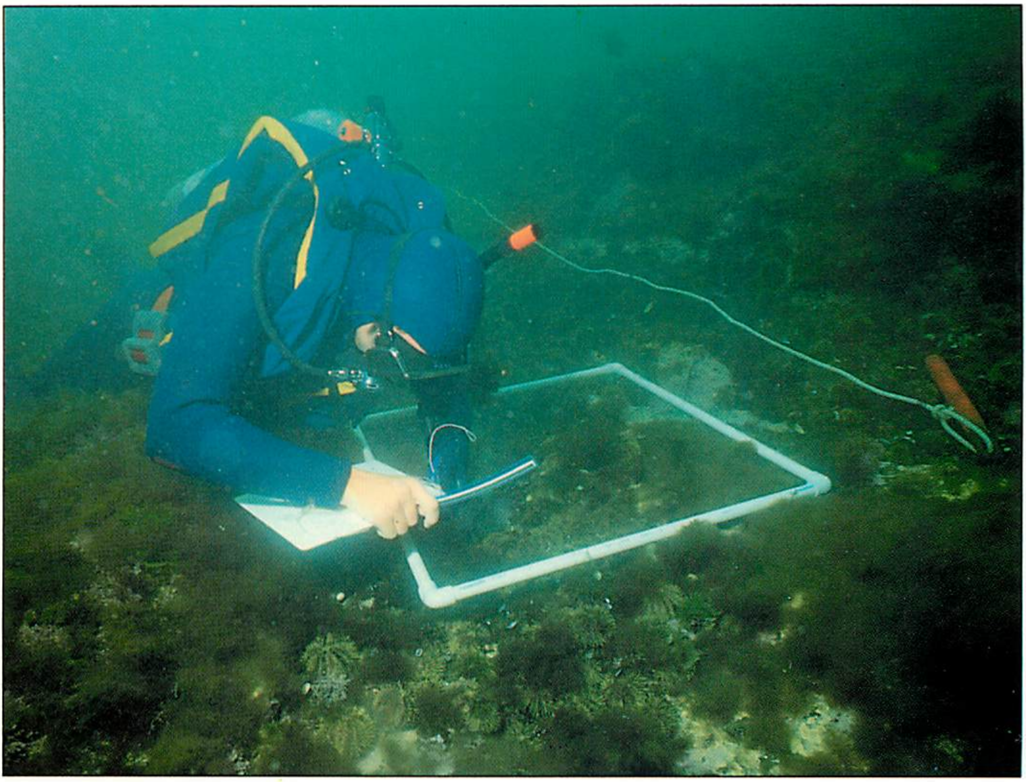
THE UNDERWATER CATALOG

A Guide to Methods in Underwater Research



James Coyer and Jon Witman

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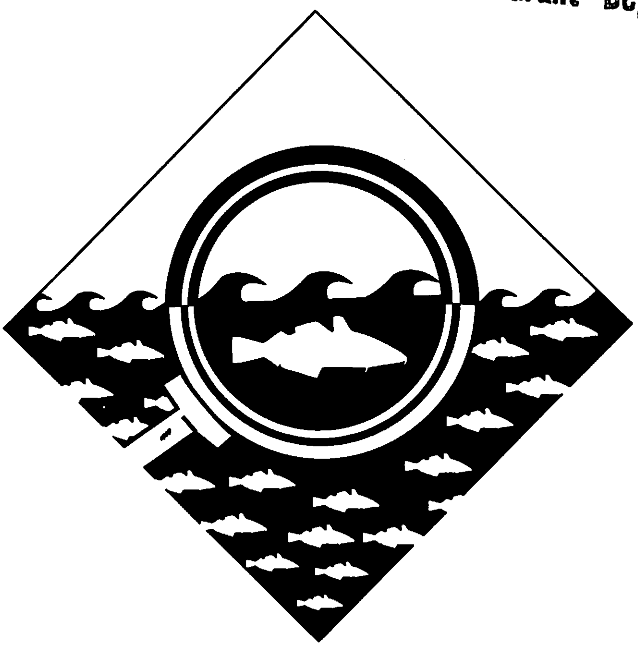

