. 6, Ocean Springs, Miss.

Gunter, G. and H.H. Hildebrand. 1951. Destruction of the fishes and other organisms on the South Texas coast by the cold wave of Jan. 28-Feb. 3, 1951. **Ecology** 32:731-735.

Gunter, G. and W.E. Shell, Jr. 1958. A study of an estuarine area with water-level control in the Louisiana marsh. **Proc. La. Acad. Sc.** 21:5-34.

Hall, G.E. 1974. Sampling reservoir fish populations with rotenone. In: R. Welcomme, ed. **Symposium on the methodology for the survey, monitoring and appraisal of fishery resources in lakes and large rivers.** 249-259 p.

Herke, W.H. 1971. Use of natural and semi-impounded Louisiana tidal marshes as nurseries for fishes and crustaceans. Ph.D. Dissertation, LSU. 264 p.

Herke, W. H., E. E. Knudsen, B. D. Rogers, and V. L. Pringer. 1985. Effects of a fixed-crest water-control structure on the abundance of the fish and crustaceans migrating from a shallow marsh nursery toward the Gulf of Mexico. **Estuaries** 8(2A):21A.

Leavell, C. 1971. **Legal aspects of ownership and rise of estuarine areas in Georgia and South Carolina.** Inst. of Gov., Univ. of Georgia, Athens, 111 p.

Lennon, R. E., J. P. Hunn, R. A. Schnick, and R. M. Burress. 1970. **Reclamation of ponds, lakes and streams with fish toxicants: a review.** FAO of the U. N., Fish. Tech. Pap. No. 100:1-99 p.

Luebke, D. A. 1973. The culture of some marine fishes in ponds receiving heated discharge water from a power plant. M. S. Thesis, Texas A&M University. 213 p.

Luebke, Richard W. and Kirk Strawn. 1973. The growth, survival, and feeding behavior of redfish (*Sciaenops ocellata*) in ponds receiving heated discharge water from a power plant. World Mariculture Society Workshop. 143-153 p.

Lunz, G. Robert. 1956. Harvest from an Experimental One-acre Salt-water Pond at Bears Bluff Laboratories, South Carolina. **Prog. Fish. Cult.** 18:92-94 p.

Lunz, G. R. 1958. Pond cultivation of shrimp in South Carolina. **Proc. 10th Annu. Sess. Gulf Caribb. Fish. Inst.** 1957:44-48 p.

Mendelssohn, I. A. and E. D. Seneca. 1980. The influence of soil drainage on the growth of salt-marsh cordgrass *Spartina alterniflora* in North Carolina. **Estuarine and Coastal Marine Science.** II:27-40 p.

Mercer, L. P. 1984. **A biological and fisheries profile of red drum, *Sciaenops ocellatus*.** Special Scientific Report #41. North Carolina Dept. of Nat. Res. and Comm. Dev., Div. Mar. Fish.

Morgan, P. H. 1974. A study of tidelands and impoundments within a three-river delta system - the South Edisto, Ashepoo, and Combahee rivers of South Carolina. M. S. Thesis, University of Ga. 92 p.

Neely, W. W. 1960. Managing *Scirpus solustus* for ducks.

Proc. Southeast. Assoc. Game and Fish Commissioners. 14:30-34 p.

_______ 1968. Planting, disking, mowing, and graining. Pages 212-221 p. In: J. D. Newson (ed.). **Proc. Marsh Estuary Mgt. Symp.** LSU, Baton Rouge.

_______ and M. Fanning. 1973. Comparison of the productivity of *Spartina alterniflora* and *Spartina cynosuroides*-in Georgia coastal marshes. **Bull. Georgia Acad. Sci.** 31(1):1-12 p.

Overstreet, R. M. 1983. Aspects of the biology of the red drum. *Sciaenops ocellatus*, in Mississippi. **Gulf Research Reports**, Supplement 1, 45-68 p.

Perret, W. S., J. E. Weaver, R. C. Williams, F. L. Johason, T. D. McIlwain, R. C. Raulerson and W. M. Tatum. 1980. Fishery profiles of red drum and spotted seatrout. Gulf States Mar. Fish. Comm., Ocean Springs, MS. No. 6, 60 p.

Perry, Guthrie W., Jay V. Huner, and James V. Avault, Jr. Culture Studies with Malaysian Prawn in Unfed Brackish Water Ponds. **Proc. Ann. Conf. S. E. Assoc. Fish and Wildl. Agencies.** 34:215-222 p.

Perry. W. G. 1981. Seasonal abundance and distribution of brown and white shrimp in a semi-impounded La. coastal marsh. **Proc. La. Acad. Sci.** 44:102-111 p.

Post, G. 1958. Time versus water temperature in rotenone dissipation. **Annual proceedings of the Western Association of Game and Fish Commissioners.** 38:279-284 p.

Provost, Michael B., et al. Production and Utilization of Waterfowl Foods in Brackish Water Impoundments in South Carolina. **Proc. Ann. S. E. Assoc. Fish and Wildl. Agencies.** (32):60-70 p.

Reeves, C. E. Graham. 1983. Management of Brackish Water Impoundments in the Santee River Delta. Private Document.

Rogers, B. D., W. H. Herke, and E. E. Knudsen. 1987. **Investigation of a Weir-design Alternative for Coastal Fisheries Benefits.** La. Coop. Fish Wildl. Res. Unit. 98 p.

Rose, C. D., A. H. Harris, and B. Wilson. 1975. Extensive culture of penaeid shrimp in Louisiana salt-marsh impoundments. **Trans. Amer. Fish. Soc.** Vol. 104(2):296-307 p.

Schnick, R. A., F. P. Meyer, and D. L. Oray. **A guide to approved chemicals in fish production and fishery resource management.** Univ. of Arkansas Coop. Ext. Serv. 24 p.

_______ and F. P. Meyer. 1978. **Investigations in Fish Control: Registration of Thirty-three Fishery Chemicals: Status of Research and Estimated Costs of Required Contract Studies.** U. S. Fish and Wildl. Serv. 86 p.

Shiriman, J. V., D. E. Colle, and D. F. DuRant. 1981. Efficiency of rotenone sampling with large and small block nets in vegetated and open-water habitats. **Trans. Am. Fish. Soc.** 110:77-80 p.

Simmons, E. G. 1962. The effects of a hard freeze on marine life. Texas Parks and Wildlife Dept., **Texas Game and Fish Magazine.** April, 1962. 2 p.

Simpson, H. James, Hugh Ducklow, Bruce Deck, and Harry Cook. 1983. Brackish-water Aquaculture in Pyrite-Bearing Tropical Soils. **Aquaculture,** 34:333-350 p.

Theiling, Dale L. and Harold A. Loyacano, Jr. 1976. Age and Growth of Red Drum from a Saltwater Marsh Impoundment in South Carolina. **TAFS.** 105(1):41-44 p.

Trimble, W. C. 1979. Yield trails for red drum in Crawfish-water ponds, 1976-1977. **Proc. Annu. Conf. Southeast Assoc. Fish Wildl. Agencies.** 33:432-441 p.

Wechasitt, S. 1984. Cleaning of interior canals of brackish-water ponds. In: **Malaysia Coastal Aquaculture Development,** FAO of the U. N. 225-232 p.

Yokel, B. J. 1966. A contribution to the biology and distribution of the red drum, *Sciaenops occelatus*. M. S. Thesis, University of Miami, Coral Gables. 160 p.

_______ 1980. A contribution to the biology and distribution of the red drum, *Sciaenops occelata*. (Abstract). In: **Colloquium on the biology and management of red drum and seatrout.** Gulf States Marine Fisheries Comm., No. 5:7-8 p.

Characteristics of a Freshwater Red Drum Sport Fishery

Richard W.Luebke
Heart of the Hills Fisheries Research Station
Junction Star Route, Box 62
Ingram, Texas 78025

Abstract

Following low-density stockings in 1976 and 1980, fingerling red drum (*Sciaenopsocellatus*) were stocked annually from 1981 through 1986 at 231- 1,115/hectare (93-451/acre) to enhance the sport fishery in Victor Braunig Reservoir, a 546-hectare (1,350-acre) heated reservoir near San Antonio, Tex. Access point creel surveys were conducted from March through November, 1982 through 1986 to monitor the red drum sport fishery. The portion of the overall fishing pressure attributable to red drum anglers ranged from 17.5 percent in 1982 to 41.8 percent in 1985. Fishing pressure directed toward red drum increased dramatically from 57.92 hours/hectare (23.44 hours/acre) in 1982 to 387.57 hours/hectare (156.85 hours/acre) in 1986.

Total harvest of red drum also increased from 7.74 kg/hectare (6.91 pounds/acre) in 1982 to 96.52 kg/hectare (86.18 pounds/acre) in 1986. Harvest rates for anglers seeking red drum ranged from 0.050 to 0.140 red drum/hour. Mean total length and weight of harvested red drum ranged from 404 mm (15.9 inches) and 0.71 kg (1.57 pounds) in 1982 to 730 mm (28.7 inches) and 4.21 kg (9.28 pounds) in 1985. During the five years creel surveys were conducted, anglers harvested an estimated 45,718 red drum that weighed 121,822 kg (268,567 pounds).

Fee Fishing Potential for Red Drum

Billy Higginbotham
Wildlife andFisheries Specialist
Cooperative Extension Program
Overton, Texas 75684-0038

The commercial production of red drum is still in its infancy. As a result, the utilization of this recreationally and commercially valuable species in a fee fishing enterprise is largely unexplored. No known fee fishing ponds for red drum currently exist; however, as technology develops for large scale production, fee fishing may be one alternative marketing technique. Potential advantages for utilizing red drum in fee fishing include its value as a highly regarded food fish, high nutritional value and tolerance for waters containing a wide range of salinity. Red drum demonstrate similarities to catfish for use in fee fishing since they appear able to adapt to crowding and readily accept a pelleted feed.

The economic importance of the recreational red drum fishery in Texas was demonstrated by the ban on commercial fishing for this species recommended by Texas Parks and Wildlife in 1981. Successful introductions in freshwater impoundments have created a substantial interest among many anglers who were previously unfamiliar with the species. As a result, the demand for red drum continues to exceed the current available supply.

The potential for developing a red drum fee fishing enterprise is possible on an extensive (low density), semi-intensive, or intensive (high density) basis. The extensive or semi-intensive basis is normally employed where sportfish species are stocked in existing ponds and lakes in conjunction with forage species.

This may mean a lower level of business since fewer of the target species are usually present. Higher entry fees are usually charged since management is aimed at fewer but larger (trophy) fish. These techniques can be utilized in existing lakes, but to ensure overwintering of red drum, minimum temperature requirements of the species must be met. Intensive management potentially offers the most profitable method of operating a fee fishing enterprise. However, since little information exists on how red drum react to angling pressure in small ponds (typical of intensive operations), their "catchability" as compared to traditional fee fishing species such as rainbow trout or channel catfish remains unknown. This question should certainly be addressed in commercial production ponds prior to establishing fee fishing enterprises. Nevertheless, techniques primarily aimed at intensive management are discussed in this chapter since this method appears to have the most profit potential at the present time.

Culture and Maintenance Requirements

Since detailed red drum production and maintenance requirements are discussed elsewhere in these proceedings, only a brief review of factors affecting the red drum in a fee fishing enterprise will be outlined.

The potential red drum fee fishing enterprise must meet the needs of the fish in order to maintain health; stressed or moribund fish do not bite well. The requirements for red drum maintenance in a fee fishing pond are no less stringent than those necessary for intensive production techniques in culture ponds.

Water quality is of utmost importance, especially if freshwater ponds are utilized since red drum cannot survive in low ionic strength freshwater. In order to maintain the requirements of the fish, water hardness should be at least 100 ppm and chlorides at least 150 ppm (for further information on environmental requirements, refer to Bill Neill's paper in the previous section). Ponds filled with water from a source that does not meet these requirements can be corrected using agricultural limestone to increase water hardness and agricultural grade salt to increase the chloride concentration present. While the cost of these additives for water quality management are debatable, both have proven cost effective in the commercial catfish industry. Saltwater ponds can also be utilized to maintain red drum. If necessary, a mixture of salt and freshwater can be flowed into a pond to provide the proper hardness and chloride concentrations.

Water quantity is also important for maintaining high densities of red drum. A minimum of 50 gallons per minute per surface acre of pond is necessary for water exchange and to compensate for evaporation and seepage.

Water temperature is another important water quality parameter for consideration. Red drum are incapable of surviving prolonged water temperatures below 8-10 C (further background provided in Environmental Requirements of Red Drum) and feeding (important in a fee fishing operation) also ceases at temperatures around 10 C. Because of this species' cold intolerance, fee fish enterprises will be limited to the warmer months due to potential winterkill, or to those geographical areas where overwintering is possible due to mild winters or the availability of a warm water source.

Oxygen content becomes particularly critical for intensive operations where 4,000 pounds (or more) per surface acre are maintained during the warm months. Emergency aeration equipment, (ie. paddlewheel agitators) may become necessary to maintain oxygen content above the critical 3 ppm level.

Fee fishing operators, like aquaculturists, must become familiar with analysis procedures to monitor levels of chlorides, hardness, oxygen and temperature. Water testing should become a routine part of the fee fishing operator's management plan. The ability to recognize fish health problems due to inferior water quality, diseases and parasites is essential in order to minimize fish losses.

As previously discussed, one advantage that red drum demonstrate as a potential species for fee fishing is its acceptance of pelleted feed. The fish will require a maintenance diet to remain healthy in the fee fishing pond. For example, in most successful catfish fee fishing ponds, fish are fed a maintenance diet of 1 percent body weight at optimum water temperatures. Few operators feed fish at a level to facilitate weight gain, but rather supply just enough ration once or twice per week to maintain their fish in good condition. For red drum, the same rations fed in grow out ponds should be utilized in fee fishing ponds. The operator should plan feedings well in advance of (or following) high use times by clientele (ie. weekends) to be sure the fish will bite readily.

Fee Fishing Pond Design

Successful fee fishing ponds must contain many of the same design features utilized in commercial growout ponds. A soil type containing suitable clay content to prevent excessive seepage is one important parameter in selecting a site. In addition, the topography of the site will affect construction costs. The ponds must also be constructed in conjunction with a water source capable of delivering the necessary water quantity and quality to meet the requirements of the species.

All fee fishing ponds should be constructed to allow complete pond draining via a bottom drainpipe. To facilitate gravity flow draining, the land immediately outside the pond levee should be slightly lower than the deepest part of the pond. Drainpipes of four to six inches in diameter are sufficient for one to two surface acre ponds.

The bottom drainpipe allows the removal of the poorest quality water present in the fee fishing pond. Unconsumed feed and metabolic wastes can be flushed from the pond on a routine basis to maintain adequate water quality. The drainpipe also allows for pond lowering to facilitate pond treatments for weed and disease control if necessary.

The pond should also be harvestable by seining. This may become particularly important in geographical areas where red drum must be removed and sold prior to the onset of cold weather to prevent winterkill losses. The pond should slope smoothly from a depth of 4 feet at the shallow end to the deep end (6 to,8 feet) where the bottom drainpipe is located.

The ideal size of a fee fishing pond is debatable. Most fee fishing ponds that are intensively managed are 0.75 to 1.50 surface acres in size. The pond should not be so large that the operator cannot monitor all angler activity. Rectangular shapes are the most popular and provide more shoreline length per surface acre (vs. square ponds) to allow each angler more individual space if desired.

Pond levees should be wide enough to allow vehicular traffic for pond stocking, harvest by seining and routine maintenance. A 3:1 levee slope toward the pond with a 2:1 slope on the outside levee is sufficient. The slope should not be too steep or the establishment of vegetative cover becomes difficult and levee erosion may result.

Establishing a Fee Fishing Enterprise

The principles for establishing a successful fee fishing operation are the same whether the target species is channel catfish, rainbow trout or in this case, red drum. The marketing of fish through fee fishing fills a niche by providing a convenient opportunity for anglers to practice their sport. The competition for access to quality fishing waters, especially near populated areas, has further increased this demand.

The most successful fee fishing operators realize that they are marketing much more than just the fish. The value of the recreational experience received by the angler far outweighs the value of the fish alone. A recreational experience consists of five basic parts: anticipation, travel to site, on-site experience, travel from the site and recollection of the overall experience. The fee fishing enterprise should satisfy this experience to the greatest extent possible.

Although the value of the fish alone does not always figure prominently in the overall recreation experience achieved, fee fishing enterprises should strive to provide consistently good catches or business will eventually fail.

The prospective fee fishing entrepreneur should conduct an informal study to determine if a need exists for the service to be offered. The immense popularity of the red drum as a sportfish may preclude the necessity of this step in some areas. Nevertheless, in most locations consumer acceptance and potential competition from other recreational service operators should be investigated. This may be accomplished by surveys of potential clientele groups as well as population trends in a potential area.

Location of the site is the most important physical aspect of a successful fee fishing enterprise. Assuming that a market for fee fishing exists, convenient access to the site is important for gaining clientele acceptance. Locations on major highways near intersections (areas of high traffic) are preferred, however, many successful fee fishing enterprises exist away from major thoroughfares as long as they have good accessibility. As previously discussed, the specific site of the enterprise must also meet the aquaculture requirements for water quality and quantity, topography and soil type necessary for pond construction.

The fee fish operator has three potential sources of fish for stocking. These include (1) spawn, hatch and raise fish on the premises, (2) purchase fingerlings and grow them out to harvestable size or (3) purchase harvestable size fish. The first two alternatives would be most feasible in conjunction with a redfish production operation. The third alternative may be more expensive in actual fish cost but requires less pond space and facilities than the other two alternatives.

Management of a Fee Fishing Enterprise

Poor management is the number one cause of failure of fee fishing enterprises. In addition to possessing aquaculture skills, the operator must possess managerial expertise and public relations skills. The entire enterprise should be operated under the premise of marketing a recreational experience and should therefore be capable of selling the pond operator as well as the facilities as a whole to clientele.

Design aspects of the fee fishing facility are also important for the successful management of the overall operation. A parking area located away from the ponds should be provided. Access from the parking area must be controllable. Landscaping, fencing and pathway designation can be used to control angler movement into and out of the property.

Access from the parking area to the ponds is usually through a "service area" or building where fees are assessed, etc. Refreshments, bait, tackle, restrooms and fish cleaning facilities should be available in the service area. In addition, troughs or vats for holding live fish should be provided for walk-in customers and anglers wishing to purchase fish to supplement their catches.

The determination of fee assessment is of paramount importance to the success of a fee fishing enterprise. The most common types of fee assessments include the annual fee, daily fee, weight of fish fee, and a combination of the daily and weight of fish fee. The latter is the most commonly utilized technique among fee fish enterprises in Texas. While the daily entry fee varies greatly, the weight of fish fee falls between the wholesale cost paid by the operator and the retail cost of the fish within the marketing area. Walk-in customers are normally charged the prevailing retail price. In addition, most pay lake operators offer processing services to their clientele, usually on a cost per pound basis.

Additional Considerations

As previously discussed, the fee fish operator must wear many different hats and the fee fishing enterprise must provide much more than just a pond with fishing available.

A study of fee fishing enterprises in Kentucky revealed that approximately 50 percent of all clientele were adult males, however, almost all of the remaining user group was made up of family units. In order to cater to these clientele, picnic areas and a playground for children can greatly enhance the overall recreational experience.

Safety of clientele is also a consideration and life saving equipment should be available at all ponds. Liability has become a major concern for all property owners, but the owner becomes particularly liable for accidents where a fee is charged for entrance as is the case in fee fishing. The fee fishing operator should take all reasonable steps necessary to avoid negligence. Liability insurance is considered essential by many fee fishing operators.

Advertisement, as with any business, can greatly enhance the success of a fee fishing enterprise. Newspaper ads, television spots and billboards can all supplement word of mouth advertising. Some operators further promote their business by tagging a few "trophy" fish in the pond and offering prizes to anyone able to catch one.

The legal aspects of operating a fee fishing enterprise are very similar to any other aquaculture operation. Permits for surface and groundwater rights are often necessary from the State in addition to county or municipal regulations. If saltwater is utilized and then discharged from the ponds, a permit from the Texas Department of Water Resources is necessary. In addition, the marketing of red drum will require a fish farming license available from the Texas Department of Agriculture. Since most fee fishing operations also sell bait and tackle, an additional license from Texas Parks and Wildlife Department is required. Furthermore, employees of the enterprise involved in the cleaning and processing of fish should obtain a state health card as well as adhere to any local regulations regarding food handling and processing.

Potential Problems

Obviously, the development of red drum fee fishing as a viable industry clearly hinges upon the success of red drum aquaculture in general. A reliable supply of healthy fish must be available within a reasonable distance of the fee fishing site. Although the basic criteria for hauling red drum are known, live hauling large poundages necessary for stocking fee fishing ponds will require additional investigation. Furthermore, the fish must be available at a cost that will allow the fee fishing enterprise to remain competitive with other recreational enterprises within a given market area.

Finally, the prospective owner of a red drum fee fishing enterprise should be aware that no privately owned operation of this type currently exists in the United States. While this may suggest an available market, success or failure of the venture depends upon one's ability to meet the needs of the fish and the needs of prospective clientele interested in a quality recreational experience.

Recommended Procedures

1. Verify "catchability" of red drum in commercial production ponds prior to establishment of fee fishing enterprises.
2. Perform market study to determine consumer acceptance of red drum and to identify potential competition from other recreational enterprises.
3. Select site suitable for both aquaculture (water quality and quantity, topography and soil type) and access by clientele.
4. Apply for appropriate permits from state and local authorities regarding water use and discharge permits, license requirements, etc.
5. Select appropriate pond design and management methods (water quality management, feeding procedures, etc.)
6. Design overall facility layout for handling flow of customers. Include a service area design to provide room for holding and processing fish, sale of bait, tackle, refreshments, etc.
7. Choose source for stocking fish.
8. Assure liability coverage.
9. Advertise and promote facility in local media.
10. Charge based on combination of entry fee and catch. Record keeping of receipts and expenses is of utmost importance to adjust fees as necessary.

Literature

Chamberlain, George. Red Drum Aquaculture Status. **Coastal Aquaculture**, Volume III, No. 3. Corpus Christi, Texas. December 1986.

Cremer, Michael, Steven Mims and Gregory Sullivan. Pay Lakes as a Marketing Alternative for Kentucky Fish Producers. **Aquaculture and Natural Resources** Bulletin No. 8. Frankfort, Kentucky. 1984.

Glaser, Stanley. Central Texas Fish Ponds. **Proceedings of the Annual Conference of the Fish Farmers of Texas.** College Station, Texas. 1974.

Higginbotham, Billy. Current Fee Fishing Trends and Techniques in Texas. **Proc. Ann. Conf. Fish Farms of Texas.** 1987. 4 pp.

Higginbotham, Billy. Facility and Equipment Needs for Texas Fee Fishing Operations. **Proc. Ann. Conf. Fish Farms of Texas.** 1988. 4 pp.

Huner, Jay V. Fee Fishing - Look Before You Leap. **Farm Pond Harvest**, Volume 14, No. 4. Momence, Illinois. 1985.

Johnson, Pershing. Big City Fish Ponds. **Proceedings of the Annual Conference of the Fish Farmers of Texas.** College Station, Texas. 1974.

Lee, Jasper S. Operating Recreational Marketing Facilities. **Commercial Catfish Farming.** Danville, Illinois. 1973.

Watt, Carson E. Location and Design of a Commercial Fish-Out Operation. **Proceedings of the Annual Conference of the Fish Farmers of Texas.** College Station, Texas. 1974.

Semi-intensive Grow out in Saltwater

J. Stephen Hopkins
Manager
Waddell Mariculture Center
P.O. Box 809
Bluffton, South Carolina 29910

To farm significant quantities of red drum, it appears that semi- intensive culture (Trimble, 1979; Hopkins *et al.,* 1987) will be the methodology of choice for most ventures. Semi-intensive culture combines the relatively low-capital costs characteristic of extensive culture with consistent production levels high enough to base corporate-level businesses upon. Semi-intensive production techniques are more forgiving than intensive systems. The semi-intensive pond environment provides only a small amount of natural food items, but there is apparently enough to supply the essential nutrients needed to make up for deficiencies in the prepared rations. In the semi-intensive pond environment, metabolities seldom accumulate to toxic levels so water quality management is largely a matter of dissolved oxygen management.

Outline of Production Scenario

It should be noted at the outset of this paper that the hypothetical, semi-intensive, production scenario proposed is intended only as a useful point of departure. Refinement of these methods will almost certainly occur as current information gaps are filled. Selection of this scenario is based upon several premises: (1) out-of-season spawning in the spring will result in improved survival and lower operating expenses, (2) a three-pound fish is highly desirable in the market place, (3) a pond biomass of 5,000 pounds per acre is easily attained with supplemental aeration and water exchange and up to 20,000 pounds per acre has been demonstrated in small ponds, (4) a three-phase production system where fish are progressively thinned out into larger ponds will maximize the available space and provide better accountability, and (5) adequate feeds are available.

Out-of-Season Spawning

Out-of-season spawning promises to reduce semi-intensive growout operating expenses and result in increased overall survival of the fish. As noted previously in this manual, the natural spawning season for red drum is in the late summer and early fall. Also as noted, the simplest and most effective method of producing fingerlings is to stock fry into ponds with an induced zooplankton bloom. As the autumn-spawned fingerlings grow, the zooplankton concentration declines due to fish grazing and falling water temperatures. This is a critical point in the production cycle as the fish must learn to accept dry rations rapidly to compensate for the reduced concentration of natural food items. Many juveniles starve or are cannibalized during this time.

If environmental conditions for captive broodstock are manipulated to induce spawning in the spring (March for Texas and April for South Carolina), survival of juveniles may be significantly increased. Fry spawned in the spring pass through this critical development period when water temperatures are similar to those encountered in the natural spawning season, yet the availability of natural food items is increasing rather than decreasing. It is our experience that winter survival is excellent for juveniles that are weaned onto dry feeds. However, survival of small unweaned fingerlings that are the product of autumn spawnings have a survival rate of only about 50 percent through their first winter.

The other major benefit of out-of-season spawning in the semi-intensive production scenario is the lower operating costs associated with aeration and water exchange. As noted below, fish will be maintained in each of two growout phases for one year, from one spring until the next. The pond biomass increases as the year progresses. During the summer, the biomass will be relatively low. Thus, the most stressful growing conditions — the combination of high biomass and high summer water temperature with low dissolved oxygen saturation — will be avoided. Since sizing two of the capital costs, aeration equipment and pumping equipment, as well as the operational costs for aerators and pumps, are based upon the most stressful environmental conditions expected, their sizing and usage are minimized by out-of-season spawning. It is expected that the dissolved oxygen levels will be the lowest from late June through early September. Were a natural spawning season used, the pond biomass would be at least twice as large during this critical period. Supplemental aeration and water exchange requirements may be as much as twice as high under these conditions.

Three-Phase Production System

A single-phase system where fry are stocked into a pond and raised to the three-pound market size is not practical from an accountability stand point. The survival of fry to fingerlings is the most highly variable segment of the production cycle. In a single-phase system, the number of fish in a pond will not be known. This will make feeding rate calculations difficult and inaccurate. The farmer will have no accurate measure of the biomass within the pond. Consequently, the pond may end up being overstocked and produce below normal growth rates or be understocked and not effectively utilize the facility.

In order to maximize pond space with minimal handling of the stock, a three-phase, semi-intensive production system should be utilized. In Phase-I, fry will be grown to fingerling size in small ponds. Phase-II will raise fingerlings to 1 pound yearlings in ponds of 5 acres or less. Phase-III will raise the 1 pound yearlings to market size in ponds of 20 acres or less. These are the largest pond sizes deemed feasible, based upon the catfish industry experience, although it must be stressed that field tests have not yet been conducted with ponds of this size. The relative amount of acreage under water in the Phase-II and Phase-III units should be maintained at a ratio of 1:4, respectively (Table 1).

The rate of return on investment will be significantly higher using a three-phase system as opposed to a two-phase system (Rhodes *et al., 1987* [economic section]). It is possible to transfer Phase-I fingerlings directly to a two-year growout pond. However, the disadvantage of a two-phase system is that the pond space is not effectively used during the first year of production. The biomass in the pond at the end of the first year will be 1,000 to 1,500 pounds per acre in a two-phase system. A three-phase system will have about 5,000 pounds per acre at the end of one year. The additional labor and equipment needs of a three-phase system will be more than offset by the savings in capital construction costs. By using a three-phase system, a 35 percent reduction in pond acreage can be realized in the production of an equal amount of fish compared to a two-phase system. This additional acreage represents a significant investment in land and pond construction costs. Another disadvantage of the two-phase system is the build-up of organic sludge on the pond bottom. This organic sludge is an oxygen sink and may inhibit the ability of fish to feed effectively off the bottom. In a three-phase system, the pond is drained and some of the organic sludge is allowed to oxidize after each 12-month period.

For the purpose of preliminary cost analysis, two farm sizes are visualized using this scenario. One farm size would have (1) three 0.5-acre fingerling ponds, (2) six 5-acre yearling ponds and (3) six 20-acre growout ponds. The other farm size would have (1) three 0.5 acre fingerling ponds, (2) five ponds at 2.5 acres and (3) five ponds at 10 acres.

Table 1. Summary of Three-Phase Production Scenario for Red Drum Farm.

Production Phase	Phase Duration	Harvest Size	Relative Pond Size
I (fingerling)	2 mo.	1/500 lb.	0.02
II (yearling)	12 mo.	1 lb.	0.25
III (growout)	12 mo.	3 lb.	1.00

As discussed by Haby and Cuenco, the most highly desirable market size of red drum is three to seven pounds, and certainly above two pounds. Depending upon the stocking density and management of the pond, a fish ranging from three-quarters to one-pound can be produced in the first year. During the second year, the average size increases to about three pounds. Scheduling of the production cycle, the predictability of the crop and maximizing the farm production is facilitated by running a 12-, 24- or 36-month cycle. Thus, a two-year crop producing 3,000 to 5,000 pounds per acre of three-pound fish is suggested. As better feeds and more biological and marketing information is collected, the cost effectiveness of one- and three-year crops should be investigated as well.

Stocking Density, Growth and Survival

Phase-I

Fry, ready to feed, will be stocked into small fingerling production ponds at a rate of 400,000 to 500,000 per acre. After 45 to 60 days, about 100,000 fingerlings will be harvested per acre.

The size of the fingerlings at harvest is largely dependant upon temperature and the amount of forage present. Fingerlings smaller than 1.5 inches are difficult to handle and may not produce consistent survival in Phase-II.

In the example farm, projected fingerling survival will provide twice as many fish as are needed to stock the Phase-II ponds. This provides a level of security in case there is a problem with the quality of the fry or the zooplankton bloom does not develop correctly. Since there may be a strong market for fingerlings, the integrated farm may produce excess fingerlings for sale to other operations. Small farms and farms that are not adjacent to water with acceptable salinity ranges will purchase fingerlings from other producers, thus elimating Phase-I. Many semi-intensive growout sites will not have the proper salinity levels for fingerling production.

Phase-II

The growth rate of red drum in semi-intensive ponds is somewhat density dependent. Production levels as high as 11,000 pounds per acre of yearling fish have been demonstrated. However, a Phase II stocking rate of 5,000 to 6,000 fish per acre is currently recommended due to management and economic factors. In addition to stocking density, growth during Phase-II is influenced by temperature, water quality and food availability. A typified semi- intensive growth curve under adequate water quality and feed conditions is shown in Figure 1. Annual water temperature conditions, typical of South Carolina, are also shown. The period of no growth due to low temperature and the resulting growth rate must be adjusted accordingly for locations that are more or less temperate than South Carolina. The site-selection chapter of this manual may be used to correct the temperature curve for other locations.

Survival in the yearling pond should be very high if the fingerlings are healthy when stocked and there are no major disease or predator problems. The disease/parasite problem of primary concern is an infestation of the dinoflagellate *Amyloodinium* sp., which is discussed elsewhere in this manual. *Amyloodinium* sp. presents an extremely serious threat to the viability of commercial production as an entire pond of fish can be lost over a period of only a day or two. The chemotherapeutic COPPER CONTROL used at a level of 9 ppm of copper ion for 24 hours has been found to be a very effective treatment in tanks although its use has not been approved for redfish and may be cost prohibitive. In addition, it is difficult to keep the copper in solution for the required 24 hours, because it tends to bind to organic matter in the water and the pond soil. Successful treatment of *Amyloodinium* sp. in a pond has not yet been demonstrated although pond flushing appears to help (Trimble 1979). Research ponds have typically had yearling survival in excess of 95 percent. For the purpose of cost projections and feed rate calculations, a survival of 90 percent should be assumed. The mortality of juvenile and larger red drum is usually apparent to the observant culturist. The only fish that die and cannot be accounted for are those taken by cormorants, osprey and other large predators.

Phase-III

The effect of stocking density upon growth in the final production phase is not as well defined. The fragmentary evidence from red drum growout trials and experiences with other fish indicate that the growth rate depicted in Figure 1 is easily attainable in ponds producing 3,000 to 4,000 pounds per acre at harvest. During the first few years of semi-intensive production, a stocking rate of 1,000 to 1,500 fish per acre is suggested to meet the targeted three-pound market size. It is likely that, as the farmer gains experience and the quality of available feeds is improved, higher stocking rates and production levels will be possible. In addition, the red drum farmer may wish to alter the stocking density based upon the market demands for various fish-size categories.

Survival in the Phase III growout pond is expected to be near 100 percent in the absence of disease outbreaks. For cost projections and feding rate determinations, a survival rate of 95 percent should be used unless dead fish are found. Osprey predation is still possible yet insignificant during the first half of the Phase-III production cycle. Particular sites may have other predatory creatures.

Pond Description and Harvesting Procedures

Phase-I

It is advisable to have two to five fingerling ponds with a combined area of one acre for every 10 to 15 acres of Phase-II ponds. Having several fingerling ponds will facilitate handling of the fingerlings as they are transferred to the Phase-II ponds. In addition, having several ponds eliminates the need to stock fry of different ages into one pond thereby reducing cannibalism and making the fingerling size distribution narrower at harvest. After the first fingerling production period, these ponds may be used (1) to produce fingerlings for sale, (2) to hold fish seined from Phase-III grow-out ponds prior to marketing or (3) to restock for yearling grow out.

The fingerling ponds must be built with a fairly steep bottom slope of at least 1 percent. The steep grade facilitates complete and rapid drainage at harvest and ensures that the fingerlings do not become stranded in depressions in the pond bottom. The steep slope also facilitates predator and competitor control before the pond is fertilized and filled.

If the pond is to be used to hold fish going to market temporarily, the barrel should be oversized, perhaps 10 to 12 inches for 0.5 acre ponds, to enable the pond to be drained in just a few hours. The inlet piping should be 6 inches so that the pond can be filled rapidly to accept another group of fish. Splash pads at the water inlet and copious rip-rap around the drain outlet will be needed to facilitate rapid draining and refilling.

The design of the riser screen is critical to the effective operation of the fingerling pond. The screen must have enough surface area to allow rapid drainage without trapping fish against the mesh. The screen should be removable yet must fit tightly.

The Phase-I pond should be located as far as possible from trees as leaves and other such debris that blows into the pond can make handling and enumeration of the fingerlings difficult at harvest.

A harvest basin should be built in front of the drain riser. The harvest basin greatly facilitates the safe and rapid handling of the fingerlings. A variety of harvest basin designs exists in fish hatcheries that routinely handle mass numbers of live fingerlings for stocking purposes. The top of the basin wall should

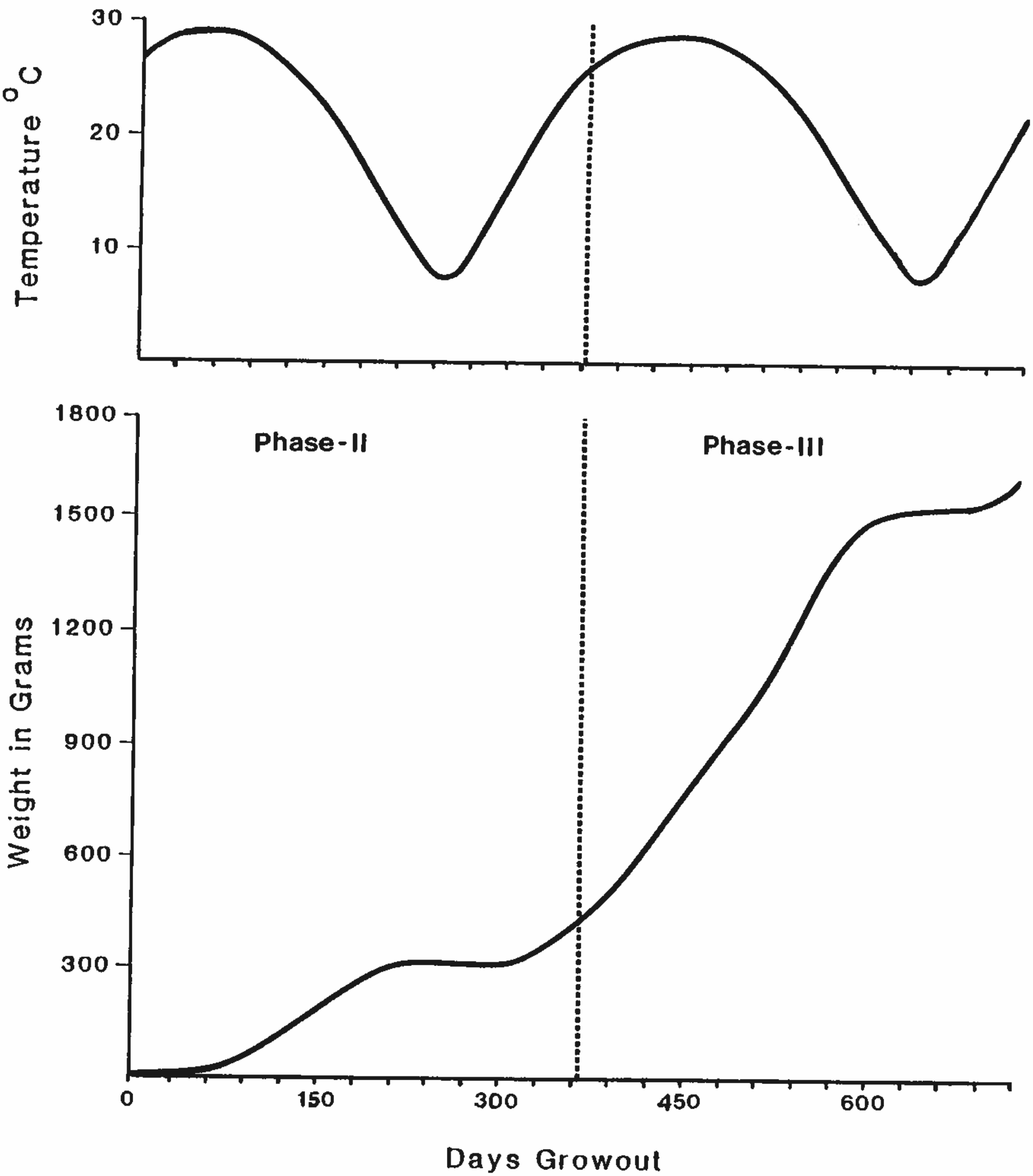

Figure 1. The growth rate of red drum during Phase-II and Phase-III of production cycle and the effect of water temperature on growth.

be flush with the pond bottom. A saltwater inlet pipe should be run to the harvest basin so that water can be run through the basin while the fingerlings are being collected. This will maintain water qualilty in the basin and ease the frantic nature of the fingerling harvest.

Fingerlings will be transferred to the Phase-II ponds during spring or early summer. It is safer to make all transfers before the water temperatures rise in the summer. Fingerlings will be graded to pool or eliminate the larger cannibals and stocked into the yearling ponds at a rate of 5,555 fish per acre.

Phase-II

The yearling ponds will have to drain fairly rapidly to facilitate transfer of livestock. A bottom slope of 0.2 to 0.5 percent is suggested. The barrel size on a typical 2.5-acre or 5-acre yearling pond should be 16 or 20 inches, respectively, to get total drainage in less than 24 hours.

For a variety of reasons, a nearly square pond may be most efficient and cost effective. Square ponds have less perimeter berm to construct per surface acre. A square pond is easier to mix with aeration equipment as there is less tendency for water to short-circuit the complete gyre. Long, narrow ponds will work and may be necessary if the cost of the inlet and outlet canals is very high or the property has an unusual shape. Long, narrow ponds may be excessively deep in one end or excessively shallow at the other end to maintain a drainable slope. The aeration rates being recommended impart enough energy into the pond to set up a gyre to mix the pond totally when needed.

The inlet pipe for the yearling pond should be in the order of 12 and 18 inches for 5 or 10 acre ponds, respectively. This pipe should provide up to a 25 percent water exchange per day, should this much water be needed to alleviate a crisis situation.

A concrete harvest basin is not necessary in the yearling pond although a slightly deeper area around the drain structure will facilitate handling the fish.

Some specialized equipment will be necessary to safely and effectively transfer the yearlings to the Phase-III pond. The pond will be partially drained and the fish seined with equipment similar to that used for commercial catfish culture. The fish will be confined in floating mesh live cars with about 1,000 to 2,000 fish per unit. The live car will be constructed of small knotless mesh to minimize damage to the fish. A specially designed flat bed truck with a boom and tank mounted on the back will be needed. The boom will have a winch and cable arrangement with a scale on the terminal end. A live car will be picked up by the boom, the weight of the fish determined, and the entire live car with fish placed in the tank with oxygenated water. The truck would drive to the edge of the Phase-III pond and hoist the live car into the pond to the fish released. A representative group of fish would be used to determine the average weight for use in the survival rate calculation.

Phase-III

The grow-out ponds will not have to drain rapidly, unless they are designed for multiple use, e.g. to double as shrimp grow-out ponds. A bottom slope of 0.2 percent and an outlet pipe of 18 or 24 inches for 10- and 20-acre ponds, respectively, would be adequate for slow draining. The inlet pipe would need to be 12 and 18 inches for the 10- and 20-acre ponds, respectively, to facilitate a 25 percent water exchange in crisis situations. Once again, a nearly square pond is the most cost effective.

Since complete harvesting may not be a prime consideration when sending fish to market, the harvesting procedure need not be as intense as the transfer from the yearling ponds. Typical catfish seining and brailing techniques will work well. Final draining may still be advisable as unharvested fish may be above the prime market size by their third year. A sump near the pond drain will not be necessary .

Cull harvesting may actually begin much earlier in the year if the market conditions so dictate. Some two-pound fish, the minimum market size, will be available in September. A typical size distribution of fish 22 months after fingerling stocking is depicted in Figure 2.

Feeding

Phase-I

The natural succession of pond fauna during the spring and early summer is ideally suited to fish production on farms adjacent to estuaries of the southeastern United States. The pond is fertilized and filled as per the procedures outlined previously in this manual. The phytoplankton bloom that develops provides a large forage base for rotifers, which are the proper size food item for the fish fry when they are stocked. As the fry grow and require larger prey items, a population of copepods develops. When the fish are ready to take still larger prey, an abundance of water boatmen (family Corixidae) are available. Grass shrimp, *Palaemonetes* sp., grow up rapidly in the pond and provide some natural forage for juveniles later.

By this time, the fish destined for semi-intensive grow out should be weaned onto dry rations. However, the availability of some of these larger natural forage items may reduce the incidence of cannibalism. It may be advisable to begin applying crumbled dry rations several weeks after stocking. While this material is not readily accepted initially, the fish will slowly begin the weaning process and uneaten feed will act as additional organic fertilizer to maintain productivity of natural food items.

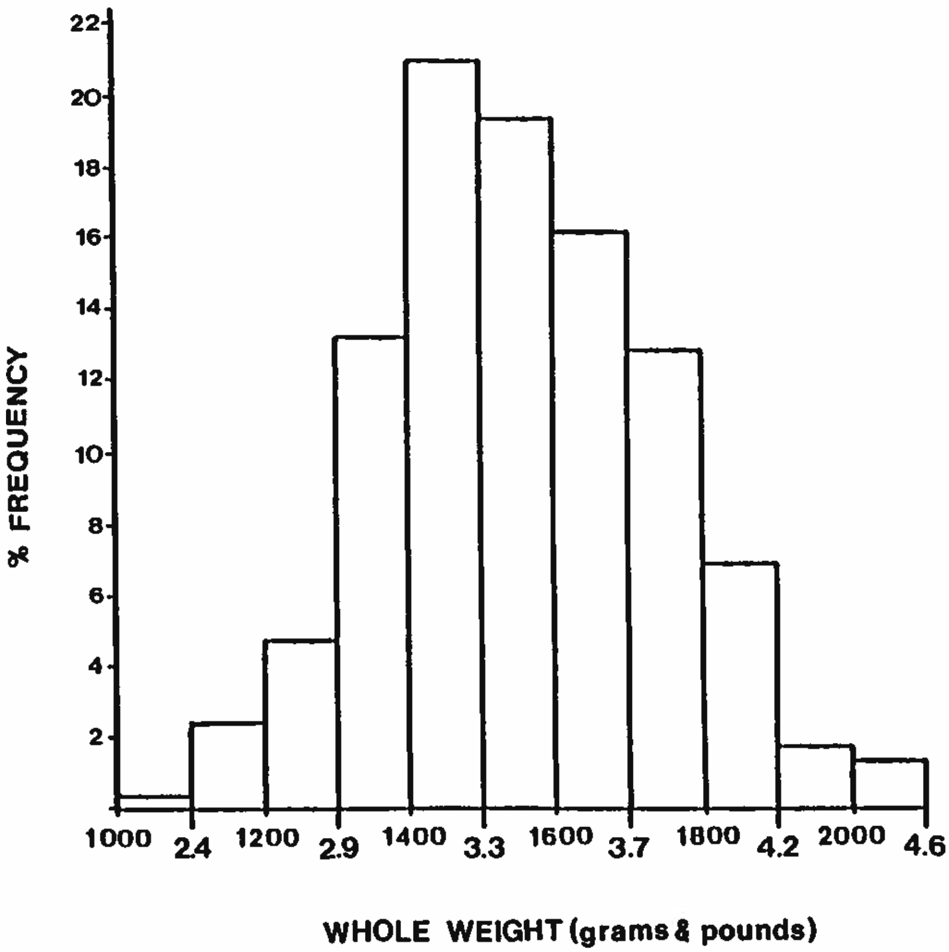

Figure 2. The size distribution of red drum in a Phase-III production pond after 10 to 12 months.

Phase-II

Feeds that are accepted by and promote growth in red drum are on the market. However, as was discussed in an earlier section, the process of defining the dietary requirements of the species has only recently begun. Due to the immediate interest in commercialization, "best-guess" diet formulations have been proposed and several off-the-shelf rations that were developed for other species have been compared. Formulations that, in tank studies, appear to produce faster growth than those used in semi-intensive pond culture to date, are now available. The most effective method of applying feed for a semi-intensive red drum pond has not been determined. We do know that the fish will come to feeding stations and can even be trained to use demand feeders. However, large ponds have never been tested so we can only assume that feeding from the bank is a satisfactory technique. It is expected that feed blowers operated from the bank would be used to distribute food around the perimeter of the pond.

Feed conversion rates for red drum are not well defined and present a major information gap that hinders commercialization of semi-intensive culture. To date, semi-intensive grow-out trials have tended to overfeed red drum to ensure that the system is not being feed limited. As a result, the optimum feed conversion and the trade-off between feed conversion and growth rate is unknown. Reported Phase-I feed conversions have ranged from 2.8:1 to 5.6:1. We have only one Phase-II grow-out trial to base our recommendations on. This trial had a feed conversion of 3.3:1. It is generally felt that the feed conversion ratio can be significantly decreased. As mentioned elsewhere in this manual, formulations yielding a feed conversion ratio of 1.8:1 are believed to feasible. With experience, a feed conversion ratio of about 2.5:1 should be achievable. However, it is reasonable to expect that the feed conversion will have dropped to 2.0:1 within five years as a result of improvements to the pond management practices and feed formulations.

Aeration

Semi-intensive stocking rates require supplemental aeration, particularly during the warmer parts of the year. The high biomass of fish and the daily addition of nutrients in the form of fish feed result in an enormous oxygen demand. Oxygen usually becomes the first limiting factor to increased production in semi-intensive systems.

Aeration devices perform two basic functions: (1) direct dissolved oxygen elevation through the splashing of water into the air and bubbling air through water, and (2) indirect dissolved oxygen elevation through water mixing which, in turn, increases atmospheric diffusion of oxygen across the entire pond surface. Of course, the splashing and droplets formed increases the surface area of a small volume of water thereby elevating the dissolved oxygen. The vertical mixing is important in that it pulls the surface film of oxygen saturated water down into the water column. The diffusion rate of oxygen across the water surface is inversely proportional to the degree of oxygen saturation. Therefore, the diffusion rate from the atmosphere is maximized by continually replacing the surface film with water of low oxygen content from below. It is believed that mixing may be a more cost effective and energy efficient method of raising the overall level of dissolved oxygen in the pond.

There are a variety of devices that perform one or both of these functions. One of the most common types is the paddlewheel aerator.

The spinning of the paddlewheel rotor creates turbulence and throws water into the air. Horizontal currents produced by the rotor circulate the water with elevated dissolved oxygen. The horizontal currents also generate vertical mixing in the shallow fish pond. The relative degree to which a paddlewheel accomplishes the two basic functions of splashing and mixing is controlled by the speed of the rotor. With high-speed units, operating at about 100 rpm, much of the electrical energy from the motor is used to throw water into the air. The paddlewheel does more mixing and less splashing when the rotor turns at a slower speed. Units designed for slower speeds put more paddle surface into the water at the rated horsepower. Therefore, more of the electrical energy goes into moving water and mixing the pond as a whole than in splashing water and creating a localized area of elevated dissolved oxygen.

The catfish industry tends to use paddlewheels or other aeration devices, which do a lot of splashing and breaking up of the water to create localized areas of elevated dissolved oxygen concentrations. This works well for catfish as the fish will sense an oxygen gradient and position themselves around the oxygen haven created by an aeration device during times when the oxygen levels in the pond as a whole are low.

Shrimp, on the other hand, do not respond to an oxygen haven and will be found uniformly distributed across the pond surface during periods of low dissolved oxygen concentrations, even when aeration equipment is running. Therefore, the shrimp farmer uses equipment whose prime purpose is mixing rather than splashing. The two major areas where shrimp production is at a level where aeration/mixing equipment is required are Taiwan and Japan. Taiwan is using 100 rpm paddlewheel units. Japan has now completely shifted from paddlewheels to submerged-propeller type devices.

At this point, we do not know if the aeration philosophy for red drum culture is more akin to that of catfish or shrimp, the two species with which we have the most experience. There are some indications that red drum will migrate toward an oxygen haven but the lethal oxygen concentration is probably higher than it is for catfish. Catfish aeration in systems designed to produce 3,000 to 5,000 pounds per acre is generally in the order of one horsepower per acre. Shrimp ponds with similar production levels use two to three horsepower per acre.

Aeration rates for red drum ponds at various stocking densities have not been pin-pointed as yet. In addition, actual aeration requirements in terms of the amount of time the unit is run and the operational expense is strongly influenced by climatic conditions and phytoplankton succession in the pond. To be safe, the production scenario discussed here should include two horsepower per acre in the yearling ponds and one horsepower per acre in the grow-out ponds. No supplemental aeration will be required in the Phase-I fingerling production ponds.

The real expense of aeration is in the operation of the units rather than the amortized capital costs of the equipment. While these suggested rates may be a slight overkill, it is believed to be an inexpensive form of insurance. This recommendation is based upon the use of high-speed paddlewheel devices. Horsepower requirements will probably be higher for bubble-up or fountain type equipment. Similarly, engineering studies may reveal that less horsepower is required if low-speed paddlewheels or submerged-screw type units are employed.

Water Exchange

Water exchange in red drum culture may be relatively high compared to traditional catfish culture. Catfish operations are generally water limited as the ground water is expensive to extract in terms of both capital and operating costs. It is generally impractical to exchange water at a rate of more than a few percent per day on commercial catfish farms and a beneficial effect of water exchange at rates of up to 4 percent per day has not been demonstrated. Limited water resources and pumping capacities are a part of the reason catfish farmers are shifting toward smaller production units (10-acre ponds instead of 20- to 40-acre ponds). The inability to manage ponds in a crisis situation, such as a phytoplankton bloom "crash," makes small ponds more practical. By diverting all the pumping capacity to a single small pond, a higher exchange rate can be achieved. Overall losses can be minimized by using a larger number of smaller pond units.

Coastal red drum farms will have abundant supplies of estuarine water with relatively low pumping head. However, these abundant supplies of water bring with them federal, state and sometimes local regulations on the quality of the effluent. In addition, seepage of saltwater into the ground may be regulated in some locals with certain soil types. The regular exchange of pond water not only dilutes the algae bloom and mediates dissolved oxygen fluctuations, but also results in an overall improvement of the effluent quality. More importantly, the ability to rapidly flush a pond during a crisis situation may save a crop.

The main pumping system should be capable of exchanging up to 5 percent of the total volume on the farm per day or 30 percent of the volume of an individual pond per day, whichever is larger. Thus, farms with a small number (less than five) of large ponds will need more pumping capacity per acre. This level of pumping capacity and the availability of supplemental aeration equipment should be sufficient to prevent mortalities even after a massive algae crash. Water quality issues are discussed elsewhere in this manual.

Literature Cited

Hopkins, J.S., A.D. Stokes, P.A. Sandifer, R.A. Smiley and T.R. McTeer. 1987. Spawning and growout trials with red drum in South Carolina. Proc. Ann. Conf. Southeast. Assoc. Fish and Wildl. Agencies. (in press)

Trimble, W. C. 1979. Yield trials for red drum in brackishwater ponds, 1976-1979. Proc. Annu. Conf. Southeast. Assoc. Fish and Wildl. Agencies. 33:432-441.

Semi-Intensive Pond Management Methods

Alvin D. Stokes
Waddell Mariculture Center
P. O. Box 809
Bluffton, South Carolina 29910
and
Theodore I.J. Smith
Marine Resources Research Institute
P.O. Box 12559
Charleston, South Carolina 29412

To produce commercial quantities of red drum, many farmers will probably choose semi-intensive pond management methods. These management methods have been demonstrated to yield rapid growth and high survival under experimental conditions. For example, experimental semi-intensive yields of 3,000 to 4,000 pounds of red drum per acre have been produced in 22 months. The three-phase production system previously described will allow the pond manager to implement semi-intensive management methods more effectively. Utilizing the year-long Phase II and Phase III scenarios versus the two-year growout system, the manager can more efficiently dry and fertilize ponds, regulate water quality, monitor feed rates, and schedule income.

Pond Preparation

Draining and Bottom Management

Ponds should be drained, allowed to dry, and plowed according to the production schedule. This procedure allows organic matter to oxidize and decompose, thereby reducing the sediment demand on oxygen once the ponds are refilled.While the pond bottom is drying, soil samples can be analyzed by extension service personnel to determine soil pH. If the soil pH is below 6.5, liming may be required. Extension service personnel can assist with liming recommendations for pH adjustment.

Fertilization

Pond managers fertilize their ponds with either inorganic fertilizers, organic fertilizers, or a combination of the two, to initiate phytoplankton production. Natural pond fertility varies from site to site; therefore, the effect of each application rate should be closely monitored to avoid over-fertilization and the resulting poor water quality. This is especially true in areas where soil and water contain high nutrient levels.

Inorganic (or chemical) fertilizers are widely used in finfish culture. As these generally have a high nutrient value, production ponds may only require relatively small quantities to induce phytoplankton production. Inorganic fertilizers are identified according to their percentage composition (by weight) of nitrogen, phosphorus, and potassium (N-P-K). Popular inorganic fertilizers such as 20-20-5, 20-15-5, 10-34-0, phosphoric acid (0-54-0), urea (42-0-0), and triple super phosphate (0-46- 0) are commonly used in finfish culture throughout the United States.

Organic fertilizers used in finfish culture in the United States consist mainly of plant matter such as cotton seed meal, Bermuda grass pellets, rice bran, etc. These organic fertilizers contain a lower nutrient value when compared to inorganic fertilizers; therefore, pond managers must apply larger quantities to achieve comparable phytoplankton production.

The application of both inorganic and organic fertilizers has proven to be an effective method of establishing healthy phytoplankton blooms in semi-intensive red drum reproduction ponds. Pond managers should realize the important contributions of both types of fertilizers. Inorganic fertilizers provide the nutrients that phytoplankton require. Organic fertilizers provide limited nutrient value for phytoplankton, but more importantly serve as a food source for forage organisms. In South Carolina, semi-intensive red drum production ponds are fertilized during Phase II and III with a combination of liquid 10-34-0 inorganic fertilizer (0.5 gal/acre) and Bermuda grass pellets (300 - 500 lbs/acre) to initiate phytoplankton and forage-organism production.

Role of Phytoplankton

Healthy phytoplankton production in red drum ponds is important for several reasons. Phytoplankton biomass increases the absorption of energy from the sun. This warms and stabilizes water temperatures for stocking. In addition, phytoplankton blooms can shade out unwanted plants and filamentous algae, encourage forage organisms by inducing zooplankton production, and produce dissolved oxygen through photosynthesis.

Conversely, phytoplankton production can also

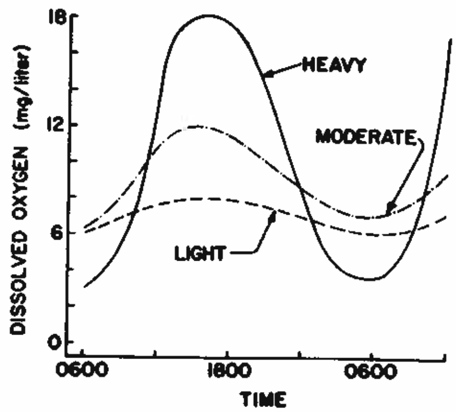

Figure 1. Diel fluctuations of dissolved oxygen concentrations in surface water of ponds with different densities of phytoplankton (from Hollerman, 1983).

produce adverse effects. Dense blooms can form when the water becomes enriched with excess feed and metabolic wastes. These dense phytoplankton communities lower dissolved oxygen levels at night with their increased respiration rates. Other negative effects include thermal stratification and phytoplankton crashes caused by dense shading. By measuring light penetration with a Secchi disc, pond managers can prevent these water quality problems through the use of appropriate aeration and water exchange practices.

Screening Water Inflow and Filling

Water used for filling and exchange should be screened (i.e. with a 400 micron mesh sock) to prevent the entry of predator and competitor species. Introduction of competitors in a grow-out pond could reduce feeding efficiency (thereby affecting feed conversion and growth rates), impair water quality, and impede harvest and restocking procedures. When filling the ponds, the "puddle method" is recommended. This allows sunlight to penetrate pond water, thereby rapidly inducing phytoplankton production and stabilizing water temperatures.

Water Quality Management

Dissolved Oxygen

Dissolved oxygen concentration is usually the most critical environmental parameter measured by the pond manager. During a daily cycle, dissolved oxygen concentrations normally rise to supersaturated levels during the day by photosynthesis and drop (usually not to critical levels) by diffusion and respiration at night. However, as the fish biomass increases and metabolic wastes and excess feed accumulates, the increased demand for dissolved oxygen at night can lower dissolved concentrations to critical levels by morning.

Pond managers familiar with this cycle measure surface and bottom dissolved oxygen concentration daily at sunrise when dissolved oxygen concentration levels are lowest. By watching this trend from day to day, critical dissolved oxygen levels can be avoided by managing feed, aeration and water exchange rates. In the event that early-morning dissolved oxygen levels are 3.5 mg/l or less, pond managers should be alert for further decreases in dissolved oxygen levels. If early-morning dissolved oxygen levels fall below 3.0 mg/l, pond managers should aerate, exchange water and reduce feed if necessary until morning D. O. levels improve.

Temperature

Daily morning surface and bottom temperature are important data for semi-intensive pond management. Water temperatures affect respiration rates and dissolved oxygen saturation values. Feed rates are also adjusted according to water temperature. Red drum growth data suggest that red drum grow faster in warmer months when water temperatures are between 79°and 86°F (26°and 30°C) and that very little growth occurs in the winter months or in the summer months when the temperatures exceed 86°F (30°C). These temperature data are useful in determining whether to increase or decrease feed rates. In addition, differences in surface and bottom water temperatures may alert the pond manager to the formation of thermal stratification. If there is a temperature difference of 3°F (1.6°C) or greater, proper aeration and water exchange should be implemented to avoid an associated low oxygen situation in the bottom water stratum.

Water Exchange

Water exchange is a vital part of a balanced semi-intensive pond management program. Daily water quality data will alert the manager to the need for water exchange to improve dissolved oxygen concentrations, reduce phytoplankton density, eliminate thermal stratification, and improve pond water quality in general. During the months of May through October when red drum growth and feed rates are the most intensive, a daily water exchange rate of 5 to 10 percent may be required (12 - 25 gpm/acre-foot). These rates are reasonable; however, a well-designed semi-intensive pond system will have the ability to exchange water at much higher rates for emergency situations.

Aeration

Pond aeration may be achieved by three aeration methods: injecting air into the water, spraying water into the air, and mixing. Mixing water appears to be the most effective method for increasing dissolved oxygen levels in pond bottom water. It also encourages phytoplankton production and prevents thermal stratification. Paddlewheel aeration for semi-intensive red drum production has been successfully demonstrated. Rates of 2 hp/acre for Phase II and 1 hp/acre in Phase III production ponds are recommended.

Aeration units should be placed around the perimeter of the pond, directing water flow parallel to the pond bank. This movement will improve mixing and create a circular motion. Organic matter and debris will then collect in the center of the pond for removal when the pond is drained and dryed.

Sampling, Grading and Harvesting

Sampling

Once the red drum are stocked into Phase II and III, specific pond management practices should be implemented. Every 28 days, growth should be estimated by seine sampling 25 fish from each pond. All fish should be transported in aerated pond water and anesthetized (discussed in Transportation and Acclimation of Red Drum Fingerlings) at the sampling station to reduce handling stress. Individual fish should be weighed to the nearest gram to determine mean fish size. After sampling, the fish can be revived and returned to the pond.

Phase II

Red drum fingerlings harvested from Phase I ponds should be graded. This will eliminate the large fish, thereby making the estimated stocking rates of Phase II ponds more accurate. A random sample of 25 to 50 fingerlings should be adequate to determine mean fish size. Red drum fingerlings can then be stocked into Phase II ponds by weight at an estimated rate of 5,000 to 6,000 fish/acre.

Phase III

After approximately 10 months, the yearling red drum are harvested from Phase II ponds and restocked into Phase III ponds. These 3/4 to 1 pound fish should be stocked at a rate of 1,000 - 1,500 fish/acre. A random sample of 50 fish should be adequate to determine mean fish size for restocking and to estimate survival and feed conversion from Phase II. With good semi-intensive pond management, a harvest of 3,000 - 4,000 lbs/acre can be achieved from the Phase III ponds in 10 months.

Feeds and Feeding

Feed Type

Red drum reared for 22 months in a semi-intensive pond production study in South Carolina were fed a 38 percent protein trout chow from Zeigler Brothers. At harvest, the red drum averaged 3.4 pounds and had a survival rate of 95 percent. Overall feed conversion (feed-to-fish) for Phase II and III was as high as 4.2:1. However, feed rates were artificially elevated to identify the growth potential for red drum produced in semi-intensive production ponds.

Pellet Size

The trout chow pellet size was increased several times during Phase II. With an average size of 11.0 grams, redfish juveniles were provided with 1/8-inch pellets. When the average fish size reached 52.0 grams, pellet size was changed to 3/16-inch. After attaining an average weight of 237.0 grams eight weeks later, red drum were fed 1/4-inch pellets throughout the remainder of Phase II. Quarter-inch pellets were also provided to the red drum in Phase III.

Feeding Rates

Feed rates are estimated in a number of ways. In South Carolina, we utilize growth curves and survival rates determined from previous semi- intensive red drum production studies. With this information, pond managers can project a change in total biomass from one sample to the next (28 days) and employ a feed-to-fish conversion rate of 2:1 for the change. In addition, daily shifts in water quality parameters—such as temperature and dissolved oxygen levels—should be monitored for their potential effect on the feeding habits of red drum.

Parasites

Although there are dozens of diseases and parasites that may affect red drum, one of the most undesirable is an outbreak of the ectoparasitic dinoflagellate, *Amyloodium ocellatum* (detailed description provided in Recognition and Control of Diseases Common to Grow-out Aquaculture of Red Drum). This parasite, when uncontrolled, can wipe-out a semi-intensive production pond. Since the parasite attaches to the gills and interferes with respiration, infected fish may gather near the pond surface, aeration devices, and water inflow structure in search of higher dissolved oxygen levels. Fish also may appear to "cough" water in an attempt to flush the gills. Observing this behavior in ponds may be difficult due to phytoplankton density; therefore, pond managers may want to sacrifice a fish periodically to check for this parasite. Treatment with chelated copper maintained at a level of 4 - 6 mg/1 for 24 hours should be sufficient to control this parasite. However, the ability to maintain the proper dose in production ponds is difficult due in part to the absorption of copper into pond muds.

Conclusion

Red drum farmed in ponds stocked at a rate of 5,000 to 6,000 fish/acre in Phase II and 1,000 to 1,500 fish/acre in Phase III must have a proper management program to assure good survival, growth rates and feed conversions. Ponds must be properly prepared (draining, liming, fertilization, etc.) and man-

Table 1. Example of feeding and management schedule for red drum production ponds during Phase II and Phase III.

Month	Mean Fish Size (g)	Water Temp. (°F)	Percent Feed (2:1)	Feed Pellet Size(")
	Phase II			
March (Drain, soil analysis, lime, fertilize)				
April (Fill by puddle method)				
May (Stock fingerlings)				
June (Stock fingerlings @ 0.4 g)			200.0	
July	11.0	80.6	26.5	1/8
August	52.0	87.8	6.5	3/16
September	99.0	77.0	10.0	3/16
October	237.0	75.2	0.7	1/4
November	260.0	60.8	0.6	1/4
December	280.0	58.2	0.2	1/4
January	287.0	63.0	0.3	1/4
February	298.0	51.8	0.1	1/4
March	304.0	62.6	0.6	1/4
April	330.0 (Harvest)	77.0		
	Phase III			
March (Harvest, drain, dry, fertilize)				
April	330.0 (Fill and restock)		2.5	1/4
May	450.0	80.6	4.0	1/4
June	690.0	86.0	0.5	1/4
July	737.0	82.4	2.5	1/4
August	1000.0	86.0	1.5	1/4
September	1198.0	78.8	1.0	1/4
October	1395.0	77.0	0.5	1/4
November	1500.0	68.5	0.3	1/4
December	1560.0	50.0	0.2	1/4
January	1580.0	51.8	0.0	1/4
February	1580.0	57.2	0.1	1/4
March	1600.0 (Harvest)	59.0		
April (Fill and restock)				

aged. Water quality must be monitored (dissolved oxygen levels, temperature, etc.) and improved when necessary by water exchange and aeration. A good sampling and feed management program is also required. In the end, a well-managed semi-intensive pond can yield high production with competitive efficiency.