James Coyer and Jon Witman

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Cover: Student from Underwater Research course
at Shoals Marine Laboratory surveying urchin
populations with a 0.25 m² quadrat along a transect line.
Photo by J. Witman

printed on recycled paper

Foreword

The need for an "Underwater Catalog" originated during our Underwater Research course at the Shoals Marine Laboratory when we realized that our lectures on techniques occured at the expense of valuable field time. As we began to think about the various methods we had learned over the years, we realized that most were obtained by word-of-mouth through the "grapevine" linking marine labs throughout North America. Consequently, one of our objectives in compiling the Catalog is to preserve and distribute methodology in a written format (albeit informal), so as to accelerate the process of implementation and modification. Ultimately, of course, we hope that the Catalog will reduce the amount of time devoted to methodology and increase the amount of time underwater for hypothesis testing.

We are indebted to the contributors listed in Appendix III, many of whom provided long and detailed descriptions which easily could have been included as separate Appendices. In most cases, the names of contributors appear in parentheses next to the method. Although the contributors may not be the original developers of a technique, they have used the technique regularly; interested readers are encouraged to contact them directly for more information. Products listed in the catalog are for reference only and should not be regarded as endorsements. In addition to the numerous contributors, we are especially indebted to J.B. Heiser and Christine Bogdanowicz of the Shoals Marine Lab and also to Gary Davis and Don Harper of AAUS for their support and encouragement.

Obviously, the Catalog can never be complete and always will require revision. Toward this end we depend on you. As you experience success or failure with various methods described herein or developed by you, please send descriptions to us so that we may make appropriate revisions.

The ways and means employed by underwater researchers are frequently ingenious. We look forward to future innovations.