Literature Cited

Hollerman, W. D. 1983. Pond management: water quality criteria and control. *In* **Aquaculture in Dredged Material Containment Areas**. Miscellaneous Paper D-83-2, U.S. Army Engineer Waterways Experiment Station, Vicksburg, Mississippi. pp. 121-129.

High Density Recirculating Growout Systems

C.R. Arnold and Bart Reid
The University of Texas at Austin
Marine Science Institute
Port Aransas, Texas 78373
and
B. Brawner
Red Ewald, Inc.
P. O. Box 519
Karnes City, Texas 78118

The growing of aquatic organisms in the United States is a relatively new industry. The common method of growing organisms is the use of earthen ponds, (Nash, 1988; Kerby *et al.*, 1987; Sandifer *et al.*, 1987) but with the high prices of land, as well as the scarcity of land suitable for aquaculture, a new concept in growing aquatic organisms is necessary. Just like the chicken industry, which has gone to high density growout, the growing of aquatic organisms also will probably follow. The development of such a system to grow aquatic organisms at very high density has been limited because of the difficulty in controlling water quality (Mock *et al.*, 1977). New concepts in water quality control, in food and feeding methods, and in pumping water have been developed in recent years, which have made possible the idea of growing aquatic organisms at very high density.

System Design

The basic components of our high-density system are the tank that holds the aquatic organisms, the filter that removes the waste products and maintains water quality, and the air supply that is the energy driving this system. The tank can be of any size or shape, but raceways (rectangler tanks) are more efficient for water flow, even distribution of food, maintenance and floor space considerations. The raceway described here is 45 feet long, 8 feet wide and 4 feet deep. The filter is 16 feet long, 4 feet wide and 4 feet deep, the air is supplied by a blower that produces high volume at low pressure, (3 to 5 PSI).

The water flows from the tank into the filter box by gravity, through the filter horizontally, and from the filter box back into two 4 inch PVC pipes to airlift pumps that force the water into the tank. The filter box has 16 plates, which are lined with a polyester air-conditioning filter material. The plates have 4-inch holes cut in them accounting for about 50 percent of the surface area. The filter material is attached to these plates using a silicon sealant. Air is injected between each plate, which is necessary for oxygenation to maintain a good bacteria population on the filter media. This bacterial action depletes the dissolved oxygen content of the water passing through the filter tank. To replace this loss, air is bubbled into the spaces between each filter plate using air stones or porous tubing fixed to tank's bottom. Nylon twine is used to secure the aerators by passing it through a fiberglass eye and then tying it off at the tank's top in another fiberglass eye. This allows the aerators to be pulled up for servicing and then repositioned without having to drain the tank. In setting up the filter it is very important to make sure the top of the filter will be at the same level as the water level in the raceway. This will insure that the water level in the filter will be high enough to cover the entire filtering surface area, which increases the efficiency of the filter.

Aeration and Circulation

Air is important in the raceway as well as the filter box. Air can be supplied using either air stones, porous air tubing, or airlift pumps attached along the side of the raceways. Airlift pumps set up a current as well as aerate the water. Again, the driving force that makes this filter and raceway system work is the airlift pumps. They pump the water from the filter back into the raceway where it gravity feeds back into the filter box so that a closed system is set up and the only working parts are the airlift pumps. Air is supplied at 3 psi by air blowers. The advantage of using blowers instead of compressors is the delivery of large volumes of air at low pressure, eliminating the need for elaborate regulators. The air is piped in 2-inch PVC to the raceway area where it enters a 3-inch PVC pipe loop that circles the raceway. The loop stores air and allows its delivery at equal pressure to all points of the loop. This pipe is contained in an overhead rack providing easy availability for both maintenance and use. Holes drilled in the loop every 2 feet and tapped for one-half inch pipe to accept one-half inch ball valves are used to regulate flow to aerators in different locations. This makes modifications in tank aeration or air flow very easy to accommodate.

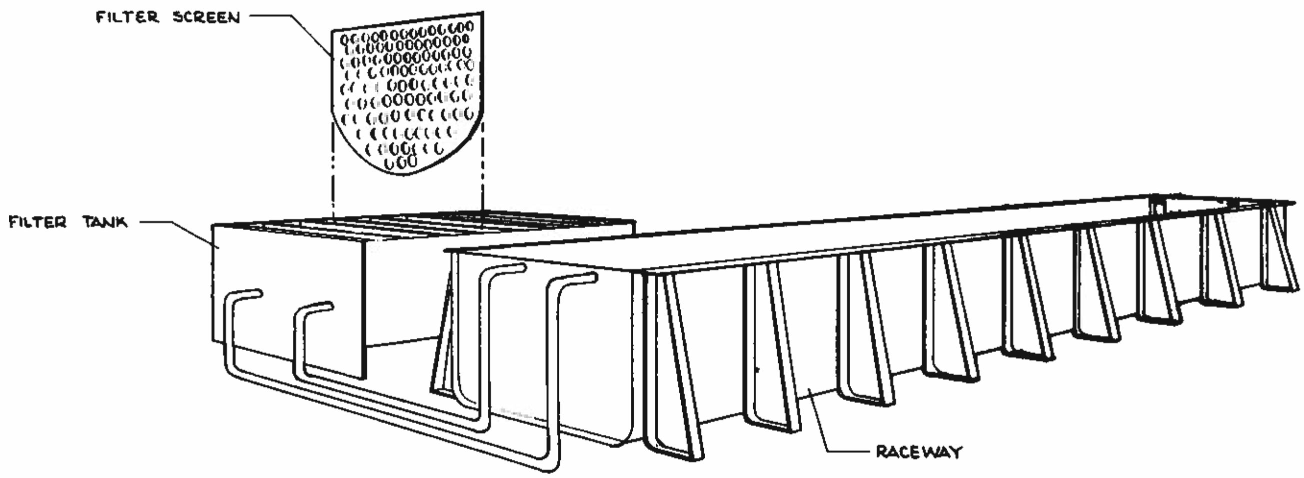

Figure 1. Schematic diagram of a high density, recirculating grow-out system.

Emergency Oxygen System

In the event of electrical or equipment failure, an emergency oxygen supply has been installed on the raceway. This system is controlled by a normally open solenoid valve which prevents the oxygen from being used until needed. Should power fail, the valve opens, allowing oxygen to fill the emergency system. If air pressure fails and power remains on, a pressure switch cuts power to the solenoid valve to permit oxygen to be released. Upon its release, the oxygen enters a three-quarter inch PVC pipe that supplies six 4-inch airstones in the raceway. This pipe is also contained in the overhead rack as is the water supply pipe and air loop.

Feeds and Feeding

One of the most critical areas of any raceway operation is the selection of the food and the feeding regime of the organisms. Selection of the food should be based on a sufficiently high protein diet with as little fiber as the organisms can tolerate. An over abundance of fiber and other indigestible materials will clog the filters to the point that they will physically prevent water flow. The food should be fed in small amounts continuously over a long period of time. Feeding once, twice or three times a day injects large amounts of food into the systems that is not as effectively utilized as the same amount of food fed in small portions on a continuous basis. Equally important is the observation of the feeding behavior of the organisms. When it is obvious that the food is not being eaten then the ration should be decreased. The ideal situation is the food being eagerly consumed by the organisms as soon as it hits the water. Overfeeding is the primary source for the "sludge" that is commonly associated with recirculating systems, therefore maintaining a proper feed schedule eliminates this problem.

There are several ways in which food may be offered to organisms in a raceway system. Hand feeding is where an individual goes around the tank periodically during the day offering food to the organisms. This is laborious and costly, but it also provides an opportunity to observe the organisms feeding. At this time, you can determine whether the fish are eating the way they should or whether the food is only being partially eaten. This is an early indication of the well being of the organisms. Another method is automatic feeding with either time-released or continuous feeders. Generally, a prescribed amount of food is fed at predetermined times. The third type of feeding is by demand feeder. This involves placing a large amount of food in a container over the raceway allowing the fish to feed by manipulating a rod that extends into the water. The fish must be trained to utilize this type of feeder. One of the advantages is that fish feed only when hungry. A continuous feeder is often more desirable than any other type of feeding. Factors to be considered in selecting a feed are sufficient protein content, good palatability, and a diet designed to meet the nutritional needs of the organism under culture. Nutritionally correct diets formulated specifically for redfish are needed to replace the trout and catfish feeds currently in use for redfish production.

Tank and Filter Operation

Once the tank and filter are in place and filled with water, the aquaculture organisms are added. At the same time the filter is inoculated with a bacteria culture containing nitrifying bacteria.

There are two important steps in the nitrifying process. The first is the conversion of ammonia to nitrite by the *Nitrosomonas* spp. bacteria, and the second step is the conversion of nitrite to nitrate (which is non-toxic) by the *Nitrobacter* spp. bacteria. Generally, when a bacterial concentrate is used for initial inoculation, it takes three to four days for the bacterial growth to reach a level where they utilize all the ammonia produced by the fish. If a bacterial concentrate is not used, then it may be three to four weeks before the bacteria catches up to the ammonia produced (Wickins, 1983). In this case, there must be sufficient water exchange in the system to maintain the ammonia level below 0.6 ppm. There is some evidence that a high concentration of ammonia is toxic to the *Nitrobacter* bacteria, so, after the ammonia levels go down, the nitrite levels may be elevated above acceptable levels (< 0.8 ppm) and the filter must then be reinoculated. Once both bacterial populations are well-established, there should not be large fluctuations in the ammonia and nitrite concentrations (Otte and Rosenthal, 1979).

Maintenance

The water flow should be maintained at approximately 75 to 80 gpm. In the operational process the filter may become physically clogged restricting the water flow. If this occurs, the filters can be removed, rinsed, and replaced. They should never be rinsed with chlorinated water, and in a seawater system they should not be rinsed with fresh water. They should not be allowed to dry out because this will kill the bacteria population. A schedule should be maintained for cleaning the filters with no more than two filter plates/week being cleaned. Occasional siphon vacuuming of the filter box to remove settled solids may also be necessary.

Monitoring Water Quality

Uneaten food and fecal material should be removed from the raceway at least weekly. Ammonia, nitrite, and pH should be monitored three times weekly. Ammonia and nitrite concentrations should not go above 0.6 ppm and the pH should be maintained between 7.7 to 8.2. The pH can be raised by adding one liter of saturated sodium carbonate per 13,500 liters of water. Bolting two filter plates together with a plastic screen and filling with crushed oyster shell helps maintain the proper pH. There should be porous air tubing or air lift pumps in the tank for aeration so the dissolved oxygen should never be a problem.

Disease in Recirculating Systems

The potential for redfish diseases (see Recognition and Control of Diseases, etc.) are inherent in any grow-out system. The advantage to raceway culture is in direct observation of the animals to determine any appearance of disease enabling the treatment of the problem in its early stages. Water chemistry can be easily adjusted to treat any potential problems that may occur.

Stocking and Growth Rates

Stocking rates for high density closed raceway systems have not been well-established. Growth rates depend on density, and much work remains to be done to establish the optimum density that will give the best growth rates at the least cost. Some of the preliminary stocking densities and growth rates are listed below.

Red Drum

There are several methods of stocking raceways: (1) Stock at high density (500 fingerlings/m3 35 to 40 mm) and separate according to size as the fish grow; (2) Stock at lower density with no separation until harvest.

Generally red drum grow at an average rate of:

hatch to 40 days	-	1 g
40 days to 200 days	-	260 g
200 days to 18 months	-	2 kg.

Fingerlings stocked at a 1 gram size (35 to 40 mm) and at a density of approximately 280/m3 had 44 percent survival after 116 days. The final fish weight ranged from 20 g to 190 g. This large growth differential could be the result of overcrowding, but was probably due to the diet.

Shrimp

Penaeid shrimp have been grown in raceways with encouraging results. The raceways were stocked with post-larvae at a rate of 1450/m3. Survival was about 40 percent and growth ranged from less than 1 gram to 10 grams in 90 days.

Conclusion

There remains much work to perfecting the closed system approach to growing aquatic organisms, but the results thus far are very encouraging and indicate that this may soon be the most economical method available.

Literature Cited

Kerby, J.H., J.M. Hinshaw and M.T. Huish. 1987. Increased growth and production of striped bass x white bass hybrids in earthen ponds. **Journal of the World Aquaculture Society** 18(1):35-43.

Mock, C.R., L.R. Ross and B.R. Salser. 1977. Design and preliminary evaluation of a closed system for shrimp culture. **Proceedings of the Eighth Annual Workshop, World Mariculture Society** pp. 333-356.

Nash, C.E. 1988. A global overview of aquaculture productions. **Journal of the World Aquaculture Society** 19(2):51-88.

Otte, G. and H. Rosenthal. 1979. Management of a closed blackish water system for high-density fish culture by biological and chemical water treatment. **Aquaculture** 18:169-181.

Sandifer, P.A., J.S. Hopkins and A.D. Stokes. 1987. Intensive culture potential of *Penaeus vannamei*. **Journal of the World Aquaculture Society** 18(2):94-100.

Wickens, J.F. 1983. Studies on marine biological filters. **Water Resources** 17(12)1769-1780.

Inland Culture of Red Drum

James T. Davis
Fisheries Specialist
Texas Agricultural Extension Service
College Station, Texas 77843

Demonstrations in West Texas indicate that red drum can survive and grow in inland ponds supplied with saline ground water. However, economic feasibility remains to be determined, because the fingerling fish stocked on June 5, 1988 did not achieve marketable size by the fall and other data indicate they will not overwinter at these locations due to cold temperatures.

This paper reports results from larval and fingerling rearing trials in West Texas and discusses general culture considerations for red drum in inland waters.

Larval Rearing

Red drum larvae, (*Sciaenops ocellatus*), were obtained from the John Wilson Hatchery of Texas Parks and Wildlife Department (TPWD) approximately 30 hours after having been spawned. The larvae were placed in two plastic bags containing about 40,000 larvae and 2 gallons of 25 ppt salinity seawater. The bags were transported to a pond near Stanton, Tx., by automobile and commercial aircraft. The entire trip requiredeight hours. On arrival, the fish were inspected visually and survival was estimated at 99 percent. Temperature of the water in the hauling bags was 23 °C and surface water temperature was 27 °C. The receiving ponds had been treated with fertilizer. The lone exception to the TPWD recommended regime was the fish arrived after water had been in the pond 45 days rather than the recommended 15 days after filling.

Harvest was attempted on May 22 after the fish had been in the ponds 23 days. The equipment available was of too large a mesh size to capture the animals. On June 5, seining of the pond revealed that many fish had survived; however, field personnel were unable to determine the actual survival percentage, but estimated survival of less than five percent. They did report a very dense insect population, which may have had an impact on red drum survival.

Fingerling Rearing

Fingerlings 3/4 to 1 inches in length were secured from the Perry Bass Hatchery of Texas Parks and Wildlife and transported in truck-mounted hauling tanks by TPWD personnel. Seven ponds were stocked in the vicinity of Stanton and three ponds in the vicinity of Grand Falls. Fish were transported in water with 25 ppt salinity and about 25 °C. Survival on the truck was reported to be fair to good.

One series of ponds, (four near Stanton) were cut off from the road by high water. This required the fish be transferred to different hauling tanks and moved on a trailer towed by a farm tractor to the pond site. This extra handling further stressed the fish and mortalities were reported as high at the pond. The aquatic insect population at this location was extremely high and they began to prey on the fingerlings immediately. In addition, recent high waters had allowed sheepshead minnows (*Cyprinodon variegatus*) to spread into the ponds. These fish also began to prey on the fingerlings. Water quality data were kept on the ponds and test seining conducted at two-week intervals. Survival was very low and at final harvest less than 50 fish were harvested from the 3 acres of ponds from a total of 4,000 stocked.

Three ponds were stocked at a second site near Stanton. Mortalities noticed at the time of stocking were minimal. Data on water temperature and quality were maintained and test seining was conducted on a monthly basis. Insects were also a problem at this location but not in the numbers found at the first location. One of the ponds was plagued with hypersaline water during the first three weeks and no fish were found in the pond at the end of six weeks. A second pond developed very low dissolved oxygen and though fish survived for at least 12 weeks, the survival rate was less than 1 percent. The third pond was stocked with 2,000 fingerlings and survival was good for the majority of the season. Low dissolved oxygens occurred on one occasion in late August and again in late September. When the ponds were completely seined in early October, approximately 300 of the fish had survived and reached an average length of 8 inches.

All 15,000 of the fish delivered to the four Grand Falls ponds arrived in very poor condition due to a malfunction of the hauling equipment. Growth was very poor due to faulty feed used for the first 90 days. Subsequently, the fish grew rapidly and reached an average length of 6 inches by the harvest date of October 8. Fish were left in these ponds for overwintering.

Aquaculture Considerations at Inland Sites

Saline Water Disposal

A primary concern for anyone considering aquaculture utilizing saline waters in inland areas is potential regulatory constraints of discharging saltwater into surface streams. In most areas this will require a special permit from the State Water Commission, which may be difficult to obtain. In Texas, underground salt water is under the regulation of the Railroad Commission, but its discharge is regulated by the Texas Water Commission. This means the use of injection wells requires a permit from one agency, but to discharge into streams requires concurrence from both agencies. The salinity level of the water will be the primary constraint and dilution may be a viable alternative in some instances. Generally, percolation or evaporation are not viable disposal solutions.

Ionic Composition

Another question that often arises about the use of inland saline waters is the ionic composition required. We do not have enough information to say exactly what the limiting levels of specific ions are because red drum are adapted to salinity levels and ionic components of water in the Gulf of Mexico and the South Atlantic Coast. The more nearly the water contains these exact constituents the better the fish should perform. Normally, exact duplication is not possible in a natural inland system; therefore, we have developed general guidelines as follows:

- Total salts—not to exceed 40,000 ppm
  —at least 6,000 ppm
- Chlorides to be at least twice concentration of sulfates
- Calcium to exceed 150 ppm
- Chlorides to exceed 250 ppm
- PH between 6.0 and .5

Plankton Colonization

Questions frequently arise about the potential problems of inducing a plankton bloom in saline water located far from the coast. Dr. William Clark, limnologist for the Department of Wildlife and Fisheries Sciences at Texas A&M University, has developed a technique for determining the probable requirements and responses of a given body of water to fertilization with nitrate and/or phosphate fertilizer. This is the single best criterion we have seen to determine efficacy of fertilization for a plankton bloom.

Processing and Marketing

Another consideration is where the fish will be marketed. At this time there are no processing plants within a reasonable hauling distance of many installations. A new producer must check out a possible market and determine in what form the fish are perferred by the buyers. Generally, gilled and gutted is the preferred form. Some special requirements may be gutted only, filleted, skinned and so forth. At most inland locations in Texas, fresh seafood is at such a premium that product form is not a major marketing constraint at this time. As more fish are grown in these areas, this will change and better processing conditions and products will be required.

General Aquaculture Considerations

Consideration should be given to general requirements for a successful aquaculture operation. During most of the year, a fish farm will require that the owner, manager or trusted employee be present at the site everyday. During the summer months, this should be before and immediately after daylight each day. Anything less than this will eventually result in a loss of a major portion of the crop. Proper construction has been discussed elsewhere and will alleviate many problems as will proper feeding regimes; but nothing replaces constant, vigilant attention to the production complex.

Fertilization and Feeds

We recommend initiation of the TPWD fertilizer program in all ponds 15 days prior to delivery of fry or fingerlings. **Caution.** Do not over fertilize. If fish are fed, one application of fertilizer will be sufficient. Uneaten feed and fish droppings will add the required fertility.

There are many commercially available catfish feeds on the market at this time. Make sure that the one you select will be readily available throughout the growing season. Several farmers have been caught short of feed by buying from small mills that fail to have adequate supplies on hand. Experience indicates that changing brands of feed often results in failure to feed (and therefore reduced growth) for three to seven days.

If several feeds are readily available, then the analysis of the feed and its constituents should be studied carefully. There should be from 30 to 35 percent crude protein in the feed with at least one-half animal protein or fortified vegetable protein. There should be a minimum fat content of 6 percent and a maximum fiber of 15 percent in the feed. The feed should be pelletized and the pellets should remain relatively intact for 10 minutes in water. If a choice of pellet sizes is available, consideration should be given to feeding smaller pellets to fish under 12 inches and those just starting to feed. Meal type feeds are recommended for fry and small fingerlings.

Certain other factors should also be considered in selecting the feeds you will use. Some feed mills, due to lack of quality control, allow their feeds to vary widely from batch to batch. This often results in poor growth of fish and many days "off feed." If you are in doubt about a particular batch of feed, have it analyzed by a qualified laboratory. Most feed mills are good, but a bad batch of feed can result in significant losses. Another indication of poor quality control in feed is a large percentage of "fines" in the bottom of a sack of pellets. The decay of these fines in ponds is often the forerunner of serious oxygen problems.

Considerable discussion has arisen over the relative merits of floating versus sinking feeds. It is important to indicate that red drum can and do feed naturally either on the surface or on the bottom. Many indirect benefits have been ascribed to feeding floating pellets. Major values claimed include: better food conversion, better regulation of amounts of feed, and better disease and parasite control because you can see the fish feed. The most important deterrent is the $30-$50 per ton added cost.

For the beginning fish farmer, all of these claims do have some merit. The deciding factor is, are you willing to pay extra for feed in order to see the condition of your fish daily. At least four major feed producers are marketing floating catfish feed in the state and new operators should consider using their feeds.

Feeding

After the type and kind of feed is selected there are still several decisions to be made. Most farmers feed their fish manually. For optimum growth, fish should be fed approximately 3 to 5 percent of their body weight each day until they reach one pound. The 3-percent rate is most widely used. Above one pound, 2 percent of their body weight is adequate. To determine the amount of feed required, some of the fish may be caught monthly or preferably biweekly. Use short seines for this purpose. About 100 fish should be weighed to get a good sample.

To calculate the amount of feed required, multiply the average weight of the fish caught times the number of fish in the pond times 0.03. The resultant figure is the feed needed daily for the next feeding period. Do not attempt to use ready-made feeding tables. They cannot foresee the events that will have taken place in your pond. At best, these short cuts will fail to assure maximum growth. At worst, they either starve the fish to death or kill them by overfeeding, causing an oxygen depletion. A short cut is to feed the fish what they will consume in 15 minutes. This works well for catfish and, although unproven, apparently is satisfactory for red drum.

Under the best conditions, fish will double their weight each month. Therefore, increasing the feed daily in large ponds should be considered. Remember: Calculation of a certain percentage of body weight does not mean this is exactly what the fish will eat. A continuous check must be made to assure that all the feed in consumed. We use metal underwater feeding trays with sinking feed and check them at the end of one hour to ascertain the food consumption rate. If food remains, a lower feeding rate should be used. If the trays are empty, a higher rate may be desirable. Certain other general rules should be followed:

- Do not feed in the rain as fish will normally not feed on the surface at that time.
- Cut feeding rate by 1/2 when water temperature exceeds 90°F. Growth rate declines and dissolved oxygen levels are critical at these temperatures.
- Cease feeding when more than four consecutive days of overcast weather have occurred. Oxygen levels are critical under these conditions. Feeding may be resumed after the first day of sunshine.
- Avoid feeding during periods of high humidity and/or low pressure. Oxygen tension is low and fish tend to be sluggish.
- Evening is the preferred time to feed during winter months and early morning feeding is preferred during the summer. If maximum growth is desired, split the feeding between late evening and early morning.
- Feed in waters less than 3 feet in depth. Deep waters tend to be oxygen-depleted and fish will not feed well.
- At least 10 percent (preferably 25 percent) of the pond area should be covered when feeding. This reduces competition and helps to assure all fish are fed and therefore fewer runts occur.
- Do not exceed 30 pounds of feed per acre per day during July, August and September. Higher feeding rates usually cause excessive oxygen depletion.
- Red drum do not feed well below 60 degrees. Feeding should be limited to two days per week and one percent of body weight. Feed then on warm, sunny afternoons. Remember fish can live for long periods of time without feed when temperatures are below 45 degrees.
- If fish stop feeding, determine the cause immediately and correct it. Don't resume feeding until the adverse conditions present are corrected.
- Do not feed for at least 24 hours before harvesting the fish. The fish will handle better with fewer stress problems.

Recently, considerable interest has been expressed in automatic feeding devices. Due to high labor costs this interest is commendable. At the same time, unattended feeding is extremely dangerous. An automatic time-scheduled machine cannot ascertain if fish are feeding. Therefore, it will continue to feed at a set time that may result in overfeeding and an oxygen depletion and loss of fish. Demand feeders can also be used but are subject to interference from ducks, turtles, raccoons, etc.

All in all, the best treatment for fish is keep them healthy. The best formula for this is (1) buy clean fish, (2) keep them clean, (3) feed them well, but not excessively, daily, (4) supply them with plenty of fresh water, (5) don't overcrowd.

Harvesting, Holding, and Hauling

Before you start to harvest you must locate a market. Fish are a highly perishable item and there can be a temporary over supply on the market. Therefore, harvest only when you have a market available.

What is needed for harvesting? The first necessity (unless the buyer is on the bank) is holding facilities. Earlier we mentioned holding ponds. The size of these ponds depends largely on the market available but a general guide is to have ponds large enough to hold at least the equivalent of two but no more than five truckloads. For small farmers catering to the local trade and using pickups to haul, 1/2-acre ponds are adequate. When the water is cool, fish can be held at rates of 10,000 pounds per acre. In warmer weather this is impossible and 4,000 pounds is probably the maximum.

Prior to hauling, the fish should be held off feed for at least 24 hours. This allows the gut to be emptied prior to loading. If available, the fish should be held in vats with running water and plenty of aeration. The vats can be made of wood or concrete. Each has certain advantages and disadvantages. Wood must be painted with a waterproof paint (lead-free) or coated with fiberglass to promote better cleaning and prevent rotting. Concrete troughs are tough and durable but must be painted and are undesirable as the fish tend to abrade or injure themselves very quickly. One way to prevent these abrasions is to line the troughs with polyethylene film. The use of holding ponds or vats goes a long way towards alleviating the press of time schedules for live fish deliveries.

When holding facilities are available, then it is time to begin harvesting. Here again the money spent to do a good job on construction means more profit to the grower. With good construction, two types of harvesting are available, partial and complete.

In partial harvesting, seines are used to encircle the fish. Usually the fish are fed to draw them into a small area. The numbers of fish needed dictates the length of the seine and/or the number of hauls required. The seines used vary in mesh size according to the size of fish being captured. For fish of 1 pound or larger, a 1-inch mesh seine works quite well. The seine should be made of nylon and 1/3 to 1/2 deeper than the water to be seined (i.e. for 3 foot deep water use a seine from 4 to 5-1/2 feet deep). The cork or float line should have floats at least every 2 feet. The bottom or lead line is usually of 30-40 strand jute twine. This type of bottom line lies close to the bottom contours and still does not sink into a soft bottom or cut into high ridges on the bottom. The use of trammel or gill nets for harvesting is discouraged because fish may be killed or injured.

Total harvest is usually accomplished by seining the entire pond and then drawing the pond down to complete the seining and harvesting. The seines used are similar to those described above except much longer, of course. Successful seining of a large pond is not a job for amateurs.

It is a lot of hard work. Most haul seines are now made to be pulled by wenches and/or farm tractors. Nonetheless, it is still heavy work. As the seine nears the bank, the fish must be removed from the water and placed in hauling containers.

A word of caution should again be inserted here. If a 20-acre pond is drawn down to the catch basin, particularly in hot weather, it becomes necessary to immediately provide holding space for 25,000 to 40,000 pounds of live fish. This can be a real problem. Decomposing dead fish are of little value on present-day markets.

Hauling tanks are made of fiberglass or over a wood frame. They come in various sizes and shapes. We will be glad to furnish a list of suppliers. Several features are desirable when buying or building new hauling tanks. There should be provisions for agitators (12 volt and 110 volt) with at least 1/4 of the agitator paddle and basket exposed.

There should be a bottom drain of sufficient size to permit passages of the largest fish to be hauled. The drain should have no sharp turns or corners and be free from projections that might injure the fish.

The bottom of the tanks should be sloped towards the drain. If possible the tank should have double-wall construction with the space between filled with insulating material. The tops of the tank should have vents for the release of foul air and air inlet scoops should be installed, if possible. In addition, provision should be made for the use of ice above or in the hauling tanks.

For the novice hauler, certain precautionary words are in order. It is generally agreed that water should be changed in hauling tanks every 24 hours. To eliminate potential ammonia and nitrite problems, well water with a high iron content (over 2 ppm) should be avoided, too. Chlorinated water will kill red drum.

Financial Analysis of Commercial Red Drum Aquaculture Enterprise

Raymond J. Rhodes
Industry Economist
Division of Marine Resources
South Carolina Wildlife & Marine Resources Dept.
P.O. Box 12559
Charleston, South Carolina 29412
and
Dewayne Hollin
Marine Business Management Specialist
Sea Grant College Program
Texas A&M University
College Station, Texas 77843-4115

This chapter discusses techniques for estimating major capital and operating costs to assist the potential investor in preparing a preliminary financial analysis of a commercial red drum (*Sciaenops ocellatus*) farming project. By applying projected cost and revenue information for a specific project, one will be able to estimate the approximate investment cost and profitability for a proposed red drum operation.

To facilitate understanding of the financial analysis methods used herein, output of a simple Lotus 1-2-3 (V2.01) spreadsheet model of a red drum hatchery and growout operation has been included (see appendices.) The coefficients and variables used in these models have been used throughout the text.

This only emphasizes one aspect of an overall business proposal (financial data and analysis). A comprehensive aquaculture business proposal contains many other elements, including market description, a discussion of marketing strategy and a management plan (Rhodes, 1983). Due to recent interest in starting small businesses, there are many good publications on preparing formal business plans (e.g. Owen *et al.*,1986 and Osgood, 1983). In addition, these references often contain information on locating and raising start-up capital.

Background Information

Four Important Factors for Success

As in commercial shrimp farming (International Financial Corporation, 1987), four factors critical to the success of a commercial aquaculture venture need to be considered prior to initiating a detailed financial appraisal: Site selection, management, availability of fingerlings, and project design. As other chapters have indicated, the importance of site selection cannot be overemphasized.

Further, the success of any organization is always dependent on its technical and management personnel. Managers with experience in other aquaculture enterprises (e.g. catfish or shrimp farming) may need to be recruited. In addition, a commitment by local education institutions (e.g. colleges, vocational education programs) to train and/or upgrade aquaculture technicians might be considered when evaluating a site.

Availability and quality of red drum fingerlings are essential to any commercial growout operation. Consequently, the decision to initially purchase fingerlings versus starting a red drum hatchery operation will need to be addressed. If a hatchery operation is to be started, then the availability of red drum broodstock will be crucial.

Last but not least, the farm's design and construction management should involve professionals (e.g. engineering firms) experienced in the pre-qualification of contractors and construction supervision. Professional support and close project monitoring can not only facilitate timely completion of construction, but can lead to significant savings in capital and operating costs. These professionals may also be critical in mitigating serious delays in the site permitting process.

Critical Performance Factors

In this chapter, production of red drum fingerlings will be based upon methods proposed by Hopkins (page 170) and Stokes and Smith (page 178) using a three-phase system. These semi-intensive production techniques require relatively high stocking densities, continuous feeding, and a high rate of water exchange. This results in high poundage yields per unit of land compared to extensive cultivation in wetland impoundments.

Table 1. Performance Characteristics for a 2-acre Red Drum Hatchery Operation

Fingerling Ponds:	
Stocking Density, fry per acre:	500,000
Duration of Fingerling Production Cycle, days:	45
Projected Survival Rate:	25 percent
Size of Fingerling Ponds, acres:	1/2
Number of Fingerling Ponds:	4
Total Fingerling Ponds Acreage:	2
Projected Yield, number of fingerling/acre:	125,000

The major financial assumptions for the hatchery portion of the hypothetical commercial red drum aquaculture enterprise include the following:

Depreciation Treatment: Modified Accelerated Cost Recovery System with no IRS Section 179 deductions.

Income Tax Rate:

Federal: Corporate rate effective after July 1, 1987

State: 6 percent of net income

Type of Capital (Debt to Equity Ratio): 100 percent equity with opportunity costs set at 10 percent.

Inflation Rate: 3 percent compounded annually after year 1

Land Costs: $3,000 per acre for 4 acres

Table 2. Example of Performance Characteristics for a Hypothetical Red Drum Semi-intensive Farm in South Carolina.

Yearling Ponds:	
Stocking Density, fingerlings per acre:	5,882
Duration of Yearling Cycle, months:	10
Projected Survival Rate:	85.0 percent
Size of Yearling Ponds, acres	5.0
Number of Yearling Ponds:	6.0
Total Yearling Pond Acreage:	30.0
Projected Yield, number of fish/acre:	5,000
Growout Ponds:	
Stocking Density, yearlings per acre:	1,250
Duration of Growout Cycle, months:	10
Projected Survival Rate:	95.0 percent
Size of Growout Ponds, acres:	20.0
Number of Growout Ponds:	6.0
Total Growout Pond Acreage:	120.0
Average Harvest Weight, pounds:	3
Projected Yield, pounds per acre:	3,563
Other Characteristics:	
Feed Conversion Ratio (FCR)*: (Pounds of Feed: Pounds of Live Red Drum)	2.5
Average Paddlewheel HP/Growout acre:	1.0
Average Paddlewheel HP/Yearling acre:	2.0

*The FCR is projected to decline 0.1 per year after Year 1 until it reaches 2.0 in Year 6.

Table 3. Major Financial Assumptions for a Hypothetical Semi-intensive Red Drum Farm in South Carolina.

Depreciation Treatment: Modified Accelerated Cost Recovery System (MACR) (No IRS Section 179 deduction was used)

Income Tax Rate:
Federal: Corporate rate effective after July 1, 1987
State: 6 percent of net income
NOTE; Alternative Minimum Taxes, Net Operating Losses and other tax treatments affecting corporate income taxes were not considered in this analysis.)

Type of Capital (Debt to Equity Ratio): 100 percent equity with opportunity costs set at 10 percent (Discount Rate)

Inflation Rate: 3 percent compounded annually after Year 1

Land Cost: Leasing at $75 per acre per year.

Basic performance characteristics are needed to make an initial evaluation of the proposed project's profitability. These parameters are critical because the directly influence both start-up (e.g. capital) and operation costs. For example, selection of the survival rates in the growout phase will affect operating expenses (by affecting the fingerlings needed, and hence the hatchery capacity), and revenue (by the total harvested pounds).

Performance characteristics for a hatchery and growout operation will be discussed separately.

Hatcheries

The hatchery consists of 1) a spawning and hatching facility to produce redfish larvae and 2) a small pond system to grow 2- to 3-day-old larvae into 2 inch fingerlings in a 45-day period using the fertilized pond method (see Colura, page 78). The pond component of the hatchery represents Phase I of the three-phase production system. Keys to success of this phase include: 1) good predator control in fertilized ponds; 2) stocking fry at the proper state of the zooplankton bloom; and 3) preventing low dissolved oxygen levels.

Predator control is achieved by sun-drying the pond between harvests of fingerling crops, and filtering—with fine-mesh filter bags—to remove waterborne predators when refilling. Larvae will be stocked between 10 and 14 days after initial pond filling and fertilization to ensure optimum zooplankton densities. Dissolved oxygen is maintained above 3 ppm by controlling fertilization rates, water exchange and aeration, as needed.

Critical performance characteristics for this type of hatchery include stocking density, length of production cycle, survival rate, pond size, total production acreage, and projected yield. Table 1 provides the parameters used during the fingerling production analysis.

Major financial assumptions include depreciation schedule, tax treatment, capital structure, inflation rate and land costs.

Grow out

Growout production is based on Phases II and III of the three-phase system: a) Phase II, stocking of red drum fingerlings in yearling (nursery) ponds; and b) Phase III, transferring 12-month-old red drum into characteristics for this type of system (Table 2) are used for this growout analysis along with other assumptions listed in the Appendices. These parameters will be used during the growout analysis illustrated in this chapter along with other performance assumptions listed in the chapter's Appendices.

Major financial assumptions are provided in Table 3. The internal rate of return (IRR) was calculated with all end-of-period cash flows. Equipment, pond construction costs and other capital expenditures were depreciated (Table 3) using the current Internal Revenue Service (IRS) depreciation rules under the Modified Accelerated Cost Recovery System (MACRS), which generally applies to all property placed in service after Dec. 31, 1986 (IRS, 1986). Estimated income taxes were calculated using new tax rates effective for corporate tax years starting on or after July 1, 1987, and the S.C. Tax Commission's corporate income tax rate of six percent.

Precautions and Limitations

Techniques used in this chapter do not include detailed cash flow analysis of the growout operation. Any such analysis should include monthly projected cash flow statements for at least the first two years and projected quarterly cash flow statements for the first three years or longer (Rhodes, 1984).

Because commercial red drum techniques are new and rapidly changing, current state-of-the-art knowledge about red drum farming could soon be obsolete. Growout techniques, hatchery technology, feed formulations and marketing strategies have not, in many cases, been subject to the test of the private

Table 4. Estimated Initial Capital Investment for a Proposed 4-acre Red Drum Hatchery Facility with 4 1/2-acre Fingerling Ponds.

	Quantity		Total Cost
Land (acres)	4	$3,000/acre	$12,000
Pond Construction			
1/2-acre ponds	4	$3,500/pond	$14,000
Hatchery			
Building	1,800 sq ft	$25/sq ft	$45,000
Parking Lot	1/4-acre gravel		$ 1,000
Equipment			
Major Hatchery Equipment (Appendices Table A1)			$32,100
Misc. Furniture/Equipment			$ 2,500
Broodstock (10 fish at $125 each)			$ 1,250
		Total Capital Costs	$107,850

sector. Consequently, physical performance characteristics and production methods described herein should be considered preliminary. Moreover, many of the physical parameters are interdependent. For example, the harvest weight of a given fish is a function of several interrelated factors, including stocking density, growout days and expected feed conversion. The quantitative relationship between these many factors, especially for commercial scale operations, is unknown.

Hypothetical Red Drum Hatchery

Determining Capital Costs

The total estimated capital costs for the 4-acre red drum hatchery are $107,850 (Table 4).

Facility description

Hatchery operation is integrated into the larger growout farm. The facility is comprised of a hatchery building, containing two brood tanks, three incubators and other equipment listed on the capital equipment list, and four 1/2-acre fingerling production ponds nearby. As the facility is totally integrated, some of the pumping equipment used for the large ponds is also used for fingerling production.

The hatchery building (Figures 1 and 2) is constructed of concrete blocks on a concrete slab and contains 1,800 square feet of overall space. The building has insulation blown inside the blocks and the various rooms are partitioned and insulated. Each of the two brood tank rooms is 18 by 18 feet and contains one 12 foot diameter by 5 foot deep brood tank. Three incubators are located in the 17 foot by 18 foot incubator laboratory. There is also a 17 foot by 18 foot laboratory area for supplies, equipment and analytical work space. The additional 540 square feet is used for office space, restrooms, hallways and closets. Cost of the building, including insulation, air conditioning/heating, plumbing and interior pre-finishing is $25 per square foot, or $45,000. Adjacent to the hatchery is a gravel parking lot for 20 cars built for $1,000. The facility is located on a total of 4 acres (2 surface acres for fingerling ponds) valued at $3,000 per acre.

Four one-half acre fingerling production ponds are located adjacent to the hatchery. The ponds have a 1- to 2-percent slope to facilitate complete drainage and predator/competitor control before the pond is fertilized and filled. The ponds have 10-inch pipes for filling and draining, and a concrete harvesting basin. Ponds can be drained in 8 hours or less, and filled in 24 hours or less. When not in use for fingerling production, ponds may be used for holding fish for market or restocking for yearling grow out.

The four fingerling ponds could be built for a cost of $3,500 each, including drains, piping and concrete structures, for a total of $14,000.

Estimating hatchery costs

In addition to the construction cost of the building, parking lot and four fingerling ponds, the hatchery contains $32,100 of equipment. Hatchery equipment is shown in Table A1 (Appendix) with purchase price and economic life.

Miscellaneous office furniture and other equipment is valued at $2,5000, with an economic life of 10 years. The estimated initial capital investment for the entire hatchery facility is summarized in Table 4 and the associated depreciation in Table A2 of the appendices.

Determining Operating Costs

Hatchery operating costs were established for each of two production scenarios, one conservative (2 fingerling crops), and one ambitious (3 fingerling crops).

Scenario I: conservative production

The hatchery plans to induce the broodstock to initiate spawning in late March to produce 2 million eggs. After spawning the required eggs, the brood-

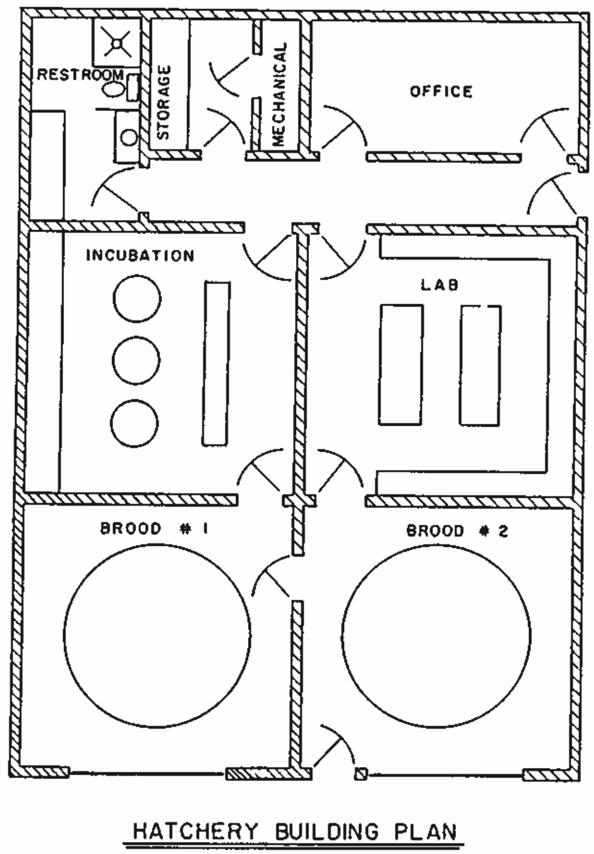

Figure 1. Plan view of 1,800-square foot red drum hatchery.

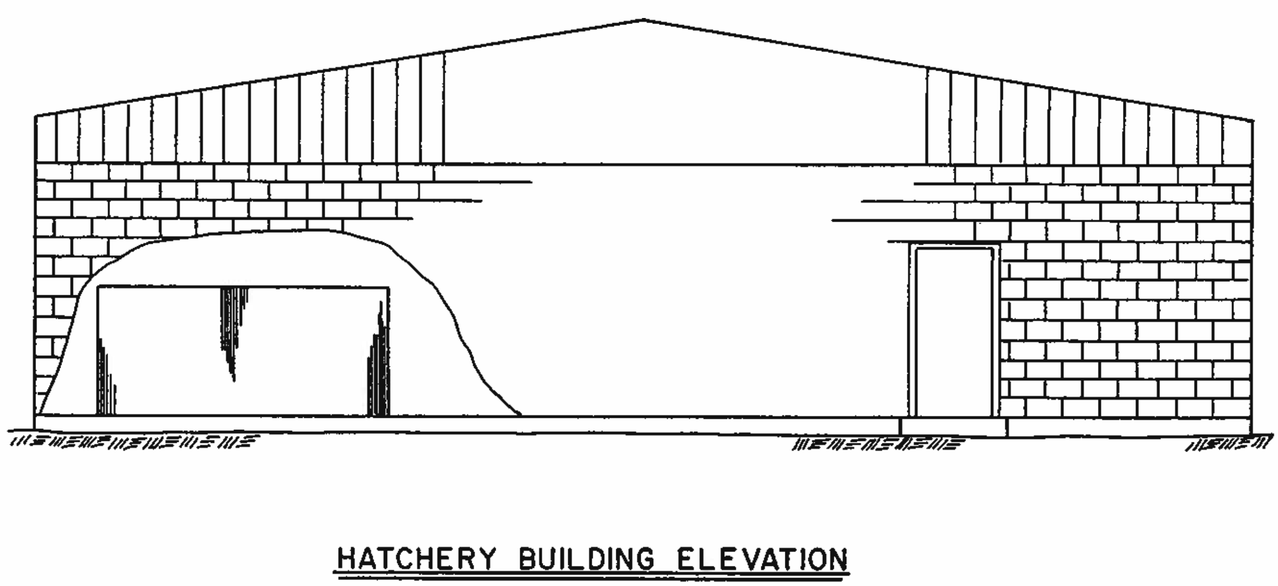

Figure 2. Elevation view of 1,800-square foot red drum hatchery.

stock would be maintained in a reproductive mode until the second cycle by periodically raising and lowering temperatures within the brood tank (as described by Roberts in Section II). The eggs produced in the first cycle would be incubated to produce 500,000 ready-to-feed fry per acre or a total of 1 million fry for stocking all four fingerling ponds. Based on fingerling production research, this stocking should produce about 250,000 2-inch fingerlings in 45 days for stocking in Phase II (yearling production). This exceeds the fingerling requirements of the larger 150 total acre farm of 175,000 fingerlings for six 5-acre yearling ponds by 75,000 fingerlings. These additional fingerlings would be sold to other farmers or for recreational stocking. The charge-off to the integrated-system farm would be \$0.08 per fingerling or \$14,000. The price for excess production would be \$0.15 F.O.B.-hatchery per fish, or \$11,250. This results in a total income to the hatchery of \$25,250.

The second spawning cycle, in late May, would yield an additional 2 million eggs to create 1 million ready-to-feed fry for stocking the fingerling ponds at 500,000 per acre, or 250,000 per 1/2-acre pond. This would produce a second crop of 250,000 fingerlings for general market distribution in July at a price of \$0.15 F.O.B.-hatchery per fish, or a total of \$37,500.

Total income to the hatchery in Scenario I would be \$62,750 for two crops between late March and mid-July.

Scenario II: maximum production

The hatchery would cycle the broodstock for a first spawn in late March to produce 2 million eggs to be incubated to produce about 1 million ready-to-feed fry for a stocking of 250,000 in each 1/2-acre fingerling pond. The 45-day fingerling growout period ending the first week of May would produce about 125,000 2-inch fingerling per acre, or 250,000 total.

As in Scenario I, 175,000 fingerlings would be used to stock the six 5-acre yearling ponds and the remaining 75,000 would be sold at \$0.15 per fish, or \$11,250. The growout facility would be charged \$0.08 per fish for the 175,000 stocked in yearling ponds, or \$14,000.

Again, a second spawn would be cycled for the first week in May to produce 2 million eggs to incubate in 48 hours into 1 million ready to feed fry for stocking in fingerling ponds at 500,000 per acre, or 250,000 per 1/2-acre pond. This would produce a second crop of 250,000 2-inch fingerlings in mid-July for distribution at a price of \$0.15 F.O.B.-hatchery per fish, or \$25,000.

Total income to the hatchery in Scenario II would be \$87,750 for three crops of fingerling between late March and early October. (Appendix Table A3).

Scenario II: Details of Analysis

As the hatchery is totally integrated into the redfish growout operation to provide redfish fry for stocking, we set this as a number one objective. The number two objective is to generate positive operating income to make the hatchery operation self-supporting. To accomplish both of these goals, we elected to work with Scenario II with three crops per year of 2-inch fingerlings. The 10-year financial analysis, including projected income statement, a discounted cash-flow analysis and depreciation schedule can be found in the appendices (Tables A3 and A4).

Direct operating expenses

Feed for broodstock, chemicals and fertilizer for fingerling ponds, and harvest labor cost are the major direct operating expenses. Feed and fertilizer costs are based on industry estimates, while harvest labor is determined by hourly wage figures used in the growout operation. In the 10-year projections, costs are increased at 3 percent each year.

Other operating expenses

All other operating expenses are set to increase at the 3 percent inflation rate. First-year costs are basic redfish hatchery operations estimates, for a 2-surface-acre hatchery including maintenance, pumping costs, utilities, salaries, transportation, depreciation, insurance, taxes and miscellaneous.

Projected Income and Financial Analysis

Projected profit and loss statement

Annual profit and loss projections are based on revenue and expenses estimates. The three-crop hatchery operation produces gross sales of \$87,750 in years one through four; \$96,525, years five through eight; and \$106,177, years nine and 10. (Production could be increased further by increasing the stocking rates, increasing the survival rate or enlarging the facility.)

In one year, net operating income before taxes is estimated at \$32,661. The combined state and federal tax rate of 21 percent reduces this to a net income after taxes of \$25,802.

Further definitions for major headings in the income statement are covered in the growout operation segment.

Comments on cash flow

Cumulative annual cash flow becomes positive during the third year; however, if evaluated from the standpoint of the time-value of money (cumulative net present value), positive cash flow does not occur until the fifth year. Annual net cash flow without consideration for the capital costs in year one is positive, but not large enough to offset the capital costs until the fifth year.

Financial analysis

Results of two basic techniques for analyzing profitability of a commercial enterprise—discounted payback period (DPP) and internal rate of return

(IRR) — will be briefly examined using the hypothetical red drum operation as and example. DPP, the number of years needed to recover the original investment in terms of discounted net cash flow, is a measure of relative liquidity (Bhandari, 1986). It should be used instead of the traditional (non-discounted) payback period (Rhodes, et al., 1987). For the hatchery operation evaluated here, the projected DPP, discounted at 10 percent, is about five years.

The IRR is the discount rate at which, over a specified time period, the present value of projected net cash flows equals the present value of invested capital; i.e., the discount rate at which the Net Present Value is zero. It is currently perhaps the single most common financial tool used to evaluate the relative attractiveness of competing investments. In general, the higher the IRR, the better the investment[1]. (More specifically, it should exceed the investor's cost of capital.)

The projected IRR for the hatchery operation was calculated for five- and 10-year periods. The five-year IRR, 28.9 percent, and the 10-year IRR, at 43.8 percent (Table A4 in appendix), are both relatively high for the project.

Growout Component

Determining Capital Costs

Capital costs for a growout operation with earthen ponds are strongly dependent upon pond size and number. Pond size, along with stocking densities, not only affect pond construction costs, but also pumping and aeration requirements. Pond construction, pumps and aerators may constitute more than 60 percent of a growout operation's capital costs (see Appendix Table A8).

Facility Description

The growout facility consists of six 5-acre yearling ponds (Phase II) and six, 20-acre growout ponds (Phase III). Each yearling pond is square and approximately 470 feet on a side with an average water depth of 4 feet. Each growout pond is also square and is 940 feet on a side with an average water depth of 4 feet.

Earthmoving costs, the largest single component of capital costs, are significantly affected by pond layout and design. Dike volume and soil shrinkage factors are major determinants of earthmoving requirements. Some dikes should be wide enough to accommodate harvesting trucks so that each pond is accessible for stocking, feeding and harvesting.

Daily pumping requirements and the type of pumping system are site-specific; they largely depend on the vertical distance the water must be lifted, local tidal conditions and stocking densities. In this analysis, two 50-hp axial-flow pumps capable of collectively delivering 14,000 gpm (with a total dynamic head of 17 feet) were selected based upon estimates of pumping capacity for about 150 acres (see Table A10 in appendix).

In the present case, water is pumped from an estuarine creek and then gravity-fed into distribution canals that also serve the hatchery. Lateral inflow and drainage (outflow) are designed for a 5 percent water exchange per 12 hours, with the system capable of exchanging up to 25 percent of one 20-acre pond's water within 12 hours. Flashboard risers would be used in both the intake and outflow pipes in each pond.

Approximately two 10-hp paddle wheel aerators would be used in each 20-acre pond, and two 5-hp paddle wheels in the 5-acre yearling ponds. Paddle wheel aeration requirements were estimated on the ratio of one paddle wheel-aerators-horsepower per growout-acre and two per yearling-acre (Table 2). Other equipment requirements would be similar to those reported for marine shrimp farming in the United States (e.g. Rhodes, et a;., 1987), except for the harvesting equipment. Harvesting equipment would be similar to those used in U.S. catfish farming.

Estimating costs

Construction and equipment costs can be estimated using information from several sources, including average statewide (i.e. South Carolina) costs provided by the Soil Conservation Service, U.S. Department of Agriculture (1986), used equipment dealers, equipment manufacturers, contractors and other aquaculture enterprises in the United States.

Earthmoving costs were based on 160,000 yd^3 at $\$1.20/yd^3$. Though satisfactory for this preliminary analysis, when preparing a detailed recommended; even a small difference in earthmoving and/or unit costs (e.g. heavy equipment operator hours) may lead to substantial differences in total earthmoving costs. (Actual bids may be based upon equipment use and operator man-hours, instead of costs per yd^3.

Determining Operating Costs

Determination of operating costs is especially important to a red drum aquaculture project because no revenue will be generated from grow out until the second year of operation. Feed and salaries will generally be the two largest expenses of the growout operation (Table 5).

Feed and fingerling costs

For preliminary cost estimates, the feed cost per pound of red drum harvested can be calculated by multiplying food cost by the feed conversion ration (FCR). Since research and experiments to improve the quality and efficiency of red drum feed are ongoing, estimated feed costs are tentative at best.

Feed cost estimates in Year 1 were based upon the total yearling harvest (i.e. pounds). In Year 2, the total

[1]The IRR should be used with caution.

Table 5. Preliminary Projected Income Statement for a Hypothetical Commercial Redfish (Red Drum) Farm (First 10 Years)

Projected Total Harvest & Yields:	1st	2nd	3rd	4th	5th	6th	7th	8th	9th	10th
Total Harvest, whole lbs. in 1,000's:	0	427.5	427.5	427.5	427.5	427.5	427.5	427.5	427.5	427.5
Average Yield, Whole lbs. per Acre:	0	3,563	3,563	3,563	3,563	3,563	3,563	3,563	3,563	3,563
Gross Sales: (in thousands)										
Redfish (Red Drum), whole fish:	$0.0	$102.6	$102.6	$102.6	$102.6	$102.6	$102.6	$102.6	$102.6	$102.6
Redfish (Red Drum), processed fish:*	0.0	481.5	481.5	481.5	481.5	481.5	481.5	481.5	481.5	481.5
Total Gross Sales:	$0.0	$584.1	$584.1	$584.1	$584.1	$584.1	$584.1	$584.1	$584.1	$584.1
Direct Operating Expenses: (in thousands)										
Fingerlings (if purchased)	$14.1	$14.1	$14.1	$14.1	$14.1	$14.1	$14.1	$14.1	$14.1	$14.1
Feed, yearlings & growout fish	48.8	133.4	127.8	122.3	116.7	111.2	111.2	111.2	111.2	111.2
Processing	0.0	30.1	30.1	30.1	30.1	30.1	30.1	30.1	30.1	30.1
Fertilizer	1.0	4.7	4.7	4.7	4.7	4.7	4.7	4.7	4.7	4.7
Total Direct Expenses:	63.8	$182.3	$176.7	$171.2	$165.6	$160.1	$160.1	$160.1	$160.1	$160.1
Gross Profit:	-$63.8	$401.8	$407.4	$412.9	$418.5	$424.0	$424.0	$424.0	$424.0	$424.0
Other Operating Expenses (30% Inflation/Year) (in thousands)										
Salaries, Manager & Technicians	$72.4	$74.6	$76.9	$79.2	$81.5	$84.0	$86.5	$89.1	$91.8	$94.5
Electricity	10.7	56.4	58.1	59.8	61.6	63.5	65.4	67.4	69.4	71.5
Wage Labor 24 man-hrs/pond	1.0	2.5	2.6	2.7	2.7	2.8	2.9	3.0	3.1	3.2
Land Leasing Fees	13.5	14.0	14.4	14.8	15.2	15.7	16.2	16.7	17.2	17.7
Total Depreciation	74.5	126.7	89.5	67.1	67.1	61.5	61.5	15.8	0.0	0.0
Business Insurance	10.0	10.3	10.6	10.9	11.3	11.6	11.9	12.3	12.7	13.0
Professional Fees	15.0	5.0	5.2	5.3	5.5	5.6	5.8	6.0	6.1	6.3
Licenses & Property Taxes	4.0	4.1	4.2	4.4	4.5	4.6	4.8	4.9	5.1	5.2
Car & Truckl Expenses	6.0	6.2	6.4	6.6	6.8	7.0	7.2	7.4	7.6	7.8
Miscellaneous Expenses	5.0	5.2	5.3	5.5	5.6	5.8	6.0	6.1	6.3	6.5
Pond & Other Maintenance	10.0	10.3	10.6	10.9	11.3	11.6	11.9	12.3	12.7	13.0
Total Other Operating Expenses:	$222.1	$315.3	$283.7	$267.1	$273.1	$273.7	$280.1	$240.9	$231.9	$238.8
Total Direct & Other Expenses:	$285.9	$497.6	$460.4	$438.3	$438.7	$433.8	$440.2	$401.0	$392.0	$398.9
Net Operating Income Before Tax	-$285.9	$86.5	$123.7	$145.8	$145.4	$150.3	$143.9	$183.1	$192.1	$185.2
Estimated Income Taxes:**	$0.0	$22.9	$38.9	$48.9	$48.7	$50.9	$48.0	$65.6	$69.7	$66.6
Net Operating IncomeAfter Tax:	-$285.9	$63.7	$84.8	$96.9	$96.7	$99.4	$95.9	$117.5	$122.4	$118.6

* The processing sales are based upon a 12% loss in round (whole) weight of the fish.
** Estimated income taxes donot include alternative minimum taxes and other taxes applicable to corporations.

poundage of yearlings raised in Year 1 was subtracted from that harvested in Year 2:

Year 1: Total Feed Expenses=$Yn^{x\ FCR\ x\ P}$

Year 2: Total Feed Expenses=

$[(G_n\text{-}Y_{n-1})+Y_n] \times FCR \times P$

where,

Y-Total Yearling Harvest, lbs.
G-Total G.O. Harvest, lbs.
P-Feed price, $/lbs.
n-Current year of operation.

(The Year 2 calculation is repeated for all subsequent years.)

In this example, the price of fingerlings, $0.08/fish, was based upon the cost of purchasing them from the parent company's hatchery.

Salaries and wages

An experienced farm manager is needed for the growout operation. For this example (Table 5), the manager and technicians would be hired in Year 1. Wages were based upon 200 man-hours at $5.00/hr in Year 1, and 24 man-hours per pond per year with 1,000 man-hours/year for all other years.

Other operating costs

Electricity cost (assuming electric motors are used on the pumps and aerators) is usually the second highest expense. Estimating electricity expenses will generally depend upon electrical consumption of the pumps and aerators kilowatt hours (KWH) used per unit, total operating time per year, and average cost per KWH (Appendices).

Other significant expenses (Table 5) include leasing, professional feed (e.g. lawyers, accountants, technical consultants, etc.) and maintenance.

Working capital requirements

Working capital approximates the difference between the inflow of revenue from sales and the outflow of cash for operating expenses for the start-up period, when cash operating costs are greater than revenues. (In practice, working capital is included in the capital budget during start-up and subsequently classified as expenses on the operating budget.)

Working capital was not estimated in this example, but it may be approximated as the sum of all operating expenses less depreciation.

Interest expenses incurred during construction, are also included as a capital expense. This project assumes the perhaps unlikely case of 100 percent equity-financing; there is consequently no interest expense.

Determining Gross Sales

It is assumed that all of the sales by the growout operation will be derived from the sale of red drum to retailers or wholesalers. In addition, a processor will prepare about 80 percent of the red drum total harvest each year (Appendices Table A5) as heads-on, gutted fish. To calculate the gross sales of processed red drum, the ex-processing plant price per pound, $1.60, should be multiplied by the net weight (heads-on and gutted) of the red drum after processing. The whole price, $1.20/lb, should be multiplied by total weight of red drum sold in the whole form. These prices are assumed to F.O.B. ex-pond (or ex-processing plant); therefore, no shipping cost has been included. (In fact, shipping and other marketing costs can be significant expenses and should be included when projecting growout operating costs.)

Projected Income and Financial Analysis

Projected annual income statement

Once expenses and revenues have been estimated, a projected income statement (profit and loss statement) can be prepared to forecast annual financial performance. The income statement (Table 7) used in this chapter represents projected current and cumulative earnings of the grow out for a given operating period. The categories used in this growout operation (Table 5) are explained as follows:

- *Total gross sales:* Revenues generated from the sale of red drum. It is assumed that all the red drum reaching marketable size in a given year are sold in that year. This may **not** be the case in an actual operation.
- *Direct operating expenses*: Variable costs that vary directly with the level of production (for accounting purposes, these may be considered as the "Cost of Goods Sold").
- *Gross profits* (gross sales minus direct operating expenses): Represent the gross profit on sales before other expenses.
- *Other operating expenses*: Usually, costs that must be met no matter what the actual stocking density. (In actuality, the stocking density will directly influence electricity consumption.) This category would include interest expenses, although, in typical income statements, interest expenses are usually listed as "other expenses" in order to highlight the cost of capital (Osgood, 1983).
- *Net operating income before taxes*: Total gross sales minus total direct and other expenses.
- *Estimated income taxes*: This is the estimated state and federal income taxes on the operations income. From an accounting standpoint, these taxes may not be completely paid in the year incurred but in a later year. (Consult an accountant regarding actual payment timing and other income tax considerations for your proposed business organization.) In this example, taxes were estimated based on a corporation in South Carolina.
- *Net operating income after taxes*: Net operating income before taxes minus estimated income taxes. This represents current profits (or losses) generated by the operation, including depreciation as an expense.

Table 6. Sensitivity of the 10-year Projected Internal Rate of Return (IRR) and Discounted Payback Period (DPP) to Changes in Performance Characteristics for a Hypothetical Red Drum Farm in South Carolina (see Table 2).

Variable		
Growout Survival Rate	**IRR (percent)**	**DPP**
98 percent	15.8	7.9 years
**95 percent	14.3	8.6
92 percent	12.8	9.4
Red Drum Processing Price		
$1.70/pound	17.6	7.3
**1.60/pound	14.3	8.6
1.50/pound	10.7	+10.0*
Feed Cost		
$0.17/pound	11.8	10.0 years
**0.15/pound	14.3	8.6
0.13/pound	16.7	7.6

*The calculation of the DPP differs slightly from the IRR due to time difference in calculating the discounted cash flow.
**Base case used in the text.

For the base scenario used in this chapter, the operation, as might be expected, does not generate profits until Year 2. This income statement is considered realistic because the possibility of underestimating expenses has been partially avoided by inflating future expenses (at 3 percent per annum). Be careful not to overestimate gross sales.

Financial analysis

For the growout operation, the projected DPP at 10 percent is less than 10 years when the ex-processor price of red drum is greater than $1.50 per pound (Table 6).

The projected IRR for the growout operation was calculated using an internal rate of return function in the microcomputer program for five-year and 10-year periods (Table 6).

Sensitivity analysis

Sensitivity analysis assesses the effect of changes in key performance parameters—such as feed costs, interest rates, wages and sales price—on variables such as the profitability indices used herein—discounted pay-back period and the internal rate of return. In this way, the more critical biological and financial aspects of the project may be identified and given special attention in both the planning and management phases.

Results for this hypothetical red drum growout operation (Table 6) indicate that the projected IRR is more sensitive to relative changes in red drum survival rates and wholesale prices than the cost of feed. For example, a 13 percent increase in the projected feed price would reduce the projected IRR about 2.5 percent, while only a 6 percent decrease in red drum survival would result in a 3.6-percent decline, nearly three times the effect of increased feed costs on the projected 10-year IRR.

Integration of a Hatchery and Growout Operation

Integration of a hatchery and growout operation within one organization could yield several benefits, including an assured and continuing source of fingerlings, improved capability to manipulate broodstock sources, more flexibility to adjust marketing strategies as the demand for fingerling and adults change, and the general pooling of resources within one organization. These potential benefits will need to be considered relative to the potential disadvantages, including the possibility that the growout operation could become a major cash flow drain on the hatchery operation during the early critical years of operations, and the likelihood of a supply-induced decline in fingerling prices in the future.

A preliminary analysis would suggest that aquaculture entrepreneurs might consider a strategy of first starting a hatchery operation before red drum fingerling supplies cause major price declines. [In addition, there is apparently a latent demand for red drum fingerlings and/or yearlings by recreational fee fishing operations in Texas and other states.] After starting the hatchery, a growout operation could be considered. Key factors in such a decision include: Market price trends for red drum, advances in growout culturing techniques and lower-cost feeds. In contrast, the U.S. fingerling market may become saturated very rapidly; consequently, a hatchery could become a significant financial liability within a short period of time.

Literature Cited

International Finance Corporation. 1987. Marine Shrimp Farming; a guide to feasibility study preparation. Aquafood Business Associates, Charleston, S.C., April 1987, 90 pp.

Bhandari, S.B. 1986. Discounted payback: A criterion for capital investment decision. J. **Small Business Mgmt.** 24(2):16-22.

Rhodes, R.J. 1987. A preliminary financial analysis of semi-intensive penaeid shrimp farming in South Carolina. **S.C. Marine Res. Ctr. Tech. Rpt.** No. 64, Charleston, S.C., April, 1987, 23 pp.

Osgood, W.R. 1983. Planning and financing your business: A complete working guide. **Inc./CBI Publications**, Boston, Mass., 260 pp.

Owen, R.R., D.R. Garner and D.S. Bunder. 1986. John Wiley and Sons, Inc., N.Y., 298 pp.

Internal Revenue Service. 1986. Tax guide for commercial fishermen. I.R.S., U.S. Dept. of Treasury, Publ. 595 (Rev. Oct. 86), 48 pp.

Rhodes, R.J. 1983. Primer on aquaculture finances: Planning for success, Part 1. **Aquaculture Magazine**, Nov.-Dec., 1983:16-20.

Weston, J.F. and E.F. Brigham. 1978. Managerial finance. The Dryden Press, Hinsdale, Ill. (6th edition), 1,030 pp.

Appendices

This material is designed to provide illustrative information only. If accounting advice or other expert assistance is required, the services of a professional person should be sought.

Table A1. Projected Hatchery Equipment Cost and Economic Life, 1987

Quantity	Item Description	Total Cost	Economic Life
1	Water storage tank (12,000 gal. capacity)	$3,000	10 years
2	Fiberglass brood tank (12 dia. x 5 depth)	3,000	10 years
2	Biodisc filters with stand	5,600	7 years
3	Incubators, 500 gal. CB tank	2,400	7 years
2	Fiberglass collection filters	500	7 years
2	Egg collectors	300	7 years
1	Sand filter HRV-24 w/4 way valve	500	7 years
3	Sterilizer, 50 gal/min UV	$1,100	7 years
1	Blower, 1 HP	500	7 years
2	Heaters for brood tanks	150	5 years
2	Chillers for brood tanks	2,000	5 years
1	Water pump, 3/4 HP	350	5 years
1	Shop Equipment/tool set	500	3 years
1	Oxygen meter and chemistry kit	700	3 years
1	Transport tank, fiberglass 1,000 gal fitted on gooseneck trailer	7,500	5 years
20	Seines/dip nets	500	2 years
	Pumping (pipes, valves and fittings)	2,000	7 years
1	Laboratory table with sink	1,500	7 years
		Total — $32,100	

Table A2. Depreciation Schedule for a Hypothetical Red Drum Hatchery (First 10 Years), 1987.