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June 1990

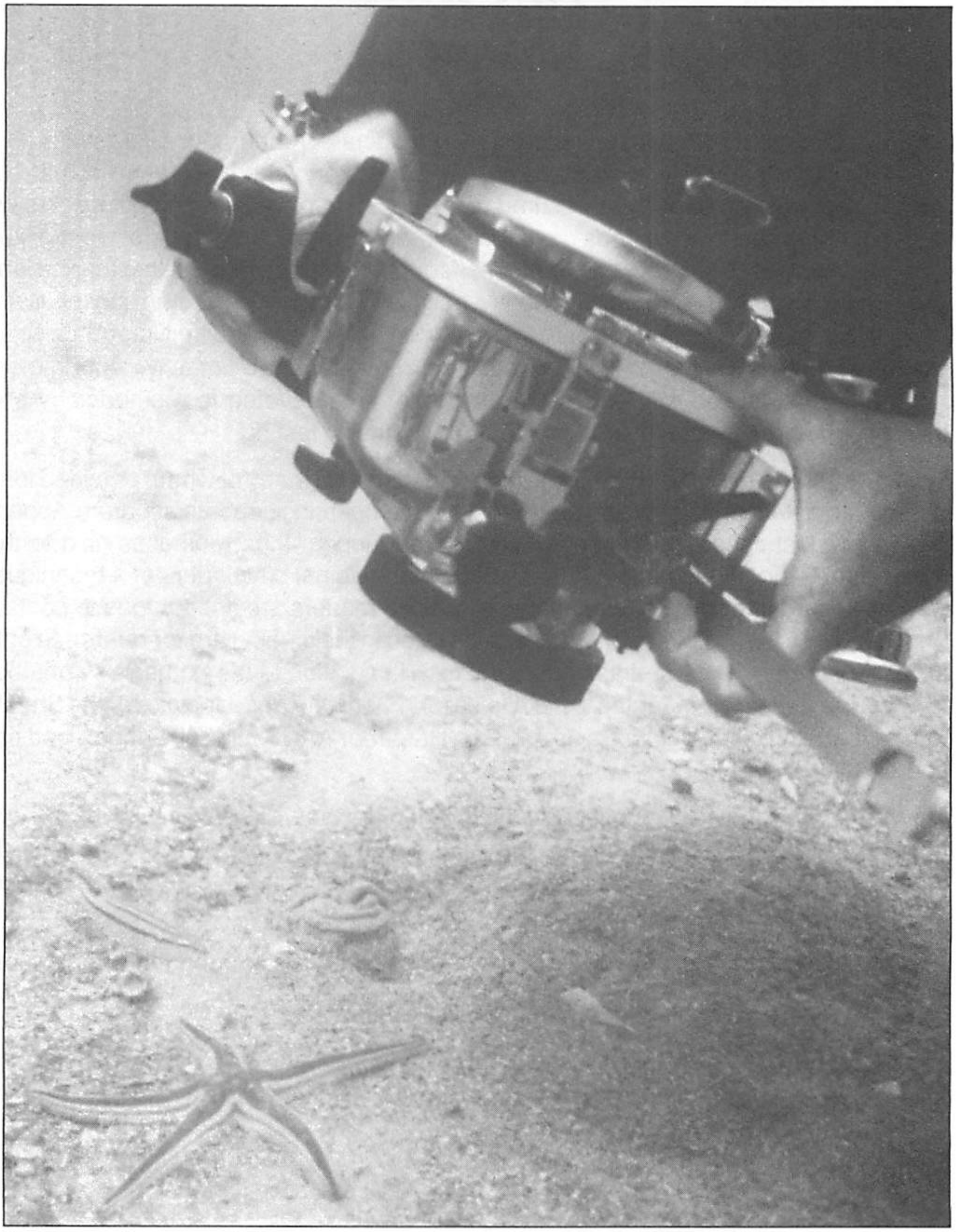

Photo by Ron Harelstad

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Underwater Record Keeping

Written

Plastic Slates

- High-impact styrene (variable thickness)
- Write directly on plastic with pencil, erase with cleanser
- Can glue plastic ruler to edge of slate for measuring
- Disadvantage: data must be transcribed after each dive

Plastic Sheets/Underwater Paper

- Advantage: can be removed from frames and filed as primary data; no transcription needed
- Frames require a PVC backboard on which paper is held with rubber bands or clamped with a PVC frame, using brass hex-head bolts and wing nuts (can use two frames and have paper on both sides of backboard); fancy frames also have thermometers, depth gauges, collecting vials, and snap-clips
- Disadvantage: sheets often fall out in heavy surge
- Types of paper/plastic sheets

  A. Nalgene Polypaper (coated): Plain
  - Available in 8.5 x 11" single sheets
  - Photocopying onto Polypaper may destroy copier, follow directions for paper or have info printed using standard printing methods
  - Order through authorized Nalgene dealer (i.e., Fisher Scientific, VWR, American Scientific Products, Sargent-Welch); no direct orders to Nalgene
  - Field notebooks made with Polypaper are available in a variety of sizes

  B. Plastic sheets: printed grid
  - Available in 8.5 x 11" sheets from Dynalab Corp., Rochester Scientific, Box 112, Rochester, NY 14692-0112 (800) 828-6595
  - Cat. #6325; $17/100, discounts on larger orders

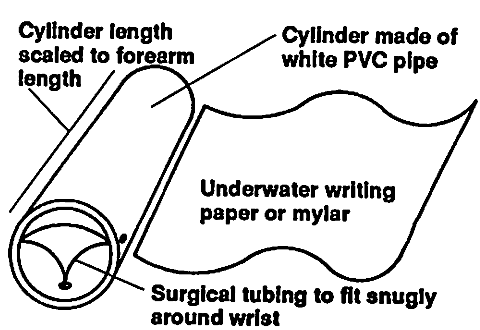
  C. Mylar (Teledar) Drafting Film (7 mil single matte)
  - Transparency allows it to be clamped over a prepared data form or template; when completed, sheets are easily removed and replaced
  - Can be erased with detergent
  - Check with drafting supply stores for local availability

  D. Kimdura (plastic sheets) (Carroll)
  - Zellerbach Paper Co., (213) 268-0200
  - Use 68# (medium weight) or 250# (heavy weight); the latter is a mill item sold in 3'x 5' sheets (cut to size)

  E. Acetate sheets for overhead projectors: must use grease pencils

Arm Slates
(frees hands when not writing)

- Steneck uses PVC cylinder scaled to forearm; an inner "cuff" of surgical



tubing fits snugly around wrist, Mylar wrapped around PVC

- See also: Foster, M.S. 1976. A mini-slate for recording information underwater. Underwater Naturalist 9:14-145. (slates with Mylar, attached to arm)