Year	1st	2nd	3rd	4th	5th	6th	7th	8th	9th	10th
31.5 Year Property: Hatchery Bldg. $45,000										
Depreciation (SLM)	$1,125	$1,125	$1,125	$1,125	$1,125	$1,125	$1,125	$1,125	$1,125	$1,125
7-Year Property: Pond Constr., $14,000 Parking Lot, $1,000 Hatchery Equip., $20,400										
7-Year Cost: $35,400										
Depreciation (DDB)	$5,062	$8,673	$6,195	$4,425	$3,151	$2,266	$1,628			
Depreciation (SLM):	$4,548	$4,548	$4,458	$4,458	$4,458	$4,458	$4,458			
5-Year Property: Office Equip., $2,5000 Hatchery Equip., $11,700 Broodstock, $1,250										
5-Year Cost: $15,450										
Depreciation (DDB)	$3,090	$4,929	$2,990	$1,792	$1,046					
Depreciation (SLM)	$2,791	$2,791	$2,791	$2,791	$2,791					
Total Cost: $95,850										
Total Depreciation (DDB):	$8,152	$13,602	$9,185	$6,217	$4,197	$2,266	$1,628			
Total Depreciation (SLM):	$8,464	$8,464	$8,464	$8,464	$8,464	$5,673	$5,673	$1,125	$1,125	$1,125

Table A3. Projected Income Statement for a Hypothetical Red Drum Hatchery (First 10 Years)

Projected Harvest/Yields:	1st	2nd	3rd	4th	5th	6th	7th	8th	9th	10th
Total Harvest, x 1,000*	750	750	750	750	750	750	750	750	750	750
Yield, fingl/A x 1,000	375	375	375	375	375	375	375	375	375	375
Gross Sales										
Red Drum Fingerlings	$87,750	$87,750	$86,750	$87,750	$96,525	$96,525	$96,525	$96,525	$106,177	$106,177
Direct Operating Expenses										
Feed	$1,400	$1,442	$1,485	$1,530	$1,576	$1,623	$1,672	$1,722	$1,733	$1,827
Chemicals/Fertilizer	$2,700	$2,781	$2,864	$2,950	$3,039	$3,130	$3,224	$3,321	$3,420	$3,523
Harvest Labor	$14,400	$14,832	$15,277	$15,735	$16,207	$16,694	$17,194	$17,710	$18,241	$18,789
Total Direct Expenses:	$18,500	$19,055	$19,627	$20,215	$20,822	$21,447	$22,090	$22,753	$23,435	$24,138
Gross Profit	$69,250	$68,695	$68,123	$67,535	$75,703	$75,078	$74,435	$73,772	$82,742	$82,039
Other Operating Expenses:										
Repairs/Maintenance	$800	$824	$849	$874	$900	$927	$955	$984	$1,013	$1,044
Pumping Cost	$3,100	$3,193	$3,289	$3,387	$3,489	$3,594	$3,702	$3,813	$3,927	$4,045
Utilities	$2,625	$2,704	$2,785	$2,868	$2,954	$3,043	$3,134	$3,228	$3,325	$3,425
Salaries	$10,000	$10,300	$10,609	$10,927	$11,255	$11,593	$11,941	$12,299	$12,668	$13,048
Car and Truck Expenses	$3,600	$3,708	$3,819	$3,934	$4,052	$4,173	$4,299	$4,428	$4,560	$4,697
Total Depreciation	$8,464	$13,602	$9,185	$8,464	$8,464	$5,673	$5,673	$1,125	$1,125	$1,125
Insurance	$1,500	$1,545	$1,591	$1,639	$1,688	$1,739	$1,791	$1,845	$1,900	$1,957
Taxes	$1,000	$1,030	$1,061	$1,093	$1,126	$1,159	$1,194	$1,230	$1,267	$1,305
Other	$5,500	$5,665	$5,835	$6,010	$6,190	$6,376	$6,567	$6,764	$6,967	$7,176
Other Operating Exp.:	$36,589	$42,571	$39,023	$39,197	$40,119	$38,278	$39,256	$35,715	$36,753	$37,822
Direct and Other Exp.:	$55,089	$61,626	$58,649	$59,412	$60,941	$59,724	$61,346	$58,468	$60,188	$61,960
Net Operating Income B/Taxes	$14,161	$7,069	$9,474	$8,122	$14,762	$15,354	$13,089	$15,304	$22,554	$20,079
Estimated Income Taxes:	$2,974	$1,485	$1,990	$1,706	$3,100	$3,224	$2,749	$3,214	$4,736	$4,217
Net Operating Income A/Taxes:	$11,187	$5,585	$7,484	$6,417	$11,662	$12,130	$10,341	$12,091	$17,817	$15,862

*250,000 fingerlings x 3 cycles/year (See text).
Inflation rate: 3 percent annual.

Table A4. Discounted Cash Flow (DCF) Analysis for a Hypothetical Red Drum Hatchery (First 10 Years).

Year	1st	2nd	3rd	4th	5th	6th	7th	8th	9th	10th
Capital Costs:										
Land	$12,000	$0	$0	$0	$0	$0	$0	$0	$0	$0
Construction	$60,000	$0	$0	$0	$0	$0	$0	$0	$0	$0
Equipment	$34,600	$0	$0	$0	$0	$0	$0	$0	$0	$0
Broodstock	$1,250	$0	$0	$0	$0	$0	$0	$0	$0	$0
Total Capital Costs:	$107,850	$0	$0	$0	$0	$0	$0	$0	$0	$0
Cash Sources:										
Net Operating Income A/T:	$11,187	$5,585	$7,484	$6,417	$11,662	$12,130	$10,341	$12,091	$17,817	$15,862
Depreciation:	$8,464	$13,602	$9,185	$8,464	$8,464	$5,673	$5,673	$1,125	$1,125	$1,125
Total Cash Sources:	$19,651	$19,187	$16,669	$14,881	$20,126	$17,803	$16,014	$13,216	$18,942	$16,987
Net Cash Flow without Capital Cost	$19,651	$19,187	$16,669	$14,881	$20,126	$17,803	$16,014	$13,216	$18,942	$16,987
Net Cash Flow with Capital Cost	($88,199)	$19,187	$16,669	$14,881	$20,126	$17,803	$16,014	$13,216	$18,942	$16,987
Cumulative Annual Cash Flow	($88,199)	($69,012)	(52,343)	($37,462)	($17,336)	$467	$16,480	$29,696	$48,638	$65,625
Cumulative Net Present Value	($89,985)	($74,128)	($61,605)	(51,441)	($38,944)	($28,895)	($20,677)	($14,512)	($6,479)	$70

After Tax Disc. 10% 10-Year NPV $70]

NOTE: Tax treatments using net operating losses (NDL) may increase the IRR and NPV.

Projected 5-YR IRR	-8.18%
Projected 10-YR IR	13.08%
Projected Disc. Payback Period	9.99 (Years)

*Interest income from cash reserves are not included in this financial analysis.

Table A5. Preliminary Assumptions and Performance Characteristics for a Hypothetical Red Drum Farm in South Carolina.

Pond Sizes (Water Acres):

Starting with redfish fry (Yes=1; No=0): 1 Ratio of Growout Ponds to Yearling Ponds: 1
Size of Growout Pond, Acres: 20.0 Yearling Pond: 5.0 Desired Stocking Density of Growout, Numbers/A: 1,250
Desired Number of Growout Ponds: 6 Expected Survival (%) in Yearling Ponds: 85.0%
Expected Fry Survival (%) in Ponds: 25.0% Size of Fry Ponds: 0.5 Number of Fry Ponds: 3

Projected Acreage and Yield Requirements

Total Growout Acres: 120 Required Number of Yearling Ponds: 6.0 Total Yearling Acress: 30
Yearling Yield, #/A: 5,000 Required Fingerling Stocking Rate: 5,882 Total Fry Required: 0.0 (Thou.)
Number of Fry Ponds: 1.0 Required Fry Stocking Density: 0.0 (Thousands)

Other Assumptions	**1st**	**2nd**	**3rd**	**4th**	**5th**	**6th**	**7th**	**8th**	**9th**	**10th**
Fingerlings Required, in 1,000's	176.5	176.5	176.5	176.5	176.5	176.5	176.5	176.5	176.5	176.5
Growout Survival Rate (1.5-2 yr. fish):	95.0%	95.0%	95.0%	95.0%	95.0%	95.0%	95.0%	95.0%	95.0%	95.0%
Yearling Transfer Size, lbs	1	1	1	1	1	1	1	1	1	1
Average Harvest Size, lbs:	0	3	3	3	3	3	3	3	3	3
Projected Yields and Acreage										
Yearling Yield, lbs/acre:	5,000	5,000	5,000	5,000	5,000	5,000	5,000	5,000	5,000	5,000
Total Yearling Harvest, 1,000's lbs:	150.0	150.0	150.0	150.0	150.0	150.0	150.0	150.0	150.0	150.1
Average Growout Yield, lbs/acre:	0	3,563	3,563	3,563	3,563	3,563	3,563	3,563	3,563	3,563
Total Water Acreage in Prod/Yr	30.5	150.5	150.5	150.5	150.5	150.5	150.5	150.5	150.5	150.5
Total Growout Harvest, 1,000's lbs:	0.0	427.5	427.5	427.5	427.5	427.5	427.5	427.5	427.5	427.5
Land Cost, $ per Acre:	3,000	3,000	3,000	3,000	3,000	3,000	3,000	3,000	3,000	3,000
Leasing Cost, $ per Acre:	$75	$75	$75	$75	$75	$75	$75	$75	$75	$75
Land Used (120% of water acres):	180.6	180.6	180.6	180.6	180.6	180.6	180.6	180.6	180.6	180.6
Percentage of Land Leased	100%	100%	100%	100%	100%	100%	100%	100%	100%	100%
Percentage of Land Purchased	0%	0%	0%	0%	0%	0%	0%	0%	0%	0%
Estimated Earthmoving, 1,000's	160	0	0	0	0	0	0	0	0	0
Fingerling Cost, $/fish:(See Text)	$0.08	$0.08	$0.08	$0.08	$0.08	$0.08	$0.08	$0.08	$0.08	$0.08
Feed Costs, $ per lb. (Bulk):	$0.13	$0.13	$0.13	$0.13	$0.13	$0.13	$0.13	$0.13	$0.13	$0.13
Feed Conversion Ratio:	2.5	2.4	2.3	2.2	2.1	2.0	2.0	2.0	2.0	2.0
Fertilizer, $ per lb.:	$0.06	$0.06	$0.06	$0.06	$0.06	$0.06	$0.06	$0.06	$0.06	$0.06
Ferlilizer Ratio, lbs./acre:	500	500	500	500	500	500	500	500	500	500
Pumping Days per Year:	110	110	110	110	110	110	110	110	100	100
Average Pumping Hours per Day:	12	12	12	12	12	12	12	12	12	12
KWH Consumption per Pump:	50	100	100	100	100	100	100	100	100	100
Number of Operating Pumps/Season:	1	2	2	2	2	2	2	2	2	2
Avg. Paddlewheel HP/Growout Acre:	1	1	1	1	1	1	1	1	1	1
Avg. Paddlewheel HP/Yearling Acre:	2	2	2	2	2	2	2	2	2	2
Aeration Days per Year:	60	60	60	60	60	60	60	60	60	60
Average Aeration Hours per Day:	24	24	24	24	24	24	24	24	24	24
Avg. Aeration Hours/Year/Wheel:	1,440	1,440	1,440	1,440	1,440	1,440	1,440	1,440	1,440	1,440
KWH Consumption/Growout Aerator	10	10	10	10	10	10	10	10	10	10
KWH Consumption/YearlingAerator:	5	5	5	5	5	5	5	5	5	5
No. of Operating Paddlewheels:	12	24	24	24	24	24	24	24	24	24
Electricity Cost, $ per KWH:	$0.07	$0.07	$0.07	$0.07	$0.07	$0.07	$0.07	$0.07	$0.08	$0.07
Harvest Percent Processed	80.0%	80.0%	80.0%	80.0%	80.0%	80.0%	80.0%	80.0%	80.0%	80.0%
Processing Cost,$ per lb.:	$0.10	$0.10	$0.10	$0.10	$0.10	$0.10	$0.10	$0.10	$0.10	$0.10
Processing Yield:	88.0%	88.0%	88.0%	88.0%	88.0%	88.0%	88.0%	88.0%	88.0%	88.0%
Whole Price, $ per lb.:	$1.20	$1.20	$1.20	$1.20	$1.20	$1.20	$1.20	$1.20	$1.20	$1.20
Processed Price, $/net lb. gutted:	$1.60	$1.60	$1.60	$1.60	$1.60	$1.60	$1.60	$1.60	$1.60	$1.60
Labor Wages, $ per hour:	$5.00	$5.00	$5.00	$5.00	$5.00	$5.00	$5.00	$5.00	$5.00	$5.00
Annual Inflation Rate:	3.00%	3.00%	3.00%	3.00%	3.00%	3.00%	3.00%	3.00%	3.00%	3.00%

* It is assumed that all the land needed for the farm will be leased and/or purchased in Year 1.

Table A6. Preliminary Projected Income Statement for a Hypothetical Commercial Redfish (Red Drum) Farm (First 10 Years)

Projected Total Harvest & Yields:	1st	2nd	3rd	4th	5th	6th	7th	8th	9th	10th
Total Harvest, whole lbs. in 1,000's:	0	427.5	427.5	427.5	427.5	427.5	427.5	427.5	427.5	427.5
Average Yield, Whole lbs. per Acre:	0	3,563	3,563	3,563	3,563	3,563	3,563	3,563	3,563	3,563
Gross Sales: (in thousands)										
Redfish (Red Drum), whole fish:	$0.0	$102.6	$102.6	$102.6	$102.6	$102.6	$102.6	$102.6	$102.6	$102.6
Redfish (Red Drum), processed fish:*	0.0	481.5	481.5	481.5	481.5	481.5	481.5	481.5	481.5	481.5
Total Gross Sales:	$0.0	$584.1	$584.1	$584.1	$584.1	$584.1	$584.1	$584.1	$584.1	$584.1
Direct Operating Expenses: (in thousands)										
Fingerlings (if purchased)	$14.1	$14.1	$14.1	$14.1	$14.1	$14.1	$14.1	$14.1	$14.1	$14.1
Feed, yearlings & growout fish	48.8	133.4	127.8	122.3	116.7	111.2	111.2	111.2	111.2	111.2
Processing	0.0	30.1	30.1	30.1	30.1	30.1	30.1	30.1	30.1	30.1
Fertilizer	1.0	4.7	4.7	4.7	4.7	4.7	4.7	4.7	4.7	4.7
Total Direct Expenses:	63.8	$182.3	$176.7	$171.2	$165.6	$160.1	$160.1	$160.1	$160.1	$160.1
Gross Profit:	-$63.8	$401.8	$407.4	$412.9	$418.5	$424.0	$424.0	$424.0	$424.0	$424.0
Other Operating Expenses (30% Inflation/Year) (in thousands)										
Salaries, Manager & Technicians	$72.4	$74.6	$76.9	$79.2	$81.5	$84.0	$86.5	$89.1	$91.8	$94.5
Electricity	10.7	56.4	58.1	59.8	61.6	63.5	65.4	67.4	69.4	71.5
Wage Labor 24 man-hrs/pond	1.0	2.5	2.6	2.7	2.7	2.8	2.9	3.0	3.1	3.2
Land Leasing Fees	13.5	14.0	14.4	14.8	15.2	15.7	16.2	16.7	17.2	17.7
Total Depreciation	74.5	126.7	89.5	67.1	67.1	61.5	61.5	15.8	0.0	0.0
Business Insurance	10.0	10.3	10.6	10.9	11.3	11.6	11.9	12.3	12.7	13.0
Professional Fees	15.0	5.0	5.2	5.3	5.5	5.6	5.8	6.0	6.1	6.3
Licenses & Property Taxes	4.0	4.1	4.2	4.4	4.5	4.6	4.8	4.9	5.1	5.2
Car & Truckl Expenses	6.0	6.2	6.4	6.6	6.8	7.0	7.2	7.4	7.6	7.8
Miscellaneous Expenses	5.0	5.2	5.3	5.5	5.6	5.8	6.0	6.1	6.3	6.5
Pond & Other Maintenance	10.0	10.3	10.6	10.9	11.3	11.6	11.9	12.3	12.7	13.0
Total Other Operating Expenses:	$222.1	$315.3	$283.7	$267.1	$273.1	$273.7	$280.1	$240.9	$231.9	$238.8
Total Direct & Other Expenses:	$285.9	$497.6	$460.4	$438.3	$438.7	$433.8	$440.2	$401.0	$392.0	$398.9
Net Operating Income Before Tax	-$285.9	$86.5	$123.7	$145.8	$145.4	$150.3	$143.9	$183.1	$192.1	$185.2
Estimated Income Taxes:**	$0.0	$22.9	$38.9	$48.9	$48.7	$50.9	$48.0	$65.6	$69.7	$66.6
Net Operating IncomeAfter Tax:	-$285.9	$63.7	$84.8	$96.9	$96.7	$99.4	$95.9	$117.5	$122.4	$118.6

* The processing sales are based upon a 12% loss in round (whole) weight of the fish.
** Estimated income taxes donot include alternative minimum taxes and other taxes applicable to corporations.

Table A7. Discounted Cash Fl.ow (DCF) Analysis for a Hypothetical Redfish Farm in South Carolina

	YEAR 1	2	3	4	5	6	7	8	9	10
Capital Costs:										
Construction	$292.6	$0.0	$0.0	$0.0	$0.0	$0.0	$0.0	$0.0	$0.0	$0.0
Equipment	226.0	$0.0	$0.0	$0.0	$0.0	$0.0	$0.0	$0.0	$0.0	$0.0
Total Capital Costs:	$518.6	$0.0	$0.0	$0.0	$0.0	$0.0	$0.0	$0.0	$0.0	$0.0
Cash Sources:										
Net Operating Income After Tax	-$285.9	$63.7	$84.8	$96.9	$96.7	$99.4	$95.9	$117.5	$122.4	$118.6
Depreciation:	74.5	126.7	89.5	67.1	67.1	61.5	61.5	15.8	0.0	0.0
Total Cash Sources:	-$211.4	$190.4	$174.3	$164.0	$163.8	$160.9	$157.4	$133.3	$122.4	$118.6
Net Cash Flow without Year 1 Capital Costs	-$211.4	$190.4	$174.3	$164.0	$163.8	$160.9	$157.4	$133.3	$122.4	$118.6
Net Cash Flow with Year 1 Capitol Costs	-$730.0	$190.4	$174.3	$164.0	$163.8	$160.9	$157.4	$133.3	$122.4	$118.6

After tax discount rate	10.0%	10-year Net Present Value	$123.
NOTE: Tax treatments using net operating losses (NOL) may increase the IRR and NPV.		Projected 5-yr Internal Rate of Return	-2.13%
		Projected 10-yr Internal Rate of Return	16.68%

	1	2	3	4	5	6	7	8	9	10
Cumulative Annual Cash Flow:	-$730.0	-$539.6	-$365.4	-$201.3	-$37.5	$123.4	$280.7	$414.0	$536.4	$655
Cumulative Net Present Value:	-$710.8	-$553.4	-$422.5	-$310.5	-$208.8	-$118.0	-$37.2	$25.0	$76.9	$122.6
Projected Payback Period:										
Projected Discounted Payback Period	NA	NA	NA	NA	NA	NA	NA	7.59834	7.51897	7.31874

*Interest income from cash reserves are not included in this financial analysis

**The initial investment used in calculating the IRR and NPV does not include "Operating Capital". The IRR and NPV were calculated using the "Net Cash Flow:" at the end of each period when calculating the NPV, IRR and DDP.

NOTE: Operating Capital for pre-construction tasks (e.g., permitting) and the first two years are not included in this analysis.

Table A8. Depreciation Schedule for a Hypothetical Commercial Redfish (Red Drum) Farm in South Carolina, 1987.

31.5-Year Property:		(X 1,000)								
Trailer	$12.0									
Total:	$12.0									
Depreciation (SLN):	$0.3	$0.3	$0.3	$0.3	$0.3	$0.3	$0.3	$0.3	$0.3	$0.3
7-Year Property:		(X1,000)								
Pond Construction $1.20 /cu.yd	$192.0									
Paddlewheels 5HP,No: 12	21.8									
Paddlewheels 10HP,No: 12	27.0									
Water Control Structures, Total 24	60.6									
Seawater Pumps 50 HP 2	25.0									
Electrical Wiring	13.4									
Harvesting Equipment & Seines	20.2									
Pump Inlet & Base Construction	40.0									
Feed Equipment	5.0									
Feed Storage Bins	9.0									
Tractor with Accessories (Used)	12.0									
ATC cycles	5.0									
Lab and Pond Monitoring Equipment	5.0									
Storage & Work Shed	5.0									
Shop Equipment	10.0									
Office Equipment	2.0									
Miscellaneous Equipment 5%	22.6									
Total 7-year Property Cost:	$475.6	$0.0								
Depreciation (DDB):	$67.9	$116.5	$83.2	$59.4	$42.4	$30.3	$21.7	$15.5		
Depreciation (SLN):	$61.1	$61.1	$61.1	$61.1	$61.1	$61.1	$61.1			
5-year Property										
Used Harvesting Boom Truck	$25.0									
Office Machinery	$6.0									
Total 5-Year Property Cost	$31.0									
Depreciation (DDB):	$6.2	$9.9	$6.0	$3.6	$2.1	$1.3				
Depreciation (SLN):	$5.6	$5.6	$5.6	$5.6	$5.6					
Total Cost:	$518.6	$0.0	$0.0	$0.0	$0.0	$0.0	$0.0	$0.0		
Total Depreciation (DDB):	$74.1	$126.4	$89.1	$63.0	$44.6	$31.6	$21.7	$15.5		
Total Depreciation (SLN)	$67.1	$67.1	$67.1	$67.1	$67.1	$61.5	$61.5	$0.3		

NOTE: Equipment with a 5- or 7-year depreciation period are assumed to be kept in operation for at least 10 years.

Table A9. Preliminary Equipment List and Construction Cost Estimates for a Hypothetical Red Drum Farm in South Carolina, 1987.

Water Control Structures:		
Intake Pipes, Growout ponds: Dia., in. 18 Length: $40 Cost/ft: $21.00 Total Number of Pipe Sections:	6	$5,040
Intake Pipes, Yearling ponds: Dia., in. 10 Length: $40 Cost/ft: $13.00 Total Number of Pipe Sections:	6	3,120
Intake Risers, Growout ponds: Size, in. 18 $/Unit: $1,100 Number:	6	6,600
Intake Risers, Yearling ponds: Size, in. 10 $/Unit: $530 Number:	6	3,180
Outflow Pipes, Growout ponds: Dia., in. 24 Length $40 Cost/ft: $25.00 Total Number of Pipe Sections:	6	6,000
Outflow Pipes, Yearling ponds: Dia., in. 18 Length: $40 Cost/ft: $25.00 Total Number of Pipe Sections	6	5,040
Outflow Risers, Growout ponds: Size, in. 24 $/Unit: $1,300 Number:	6	7,800
Outflow Risers, Yearling ponds: Size, in. 18 $/Unit: $1,100 Number:	6	6,600
Yearling Harvest Basins, Yearling ponds: Dia., ft. 20 Length 10 Width Thickness, in: 4.0 Concrete, cu.yd.:	7.4	
Number: 6 $/cu.yd.: $49.00		2,178
Canal Pipes: Dia., in. 36 Lenth: $70 Cost/ft: $36.00 Total Number of Pipe Sections	2	5,040
Labor: $10.00/hour 800 hours		8,000
Miscellaneous Labor and Materials		2,000
Total Cost of Water Control Structures		**$60,598**
Harvesting Equipment:		
Haul Seine, Growout ponds: Length: 800 ft @ $3.10/ft		$2,480
Haul Seine, Yearling ponds: Length: 400 ft @ $2.90/ft		1,160
Live Cars Number: 4 @ $500.00/unit		2,000
Loading Nets, 54 inches (round): Number: 3 @ $110.00/unit		330
Hydraulic Loading Scales:		1,400
Seine Reel:	3,800	
Net Room:	5,000	
Holding Tanks on Harvest Truck:		2,000
Miscellaneous Equipment (e.g., dip nets):		2,000
Total Harvesting Equipment Costs:		**$20,170**
Electrical System for Ponds and Pumps:		
Total Stations: 6 Cost/Station: $1,900		$11,400
Miscellaneous Labor and Materials:		2,000
Electrical System Total Cost:		**$13,400**

Table A10. Pumping Capacity Calculations for a 152-acre Red Drum Farm in South Carolina.

Total pond water acreage	150
Average water depth, feet	4
Total acre-feet of water	600
Total acre-feet of water	600
Daily water exchange rate	5.0 percent
Water exchanged per day, acre-feet	30
Water exchanged per day	30
Gallons of water per acre-feet	325,850
Water exchanged per day, gallons	9,775,500
Required water exchange per day	9,775,500
Average period of daily pumping, hours	12
Required water exchange per hour	814,625
Required water exchange per hour	814,625
Pumping minutes per hour	60
Required water exchange per minute, GPM	13,577
Gallons per minute, rounded	14,000
Required gallons per minute	14,000
Gallons per minute per pump	7,000
Number of pumps needed*	2

*Although one pump can be used to generate water requirements, it is suggested that at least two pumps be purchased. With two pumps the water exchange rate can be controlled easier and the water system is not dependent on one pump.

Marketing Opportunities for Farm-Raised Red Drum

Michael G. Haby
Seafood Marketing Specialist
Texas Marine Advisory Service
Port Aransas, Texas 78373

Prior to 1985, red drum (*Sciaenops ocelllatus*) had been traditional seafood fare in the Gulf states, with annual supplies averaging 3.1 million pounds between 1960 and 1984. Most red drum originated from the vast bay complex abutting the Gulf of Mexico, and market prices historically reflected a preference for fish weighing three to seven pounds. However, between 1985 and 1987, blackened redfish became the seafood meal of choice in some regions of the country. In response to demand for blackened redfish, many seafood distributors began offering red drum to their customers.

Since landings of red drum from traditional sources (i.e. estuarine areas) were relatively constant, a Gulf purse seine fishery began which targeted red drum found in offshore waters. Because purse seines are so effective, landings from the oceanic Gulf increased dramatically, pushing annual harvests of red drum to historic levels (13.5 million pounds for the first six months of 1986). These larger red drum harvested offshore became the major source of supply for out of state markets, and in 1986, red drum taken offshore via purse seine comprised 60 percent of the total domestic harvest.

This intense fishing pressure was short-lived, and by 1987, federal fishery managers closed the Gulf purse seine fishery to directed fishing for red drum. By 1988, most Gulf states followed Texas' lead by enacting legislation designed to institute red drum as a gamefish. Simultaneously, the euphoria surrounding blackened redfish rapidly diminished as opportunities to harvest wild stocks were eliminated. In 1989 the Texas Legislature guaranteed the market for farm-raised red drum by prohibiting its sales unless farm-raised origin could be documented.

With most red drum allocated to sportsfishing interests, prospective culturists have continued to view red drum as a species that has demand already built in. Of course, the major problem has been cost effective over-wintering strategies required to produce a saleable food fish. While most of the current emphasis on red drum aquaculture is on refinements of production technology, two marketing issues need to be addressed. These are: a) what do historical data suggest about future opportunities for redeveloping the red drum market and b) what producer marketing strategies are necessary to insure the success of red drum culture.

Historical Characteristics of the Red Drum Market

In the fall of 1986, characteristics of the red drum market in Texas (i.e. current operating strategies, expectations of future sales volumes, market form/size preferences, etc.) were assessed via mail survey (Haby and Cuenco, 1987). These findings were collected while red drum were readily available. As such, some of these data are irrelevant today, but other components of the research provide insight into the how the red drum market may look in the future.

Market Form and Size Preferences

Data on preferred size of red drum was skewed in deference to the wild-caught supplies that were currently available at the time the survey was administered. Based on 1986 survey findings, Texas seafood firms (dockside dealers, processors, wholesalers and specialty shop owners) most preferred a gilled and gutted market form weighing three to five pounds. Seafood buyers for corporate food retailing firms indicated a preference for fillets weighing at least .81 pound.

Size preference is often a function of what is currently available, and available sizes can change. For example, catfish producers are finding that raising catfish greater than two pounds can result in a net loss due to slower growth. Therefore, the average size of catfish fillets should be expected to drop somewhat as culturists seek to maximize their net returns by harvesting smaller fish.

When asked about using fish weighing .75 pound to 1.5 pounds once the head, fins and entrails were removed (i.e. a yearling red drum not requiring over-wintering), only corporate supermarket buyers indicated any interest. At the time, it was concluded that the popularity of skinned, headed and gutted freshwater catfish currently marketed through food retailing establishments could have served as a model for this unique, positive response.

Historically, most red drum have been marketed fresh. Despite this, wholesale distributors and supermarket buyers alike expressed an interest in using frozen, processed products in the 1986 survey. From a long-term, market development standpoint, it is important to note that most national, full-line distributors are generally more interested in frozen, processed products since availability is better assured. For example, in a recent assessment of current seafood processing, product procurement and distribution practices in Texas, the seafood manager of one national, full-line distributor indicated that 50 percent of all their divisions would like to offer red drum products if constant supplies could be insured (Haby and Gryska, 1990).

Since production via aquaculture will probably be seasonal and sporadic, and interest in frozen, processed red drum products should be viewed positively. Red drum typically contain only 1 to 2 percent fat, thus product quality can be maintained for several months while in frozen storage. This was supported by taste panel assessments of previously frozen red drum which demonstrated that a) the muscle is stable in frozen storage (i.e. no off odors or off flavors), and b) previously frozen products lack the earthy, gamy flavor of fresh product (Jahncke and Hopkins, 1985).

Price

Price is always the manifestation of demand and supply interaction, and therefore will continuously change as red drum aquaculture changes. In 1986, wholesale distributors reported paying an average price of $1.20 per pound, with a range $.80 to $1.40 for market sized red drum (i.e. fish weighing between 3 and 7 pounds). Today, prices for whole fish reportedly range from $1.75 to $3.00 (Linton and O'Connor, 1990).

With limited quantities currently on the market, producers may be in a position to negotiate the price for red drum. Under these circumstances, it is essential that culturists maintain accurate production cost information, since these data form the basis of sound marketing decisions (Catania and Keefer, 1988). Additionally, fish farmers need accurate estimates of expected production (i.e. total quantity and the size distribution of swimming inventory) since both production costs and expected harvest levels are required to compute break-even prices. Knowing break-even price (or a range of break-even prices computed under different expected harvests) prior to negotiation is the only way producers can be assured of a positive outcome.

Processing Yields

Yields of various market forms are of interest to aquaculturists and processors alike in establishing prices for whole fish and processed products. Once entrails and gills are removed, 88 percent of total weight remains (Harrington, *et al* 1979). Jahncke and Hopkins (1985) reported that skin on fillets cut from whole fish weighing 3.5 pounds yielded 34 percent of the total weight, while skinless fillets equaled 28 percent of total weight. Thus, two skinless fillets, each weighing approximately .5 pound, could be produced from a 3.5 pound fish.

Market Size

Little, if any, data exist about the current size of the red drum market, but assuredly it is miniscule. While nationwide exposure in the mid-1980s suggested that red drum was destined to be among the most popular of fishes, it has been and remains a specialty item; particularly when compared to other seafood products supplying the domestic market. Placing red drum into perspective, supplies of shrimp currently exceed 740 million pounds on a heads-off equivalent basis (USDC 1990). And in 1989, farm-raised catfish, produced primarily in the Mississippi delta, contributed approximately 350 million pounds to the overall seafood supply (USDA 1990). Supplies of snappers and groupers, including indigenous harvests from the Gulf of Mexico and imports, currently approaches 30 million pounds annually, with imports accounting for at least half of total supply (Gulf of Mexico Fishery Management Council, 1989).

Based on information collected between 1986 and 1990, Texas firms continue to be interested in handling red drum. In fact, as it has sporadically entered the market, specialty wholesalers and food retailers fortunate enough to be offered red drum have eagerly purchased it at prices significantly above those reported in 1986. But until the overwintering problem is solved, red drum will continue to be a specialty product, serving a small niche in the overall Gulf states seafood market. However, with the advancement and adoption of technology, the potential exists to increase the supplies of red drum.[1]

Marketing Issues Important to Future Growth of Red Drum Aquaculture

New aquaculture ventures typically involve yield risks, marketing risks, or both.[2] Currently, the major

[1]In the short run, increases in red drum supplies will be rather small. Once the basic limits to year-round culture are eliminated, production has the capacity to increase. However, this will only happen in the long run. For example, in 1975 freshwater catfish supplies from aquaculture equaled about 16 million pounds. By 1980, catfish production had nearly tripled, reaching 46.5 million pounds. But the largest production gains occurred between 1980 and 1989 when catfish production increased to roughly 350 million pounds.

[2]Various aquaculture species often have mixed amounts of associated risk. For example, a species may be practically risk-free in terms of production technology but have limited acceptance in the market. Tilapia has been a historic case in point: an organism easy to culture, but until recently, one with limited consumer appeal due to small size and unfamiliar origin.

source of risk in red drum aquaculture is crop failure (i.e. yield risk) since over-wintering is required to produce acceptable sized fish. Work continues in search of technological solutions.

Conversely, market risks are not so precisely addressed. Market risk includes such issues as price declines, and changes in preferences (Downey and Trocke, 1981). In addition, two other considerations may have a major impact upon individual profitability as well as the regional competitive advantage of producers and long-run industry growth. These are a) lack of an existing processing and distribution infrastructure and b) the strategies used by producers to market their output once cumulative production exceeds local demand (Haby and Younger, 1990).

Many have suggested that the market for red drum appears to be built in. This seems particularly true in Texas since only red drum certified from aquacultural enterprises can be sold. Projections of absolute market size in Texas defies quantification. But because red drum has been a traditional favorite, Texas should be a major market for the foreseeable future.

It is important to differentiate between having a market for a product (i.e. demand), and marketing. Demand is created for a particular product when the right product is presented, in the right form, in the right place, at the right time. This euphemism suggests that besides the product per se, the marketplace requires additional convenience embodied in the product before it can be used.

Marketing, on the other hand, is the process of adding in these various forms of convenience (often called utility) which are demanded by potential customers. The various types of convenience added once the product is produced are an essential component of value, and are important to all who buy products either for resale or ultimate use. In general, Downey and Trocke (1981) define the mix of product convenience to be a) form (e.g. converting live fish into processed products); b) time (e.g. insuring that products are available when needed as opposed to only when they are harvested); c) place (e.g. moving products from production areas to markets); and d) possession (e.g. matching buyer and seller). The amounts and types of utility required depend upon the target market characteristics (i.e. is whole fish appropriate, or do restaurants want a frozen, skinless fillet). Two elements of convenience are particularly important as the red drum market is redeveloped. These are obtaining processing necessary to meet target market needs and effective distribution of fish farm output.

Infrastructure Concerns

Processing

In Texas, seafood processing capacity has expanded in concert with growth in the wild fisheries.[3] Yet, historically Texas finfish production has been relatively insignificant when compared to that of shrimp and oysters. As a result, current finfish processing capacity in Texas consists of relatively small, inefficient operations diffused throughout the entire production and marketing system (although several isolated exceptions exist). Finfish processing is commonly done manually on a custom basis by either producers, small-scale processors of wild-caught estaurine fishes, or even by firms classified as mid-level handlers.

Procurement and Distribution

The specialty wholesaler strictly focuses on the seafood product line. These firms are noted for their sourcing expertise, and are judged successful based on how well they can procure the product mix requested by their accounts. Specialty wholesalers in Texas usually focus on assembling fresh products from various sources and distributing them. These firms may establish purchasing arrangements with producers that may also include post-production services such as farm pick-up. Seafood distributors have historically filled the gap in Texas finfish processing capability. Not surprisingly, Texas wholesalers themselves reported processing 75 percent of red drum fillets they sold (Haby and Cuenco, 1987). Thus, for the foreseeable future it appears that specialty seafood wholesalers are capable and willing to process fish farm output into the market forms demanded by the various customer types.

In contrast to the specialty wholesaler, the full-line distributor generally handles a variety of product lines in addition to seafoods. This is particularly true of those distributors who target the food service sector as their primary market. Since the full-line distributor inventories several thousand unrelated items required by food service operators, specialization of corporate skills has focused on automation and development of management systems designed to facilitate order picking, overall inventory management and cost minimization. As such, most full-line distributors prefer to purchase products in market forms immediately usable for food service establishments. Thus, processors, as opposed to producers, are the full-line distributors' major suppliers of seafood products.

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[3]Today it is generally concluded that most world fisheries, including the Gulf of Mexico shrimp fishery, are fully utilized, suggesting little if any real growth in future production from wild sources. However, the offshore Gulf shrimp fishery sustained massive growth after World War II as offshore stocks were discovered, and appropriate gear was designed. During this period, processing capacity grew dramatically. Today, Texas still claims one of the largest, most progressive shrimp processing infrastructures in the world. Many processors handle not only indigenous harvests, but rely on imported product as well.

Producer Marketing Srategies

As the supply of farm-raised red drum increases, the approach taken by producers in marketing fish farm output should also change. Initially, most red drum produced via aquaculture will be sold by the producer on a direct basis to customers within a relatively small geographic area. These products will contain limited amounts of convenience (i.e. possibly pond-side sale, distribution of unprocessed products, etc.).

However, as technological limitations are eliminated and supplies of red drum expand, serving a larger market will require more utility in the product such as timely deliveries of processed products. Once output is offered to a larger market, products will flow through customary marketing channels. These channels are, in part, dictated by the product forms required by retail interests (food service and food retailing establishments).

In the long run, farm-raised red drum production may be sufficient to enter selected markets besides those in the Gulf states. Once this production level occurs, dedicated processing should follow. At that time, selected full-line distributors will begin carrying frozen, processed red drum products, such as fillets.

Requirements for pre-determined types and amounts of form, place, time or possession utility are established by the market. However, the entities within the marketing system which add this utility is a matter of economic choice. That is, if one entity can perform a function more effectively (i.e. provide better service) and/or more efficiently (i.e. less expensively) than another, then he will specialize in completing that function. Therefore, producers are faced with the option of either using the existing marketing system or becoming the marketing system.

Obviously producers can sell fish farm output to others within the marketing system such as wholesale distributors who subsequently process and distribute it. Alternatively, culturists themselves can assume these post production responsibilities in addition to producing fish. To do this, vertical integration is required.

When a business (such a a fish farm) vertically integrates, it essentially chooses to take on additional functions besides its primary one of producing fish. In assuming other functions, required management time always increases and the skills necessary for success in the new function(s) are typically completely different from production acumen. Depending upon the function(s) to be performed, additional investment in processing equipment and/or distribution facilities may be needed. As well, cash needs of the business will increase, thus necessitating additional borrowings. Concomitantly, lenders will require detailed business plans that fully evaluate a variety of economic scenarios to determine risk or exposure. These scenarios should focus on general trends and industry outlook as well as considerations unique to the enterprise (Klinefelter *et al* .,1990).

Therefore, the decision to add various levels of convenience should be the result of careful research to ascertain a) whether, and to what extent, these services are currently being performed by competitors, and b) economic/financial analysis to estimate costs (both monetary and management time) of performing additional services and the additional returns. This planning phase is crucial because despite the additional demands placed on manager and operation alike, vertically integrating to complete marketing services such as processing and distribution does not guarantee additional profits to the producer.

Summary and Conclusions

The prospect of culturing a traditional favorite no longer available from wild sources continues to excite potential aquaculturists. And while there is virtually no red drum on the market, that which has entered sporadically has been eagerly purchased. This suggests that in traditional consumption areas for red drum, such as the Gulf states, the market could be redeveloped through aquaculture.

Until technological solutions to over-wintering are addressed, red drum aquaculture in open ponds carries enormous risk of crop failure as producers attempt to produce fish that approach the lower bound of the historic, preferred size of three pounds. In the short run however, producers may be able to market smaller sized fish. Note that in the 1986 survey, some food retailers indicated interest in selling yearling red drum (i.e. pan-ready products weighing .75 to 1.5 pounds). Opportunities to sell yearling red drum should be explored since this could have a substantial impact on the attractiveness of investing in commercial red drum culture as well as reducing the risk of crop failure.

The two essential components of utility required to market red drum beyond the local level are processing and distribution. Finfish processing and distribution functions are currently provided by specialty wholesalers in Texas. Therefore, the best access to retail interests is through this business type. By no means should the serious aquaculturist who is developing a plan to market farm output overlook this segment of the food industry.

References

Catania, P. and N. Keefer. 1988. **A Marketing Handbook for Producers.** Board of Trade of the City of Chicago. Chicago, Ill. 77p.

Downey, W. D. and J. K. Trocke. 1981. **Agribusiness Management.** McGraw-Hill New York, N.Y. 459 p.

Gulf of Mexico Fishery Management Council, 1989.

Amendment Number 1 To The Reef Fish Fishery Management Plan, Tampa, Fla. 356p.

Haby, M. G. and M. L. Cuenco. 1987. Red Drum Marketing Opportunities. *In* Chamberlain, G. W., R. J. Miget and M. G. Haby, eds. Manual on Red Drum Aquaculture. Preliminary Draft of Invited Papers Presented at the Production Shortcourse of the 1987 Red Drum Aquaculture Conference on June 22-24, 1987, Corpus Christi, Tex. 350p.

Haby, M. G. and P. Gryska. 1990. Processing and Marketing. *In* Chamberlain, G. W. ed. Texas Aquaculture: Status of The Industry. Review Draft for the 1990 Texas Aquaculture Conference January 30 - February 1, 1990, Corpus Christi, Tex. 133p.

Haby, M. G. and W. R. Younger. 1990. **Texas Crawfish Aquaculture: An Analysis of a Producer Survey Focusing on Industry Practices, Conditions, Marketing Plans and Ideas for the Future.** Texas Agricultural Extension Service, 30p.

Harrington, R. A., G. C. Matlock and J. E. Weaver. 1979. **Length-Weight and Dressed-Whole Weight Conversion Tables for Selected Saltwater Fishes.** Texas Parks and Wildlife Department, Coastal Fisheries Branch, Management Data Series Number 6, 64p.

Jahncke, M. and J. S. Hopkins. 1985. Comparison of Wild and Pond Raised Redfish with Respect to Proximate and Fatty Acid Composition and Sensory Evaluations, National Marine Fisheries Service, Southeast Fisheries Center, Charleston, S.C. 5p mimeo.

Klinefelter, D., S. Hanson and W. Wilcox, 1990. Financing. *In* Chamberlain, G. W. ed. Texas Aquaculture: Status of The Industry. Review Draft for the 1990 Texas Aquaculture Conference January 30 - February 1, 1990, Corpus Christi, Tex. 133p.

Linton, T. and R. O'Connor. 1990. Red Drum. *In* Chamberlain, G. W. ed. Texas Aquaculture: Status of The Industry. Review Draft for the 1990 Texas Aquaculture Conference January 30 - February 1, 1990, Corpus Christi, Tex. 133p.

U. S. Department of Agriculture, National Agricultural Statistics Service. 1990. **Catfish, May 1990.** U.S. Government Printing Office. Washington, D.C. 2p.

U.S. Department of Commerce, National Marine Fisheries Service. 1990. **Fisheries of The United States, 1989.** U.S. Government Printing Office. Washington, D.C. 111p.

APPENDIX A

Annotated Bibliography of the Red Drum (*Sciaenops ocellata*)

Thomas L. Linton, John H. Clark and Jane M. Boslet
Department of Wildlife and Fisheries Sciences
Texas A&M University
College Station, Texas 77843

Age and Growth

Theiling, Dale L., and Harold A. Loyacano, Jr. 1976. Age and growth of red drum from a saltwater marsh impoundment in South Carolina. **Trans. Am. Fish. Soc.** 105(1):41-44.

Adult and juvenile red drum, *Sciaenops ocellata* (Linnaeus), confined to a saltwater marsh impoundment at South Island. Georgetown County, South Carolina, were studied to determine their length-weight relationship, age according to otoliths, and the time of annulus formation in the otoliths. The equation $\log W = -1.29596 + 2.74031 \log L$ was determined for the relationship of length to weight. Sectioned otoliths revealed no annulus formed in fish up to 1,070 g and 38.5 cm in standard length, suggesting that no annulus was formed the first spring after hatching in September through November. Annuli were formed in spring (April-May). The largest fish belonged to the 1966 and 1967 year classes, which were in the marsh when the impoundment was formed in May 1968. Reading otoliths before they were cut produced erroneous results.

Wakefield, C. A. and R. L. Colura. 1982. Age and growth of red drum in three Texas bay systems. **Ann. Proc. Tex. Chap. Am. Fish. Soc.** 5: 77-87.

Age and growth of red drum (*Sciaenops ocellatus*) in Lower Laguna Madre, Matagorda, and Galveston Bay systems in Texas were described using the scale method. One annulus was formed each year and annulus formation was completed by March or April. Growth was similar for fish in lower Laguna Madre and Matagorda Bay systems but Galveston Bay system fish had a larger L_{∞}. Calculated lengths at age I were similar to those reported by others, but shorter at later ages. The scale method was easily applied and shows potential, but requires additional validation before it can be used in red drum management programs.

Behavior

Guest, W. C. and J. C. Lasswell. 1978. A note on courtship behavior and sound production of red drum. **Copeia** 1978(2):337-338.

Male red drum darkened in color and began drumming during prespawning courtship. Drumming intensified until spawning then decreased markedly. Spawning behavior was observed also.

Owens, D. W., K.Jones and B. J. Gallaway. 1981. Brine avoidance/attraction bioassay on redfish. In: W. B. Jackson (ed.) **Shrimp and Redfish studies: Bryan Mound brine disposal site off Freeport, Texas, 1979-1981.** Vol. V (Part B). NOAA Tech. Memo., NMFS/SEFC-69.

A circular tank with a water depth of 77 cm and diameter of 5.49 m was used to monitor the behavior of the redfish or red drum (*Sciaenops ocellata*) in response to various temperature and salinity gradients. Photoelectric gates separated eight peripheral compartments from the open center of the tank and monitored fish entering and leaving the compartments. The experimentation was based on a free choice, non-conditioning design. The 100 fish used had a standard length of 42.75 + 1.44 cm (X+SE) and generated an average of 83.12+24.66 compartment entries per hour of monitoring. Redfish were determined to be most active in our system during the first six hours of the dark phase, though a minimum adjustment period of three hours was necessary for this species to begin normal exploratory behavior. A comparison of various brines made from salt dome salt and artificial sea salts (Instant Ocean) indicate that redfish did not readily discriminate among these solutions as determined by the activity measures employed. In addition, brines made with Brazos River water as a diluent were not distinguished from brines made from tap water as a diluent. Redfish neither avoided nor were attracted to the 2 ppt brine gradients, as compared to control compartments receiving no brine.

In separate experiments, attraction/avoidance for brines at each of three different gradient strengths (0.5, 1.0 and 2.0 ppt) were tested on fish acclimated at three different ambient temperatures (15°, 20°, 25° C). In addition, in the same protocol, brine heated to 2°C above background was compared to unheated brine using separate compartments. Redfish at colder acclimations of 15° and 20° appeared attracted to heated brine only if the saline gradient in the test compartments was low (0.5 ppt above background). At an acclimation temperature of 25°C during fall, the unheated brine appeared attractive compared to both the heated brine and the appropriate controls. A separate experiment conducted at 25°C acclimation during spring indicated a similar trend, but the results were not statistically significant. We hypothesize that redfish may use saline gradients as part of their cueing system for movements from estuaries to the open Gulf, as well as for localized movements as required for behavioral thermoregulation.

Direct observations also indicated that the redfish responded to slight saline gradients under certain conditions. However, the diffuser's maximum near-field concentration of approximately 5 ppt above ambient reported by Randall (1981) is not of sufficient magnitude to harm any life stage of this euryhaline species. In addition, even if redfish are attracted to the brine gradients, it is not likely that they would remain long in the area without further reinforcement of the behavior (e.g. cool water or food resources). None of the present monitoring programs involving sampling near the diffuser is using methods that will result in capture of large redfish, which would enable confirmation or rejection of the hypothesis that redfish may aggregate around the brine diffuser under certain conditions.

Owens, D. W. and K. A. Jones. 1982. Acclimation temperature and orientation to salinity gradients by the red drum. **Am. Zool.** 22(4):864.

As part of a biological impact assessment on the disposal of brines during evacuation of coastal salt domes, we studied the orientation behavior of red drum, (*Sciaenops ocellata*). Large subadult fish (42.75 + 1.44 cm SL) were acclimated in the lab to 15.20 or 25°C. Gradients of .5, 1.0 and 2.0 ppt above ambient were produced in the peripheral compartments of a 5.45 m round monitoring tank. At colder acclimation temperatures (15 & 20) the fish showed a signficant attraction to the brine if the gradient was low (.5) and the brine was heated to 2° above ambient. On the other hand, if the acclimation was 25° the fish were significantly attracted to the unheated brine at 2 ppt above ambient. Our nonconditioning protocol suggest that gradients of 1-2 ppt are biologically meaningful and that salinity gradients could be used as a component of the navigation system. Orientation to salinity gradients may facilitate behavioral thermoregulation where increasing salinity would correlate predictably with colder, deeper water. (Supported by DOE through NMES-SEEC).

Rohr, B. A. 1968. Surface schooling of red drum. **Underwater Naturalist** 5(2):21-23.

Adult red drum surfaced in a large, tightly packed school in the Mississippi Sound. Many fish were observed emerging halfway from the water in quick jumps during this period of intense feeding.

Biology

Holt, Joan, Allyn G. Johnson, C. R. Arnold, W. A. Fable, Jr., and T. D. Williams. 1981. Description of eggs and larvae of laboratory reared red drum, *Sciaenops ocellata*. **Copeia** 1981:751-756.

Egg and early larval development and pigment patterns of the red drum, *Sciaenops ocellata*, are described to 13 days after hatching. The pelagic spherical eggs had a mean diameter of 0.95 mm and usually contained one oil globules which coalesced into a single globule by 13 hours. Hatching (at 22-23 C) occurred about 28-29 hours after fertilization. Standard length at hatching was between 1.71 and 1.79 mm. Yolk-sac larvae were negatively buoyant and drifted downward (head first) about 95 percent of the time. Larvae began swimming in a horizontal position in pursuit of prey when the yolk sac exhausted and the mouth and eyes were developed. Development was temperature dependent. Length of the yolk-sac stage varied from 40 hrs at 30 C to 84 hrs at 20 C. Growth of larval red drum was rapid after they began to feed. Larvae grew from 1.74 mm mean SL at hatching to 5.11 mm mean SL at 300 hr. The larvae were fed rotifers (*Brachionis plicatilis*) and nauplii of *Artemia salina*.

Pearson, J. C., 1929. Natural history and conservation of red fish and other commercial sciaenids on the Texas coast. **Bur. Fish. Bull.** 44:129-214.

Redfish spawning season occurs mainly in October and in the Gulf actual spawning occurs close to or at the mouths of the various passes. Newly hatched redfish are carried by tidal currents into the bays and lagoons where they remain indefinitely. Redfish attain a modal TL of 340 mm after one year, 540 mm after two years, 640 mm after three years, 750 mm after four years and 840 mm after five years of growth. Maturity isn't reached before the end of fourth or fifth year, with few fish under 750 mm TL in a sexually mature condition. Food of redfish from 60-600 mm TL consists mainly of shrimps, crabs and small fish.

Simmons, Ernest G. and Joseph P. Breuer. 1962. **A study of redfish, *Sciaenops ocellata* Linnaeus and black drum, *Pogonias cromis* Linnaeus.** The University of Texas Institute of Marine Science, Publications 8:184-211.

Data on redfish, *Sciaenops ocellatus*, and the black drum, *Pogonias cromis*, are presented from 1949 to 1959. Redfish spawn in the Gulf of Mexico during fall and winter months but the exact site of this spawning is uncertain. Young fish move through passes into nursery grounds into the bays where they remain from six months to three or four years. Growth is rapid and standard lengths of 320, 530 and about 700 mm. are reached during the first three years. Spawning normally occurs at the end of the third or fourth year when the fish are 700-800 mm long but ripe fish as small as 450 mm. have been found. Although the species is euryhaline, individuals are most often found at salinity of 20-40 percent. Extreme temperatures of 3-33° C are tolerated but sudden drops in water temperature cause mortality. The food of redfish is primarily crabs, but shrimp and small fish are also consumed. More redfish are found in the bays during spring and fall months than in winter or summer, but some are present all year. Migrations are much less extensive than had been supposed and intrabay movement is relatively rare. Some schools of redfish are almost permanent residents of the Gulf while others rarely leave the bays during the first three or four years of life.

Black drum spawn in all of the bays and over any type bottom, as well as in the Gulf near passes. Most spawning takes place in February or March, but there is a prolonged or split season in May or June. Growth is less rapid than that of redfish and standard lengths of about 160, 310, and 415 mm are attained in the first, second, and third years. Tagging results indicate growth of 50 mm per year after the third year. Drum are euryhaline and have been found in salinities from 0-80 percent. At this higher salinity many are blinded or in poor condition, and spawning does not occur. There is a definite temporary movement to fresh water creeks during flood periods. Most movement is random during feeding; food consists primarily of small mollusks although vegetation, small fish, polychaete worms and shrimp are also consumed. Their habit of grubbing for food is detrimental to spawning areas of trout and nursery areas of trout, redfish and shrimp.

Parasitic copepods, primarily of the genus *Caligus* are found on both redfish and drum but are lost when salinity rises above 45 percent. Few isopods have been found on either species.

Redfish are sought by sport fishermen in the bays and in the Gulf. Drum are less actively sought although there is a substantial tourist fishery for this species. Creel census figures (Simmons, 1959-60) indicate that over one million pounds of redfish are harvested annually by sports fishermen. Commercial fishing is limited by law but about 2 million pounds of redfish and drum are harvested each year by various commercial means. Catastrophic freezes occurring about every 10 years have each destroyed more fish than have been harvested commercially for the past 50 years.

Drum are considered to be detrimental to trout and redfish populations since they destroy spawning and nursery grounds, compete for food and space and are less restricted in spawning requirements. The Laguna Madre tends to become overpopulated with this species very quickly. In an effort to reduce their effects, rough fish permits, for drum only, have been issued for certain parts of the Laguna Madre.

Condition

Harrington, R. A., G. C. Matlock, and J. E. Weaver. 1979. Standard- total length, total length-whole weight, and dressed-whole weight relationships for selected species from Texas bays. Tex. Parks Wildl. Dept., **Mangement Data Series No. 6**, Austin. 6p.

Regression coefficients for equations of the form Y = a + bX were estimated for total length (TL) as a function of standard length (SL), whole weight (WW) as a function of total length and whole weight (WW) as a function of dressed weight (DW) for red drum, black drum, spotted seatrout, southern flounder and sheepshead collected in Texas bays. All lengths were measured in millimeters and all weights in grams. Estimations for the TL-axis intercept (a) and slope (b) in the SL-TL equation (TL = a + b SL) were: red drum, 12.870 and 1.177; black drum, 18.220 and 1.181; spotted seatrout, 11.804 and 1.138; southern flounder, 8.959 and 1.175; and sheepshead, 7.486 and 1.243. Estimations for the long WW-axis intercept (log a) and slope (b) in the TL-WW equation (log WW = log a + b log TL) were: red drum, -5.085 and 3.041; black drum, -4.856 and 3.001; spotted seatrout, -5.192 and 3.062; southern flounder, -5.260 and 3.125; and sheepshead, - 4.424 and 2.869. Estimations for the WW-axis intercept (a) and slope (b) in the DW-WW equation (WW = a + b DW) were: red drum, - 7.633 and 1.134; black drum, -8.624 and 1.141; spotted seatrout, -33.388 and 1.151; southern flounder, 4.100 and 1.082; and sheepshead, -23.224 and 1.177.

Overstreet, Robin M. 1983. Aspects of the biology of the red drum, (*Sciaenops ocellatus*), in Mississippi. **Gulf Research Reports, Supplement 1:45-68.**

Several hundred specimens of the red drum from Mississippi were critically assessed. Regression equations for standard-length (SL) versus total-length differed between males and females and between small and large members of the same sex. A single regression line represented the weight-SL relationship for males with females. For condition coefficients to be helpful, fish had to be grouped at least by sex, season, and length or stage of maturity. By 12 months of age, most fish were about 30 to 32 cm SL and their distribution ranged throughout Mississippi Sound rather than being restricted to inshore bayou and marsh habitats like younger individuals. Juvenile fish tended to have a high hepatosomatic index (HSI) in winter, and adults had a low one following spawning. The gonosomatic index (GSI) was typically lower than the HSI except for gravid fish in spring and summer. The number of oocytes available for spawning appeared to be much higher than cited in previous reports. The highest calculated fecundity, occurring in a 758 mm SL fish, was 62 million using a volumetric placement method and 95 million using a gravimetric one. Extrapolated estimates for larger fish were much higher. Resting and yolk-vesicle stage oocytes typically had an irregular shape, eccentrically located nucleus, and an abundance of highly basophilic substance in the ooplasm. Throughout a seasonal histological examination, both gonads contained, often in an abundance, Periodic-acid-Schiff-positive granular leukocytes. Spawning took place in late September and October one year, but apparently has occurred over longer periods when conditions of temperature and photoperiod were appropriate. No tagged, 1-year-old fish were returned from farther than 33 km from the point of release. Adults, however, apparently migrated extensively, especially from October through April. Those adults tagged several km south of the barrier islands included individuals caught 778 km away in Texas after 746 days and 316 km away in Florida after 399 days. Another fish apparently migrated at least 120 km in inshore waters in 6 days or fewer. An estimated 25 million kg or more of red drum occurred at one time between the Mississippi River and Mobile Point, Alabama. The red drum served as a host for a variety of parasites, some capable of having an adverse effect on natural stocks, on cultured stocks, and on seafood consumers, and most species known to infect the drum are listed in a table. The red drum has succumbed to some microbial agents, low dissolved oxygen concentration, rapidly dropping temperatures, and other detrimental conditions, some unidentified or unexplained. Probably, considerable mortality can be attributed periodically to environmental or environmentally-influenced conditions and to parasitic infections.

Culture

Colura, R. L., B. T. Hysmith, and R. E. Stevens. 1976. Fingering production of striped bass (*Morone saxatilis*), spotted seatrout (*Cynoscon nebubsus*), and red drum (*Sciaenops ocellatus*), in saltwater ponds. **Proceedings of the World Mariculture Society 7:79-92.**

Striped bass (*Morone saxatilis*) production in two ponds (0.5 ha) at the Marine Fisheries Research Station, Palacios, Texas, durinig 1975 totalled 55,645 fingerlings (11,290/ha) averaging 41 mm in TL. Overall survival was 55.6 percent. Thirty thousand 7-day-old fry stocked in a 0.4 ha pond produced 50,410 fingerlings (126,000/ha) in 45 days or 1.68 kg/ha/day. Survival was 72 percent.

Spotted seatrout (*Cynoscion nebulosus*) production in nine ponds (1.23 ha) during 1975 totalled 6,230 fingerlings (5,065/ha) averaging 48.7 mm TL. Production ranged from 0.00 to 0.89 kg/ha/day. Overall survival was 3 percent ranging from 0. to 18.6 percent. Copepods comprised 55.6 percent of the diet of spotted seatrout less than 25 mm TL. Spotted seatrout 25 mm TL or greater fed primarily on polycheates (40.5 percent) and secondarily on palemonid shrimp (34.2 percent).

Red Drum (*Sciaenops ocellata*) production in 10 ponds (4.8 ha) during 1975 totalled 295,119 fingerlings (61,483/ha) averaging 39.6 mm TL. Production ranged from 013 to 2.17 kg/ha/day. Overall survival was 20 percent ranging from 1.9 to 65.0 percent. Red drum less than 25 mm TL fed almost exclusively on copepods (97.3 percent) Copepods (50 percent) and aquatic insects (45.4 percent) were the primary and secondary food items of red drum 25 mm TL or greater.

Hammerschmidt, Paul C. and Gary E. Saul. 1989. Initial Survival of Red Drum Finglerings stocked in Texas Bays During 1983. **Annual Proceedings of the Texas Chapter, Ameri. Fisheries Society,** 7:13-28. Univ. of Texas Marine Science Institute, Port Aransas, TX., USA.

Red drum (*Sciaenops ocellatus*) were stocked in the San Antonio Bay system during May 1983 and in the Corpus Christi Bay system during September and November 1983. Random samples of fish from each load of stocked fish were placed in cages to determine initial 24-h survival after release. There were no significant differences (P > 0.01) in fingerling survival between bay systems. There were, however, significant differences (P < 0.01) in survival among stocking dates, suggesting differences in the condition of fingerlings among stocking. Mean survival ranged from 62.0 + 14.8 percent on 28 May to 98.7 + 2.3 percent on 12 September. Overall survival of red drum fingerlings held in cages was 89.4 + 2.7 percent indicating that harvesting, transporting and stocking procedures currently used are adequate for survival of stocked fish. This program, therefore, has a potential for rebuilding overharvested populations of red drum.

Luebke, R. W. and Kirk Strawn. 1973. The growth, survival, and feeding behavior of redfish (Sciaenops ocellata) in ponds receiving heated discharge water from a power plant. **Proc. World. Maricult. Soc. Annual Workshop.** 4:134-154.

One hundred young redfish (*Sciaenops ocellata*) were caught by hook and line fishing and stocked in two 0.1 ha ponds. The ponds, located near Baytown, Texas on the Galveston Bay complex, received heated discharge water from the Cedar Bayou Generating Station of the Houston Lighting and Power Company. Once established in the ponds, the fish were taught to eat Purina trout chow pellets. The ponds were drained periodically and redfish were counted, weighed, and measured. Eighty-three fish were alive at the end of the study. Mean daily weight and length gain in the two ponds were 3.15 g and 2.69 g and 0.76 mm and 0.85 mm, respectively. Food conversion values ranged from 2.70 to 6.61. Mean KSL values ranged from 1.67 to 1.94.

McCarty, C. E., J. G. Geiger, L. N. Sturmer, B. A. Gregg, and W. P. Rutledge. 1986. Marine Finfish Culture in Texas: A Model for the Future. *In* R.H. Stroud, ed., **Fish Culture in Fish Management,** American Fisheries Society, Washington, D.C. 249-262.

The Gulf Coast Conservation Association—John Wilson Marine Fish Hatchery (GCCA-JWMFH) located in Corpus Christi, Texas, represents a new dimension in the production of marine finfish species. This facility was built to produce red drum (*Sciaenops ocellata*) fingerlings for stocking Texas coastal waters. During 1983, the GCCA-JWMFH produced over 7.1 million red drum fingerlings over three rearing cycles. Daily fish production averaged 2.4 kg/ha and survival averaged 40 percent During 1984, over 7.1 million fingerlings were produced over two rearing cycles. Daily fish production averaged 4.2 kg/ha and survival averaged 53 percent. This highly successful pond production of red drum was the result of a pond management program utilizing a combination of organic fertilizer (cottonseed meal) and liquid inorganic fertilizers (phosphoric acid and urea). Zooplankton populations were routinely monitored and fry were stocked when zooplankton densities were optimum. This pond management program thus insured that larval red drum were provided with suitable food items of the proper size and in sufficient densities to not only meet their metabolic demands but also to provide an excess for growth. Results of our work with manipulation of pond management timing, fertilization, inoculation of ponds with rotifers, and stocking densities have been proven successful. We are confident that as these techniques are further developed and refined, they will have a significant impact on the culture of other marine fish such as snook (*Centropomus undecimalis*), and spotted seatrout (*Cynoscion nebulosus*).

Richard, W. L. 1981. Aquaculture candidate species for Eastern North Carolina. **N. C. Univ. Sea Grant Publ.,** Rept. No. UNC-SG-WP-81-2. 11 pp.

Summary statements relating to the research and development potentials for culturing various species of finfish and crustaceans in eastern North Carolina are given. Only those species that can be maintained and grown in fresh or slightly brackish water are included because only these types of water are available in quantity at the NCSU aquaculture research and demonstration facility near Aurora, N.C. All of the species discussed possess at least reasonable potential for commercial culture in North Carolina.

Robinson, E. H. and R. R. Stickney. 1982. Texas A&M studies redfish aquaculture. **Aquaculture Magazine,** 8(5):38-39.

Article contains a brief background on red fish culture and outlines red fish culture research to be undertaken at Texas A&M. Research program consists of 1) development of an inland red fish hatchery 2) determination of nutrient requirements and feeding rates 3) development of pond rearing methods for red fish.

Stickney, R. . and J. T. Davis. 1981. **Aquaculture in Texas (USA): A status report and development plan.** Texas A&M Sea Grant College TAMU-SG-O I-XVII, 1-103.

In depth report on resource, economic and legal aspects of aquaculture development in Texas. Includes a summary of redfish culture in Texas.

Trimble, W. C. 1979. Yield trials for red drum in brackish-water ponds, 1976-1979. **Proc. Annu. Conf. Southeast Assoc. Fish Wildl. Agencies.** 33:432-441.

In 2 trials during 1976-1979, juvenile and red drum

(*Sciaenops ocellata*) were reared to marketable size (454 g) in 0.08-ha, brackish-water ponds. In trial 1, survivors from a nursery pond were stocked in 2 production ponds, fed a commercial feed, and harvested when 394 or 532 days old. Less than 1 percent of drum from the first harvest were marketable, and yield was 787 kg/ha with 210-g mean weight, 89 Twenty-one percent from the second harvest were marketable, and yield was 1,062 kg/ha with 335-g mean weight, 75 percent survival and 4.6 feed conversion. In Trial 2, red drum from a second nursery pond were stocked in periodically drained production ponds, fed commercial feeds, then harvested when 715 days old. Standing crop of drum was 2,292 kg/ha during Trial 2, and 33 percent of the drum were marketable at harvest. A 40 percent protein feed ($0.62/kg) poses an economic barrier in Alabama for mariculture of red drum dock-valued at $0.66 to $0.88/kg. Effect of 25 percent protein feed ($0.44/kg) on production of red drum was negated by a fungus epizootic.

DIET

Bass, R. J. and J. W. Avault, Jr. 1975. Food habits, length-weight relationship, condition factor, and growth of juvenile red drum, *Sciaenops ocellata,* in Louisiana. **Trans. Am. Fish Soc.** 104:35- 45.

Food habits of 568 juvenile red drum, *Sciaenops ocellata* (Linnaeus), ranging from 8.0 to 183.0 mm standard length, were determined during the time the fish utilized a Louisiana salt marsh as a nursery area. Potentially available food organisms were sampled during the 7-month study. Some degree of selectivity by juvenile red drum was demonstrated, but generally the most abundant organisms of an edible size were utilized most heavily. Changes in food with increasing size can be described in three phases: 1) red drum less than 15 mm ate zooplankton; 2) between 15 mm and 75 mm the red drum ate mostly small bottom invertebrates and the young of other fish; 3) red drum larger than 75 mm ate decapods (crabs and shrimp) and fish. Some differences between day and night feeding were found. For red drum 65 to 85 mm the dominant food eaten was grass shrimp during the day, whereas at night it was fish. The length-weight relationship was log W =-7.2052 + (4.1913) (long L). The average coefficient of condition was 1.969. Average growth per month ranged between 13.8 and 25.6 mm during the study period.

Boothby, Rea N. and James W. Avault, Jr. 1971. Food Habits, Length-Weight Relationships, and Condition Factor of the Red Drum (*Sciaenops ocellata*) in Southestern Louisiana. **Trans. Am. Fish Soc** 100:290-295.