Pencils

- Pencils with replaceable tips will insure sharp points (available in "budget" stores)
  - **A.** Bensia Rocket Pencil
  - **B.** Moon Pencil (Readi-Pencil); Product #98196, Mead Corp; Dayton, OH 45463
- Mechanical pencils with hard 3H leads
- Attach pencils to slates
  - **A.** Surgical or tygon tubing lanyards; if use tygon tubing (clear), can snugly insert trimmed plastic ruler for easy availability
  - **B.** Velcro; coat both surfaces with contact cement, let dry, coat again until tacky, then join

Recording Data at Night

- Use minimal light so as to maximize night vision and minimize effect on behavior of organisms
- Suggestions:
  - **A.** Use red lenses on dive light
  - **B.** Use chemical light (i.e. Cyalume)
    - Attach to slate and use tape to shade and direct a soft glow over slate
    - Can use chemical lights over 4-5 days, if they are frozen between dives

Tape Recorders

(Duggins, Heine, Cowen)

- Full face mask essential for clear speech
- Microphone (not voice-activated) can be wired to a cassette recorder in an Ikelite housing or to the surface via an umbilical; either method provides excellent resolution
- Manual on/off switch greatly reduces time required to transcribe data after a dive (as only data need be recorded)
- Construct system from components and underwater housings or purchase complete unit (i.e., Sound Wave Systems, 3001 Red Hill, Bldg. 1, Suite 102, Costa Mesa, CA 92626)

Underwater Epoxy

General Comments

- All epoxies need a clean surface for application and maximal retention; use putty knives and stainless "pot scrubbers" or hammer and chisel
- All epoxies stick best when first mixed, and harden best when mixed thoroughly
- All epoxies listed below are designed to cure on a wet or submerged surface; curing is temperature dependent: high temperatures will speed, lower will retard
- It is difficult to use uw epoxies for securing objects to the bottom when the epoxy must bear a load while it is curing

Sea-Goin' Poxy Putty

- Permalite Plastics, 1537 Monrovia Ave., Newport Beach, CA 92663; available in many marine supply outlets
- Advantages
  - A. Dries white, easier to see
  - B. Sticks to most surfaces (including fingers-use rubber/disposable gloves)
- Disadvantages
  - A. Somewhat harder to handle
  - B. Tends to crumble after 1-2 years
  - C. Dries less hard than Koppers, will not set-up at temperatures below freezing
- Good for marking (i.e. corners of quadrats) but does not bear a load (i.e., don't use for inserting hardware)

Koppers

(Z-Spar) A-788 Splash Zone Compound

- Koppers Co., Inc., Box 911041, Commerce, CA 90011; in L.A. contact Llewellyn Supply, 507 Figueroa, Wilmington, CA (213) 834-2508 (wholesale supplier); in Boston contact Harlow-Imrie Co., 49 Needham St., Newton Highlands, MA (617) 527-5165
- Widely used in underwater research
- Advantages
  - A. Easy to mix and work; suggest mixing with wet rubber/disposable gloves (epoxy is toxic and will stick to dry surfaces; don't mix in plastic bags as epoxy sticks to sides and is wasted)
  - B. Transport to underwater site in plastic zip-lock bags within minutes after mixing topside
  - C. Dries hard and strong, long lasting, resilient
  - D. Mixed life of 1-1.5 hours; in warm temperate conditions, will cure in 6-8 hrs, 70% firm-up within 2 hrs
  - E. Can be used and will set-up at temperatures below freezing
- Disadvantages: dries green, harder to see and relocate (although this could be an advantage in that it also will be harder for the "curious public" to find)

Petit Underwater Patching Compound

("Polypoxy")

- Borough of Rockaway, NJ 07866 or San Leandro, CA 94577
- Useable up to 1.5 hrs after mixing
- Good for marking (i.e., corners of quadrats) but does not bear a load

Helor Hi-Water Concrete Adhesive

(Engle)

- Epoxy Coatings Co., P.O. Box 1035, Union City, CA 94587 (415) 471-7800
- Used extensively in the intertidal (as it oozes into crevices easily); not as easy to use subtidally
- Sets in about 1 hour

Underwater Cement
(an inexpensive alternative to drilling)

General Comments

- Underwater cement often used to patch swimming pools; check with pool supply stores for availability
- Mix topside and transport to dive site in a plastic bag sealed with a rubber band, puncture bag and squeeze out cement
- Place into cracks or crevices (cement doesn't last as long when unsupported as a blob on an open surface)
- Insert gear-securing hardware (i.e., eyebolts, threaded stakes)
- Sets in about 30 min.