A total of 349 adult red drum (*Sciaenops ocellata*) were collected from the coastal marsh below Hopedale in southeastern Louisiana, between October 1967 and October 1968. A total of 286 fish (82 contained identifiable food items which are analyzed as to frequency of occurrence and percent of total volume. The main food items in order of occurrence were fish, shrimp, and crabs. Blue crabs, mud crabs, and penaeid shrimp were the crustaceans most frequently eaten, and at least 14 different species of fish were utilized to some degree. Food habits varied substantially from season to season. Fish was the main food item during winter and spring months. Crustaceans, crabs and shrimp combined comprised the bulk of the diet during the summer and fall months. Only slight differences in food habits were detected due to size or sex. Gonadal examination of eight adults indicated that spawning took place between September and December. The length-weight relationship and seasonal condition values were determined. Red drum of a given standard length were generally heavier than previously reported. Condition values from this study represented fish in overall good condition.

Daniels, W. H. and E. H. Robinson. 1986. Protein and energy requirements of juvenile red drum. (*Sciaenops ocellatus*). **Aquaculture** 53: 243-252.

Two studies were conducted to evaluate the effects of different dietary levels of protein and energy on growth and body composition of juvenile red drum (*Sciaenops ocellatus*) grown in a low salinity environment. In each experiment, increased dietary energy resulted in decreased growth, protein efficiency ratio, protein retention, and feed utilization. Whole body lipid tended to increase with increasing dietary lipid. Data from experiment one indicated that 35 percent protein and 1.70×10^4 kJ/g were adequate for good growth and high quality body composition (i.e., low fat and high protein) in red drum reared at 22 to 26 °C. Red drum in the second study, reared at 26 to 33 °C, grew best when fed to satiation on a 44 percent diet at dietary energy levels of 1.54×10^4 and 1.72×10^4 kJ/g. In addition, higher levels of dietary carbohydrate and lipid were associated with an increased hepatosomatic index.

Minello, T. J. and R. J. Zimmerman. 1983. Fish predation on juvenile brown shrimp, *Penaeus aztecus Ives:* The effect of simulated Spartina structure on predation rates. **J. Exp. Mar. Biol. Ecol.** 72(3):211-231.

The effect of artificial Spartina structure on the predation rates of four estuarine fish on juvenile brown shrimp (*Penaeus aztecus*) was examined under laboratory conditions. Vegetative structure reduced predation rates of red drum (*Sciaenops ocellatus*) and speckled trout (*Cynospcion nebulosus*). Pinfish and Atlantic croaker were inefficient predators, needing several strikes before successfully capturing prey. Although pinfish and speckled trout appeared to be strictly visual feeders, Atlantic croaker and red drum could apparently detect and feed upon shrimp through other sensory mechanisms. to be related to the effect of vegetative structure on predation rates.

Overstreet, Robin M. and Richard W. Heard. 1978. Food of the red drum, *Sciaenops ocellata*, from Mississippi Sound. **Gulf Research Reports,** 6(2):131-135.

Examined digestive tracts of the red drum in Mississippi Sound contained mostly decapod crustaceans. Crustaceans accounted for 34 of 59 encountered taxa, more than reported from any other region. Nevertheless, the general diet for 104 fish with food contents out of the 107 examined is similar to that reported for red drum in several other studies from other areas. In addition to crustaceans, fishes followed by polychaetes occurred as the most important items (in 99, 43, and 15 percent of the drum with food, respectively). Blue crabs occurred in even more drum than the frequently encountered penaeid shrimps. Other commercial species were negligible in the diet. Sixteen large drum from Georgia beaches were also examined; unlike those from Mississippi, many of these contained echinoderms, but not polychaetes or penaeids. We suggest that the red drum's migrations may be regulated by optimal abundance of specific types of dietary organisms.

Reid, G. K., Inglis, A. and H. H. Hoese. 1956. Summer foods of some fish species in East Bay, Texas. **Southwestern Naturalist,** 1(3):100-104.

Stomach contents are reported for 83 croaker (*Micropogon undulatus*) and 280 sand trout (*Cynoscion arenarius*); also for a few individuals each of sheepshead (*Archosargus oviceps*), sand perch (*Bairpiella chrysura*), gafftop sail catfish (*Bagre marinus*), spadefish (*Chaetodipterus faber*), flounder (*Paralichthys lethostigma*), and redfish (*Sciaenops ocellata*). Variety is characteristic in the diets of all species. Populations of these fishes depend upon an adequate supply of small fishes, shrimps, and mollusks.

Robinson, E. H., W. H. Daniels, C. D. Williams and W. A. Wurts. 1984. Diet development and environmental ion requirements for fingerling redfish (*Sciaenops ocellatus*). **Proc. of the 1984 Fish Farming Conference and Annual Convention Fish Farmers of Texas.** Texas A&M University. pp. 44-50.

An overview of redfish research at the Aquacultural Research Center at Texas A&M is given.

Williams, C. W. 1985. The optimal dietary lipid requirement and development of an effective amino-acid test diet for juvenile red drum, *Sciaenops ocellatus*. M.S. Thesis, Texas A&M University.

Studies were conducted to evaluate the dietary lipid requirement and to develop a diet suitable for use in determining the lysine requirement of juvenile red drum (*Sciaenops oscellatus*). Each experiment was conducted utilizing a brackish water (5-6 ppt) recirculating system which supplied water to a series of 40 1 aquaria. Fish were stocked at a rate of 12 fish per aquarium. In the lipid study all fish were fed the appropriate diet twice a day for a period of six weeks. Dietary lipid levels ranged from 1.7 to 18.7 percent total lipid. Fish growth, feed conversion and survival were best at dietary lipid levels between 7.4 and 11.2 percent. Dietary lipid levels of 15 percent or greater depressed growth and resulted in poorer feed conversion. Whole-body lipid levels increased as dietary lipid levels increased up to 7 percent dietary lipid, then decreased when dietary levels were elevated to 15 percent or greater. Apparently, there was some interference with lipid utilization at the higher dietary levels. Whole-body fatty acid composition was reflective of dietary lipid. Both purified and practical diets were utilized in order to determine the lysine requirement. However, none of the diets tested proved acceptable to red drum. Amino acid composition of red drum eggs is presented and it is suggested that the amino acid pattern of the eggs may be reflective of the amino acid needs of red drum.

DISEASE

Henley, M. W., and D. H. Lewis. 1976. Anaerobic bacteria associated with epizootics in grey mullet (*Mugil cephalus*) and redfish (*Sciaenops ocellata*) along the Texas Gulf coast. **Journal of Wildlife Diseases.** 12:448-453.

Anaerobic bacteria tentatively identified as species of Catenabacterium were recovered from brain, liver, kidney and blood of fish involved in a massive epizootic of grey mullet (*Mugil cephalus*) and redfish (*Sciaenops ocellata*). Pathogenicity was demonstrated for grey mullet (*M. cephalus*) and sea catfish (*Arius felis*), but not for channel catfish (*Ictalurus punctatus*) or white mice. Diseased fish were disoriented, weak and swimming at the surface of the water. Tioglycolate and salt bovine blood agar containing 40 ug/ml gentamicin were useful as selective culture media.

DISTRIBUTION

Heffernan, T. L. Survey of adult red drum (*Sciaenops ocellata*), 1973. **Tex. Parks Wildl. Dept., Coastal Fish Proj. Rep**:37-66.

Surf zone samples were collected with nets, multi-hook bottom and surface lines and rod and reel to obtain availability patterns of adult red drum (*Sciaenops ocellata*) along the Texas coast. Adult red drum did not consistently appear in the surf zone until late November when significant catches were obtained near Cedar Bayou Pass.

The rod and reel was the most efficient sampling period with 975.8 hook hours yielding 237.7 kg (524.1 lb) of game fish. The multi-hook surface line fished 7,802.8 hours and produced 23.5 kg (51.4 lg), the bottom lines fished 3,717.0 hook hours and caught 15.2 kg (33.1 lb). Gill and trammel net catches totaled 80.2 kg (176.66 lb) of game fish for 11,968 hook hours based on a 1.5 m (5 ft) net unit hour equivalent to one hook hour. Catch per hook hour was 0.24436 kg (.537 lb) for rod and reel, 0.00272 kg (0.007 lb) for surface line, 0.00363 kg (0.009 lb) for the bottom line and 0.0067 kg (0.015 lb) per hook hour equivalent for the nets.

Four adult red drum were obtained at Cedar Bayou and transferred alive to the Palacios Research Station for propagation studies.

Holt, Scott A., Christopher L. Kitting, and Connie R. Arnold. 1983. Distribution of Young Red Drums among Different Sea-Grass Meadows. **Transactions of The American Fisheries Society** 112:267- 271.

Sea-grass meadows appear to be a primary habitat for young red drums, *Sciaenops ocellatus* in south Texas estuaries. The abundance of small red drums (6-27 mm standard length) in different meadows averaged 0.10-0.80/m^2. Density estimates of young red drums showed no significant or nearly significant differences between the two types of sampling gear (benthic sled or 1.0-m^2 cage) or among sampling sites, which differed in plant height, blade density, or water depth. Small red drums were not found on large (>5 m across) nonvegetated sites; however, the ecotone between sea grass and nonvegetated bottom had significantly more red drums than did homogeneously vegetated sites. Heterogeneous sea-grass meadows, therefore, may support more young red drums than homogenous ones.

Mansueti, R. J. 1960. Restriction of very young red drum, *Sciaenops ocellata*, to shallow estuarine waters of Chesapeake Bay during late autumn. **Chesapeake Sci.** 1:207-210.

Young-of-the-year red drum, ranging from 20-90 mm total length and one to several months old were found to be restricted to a shoal estuarine habitat in Chesaeake Bay during autumn months. Extensive trawl data from deep water during autumn and winter illustrate their absence from deep water throughout the bay. It is postulated that beginning in September planktonic *Sciaenops* are carried from the Atlantic ocean spawning area into Chesapeake Bay by the net upstream movements of deep sub-surface water currents of high density. After metamorphosis to a free-swimming stage, the young restrict their activities to shallow waters which may be important but temporary nursery areas. They may then rapidly descent to the ocean in early winter. Evidence is presented to show that the spawning may begin in August off the mid-Atlantic, a month earlier than was formerly believed.

Osburn, Hal R., Gary C. Matlock, and Albert W. Green. 1982. Red drum (*Sciaenops ocellatus*) movement in Texas Bays. **Contributions in Marine Science.** 25:85-97.

Red drum movement in Texas bays was studied by means of fish tagged with internal abdominal anchor tags and subsequently released. Intrabay movement was minimal with the majority (>71 returned from the Gulf or from outside the bay in which they were tagged. Movement patterns of red drum, however, differed significantly among some bay systems. There was no detectable relation between minimum distance moved by red drum and seasons or size of fish. The previously reported mass migration of red drum in winter was not detected in this study. Texas bay systems can be considered closed systems for fish 305-625 mm TL when managing red drum stocks. Due to the restricted movement patterns, local populations of red drum could be vulnerable to intense fishing pressure.

Pafford, J. M. 1981. Seasonal movement and migration of red drum (*Sciaenops ocellatus*) in Georgia's coastal waters. **Estuaries 4(3):279-280.**

To determine seasonal and/or migratory movement patterns of red drum, (*Sciaenops ocellatus*), a mark recapture study was initiated in January 1979 using Floy FD-68-BC and the Howitt internal anchor tags. Over 200 red drum have been tagged in the Altamaha, St. Simons and St. Andrew estuarine systems to date with approximately 30 percent recovery rate. Recoveries of tagged fish provide evidence that the smaller specimens (<7kg) are found primarily on the beaches and shoals during the colder months. Only 10 percent of the recoveries migrated out of the tagging area and all indicated a definite northward movement with the maximum distance traveled of 161 km and averaging 14.5 km while the maximum time at large is presently 514 days, averaging 143.9 days.

Ross, J. L., J. S. Pavella and M. E. Chittenden, Jr. 1983. Seasonal occurrence of black drum, *Pogonias cromis*, and red drum, (*Sciaenops ocellatus*), off Texas. **Northeast Gulf Sci.,** 6:67-70.

Data are presented on the seasonal occurrence and distribution of black drum (*P. cromis*) and red drum (*S. ocellata*) off Texas. Findings suggest a persistent recurrence of black drum from December to June off Texas and a persistent recurrence of red drum in offshore waters during winter.

Wakeman, John M. and Paul R. Ramsey. 1985. A Survey of Population Characteristics for Red Drum and Spotted Seatrout in Louisiana. **Gulf Research Reports,** 8(1):1-8.

Red drum and spotted seatrout stocks were sampled from seven separate study areas along the Louisiana coast and from one estuarine area in Texas, with additional intensive temporal (monthly) and microgeographic (range of salinity regimes) samplings being carried out in one Louisiana study area. Condition coefficients, which did not appear to be affected by salinity regimes within the microgeographic sampling area, varied significantly according to study area, with Texas fish showing significantly lower condition coefficients than Louisiana fish. Von Bertalanffy growth equations were fitted and annual mortality rates were estimated to obtain preliminary estimates of yields, population numbers, and densities of these species in Louisiana.

FISHING, COMMERCIAL

Adkins, G., Tarver, J., Bowman, P. and B. Savoie. 1979. A study of commercial finfish in coastal Louisiana. **La. Dept. Wildl. Fish., Tech. Bull.,** July 1979. pp 92, ref. 97.

From January 1976 through December 1977, gill net and trammel net samples were made in coastal Louisiana in two major geographic areas. Because of differences in environmental parameters, samples were made differently in these areas, and data are presented separately. A total of 35 species were captured in gillnets, with spotted sea trout being most abundant. An average weight of 1.5 pounds was recorded, with more females than males being present. Gill nets captured fewer fish and species per effort, generally, and the animals were larger. Tagging returns revealed very limited movement for both spotted sea trout and red drum, with most movement being associated with temperature changes. Fish were found in all temperatures and salinities recorded, although spotted seatrout were more abundant in waters of 15 ppt or greater and in temperatures of 20 C or greater.

Ferguson, M. O. 1986. Characteristics of Red Drum and Spotted Seatrout Commercial Fishermen in Texas. **North American J. Fish. Mgn.** 6:344-358.

The 67th Texas Legislature directed the Texas Parks and Wildlife Department in 1981 to predict the economic impact of pending legislation banning the sale of red drum (*Sciaenops ocellatus*) and spotted seatrout (*Cynoscion nebulosus*) caught from Texas water. Commercial fishing licenses and individual finfish sales reports were used to determine numbers, ages, and sexes of licensed and reporting commercial finfish fishermen in Texas; license renewal rates; continuity of fishing activity; method of harvest; extent of licensing; reporting and harvesting violations; earnings from the finfish fishery; and economic dependence upon red drum and spotted seatrout landings. Over 1,000 commercial finfish fishermen were licensed or reported finfish sales from fiscal year 1979 to fiscal year 1981. However, less than 300 had a significant economic dependence upon red drum and spotted seatrout landings. The impact of sales ban was predicted to be greatest in the areas around the Corpus Christi and lower Laguna Madre bay systems, where many of the fishermen lived and where the majority of red drum and spotted seatrout were harvested. Statewide economic impacts were estimated to be small because commercial finfish fishermen represent less than 0.3 percent of the employed work force, and sales from red drum and spotted seatrout were less than 0.1 percent the real of disposable income.

Hefferman, Thomas L. 1973. Survey of Adult Red Drum (*Sciaenops ocellata*). Texas Parks and Wildlife Dept. **Coastal Fish Project Report**. 37-66.

Surf zone samples were collected with nets, multi-hook bottom and surface lines and rod and reel to obtain availability patterns of adult red drum (*Sciaenops ocellata*) along the Texas coast. Adult red drum did not consistently appear in the surf zone until late November when significant catches were obtained near Cedar Bayou Pass.

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Four adult red drum were obtained at Cedar Bayou and transferred alive to the Palacios Research Station for propagation studies.

GENETICS

Ramsey, P. R., and J. M. Wakeman. 1983. **Final Report, Board of Regents, Research Development Program,** Louisiana Tech Univ., 51 p.
A demographic and genetic survey of Louisiana coastal populations of red drum (*Sciaenops ocellata*) and spotted seatrout (*Cynoscion nebulosus*).

Wilder, W. R. 1986. **Ph.D. Thesis, Rice University,** Houston, Texas. **NEED REFERENCE MATERIAL**

An electrophoretic analysis of Texas coastal populations of red drum (Sciaenops ocellata), with reference to geographical sub. populations of major embayments.

HATCHERY

Anonymous. 1983. Texas biologists cross redfish and black drum. **Mar. Fish. Rev.**, 45(10-12): 72pp.

Biologists at the Texas Parks and Wildlife Department's Marine Fisheries Research Station in Palacios have successfully produced a hybrid cross between the red drum (*Sciaenops ocellata*) (redfish) and black drum (*Pogonias cromis*). They hope the hybrid will be a hardy, fast-growing sport fish. About 50,000 fertilized eggs were produced by a female black drum and a male redfish earlier in 1983. The resulting fry were placed in ponds until they reached 1 1/2 inches in length, and then were stocked experimentally in Lake Creek Reservoir near Waco.

Arnold, C. R., Bailey, W. H., Williams, T. D., Johnson, A. and J. L. Lasswell. 1977. Laboratory spawning and larval rearing of red drum and southern flounder. **Proc. Annu. Conf. Southeast. Assoc. Fish. Wildl. Agencies.** 31:437-440.

Laboratory spawning and larval rearing studies were conducted with red drum (*Sciaenops ocellata*) and southern flounder (*Paralichthys lethostioma*) from 1974-1977. Adult fish were placed in 29,000 l. 92 kl spawning tanks equipped with biological filters and subjected to photoperiods and temperatures regulated to simulate seasonal variations. Red drum spawned 52 times producing eggs. Southern flounder spawned 13 times producing eggs. Eggs were collected and incubated, and larvae were reared to fingerling size. This paper describes techniques used to produce fingerling red drum and southern flounder.

PARASITES

Ho, J. 1966. Redescription of *Echetus typicus* Kroyer, a caligid copepod parastitic on the red drum, *Sciaenops ocellatus* (Linnaeus). **The Journal of Parasitology.** 52(4):752-761.

Echetus typicus Kroyer is a copepod hitherto known only as parasitic on the red drum, *Sciaenops ocellatus* (L.). A redescription of both sexes is given, based on 14 females and two males taken from *S. ocellatus* in Apalachee Bay, Florida. A comparison with specimens in the U.S. National Museum collections shows them to be identical. The Echetinae Yamaguti, 1963, and the Mappatinae Yamaguti, 1963, two of the five subfamilies of Caligidae, are discussed in regard to their definition and range. It is suggested that, should the Echetinae be accepted on the basis of this subfamily being a group where the adult females show a highly modified fourth thoracic segment, genital segment, and abdomen, then Caligodes Heller, Parechetus Pillai, and Pseudopetalus Pillai should also be included, and the erection of the mappatinae ought to be reconsidered, for it was based on a single and questionable genus Mappates Rangnekar. Caligulus Heegaard should be included in the Caligidae, and grouped together with Mappates, if the latter has only the fourth thoracic segment free.

Iverson, E. A. and B. Yokel. 1963. A myxosporidian (Sporozoan) parasite in the red drum, *Sciaenops ocellatus*. **Bull. Mar. Sci. Gulf Caribb.** 13:449-453.

A description is given of a new species of myxosporidian parasite located in the intestine and pyloriccaeca of the red drum, caught in saline waters of south Florida.

During studies upon the life history of red drum, *Sciaenops ocellatus* myxosporidian cysts were found in the intestine and pyloric caeca. This parasite is similar to a myxosporidian described from red drum captured in North Carolina waters (Linton, 1905). Linton's description is brief without a specific name for the parasite. In this paper we provide data on the parasite from red drum caught in south Florida waters during 1961-1962, and establish a specific name.

The parasites were studied in both fresh and formalin-preserved preparations.

The study of the red drum was supported by the Burearu of Sport Fisheries and Wildlife. The authors are grateful for helpful criticism of this manuscript by Dr. Elmer R. Nobe, University of California, Santa Barbara.

Johnson, S. K. 1986. **Control of *Amyloodinium ocellatum*** (Brown, 1931), A Parasite of Marine Fishes in Aquaculture. Presented at the lst Inter-American Congress of Aquaculture, Salvador, Brasil, September 1986.

There are a number of parasitic dinoflagellates. Those invading fish were taxonomically reviewed by Lom (1981)

who disclosed that all true ectoparasitic dinoflagellates of fish were first described as members of the genus *Oodinium*. This taxon includes invertebrate parasites.

Figure 1 depicts the general structure of the parasitic state of the fish infesting genera. The genus *Crepidoodinium* has one species, *C. cyprinodontum* (Lawler 1967), which is associated with cyprinodontid fishes in brackish water environments. *Piscinoodinium* spp. are associated with a variety of freshwater fishes and the monotypic *Amyloodinium* with a variety of marine fish species. Lom (1981) reports on two other monotypic genera, one of which (*Oodiniodies* Reichenbach-Klinke, 1970) has questionable status as a true dinoflagellate and the other (*Ichthyodinium* Hollande et Cachon, 1953), which is an internal parasite of the fry of clupeoid fishes. *Amyloodinium ocellatum* (Brown, 1931) may be represented by several "strains". Future work with parasitic dinoflagellates of fishes probably will reveal more species.

Riggin, G. T. and A. K. Sparks. 1962. A new gasterostome, *Bucephaloides megacirrus*, from the redfish, *Sciaenops ocellata*. **Proc. Helminthol. Soc. Wash.** 29:27-29.

During examination of fishes from the Gulf of Mexico, many specimens of a gasterostome fluke were recovered from the redfish (*Sciaenops ocellata*). Seven hosts, which harbored twelve to several hundred flukes each, were collected at Bayou Rigaud, Grand Isle, Louisiana from December 1951 to August 1956. A single redfish collected from Alligator Harbor, Franklin County, Florida in November 1955 also harbored several hundred worms of the same species. The flukes belong to the genus *Bucephaloides* Hopkins, 1954 are herein described as a new species.

The Florida specimens, approximately half of which were flattened by coverslip pressure, were fixed in alcohol-formal acetic acid solution and stored in 70 percent alcohol until stained and mounted. Frontal, sagittal and cross sections of unflattened worms were cut at 10 microns and stained with Delafield's hematoxylin and cosin. Whole mounts of flattened specimen were stained with Delafield's hematoxylin, Ehrlich's acid hematoxylin, Semichon's acetocarmine and Borax carmine.

The Louisiana specimens were fixed in Gilson's fluid under coverslip pressure and stored in 50 percent alcohol. They were stained in Delafield's hematoxylin and alum cochineal.

Measurements of the Florida specimens are given first, followed by the measurements of those from Louisiana. All measurements are of flattened specimens and, unless stated otherwise, are in millimeters. Drawings were made with the aid of a microprojector. The following description is based on 11 specimens from Florida and three from Louisiana.

PHYSIOLOGY

Caldwell, C.A. and J.R. Tomasso. 1984. An Assessment of Stocking-Induced Stress in Fingerling Red Drum. **Proc. of the Texas Chapter, American Fisheries Soc.,** 7:1 (Abstract).

Stress induced by handling, hauling and net confinement was evaluated in red drum fingerlings (*Sciaenops ocellatus*) weighing 0.2-0.8 g. Changes in plasma glucose concentrations were used as a general indicator of stress, and changes in plasma chloride concentrations were used as an indicator of osmoregulatory dysfunction. Samples were taken at intervals during harvest and transport to stocking sites. In addition, fingerlings confined in a net for nine hours were sampled at intervals in an attempt to determine the maximum possible changes in plasma glucose and chloride concentrations. Changes in the stress indicators generally followed the same trend for both net stress and stocking procedures. Plasma glucose levels exhibited an initial increase and a subsequent decrease to or below baseline levels (50 mg/100 ml) in both net confined and hauled fish. Plasma chloride levels exhibited a prolonged elevation from baseline (117 meg/liter) in response to both net confinement and hauling. Cumulative mortality was 53 percent after 9 hours of net confinement. The degree and consistency of the red drum's physiological responses to confinement and hauling stress were less than those observed in other fish species.

Crocker, P. A., C. R. Arnold, J. A. DeBoer and G. J. Holt. 1983. Blood osmolality shift in juvenile red drum, *Sciaenops ocellatus* L. exposed to fresh water. J. **Fish. Biol.,** 23(3):315-319.

Juvenile red drum, *Sciaenops ocellatus*, measuring 40-49 mm S.L. tolerated abrupt transfer from 28 ppt. salinity sea water to freshwater (< 1 ppt.). A significant shift in blood osmolality from a level of about 250 mosmol 1 super (-1) in sea water to a new level of about 311 mosmol 1 super (-1) was experienced by fish after 48 h in fresh water. The shift was followed by a slight reduction in blood osmolality to a sustained level of 300 mosmol 1 super (-1) after 96 h exposure to fresh water. The overall effect of increased environmental calcium on blood osmotic pressure of young red drum was not significant at the level tested.

Crocker, P.A., Gotto, M., DeBoer, J.A., Holt, G.J. and C.R. Arnold 1985. Notes on the surface structure of the gill arch epithelium in juvenile red drum, *Sciaenops ocellata*, exposed to salt and fresh water. **Copeia** 1985(2):515-518.

Gill arches removed from saltwater and freshwater acclimated juvenile red drum were observed using electron microscopy. Chloride cells and other gill surface features were examined and differences between the surface features of SW and FW acclimated fish were noted.

Grier, H. J. and R. Taylor. 1984. Testicular recrudescence in the red drum, *Sciaenops ocellata*. **Am. Zool.** 24(3):22a

Spermatogenesis in wildstock red drum,*Sciaenops ocellata*, begins in July and proceeds through October. Spawning occurs during September and October. Male red drum collected from November to June were nonreproductive. Between July and October, the testes undergo dramatic enlargement as tubules increase in width and particularly in length. During this recrudscence period, histological observation of the tubules revealed a persistent clustering of spermatogonia at the distal terminis. Mitotic activity of these sperma togonia is hypothesized to produce both tubule elongation and germ cell source which is found along the tubule wall as elongation occurs. Initially, spermatogenesis occurs along the entire length of the tubule. However, as spawning approaches proximal spermtogonia will produce sperm, thereby resulting in a functional conversion in the tubule from sperm production to sperm storage. In ripe, prespawn males, tubules lack distal clusters of spermatogon resulting in a hypothesized cessation of elongation. Testicular recrudescence in *S. ocellatus* is a dynamic growth process requiring coordinated activities between cell types.

Holt, J., R. Godbout and C. R. Arnold. 1981. Effects of temperature and salinity on egg hatching and larval survival of red drum, *Sciaenops ocellata*. U.S. Nat. Mar. Fish. Soc., **Fish. Bull.,** 79(3):569-573.

The red drum, *Sciaenops ocellata*, is a sciaenid fish distributed along the eastern coast of North America from Massachusetts to southern Florida and along the Gulf coast at least as far south as Tampico, Mexico. The objective of this study was to determine the optimum temperatures and salinities for hatching and growth of red drum eggs and larvae. The best conditions for hatching and 24-h larval survival were 30 ppt. salinity and 25°C. Survival of larval red drum to 24 h was influenced by both temperature and salinity. Poorest survival was at 30°C and 15 ppt. Temperature was associated with significant differences in survival of 2-week-old larvae. The lowest temperature (20° C) resulted in reduced survival rate. The effect of temperature on larval growth rate was pronounced, growth at 20°C was much slower than at 20° or 30° C. Salinity had little influence on growth.

Lee, W.Y., G.J. Holt, and C.R. Arnold. 1984. Growth of red drum larvae in the laboratory. **Trans. Am. Fish. Soc.,** 113(2):243-246.

Larval red drums, *Sciaenops ocellata*, were raised at 24 C and 28 C in the laboratory. Length of newly hatched larvae increased steadily but their mean weights first decreased over the first 5 days (until yolk was fully resorbed), then increased rapidly. At day 15 there were significant differences in mean standard lengths between larvae grown at the two temperatures (4.8 mm at 24 C, 5.1 mm at 28 C, $P < 0.05$), larvae grown at the higher temperature were also markedly heavier (363.7 mu g versus 186.3 mu g, $P < 0.01$). Transition from the yolk-sac stage to active feeding occurred at day 5-6 when larvae were about 2.7 mm long.

Miranda, L. E., and A. J. Sonski. In press. Survival of red drum fingerlings in fresh water: dissolved solids and thermal minima. **Proceedings of the Annual Conference Southeastern Association of Fish and Wildlife Agencies.**

Laboratory bioassays were conducted to estimate lower dissolved solids and temperature thresholds of red drum (*Sciaenops ocellatus*) fingerlings. Tolerance of low total dissolved solids (TDS) was measured by subjecting fingerlings to various test concentrations for 240 hours at 21 + 1 C. Higher mortality in fresh water than in diluted sea water with similar TDS suggested that concentration of individual ions may be more important than TDS to survival of red drum in fresh water. Survival in solutions of increasing sodium chloride concentrations, but constant TDS, increased and was greater than 80 percent at chloride levels above 130 mg/l. Tolerance of low temperature was measured by exposing fingerlings to different temperature regimes in freshwater adjusted to a concentration of 150 + 5 mg/l chloride. Lower lethal temperatures ranged from 3.0 to 0.8 C when water temperature was reduced 1 C/day. When temperature was reduced 1 C/3 days and maintained at 4 C, time to death was between 48 and 168 hours. Regardless of rate of temperature decrease, feeding ceased between 9 and 5 C. Stocking of red drum fingerlings should be successful in fresh waters that have chloride concentrations exceeding 130 mg/l and temperature above 9 C.

Robertson, Lori, Peter Thomas, and Connie Arnold. 1984. Physiological Evaluation of Transportation Stress in Culture of Juvenile Red Drum. **Proc. of the Texas Chapter of the American Fisheries Soc.,** 7.

Red drum (*Sciaenops ocellatus*) has become an important species for its mariculture potential. Transportation is traumatic to juvenile red drum and elicits stress responses which can be measured by changes in plasma cortisol, glucose and osmolality. The objectives of the study were to: 1) determine the degree of stress induced in juvenile red drum by transportation procedures, 2) compare the stress responses elicited by transportation in ambient seawater versus a low salinity medium, and 3) determine the efficacy of using anesthetics during transport. Blood parameters measured were plasma cortisol, glucose and osmolality.

In one experiment, red drum (21 cm mean standard length; 2 g mean weight) were acclimated, to 32 o/oo seawater and hauled for 5.5 hours with and without 5 mg MS-222/liter. No mortalities occurred. In another experiment, red drum (13 cm and 40 g) were acclimated to low salinity seawater (4 o/oo), and were hauled for 2.5 hours with and without 25 mg MS-222/liter. Survival was 98 percent. In both experiments, plasma cortisol and glucose levels were significantly elevated during transport. However, plasma osmolality increased during seawater transport and decreased in the 4 o/oo transport. The use of MS-222 did not decrease the magnitude of the stress responses nor did it increase survival.

This study indicates that juvenile red drum can be transported in high and low salinity water with little or no mortality. However, these transport procedures rapidly induced classical stress responses, as indicated by changes in plasma cortisol, glucose and osmolality. No apparent benefits were observed by using MS-222 during transportation of juvenile red drum.

Vetter, R. D., R. E. Hodson, and C. Arnold. 1983. Energy metabolism in a rapidly developing marine fish egg, the red drum (*Sciaenops ocellata*). **Can. J. Fish. Aquat. Sci.** 40(5):627-634.

Rates of utilization of different lipid classes, glycogen, protein, and adenine nucleotides in the eggs of red drum (*Sciaenops ocellata*) were measured concurrently throughout embryonic development. Red drum eggs are small, pelagic and very rapidly developing, going from fertilization to hatching in as little as 19 hours at natural spawning temperatures. Wax esters comprised about 50 percent of the neutral lipid reserve along with triglyceride. Lipid was quantitatively the most important energy reserve supplying virtually all of the catobolic demand. Total lipid content decreased 30 percent from 23.6 to 16.6 mg/g during development. Glycogen decreased 53 percent from 0.279 to 0.103 mg/g. Protein did not contribute to catabolism. Total adenylates, primarily ATP, decreased from 564 to 271 mol/g. Adenylate energy charge decreased from 0 .87 to 0.58.

Wakeman, J. M. and D. E. Wolschlag. 1983. Time course of osmotic adaptation with respect to blood

serum osmolality and oxygen uptake on the euryhaline teleost, *Sciaenops ocellatus* (red drum). **Contrib. Mar. Sci.,** 26:165-177.

Blood serum osmolalities of juvenile red drum, (*S. ocellatus*) acclimatized at 24°C and 30 ppt. salinity and abruptly transferred to separate salinities of 40, 30, 20, 10 and 2 ppt. became stabilized at these new levels after 24 hours. Variability at the 40 and 2 ppt. salinity levels was greater than at the intermediate levels. Isotonicity was at about 11 ppt. (320 mOsm kg super (-1). There was little change in hematocrit values over the various salinity changes or time intervals. There was no indication that selective mortality removed individuals with excessive osmotic stress response.

Wurts, W., and E. H. Robinson. 1986. The effects of environmental calcium and magnesium on growth and survival of juvenile red drum, *Sciaenops ocellatus*, in marine and fresh waters. **Proceedings of the Seventeenth Meeting World Mariculture Society.** 17:37.

Two preliminary and two secondary eight week trials were conducted in 114 liter aquaria equipped with individual biofilters. Triplicate groups of juvenile red drum were used in each treatment. Well water with trace levels of calcium and magnesium was used to formulate fresh (0.56-1.2 ppt) and salt (35 ppt) waters, differing in concentrations of calcium or magnesium. Acid washed and sand was used in secondary trials to eliminate any potential source of calcium or magnesium contamination.