Speed-crete

- Tamms Industries Co., 1222 Ardmore Ave., Itasca, IL 60143

All-Crete

(Chess)

- Concrete Products Inc., 1220 Fortna Ave., Woodland, CA 95695

Underwater Drilling

By Hand

- Use hand-held star drills and heavy maul; labor intensive

Pneumatic

(Impact hammer, drill, or hammer drill)

- Pneumatic tools can generate considerable auditory discomfort underwater; use ear protection unless protected by thick wetsuit
- All pneumatic tools require downward force, so diver or drill must be heavily weighted
- Clouds of bubbles will be released which may obscure visibility and attract fishes, sound also may attract sharks
- Use masonry bits for drilling; also can use star drills welded to ends of impact hammer tools
- Types of drills (any pneumatic-type should work):

  A. Chicago Pneumatic Handrills (CP-9RR Model "B-O") (Duggins, Harrold): Chicago Pneumatics, Tool Division, Utica, NY 13501

  - Drill can be run off a standard first stage regulator fitted to scuba tanks (underwater) or supplied with air from surface; air consumption is high, so a hose diameter greater than 3/8" is required
  - Duggins can drill 8 holes, 3/8" dia, 5-6" deep, in the hardest rock from one 80 ft^3 scuba tank, and many 5/16" dia, 2-3" holes from one scuba tank; for large jobs he uses a surface-stationed 300 ft^3 tank and a 75-100 ft hose
  - Harrold can drill a 7/8" hole 15 cm deep into granite in 1 min 13 sec at 42 ft

  B. Dayton 3/8" Air Drill

  - Used to drill holes, also used air hammer to clear rocky substrata
  - Utilizes approximately 90 psi working pressure and requires approximately 4 ft^3/min at surface

  C. Sears Craftsman Air Hammer (Witman)

  - Replace chipping tool with drill chuck
  - Good for 1/4" holes

- Corrosion Prevention

  A. Flush all drills with WD-40 after the dive

  B. Store in diesel oil

Attaching Gear to Hard Substrata

Cement/Epoxy

- Use underwater cement or Koppers epoxy with inserted hardware (for hardware, check construction supply outfits or write: Northern Contractors and Industrial Supply, Box 500, Fairhaven Station, New Haven, CT 06513)

Drilling Holes

(See "Underwater Drilling", pg. 11)

- Small items or large items in areas of low current and surge (Duggins)
  - **A.** Drill holes 2-3" deep
  - **B.** Fill with Koppers epoxy and insert appropriate lengths of threaded, stainless steel rod
- Large items or areas of high current and surge (Duggins)
  - **A.** Use 5" x 3/8" "Red Head" stainless steel anchor wedges (bolts)
  - **B.** Requires hard rock for wedges to set mechanically, but once set, are very strong
  - **C.** "Red Head" wedge anchors are common in the construction industry and easy to find
- High surge areas (Carroll)
  - **A.** Place small blob of epoxy into hole, then a "red eye" (bottom of hole), followed by an expanding sleeve
  - **B.** Screw in hex-head bolt, pound in "red eye"
  - **C.** Unscrew hex-head, insert eyebolt with epoxy
  - **D.** Smooth blob of epoxy over opening of hole and base of eyebolt
  - **E.** Can remove small piece of eyebolt (before inserting) so as to hold a transect line
- Moore's A.C.A.s (Adhesive Capsule Anchors) (Rioux)
  - **A.** Consists of a glass capsule (=anchor) filled with quartz aggregate/resin
  - **B.** Capsule is placed in pre-drilled hole and an A.C.A. rod (threaded or rebar) is driven into the capsule
  - **C.** Chemical interaction of aggregate/resin forms a permanent bond between rod and substratum
  - **D.** A.C.A. capsules and rods are available in a variety of sizes, also need an A.C.A. drive unit which is attached to a pneumatic drill
  - **E.** Cures after a few hours; Rioux's group used several hundred A.C.A.s without a single failure
  - **F.** Single A.C.A.s range from $1.42 (3/8 x 2 1/2") to $22.16 (1 3/8 x 11"); single A.C.A. rods from $0.82 and $2.40 (3/4" carbon, stainless) to $12.17 and $64.00 (2 1/2" carbon, stainless); drive units from $4.00 (1/4") to $16.80 (1 1/4")
  - **G.** Write: M & E Marine Supply Co., Inc., Box 601, Camden, NJ 08101; ask for Mar-Vel Catalog ($2.00) (Moore A.C.A.s on pp. 36-37)

Electrolysis

- Will occur anytime dissimilar metals are in contact
- Avoid by using plastic when possible (plastic bolts, cable ties, etc.) or attach sacrificial zinc nuts
- If using stainless steel, make sure bolts and gear have same alloy number

Site Markers

General Location

Inexpensive Weights

- Inexpensive weights (not likely to be salvaged); use lift bags to position, but always use screw shackles when lifting heavy loads, never pelican clips (which often straighten or break under load)
  - **A.** Engine blocks
  - **B.** Parking lot bumpers
  - **C.** Railroad car wheels
  - **D.** Bags of cement (Dean)
    - Place bag in burlap bag, tape shut, and insert metal rod with loop on one end, flat plate on other
    - Puncture bags a few times to allow entry of seawater
    - Place whole unit underwater; cement will harden
    - Can also use cement bags to make artifical reefs (see: "Manipulation of Substrata and Organisms, Artificial Reefs", pg. 49)

Cables

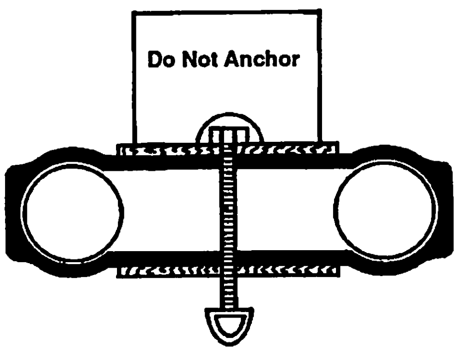
- Use discarded hydrocast cable (usually lying around most marine facilities which support oceanographic vessels) for attaching surface or subsurface buoys
- Cable is stainless steel, but not likely to be removed or salvaged (chain will be stolen)
- Attach to weights and buoys with plastic shackles (to avoid electrolysis)

Line

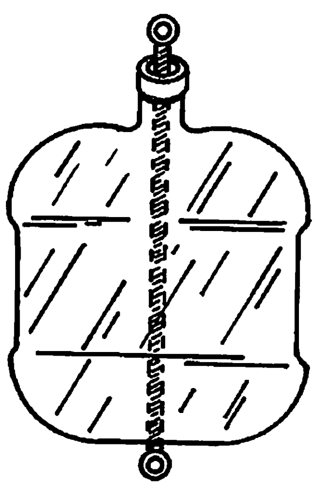
- For surface floats, use line that will sink (i.e., "Pot Warp"); avoid line that floats on surface and can foul propellers
- For subsurface floats, use high-visibility line (i.e., yellow polypropylene)
- In areas with much drift kelp, use line strong enough to resist additional drag when fouled with drift
- For chafing gear:
  - **A.** Use short length of garden hose inserted over line to protect line from chafing when tied into mooring, eyebolts, etc.
  - **B.** Use stainless steel chain-link
    - Wrap around base of boulder or eyebolt then bolt chain
    - Attach line protected with garden hose or the like

Buoys

- Surface
  - **A.** Variety are used, ranging from bleach bottles to mooring buoys



B. Inexpensive designs:
  - Tire Floats
    a. Use automobile tire with inserted and inflated inner tube
    b. Place wood disc above and below opening, screw tightly together with galvanized threaded-rod, attach nuts and eye bolt
    c. Attach sign with reflector tape to top disc (also use protruding nails on top portion to discourage gulls)
    d. Tends to flip over in rough seas
- Foam-Filled Plastic Bottles (Zimmerman)
    a. Insert a metal rod threaded at both ends into neck and base of a 5 gal. plastic water bottle
    b. Fill bottle with urethane foam (mix components and pour into bottle)
    c. Resulting foam firmly binds metal rod inside bottle; metal rod thus receives stress when attached to cable and float will last for years, even when small skiff tied off to upper rod
    d. Urethane foam kits are expensive, but can make numerous floats from one kit