Preliminary studies in salt water showed 100 percent mortality within six hours in all treatments with 60 ppm calcium (those with 400 ppm showed no adverse effects). In the secondary trial 100 percent mortality was observed within 96 hours in all reatments containing less than 200 ppm calcium. Nitrifying bacterial failed to convert nitrogenous waste products in all trace magnesium treatments.

Results from the preliminary freshwater trial indicated no apparent differences in growth or survival in treatments containing 10-300 ppm calcium. Growth suppression was observed at the 400 ppm calcium level. The secondary freshwater trial demonstrated that red drum perform poorly (4.4-13 percent survival in 96 hours) in fresh water containing trace quantities of calcium. Environmental magnesium appeared to offer no advantage for growth or survival in fresh water at levels between 1-1,200 ppm.

These results are consistent with the generally recognized importance of calcium in osmoregulation. Long term growth and survival were poor in both secondary trials. Although it has not yet been evaluated, there is a possibility that acid washing removed an essential environmental trace element previously supplied by the biofiltration gravel (pea gravel was used without acid washing in preliminary trials).

RESOURCE MANAGEMENT

Crocker, P. A., C. R. Arnold, J. A. DeBoer, and J. D. Holt. 1981. Preliminary evaluation of survival and growth of juvenile red drum (*Sciaenops ocellata*) in fresh and salt water. **Journal of the World Mariculture Society** 12: 122-134.**NEED ANNNOTATION**

Lasswell, J. L., Garza G. and W. H. Bailey. 1977. Status of marine fish introductions into the fresh waters of Texas. **Proc. Annu. Conf. Southeast. Assoc. Fish Wildl. Agencies.** 9 October 1977.

Techniques have been developed for spawning adult southern flounder (*Paralichthys lethostioma*), spotted seatrout, (*Cynoscion nebulosus*), and red drum (*Sciaenops ocellata*) and rearing their larvae for freshwater acclimation and introduction into heated freshwater reservoirs in Texas. Egg production, percentage egg fertilization, percentage hatch, percentage return of larvae stocked into laboratory aquaria and hatchery ponds, and potential for fingerling survival in fresh water were compared for the three species. Red drum was found to be the most suitable for culture and introduction into freshwater. A diagram and estimated construction cost of a laboratory facility for holding and spawning marine fishes is presented.

Lorio, W., T. Heaton and O. Dakin. 1980. The relative impact of netting and sport fishing on economically important estuarine species. **Miss.-Ala. Sea Grant Consort.**, Ocean Springs, Miss. 46 pp.

The overall objective of this study was to determine the relative impact of commercial netting and sport fishing on spotted seatrout, red drum, and Spanish mackerel. The catch was estimated by a creel census roving clerk technique. An intense controversy exists along the coasts of Mississippi and Louisiana concerning the catching of sport fish by commercial fishermen using 1,000 and 2,000 feet of nylon and monofilament gill nets and trammel nets. The sport fishermen contend that these nets, are depleting the population of spotted seatrout and red drum. Fishery biologists contend that commercial fishing pressure has had little effect on these populations; however, little data is available to substantiate the claim.

Matlock, Gary C., Patricia L. Johansen and Joseph P. Breuer. 1979. Management of red drum in a Texas estuary—a case study. **Proc. Ann. Conf. S.E. Assoc. Fish and Wildl. Agencies** 33:442-450.

In September 1974 the Texas Parks and Wildlife Commission banned the use of plastic baits on trotlines because these baits were thought to be selective for small (<500 mm) red drum (*Sciaenops ocellata*). The size of red drum landed by commercial fishermen before (1972-1974) and after (1974-1978) the ban was compared with the size of fish collected during Texas Parks and Wildlife Department trammel net surveys in order to determine whether the ban had any effect on either the commercial catch or fish availability. Fish landed by commercial fishermen were significantly larger after the ban than before; therefore, it appears that plastic baits are selective for small red drum and that the ban resulted in the desired effect. The larger red drum in the commercial landings as compared with trammel net caught fish may have resulted from such factors as fishing method (hook size and bait type), fishing location and/or culling. The reason for the increase in size of trammel net caught red drum after the ban is not clear but may involve a complex series of effects such a year class success, changes in recreational fishing pressure or general decrease in commercial harvest. While this study does not prove conclusively that the ban on plastic baits altered the size composition of commercially harvested red drum, it does indicate that the desired result was achieved. Studies of the effects of management regulations of fish populations are necessary to evaluate the effectiveness of such regulations.

Perret, W. S., J. E. Weaver, R. O. Williams, P. L. Johansen, T.D. McIlwain, R. C. Raulerson and W. M. Tatum. 1980. Fishery profiles of red drum and spotted seatrout. **Gulf States Marine Fisheries Comm.,** Ocean Springs, Miss. 65 pp.

The red drum (*Sciaenops ocellata*) and the spotted seatrout (*Cynoscion nebulosus*) support valuable commercial and recreational fisheries along the northern Gulf Coast. Since most of their life cycles are spent in state waters, regulation of the fishery has been the responsibility of state governments. The management of the fishery is discussed bearing in mind the social, economic and political factors that enter management decisions.

SPAWNING

Roberts, D. E., Jr., Harpster, B. V., and G. E. Henderson. 1978. Conditioning and induced spawning of the red drum *Sciaenops ocellata* under varied conditions of photoperiod and temperature. **Proc. World Mariculture Society,** 9:311-332.

Vitellogenesis, culminating in spawning, was artificially induced in red drum, *S. ocellata*, using photoperiod and temperature as exogenous stimuli. Maturation was induced out of season during the normal refractory period of red drum found in central Florida waters. Four regimes were compared to a control. Three regimes culminated in advanced vitello-genesis and spawning. A semi-natural regime induced spawning in 129 days, an abbreviated decelerating regime induced spawning in 83 days, and an abbreviated accelerating/decelerating regime induced spawning in 117 days. Functional maturity of females was defined by analysis of vitellogenic stage frequency and oocyte diameter frequency. Ovarian biopsies were taken by catheter at 30-day intervals during experiment 1, and 60-day intervals during experiment 2. For a 90-day period in 1976, four females spawned 8.43 million eggs. For a 100-day period in 1977, eight females spawned 4.41 million eggs. Fertilization success was always greater than 99 percent and mean hatching success was 93.4 percent for all spawns.

Roberts, D.E., Jr., Morey, L.A., Henderson, G.E. and K.R. Halscott. 1978. The effects of delayed feeding, stocking density, and food density on survival, growth, and production of larval red drum (*Sciaenops ocellata*). **Proceedings World Mariculture Society,** 9:333-343.

The starvation time, optimal feeding time, stocking density, and food density were defined for larval red drum, *Sciaenops ocellata*, in 20- and 40-1 tanks. When larvae were not offered food by the fourth day after eye pigmentation (fifth day from hatching), they experienced 100 percent mortality. The optimal initial feeding time for best survival (14 percent) was the second day after eye pigmentation. Growth and survival were affected by initial embryo stocking density and food density maintained in the tank. Stocking densities of 2, 10, and 20 embryos/1 and food densities of 1, 5 and 10 rotifers/ml were tested in 40-1 tanks. Survival and total biomass were higher when embryos were stocked at 10 embryos/1 and significantly higher when fed at 5 rotifers/ml. Growth was best when embryos were stocked at 2 embryos/l.

TOXICOLOGY

Cardeilhac, P. T., Simpson, C. F., White, F. H., Thompson, N. P. and W.E. Carr. 1982. Evidence of metal poisoning in acute deaths of large red drum (*Sciaenops ocellata*). **Bull. Environm. Contam. Toxicol.** 27:639-644.

Approximately 100 large mature red drum (*Sciaenops ocellata*, in a size range of 7-18 kg) were found dead in the Indian River and Mosquito Lagoon on the northeast coast of Florida near Titusville between 14 June and 7 July 1980. Observers reported some fish were seen coming to the surface and showing incoordinated movements just before death. There was complete loss of posture (abdomen up) and only twitching and opercular movemets present. The purpose of this report is to present evidence for metal poisoning as the cause of this mass kill.

Rabalais, Steven C., Arnold, C. R. and Nancy S. Wohlschlag. 1981. The effects of Ixtoc 1 oil on the eggs and larvae of red drum (*Sciaenops ocellata*). Symposium on the Environmental Effects of the Mexican Oil Spill, Texas Academy of Science, 7 March 1980, Corpus Christi Tx. **The Texas Journal of Science,** Vol. XXXIII, No. 1.

The toxicity of oil from the Ixtoc 1 well in the Gulf of Campeche was determined using eggs and larvae of red drum (*Sciaenops ocellata*). High mortality resulted when larvae were placed in mixtures of Ixtoc 1 oil and water. When eggs were placed in Ixtoc 1 contaminated water from the Port Aransas jetties, over half of the hatched larvae were deformed (skeletal anomalies).

Appendix B
Summary Listing of State and Federal Sources of Information and Assistance

William R. Younger
Texas Marine Advisory Service
County Courthouse, Room 326
Bay City, Texas 77414

State Agency	Service
Alabama	
Auburn University Alabama Cooperative Extension Fisheries Specialist Auburn, Alabama 36849 (205) 826-4786	Technical information and assistance on the economics, production, management, processing and marketing of aquaculturally produced fish.
Auburn University Alabama Sea Grant Extension 3940 Government Buildi ng Mobile, Alabama 36609 (205) 661-5004	Technical information and assistance on the economics, production, management, processing and marketing of aquaculturally produced marine fish.
Auburn University Department of Fisheries and Allied Aquaculture Auburn, Alabama 36849 (205) 826-4786	Technical information on marine fisheries resources.
Alabama Department of Conservation and Natural Resources Game and Fish Division Fisheries Section 64 North Union Street Montgomery, Alabama 36104 (205) 832-6307	Technical information on marine fisheries resources.
Alabama Department of Conservation and Natural Resources Claude Peteet Mariculture Center P.O. Drawer 458 Gulf Shores, Alabama 36542 (205) 968-7575	Technical information on the culture of marine fish and shellfish
Florida	
University of Florida Center for Fisheries and Aquaculture School of Forest Resources and Conservation 7922 N.W. 71st Street Gainesville, Florida 32k606 Attn: Aquaculture Specialist (904) 376-0732	Technical information and assistance on the economics, production, management, processing and marketing of culture finfish and shellfish.

University of Florida Marine Advisory Program 118 Newins-Ziegler Hall Gainesville, Florida 32611 (904) 392-1837	Technical information and assistance on the economics, production, management, processing and marketing of cultured finfish and shellfish.
University of Florida Center for Aquatic Weed School of Forest Resources 118 Newins-Ziegler Hall Gainsville, Florida 32611 (904) 392-3453	Technical information and assistance on aquatic weed control in culture ponds.
Florida Department of Natural Resources Marine Research Institute 100 Eighth Ave. South St. Petersburg, Florida 33701-5095 (813) 896-8626	Technical information on marine fish resources and their culture.
Florida Game & Freshwater Fish Commission Commercial Fisheries Section 620 S. Meridian St. Talahassee, Florida 32301 (904) 488-4066	Technical information on freshwaater fish resources and their culture.
University of Miami Division of Biology and Living Resources 4600 Rickenbacker Causeway Miami, Florida 33149 (305) 361-1236	
U.S. Fish and Wildlife Service National Fisheries Research Laboratory 7920 N.W. 71st St. Gainesville, Florida 32606 (904) 378-8181	Technical information on marine and freshwater fish resources.
U.S.D.A. Soil Conservation Service P.O. Box 1208 Gainesville, Florida 32601 (904) 377-8732	Technical information and assistance on water and soil management, including: pond design and layout; elevation and drainage surveys; soil analysis; water control recommendations.
National Marine Fisheries Service Southeast Fisheries Center 75 Virginia Beach Drive Miami, Florida 33149	Technical information on marine fish and shellfish resources and their culture.
Florida Atlantic University Department of Biological Sciences Boca Raton, Florida 33432 (305) 361-5761	Technical information on marine fish resources.
Florida State University Department of Oceanography Tallahassee, Florida 32306 (813) 893-9130	Technical information on marine fish resources.
University of South Florida Department of Marine Science St. Petersburg, Florida 33706 (813) 893-9130	Technical information on marine fish resources.

Georgia

The University of Georgia College of Agriculture Cooperative Extension Service Athens, Georgia 30602 (404) 542-3446	Technical information and assistance on the economics, production, management, processing and marketing of culture fish.
The University of Georgia Marine Extension Service Sea Grant Program Athens, Georgia 30602 (404) 542-7671	Technical information and assistance on economics, production, management, processing and marketing of cultured marine finfish and shellfish.
Georgia Department of Natural Resources Game and Fish Division Fisheries Management Section 270 Washington St., S.W. Atlanta, Georgia 30334 (404) 656-3545	Technical information on marine fish resources.
Georgia Department of Industry and Trade 2300 Peachtree St., N.W. Atlanta, Georgia 32615 (404) 656-3545	Information and assistance on marketing, finance and business development.
U.S.D.A. Soil Conservation Service Federal Office Building Athens, Georgia 30602 (404) 546-2114	Technical information and assistance on water and soil management, including: pond design and layout; elevation and drainage surveys; soil analysis; water control recommendations.

Louisiana

Louisiana Department of Agriculture Agri-Business Loan Department P.O. Box 44184, Capital Station Baton Rouge, Louisiana 70804 (504) 291-3902	Financial assistance.
Louisiana State University Cooperative Extension Service Aquaculture Specialist Knapp Hall Baton Rouge, Louisiana 70803 (504) 388-4141	Technical information and assistance in the economics, production, management, processing and marketing of cultured fish.
Louisiana State University Sea Grant Legal Program 170 Law Center Baton Rouge, Louisiana 70803 (504) 388-5391	Legal information and assistance pertaining to coastal aquaculture operations.
U.S.D.A. Soil Conservation Service 3737 Government Street Alexandria, Louisiana 71301 (318) 473-7803	Technical information and assistance on water and soil including: management, elevation and drainage surveys; soil analysis; water control recommendations; pond design and layout.
U.S.D.A. Farmer's Home Administration 3727 Government Street Alexandria, Louisiana 71301 (318) 473-7920	Financial assistance.
Louisiana Crawfish Farmer's Association P.O. Box 91544 Lafayette, Louisiana 70509 (318) 235-7072	Information on aquaculture trade association organization and operation.

Louisiana Catfish Farmer's Association, Inc. Charles Watson, President P.O. Box 267 Wisner, Louisiana (318) 724-7475	Information on aquaculture trade association organization and operation.

Mississippi

Gulf Coast Research Laboratory Fisheries Research & Development Section East Beach Drive P.O. Box 7000 Ocean Springs, Mississippi 39564 (610) 872-4256	Technical information and assistance on the culture of marine finfish.
Mississippi State University Wildlife and Fisheries Department Drawer LW Mississippi State, Mississippi 39762 (601) 325-3133	Technical information and assistance on the production, management, economics, processing and marketing of cultured finfish.
Mississippi State University Mississippi Cooperative Extension Service Fisheries Specialist P.O. Box 5405 Mississippi State, Mississippi 39762 (601) 325-3174 or (601) 686-9311	Technical information and assistance on the production, management, economics, processing and marketing of cultured finfish.
Mississippi State University Sea Grant Advisory Service 4646 W. Beach Blvd. Suite 1-E Biloxi, Mississippi 39531 (601) 388-4602	Technical information and assistance on the production, management, economics, processing and marketing of cultured finfish and shellfish.
Mississippi Dept. of Wildlife Conservation Bureau of Marine Resources Science and Statistics Division P.O. Drawer 959 Long Beach, Mississippi 39560 (601) 864-4602	Technical information and assistance on the production, management, economics, processing and marketing of cultured finfish and shellfish.

North Carolina

North Carolina State University Cooperative Extension Service Fisheries Specialist Raleigh, North Carolina (919) 737-2741	Technical information and assistance on the production, management, economics, processing and marketing of cultured finfish and shellfish.
UNC Sea Grant Program Marine Advisory Services Box 8605 North Carolina State University Raleigh, North Carolina 27695-8605 (919)737-2454	Technical information and assistance on the production, management, economics, processing and marketing of cultured finfish and shellfish.
Haywood Technical College Fishery Training Specialist Freelander Drive Clyde, North Carolina 28721 (704) 627-2821	Technical information and training on fish aquaculture.

North Carolina Marine Fisheries Commission P.O. Box 769 Morehead City, North Carolina 28557 (919) 726-7021	Technical information on marine fisheries resources.

South Carolina

South Carolina Coastal Council Summerall Center 19 Hagood St., Suite 802 Charleston, South Carolina 29403 (803) 792-5808	Counseling on the need for state and federal permits involving alteration of coastal lands or waters.
Sea Grant Marine Extension Program Aquaculture Specialist P.O. Drawer 1100 Georgetown, South Carolina 29440 (803) 546-4481	Technical information on assistance on shellfish, freshwater prawn, marine shrimp and crawfish culture.
S.C. Wildlife and Marine Resources Department Division of Marine Resources P.O. Box 12559 Charleston, South Carolina 29412 (803) 795-6350	Technical information on marine finfish culture.

Texas

Texas Marine Advisory Service Sea Grant College Program Texas A&M University College Station, Texas 77843-4115 (409) 845-7524	Technical information and assistance on the production, management, economics, processing and marketing of cultured finfish and shellfish.
Texas Agricultural Extension Service Texas A&M Research & Extension Center Route 2, Box 589 Corpus Christi, Texas 78410 Attn: Extension Mariculture Specialist (512) 265-9203	Technical information and assistance on the production, management, economics, processing and marketing of cultured finfish and shellfish.
Texas Agricultural Extension Service Dept. of Wildlife and Fisheries Science 102 Nagle Hall Collge Station, Texas 77843 Attn: Extension Fisheries Specialist (512) 845-7471	Technical information on hatchery operation and pond growout of marine finfish.
Texas Parks and Wildlife Department Coastal Fisheries Division 4200 Smith School Road Austin , Texas 78744 (512) 389-4800 or (Tx only) 1-800-792-1112	
The University of Texas at Austin Marine Science Institute P.O. Box 1267 Port Aransas, Texas 78373 (512) 749-6795	Technical information on the spawning and growout of marine finfishes.
Lamar University John Gray Institute 855 Florida Ave. Beaumont, Texas 77705 (409) 839-2229	Technical information on the economics of farming red drum in Texas.

Texas Dept. of Economic Development Small Business Revitalization Program P.O. Box 12718 Austin, Texas 77801 (512) 463-7476	Information and assistance with business plan development and the location of financial resources.
Texas Department of Agriculture P.O. Box 12847 Capitol Station Austin, Texas 78711 (512) 463-7476	Investigation of chemical contamination in aquaculture ponds from suspected agriculture, domestic, or industrial sources.
Federal	
U.S.D.A. Soil Conservation Service (usually listed-local telephone directory)	Technical information and assistance on water and soil management including: elevation and drainage surveys; soil analysis; water control recommendations; pond design and layout.
National Marine Fisheries Service Red Drum Project 3209 Frederick St. Pascagoula, Mississippi 39567 (610) 762-4591	Technical information on marine fisheries resources and brood sourcing information.
U.S. Fish and Wildlife Service National Fisheries Center Route 3, Box 700 Kearneyville, West Virginia 25430 (304) 725-8461	Technical information and training on fish aquaculture.
National Program Leader for Aquaculture Extnesion Service & Coop. State Res. Serv. U.S. Department of Agriculture Room 3871, South Building Washington, D.C. 20250 (202) 447-6014	Promotion of the overall development of the U.S. aquaculture industry through the coordination of research efforts and the collection and dissemination of fish farming information.
U.S. Food and Drug Administration P.O. Box 158 Dauphin Island, AL 36528 (205) 861-2962	Technical information on chemicals and drugs approved for use on aquaculturally produced food fish.
U.S. Army Corps of Engineers Office of the Chief of Engineers Pulaski Building 20 Massachusetts Ave., N.W. Washington, D.C. 20314 (202) 272-0001	Assistance on the permit requirements for construction and/or dredge-and-fill in or near navigable waters.

APPENDIX C

Procedures, Reagents and Standards Used for Seawater Chemistry Analyses of Nitrate, Nitrite, Ammonium-Nitrogen and Phosphate-Phosphorus*

Nitrate (NO_3-N)

Nitrate, the most completely oxidized state of nitrogen in water, is determined by the Brucine method (Jenkins and Medsker, 1974). It involves the reaction of NO_3-N with brucine sulphate in strong (13N) sulfuric acid medium. Nitrite interference is eliminated by the addition of sulphanilic acid. Salinity interference is eliminated by increasing the salinity of all samples and standards to a level above which the effects of salinity are stable. Uneven heating, or, different temperature of samples produce erratic results. Therefore, all samples (and standards) must be treated identically.

Reagents

NaCl solution — saturated solution.

Sulfuric acid — add 5 parts (v.) H_2SO_4 to 1.25 parts (v.) distilled H_2O. Cool before using.

Brucine reagent — dissolve 1 g brucine sulphate and 0.1 g of sultphanilic acid in 70 ml of hot distilled H_2O. Add 3 ml concentrated HCl and make up to 100 ml with de-ionized water.

Procedures

1. Place 10 ml of sample in a test tube.
2. Place test tubes in a rack submerged in icewater, the depth of which should be sufficient to cover the portion of the test tubes containing the samples and reagents.
3. Add 2 ml of NaCl reagent to each sample and mix by swirling.
4. Carefully pour 10 ml H_2SO_4 reagent into each sample. Incline each sample test tube for this addition.
5. Add 0.5 ml Brucine reagent to each sample.
6. Mix each sample by pouring back and forth one time in a mixing tube and replace in ice water.
7. Remove tubes from icewater and place in a boiling-water bath for 20 minutes. (A yellow color proportional to the concentration of NO_3-N in the sample develops during this time).
8. Stabilize color density by cooling samples in the icewater bath to approximately room temperature.
9. Determine sample optical density at 410 mu in a spectrophotometer, or in a filter type electrophotometer using the latter's 425 (purple) filter.
10. Compare sample optical density to standard optical density to calculate concentration.

Nitrite (NO_2-N)

The procedure used is that of Shinn (1941) as applied to seawater by Bendschneider and Robinson (1952). The procedure is based on the reaction of nitrite in saltwater with sulphanilamide in an acid solution. The resulting diazo-compound reacts with N-(1-napthyl)-ethylenediamine to form a highly colored (red) azo dye.

Reagents

Sulfanilimide solution — dissolve 5 g of sulfanilimide in a mixture of 50 ml of concentrated HCL acid and about 300 ml of distilled water.

N-(1-naphtyl)-ethylenediamine dihydrochloride solution — dissolve 0.5 g of the dihydrochloride in 500 ml of distilled water. Store in a brown bottle.

Procedures

1. Place 25 ml of sample in a 50 ml flask.
2. Add 0.5 ml of sulfanilimide reagent and mix.
3. After at least 2 min add 0.5 ml of ethylenediamine reagent and mix.
4. After no less than 10 min and no more than 120 min measure the optical density of the samples in a spectrophotometer at a wavelength of 543 mu, or in a filter-type electrophotometer equipped with a green 525 mu-filter.
5. Compare sample optical density of standard calibration curve to convert to concentration units.

From Fontaine, C.T., et al.* **The husbandry of hatchling and yearling Kemp's ridley sea turtles (*Lepidochelys kempi*). NOAA Technical Memorandum NMFS-SEFC-158, 111:34, 10 tables, 22 figures, 2 appendices.

Ammonium-Nitrogen (NH_4-N)

We used the phenol-hyprochlorite method of Solorzano (1969) as described by Strickland and Parsons (1972). The procedure involves treating samples in an alkaline citrate medium with sodium hypochlorite and phenol in the presence of sodium nitroferricyanide (nitroprusside) that acts as a catalyzer. The intensity of the blue indophenol that develops is an indication of the concentration of NH_4-N in the sample.

Reagents

Phenol solution — dissolve 20 g of crystalline reagent grade phenol in 200 ml of 95 percent v/v ethyl alcohol.

Sodium nitroferricyanide solution — dissolve 1.0 g of sodium ferricyanite in 200 ml of de-ionized water. Store in an amber bottle.

Alkaline reagent — dissolve 100 g of sodium citrate and 5 g of sodium hydroxide (analytical grade) in 500 ml of de-ionized water.

Sodium hypochlorite reagent — use Clorox.

Oxidizing solution — mix 100 ml of alkaline reagent and 25 ml of clorox. Keep this solution stoppered while not in use. Prepare fresh every day.

Procedures

1. Add 25 ml of sample to a 50 ml Erlenmyer flask.
2. Add 1 ml of phenol reagent to each sample and mix.
3. Add 1 ml of sodium nitroferricyanide reagent to each sample and mix.
4. Add 2.5 ml of oxidizing reagent to each sample and mix.
5. Allow flasks to stand at room temperature for at least 1 hour (color produced is stable for 24 hours).
6. Determine the optical density of the blue color in a spectrophotometer at 640 mu, or in an electrophotometer with a red filter.
7. Compare sample optical density to standard calibration curve to convert to concentration units.

Phosphate-Phosphorous (PO_4-P)

The procedure used to determine O_4-P in seawater is that of Murphy and Riley (1962). It is also included in Strickland and Parsons (1962). The procedure is based on the reaction of phosphate in seawater with a composite reagent containing molybdic acid, ascorbic acid, and trivalent antimony.

Reagents

Ammonium molybdate solution — dissolve 15 g of reagent grade ammonium molybdate [$(NH_4)_6MO_7O_{24}.4H_2O$] in 500 ml of deionized water. Store in plastic bottle out of direct sunlight.

Sulfuric acid solution — add 150 ml concentrated reagent grade sulfuric acid to 900 ml of de-ionized water. Allow the solution to cool and store in a glass bottle.

Ascorbic acid solution — dissolve 27 g of reagent grade ascorbic acid in 500o ml of de-ionized water. Store (frozen solid) in a plastic bottle in a freezer. Thaw for use and refreeze at once.

Potassium antimonyl-tartrate solution — dissolve 0.34 g of potassium antimonyl-tartrate (tartar emetic) in 250 ml of de-ionized water. Store in a glass, or plastic bottle.

Mixed Reagent

Mix together 100 ml of the molbydate solution, 250 ml of the sulfuric acid solution, 100 ml of the ascorbic acid solution and 50 ml of the potassium antimonyl-tartrate solution. Prepare this reagent for use daily and discard any excess. Do not store longer than 6 hours.

Procedures

1. Add 25 ml of sample to a 50 ml Erlenmyer flask.
2. Add 2.5 ml ofthe mixed reagent.
3. Mix by swirling the flask.
4. After at least 5 minutes measure the optical density of the blue color in a suitable photometer, or at 750 mu in a filter-type electrophotometer.
5. Compare sample optical density to standard calibration curve to convert to concentration units.

Laboratory Water for Analyses

The de-ionized water used in our laboratory is prepared by passing tap water, that varies from 200 to 800 ppm total solids, through a mixed bed deionizer (which is supplied, and replaced as needed, by a private contractor). Our distilled water is prepared by distilling the de-ionized water with an all Pyrex still. Comparing blank values showed that distillation of the de-ionized water did not improve its quality as far as our chemical analyses were concerned. Therefore, whenever distilled water is suggested in the analytical procedures, de-ionized water may be used provided it is produced from a mixed bed de-ionizer of the type that removes virtually all of the positive and negative electrolytes.

Preparation of Standards

Reagents used for preparing calibration standards must be readily soluble, reagent-grade chemicals, dried before use. To minimize contamination, a bottle of each is set aside for standardization purposes only. The following chemicals are used most frequently for calibration standardization at this laboratory:

1. Sodium nitrate ($NaNO_3$) for NO_3-N
2. Sodium nitrite ($NaNO_2$) for NO_2-N
3. Potassium acid phosphate (KH_2PO_4) for PO_4-P
4. Ammonium chloride (NH_4Cl) for NH_4-N

Procedures

1. Weigh a small amount of the desired chemical to the nearest mg. For convenience, the weight should be in the range of 0.1 g to 0.5 g.
2. Dissolve the weighed chemical into 1000x ml with de-ionized water, where x is numerically equal to the gram weight of the chemical. For example, if 0.235 g is the weight, it should be dissolved in 235 ml of water. When the chemical is completely dissolved and the solution thoroughly mixed (solution A), 1 ml will contain 1 mg of the chemical. Solution A is used to prepare the working standard from which calibration standards are prepared. Assume a series of NO2-N standards having a final volume of 25 ml are desired that will increase in increments of 1 u at NO_2-N/l; i. e., 1 ml standard + 24 ml H_2O = 1 ug at N/l; 2 ml standard + 23 ml H_2O = 2 ug at N/l, etc. If solution A were used undiluted, 1 ml diluted to 25 ml would contain 580 ug at NO_2-N/l. the working standard solution, therefore, is prepared by diluting 1 ml of solution A to 580 ml.

References

Bendschneider, K. and P. J. Robinson. 1952. A new spectrophotometic method for the determinations of nitrite in seawater. J. **Mar. Res.** 11:87-96.

Fontaine, C. T., K. T. Marvin, T. D. Williams, W. J. Browning, R. M. Harris, K. L. W. Indelicato, G. A. Shattuck and R. A. Sadler. 1985. **The husbandry of hatchling and yearling Kemp's ridley sea turtles (*Lepidochelys kempi*).** NOAA Technical Memorandum NMFS-SEFC-158, 111 + 34 p., 10 Tables, 22 Figures, and 2 Appendices.

Jenkins, D. and L. Medsher. 1974. Brucine method for determination of nitrate in ocean, estuarine, and freshwater. Anal. **Chem.**, Vol. 36, No. 3 pp. 610-612).

Murphy, S. and J. L. Riely. 1962. A modified single solution method for the determination of phosphate in natural waters. **Anal. Chem. Acta** 27-31-36.

Shinn, M. B. 1941. A coloriometric method for the determination of nitrite. **Ind. Eng. Chem.** [Anal. Ed.] 13(33).

Solorzano, L. 1969. Determination of ammonia in natural waters by the phenolhypochlorite method. Limnol. and Oceanogr. 14:799.

Strickland, J. D. H. and T. R. Parsons. 1972. **A practical handbook of seawater analyses.** Fish. Res. Bd. of Canada, Catalogue No. FS 94- 167.

APPENDIX D

Directions for Repairing with Fiberglass

Brian L. Brawner
Ewald Enterprises, Inc.
P. O. Box 519
Karnes City, Texas 78118

Equipment and Supply Needs

Small Repair Kit	Large Repair Kit
1 qt. resin	1 gal. resin
1 pt. acetone	1 qt. acetone
1/2 oz. catalyst	1 oz. catalyst
1 roll 4-inch mat	1 roll 50-inch mat
1 sheet sandpaper	1 sheet sandpaper

Mixing Catalyst and Resin or Putty:

Regardless of whether resin or putty is being mixed, the procedure is the same: combine 1 cup resin and 6 to 8 drops of catalyst, mix well. It is best to mix only a small amount at a time. Use old clean containers or paper cups that can be thrown away. Use old clean paint brushes or cheap paint brushes that can be thrown away. A flat stick works very well to apply putty.

Read all warning labels before using catalyst, resin or acetone.

Do not use near open flame.

Keep out of reach of children.

Repair on Large Areas

Fiberglass

Sand surface to be repaired, clean with acetone, paint resin onto surface to be repaired, cover with fiberglass material, paint with resin until all cloth is wet. Smooth and work out all bubbles. Let dry. For a smooth surface, sand. To achieve a glass-like finish apply get coat. Most colors are available.

Cement

Area must be dry. Scrub with steel brush to completely clean area to be fiberglassed. Wipe with acetone. Paint area with resin. Cover with fiberglass material. Paint with resin until all material is wet. Smooth material and work out all air bubbles. Let dry.

Applying Putty to A Small Area

Sand area to be repaired, clean with acetone, cover area with putty. Let dry.

Metal

Remove all paint and rust, etc. clean with muriatic acid. Wash with vinegar and water. (Metals that are free to paint, rust, etc. that cannot be cleaned with acid should be cleaned with acetone). Paint with resin until all material is wet. Work out all air bubbles. Let dry.

Replacing Old Fittings and Inserting New Ones

Remove old fittings. Sand surface around fittings until smooth. Insert new fittings. Work putty around new fitting. Smooth the surface with flat stick. Let dry

Wood

Area must be free of paint, clean and dry. Paint surface with resin. Cover with fiberglass material. Paint resin until all material is wet. Work out all air bubbles. Let dry

Assembling Sectional Fiberglass Tanks

1. Unroll the round floor panel on a flat, level surface.
2. Bolt side panels together and position on the floor panel so they form a circle with the required diameter in all directions.
3. Drill holes in the floor panel and bolt the side panels in place, making sure all bolts are tight.
4. Sand all surfaces at the seams where there is no gel coat and clean with acetone.
5. Mix Putty. A 1 percent mixture of catalyst should be added to putty and mixed well. Working time is about 20 minutes depending on temperature and humidity. Use clean containers or paper cups that can be thrown away. A flat stick works very well to apply putty. Place putty in all the side panel seams and floor seams.
6. Mix Resin. A 1 percent mixture of catalyst should be added to the resin and mixed well. It is best to prepare only a small amount of resin at a time. Paint the area to be fiberglassed with resin. Place a layer of fiberglass mat on a piece of cardboard and wet out with resin using a paint brush. Place

another layer of mat on top of the first layer and wet out. Remove these two layers from cardboard and place over the center of seam to be fiberglassed. Remove all air pockets and bubbles with a roller. Repeat this procedure with two more layers of mat. Each seam requires a total of four (4) layers of mat. Let dry.

7. For a smooth surface, sand to a glass-like finish and apply resin only. A 1 percent mixture of catalyst is needed in resin.
8. Leave bolts in the seams for additional strength.
9. Ten-foot and 12-foot diameter tanks require a one-piece bottom. A 20-foot diameter tank requires a two-piece bottom that must be glassed together by customer.