- Subsurface
    A. Make sure they are below propeller depth for local fishing vessels (especially lobster boats!!)
    B. Bleach bottles with plastic caps and filled with fresh water (for gentle flotation) and Spongex Bullet Buoys (from fishing supply outlets) are popular
    C. Also used in conjunction with surface buoys to keep line from abrading on bottom during low tide

Specific Location: Hard Substrates

Masonry (cement) nails

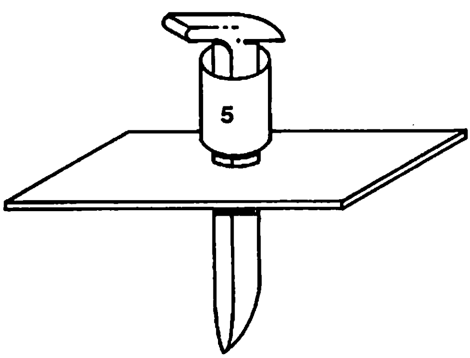
- Push nails through tags (bleach/milk bottle plastic, plexiglass, cow-ear tags, etc.) before bringing them underwater
- Simply hammer into rock
- Have lasted up to 6 yrs in warm temperate conditions before rusting away (Ambrose)

Epoxy/Cement Blobs

- Place on cleaned open surfaces or in cracks or crevices
- Leave as an artificial mound or attach identification
    A. Press into mound labeled pieces of plexiglass or equivalent
    B. Insert labeled masonry nails
    C. Insert plastic screw anchors; after epoxy/cement has cured, screw in labeled screws

Railroad Spikes
(Nelson)

- Push labeled (id number burned into plastic with soldering iron or pre-pared numbers covered with fiberglass resin) and tight-fitting PVC ring (Schedule 40) up to head of spike
- Drive spike through large square of bleach bottle plastic (for high visibility and ease of relocation) and into crack/crevice; use heavy mall
- Plastic square must be tightly-fitting so it won't spin around the spike in surge; spinning will remove rust from spike causing rapid erosion
- Will last decades in warm temperate areas, but as both spike and plastic square will become overgrown, will require periodic cleaning
- Spikes usually are available free or for minimal cost at scrap yards; a brief hike along any well-used stretch of railroad tracks will yield many specimens



Re-bar Spikes
(Carroll)

- Sharpen a length of re-bar, and pound into crevice
- Clamp a length of yellow garden hose over exposed portion for high visibility

Drill holes

- Drill holes, insert hardware (see: "Attaching Gear to Hard Substrata," pg. 12)

Specific Location: Soft Substrates

Placing Markers

- In high-energy areas, presence of markers can alter the immediate substratum (increase scouring, sedimentation, etc.), thus it may be necessary to place markers outside of study area and find specific site by triangulation;
- see: Fager et al. 1966. Equipment for use in ecological studies using scuba. Limnol. Oceanogr. 11:503-509

Earth Anchors

- Available in variety of sizes: all with loops at top and auger at bottom
- Originally designed for trailor tie-downs, available at Sears
- Insert metal rod in loop then screw into substratum

Plastic Tent Stakes

Bleach Bottles

(for low-energy areas)

- Fill bottles with sand, lay on side with at least half of bottle buried in sediment
- Attach stenciled tags for identification (do not rely upon felt-pen markings, as snails love to graze the ink)
- Work well as markers for corners of large quadrats (i.e., 10x10 m^2) and/ or in areas where large kelps could become fouled in earth anchors or other erect markers
- Will require periodic cleaning

Specific Location, Electronic Systems

- Expensive to purchase, but can be reasonable to rent

Motorola Miniranger Navigational Device

- Used by Schroeter and Dixon
- Requires 2-3 shore-based transponders which receive and return a signal from the boat
- Digital read-out gives distance in meters to shore station
- Use for:

  A. Mapping and re-locating stations

  B. Census of large organisms which are patchy and/or in very low density

    - Boat follows divers who are swimming along band transect and counting organisms
    - Miniranger reading recorded every few minutes and correlated with divers' location
    - Divers release pop-up float every 3-5 minutes, location recorded

- Cost: approximately $40,000 (Motorola Inc., Position Determining Systems, Box 2606, Scottsdale, AZ 85252)

Quadrats and Transects

General

- Widely used to determine patterns of abundance, distribution, and dispersion of organisms
- Many variations-on-the-theme, thus one should be aware of advantages/ disadvantages of each
- It is beyond the scope of this guide to suggest and/or discuss the many variations, but the following are emphasized:
  - **A.** Consult references during the planning phase (see below)
  - **B.** Collect preliminary data on both the area and organisms prior to the formulation of a sampling program (i.e., destructive or non-destructive, sample size and number)
  - **C.** Know beforehand how the data will be analyzed, as analysis will depend on method of collection
  - **D.** Replicate samples and randomize sampling; a necessity if parametric statistics will be used

Quadrats

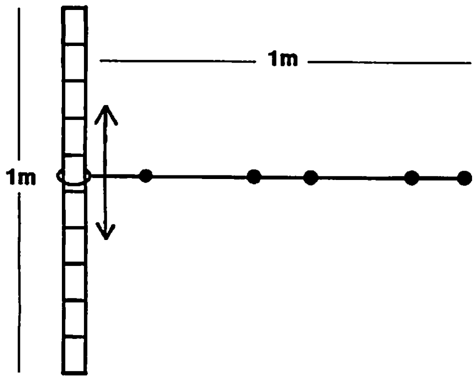
Square Quadrats

- "4-sided" quadrats
  - **A.** Small quadrats are easily made from iron re-bar; another design (Carroll) incorporates machined stainless-steel quadrats with etched numbers on one side for random-point sampling and with holes drilled in corners so quadrat can be placed over pins marking permanent location
  - **B.** Quadrats can be "strung" to subdivide the area for more accurate counts or for randomly collected subdivisions
  - **C.** Larger quadrats must be lighter and possibly collapsible for ease of transport and use
    - Aluminum stripping
      - **a.** 2" wide x 1/8" thick
      - **b.** Hinged on three corners with wing nuts or bent eyebolts
    - PVC pipes (Schedule 40)
      - **a.** Drill holes in pipes so they will sink
      - **b.** For collapsibility, thread bungy cord inside pipe
- "3-sided" quadrats
  - **A.** Essential in areas with kelp; "missing" side allows quadrat to be placed "over" attached kelps
  - **B.** Bend re-bar or aluminum rod to form sides
- "2-sided" quadrats
  - **A.** 2 adjoining "soft" sides made of line
  - **B.** Simply stretch out to make quadrat

Point Contact Quadrats
(="Random" Point Contact)

- Basic design (many variants available) consists of a 1m, knotted line (randomly or regularly placed knots) which is either attached to, or allowed to slide along, a 1 m bar
- Bar is placed at the location to be sampled and the knotted line stretched tightly from the bar
- All organisms at each knot (from substrate to 1m above the substrate) are identified
- If no organisms are present at a knot, the type of substrate is noted
- See Davis, 1985 ("References," pg. 20)

- Not truly random if same knots are used each time quadrat is employed



Circular "Quadrats"

- Reduces "edge-effect"
- Highly portable; simplest design consists of a weight with weighted length of line to circumscribe desired area.
- Particularly good in kelp beds, eelgrass meadows and other areas where rigid quadrats are hard to fit around 3-D objects.

Permanent Quadrats

- Positioning: Method I (Carroll)
  - **A.** Clean and chip hard substrata under 3 or 4 corners of positioned quadrat
  - **B.** Place blobs of epoxy onto cleaned areas
  - **C.** After epoxy sets, drill holes (to support brass positioning pins when sampling)
  - **D.** Between sampling periods, insert small cork into holes to prevent sand accumulation; when sampling, remove cork, insert brass pin, align quadrat over pins
- Positioning: Method II (Witman)
  - **A.** Drill two 1/4" holes for each quadrat
  - **B.** Insert plastic screw anchor and 3" long, 1/4" brass screw for permanent alignment pin, attach tag over a pin for reference
  - **C.** Quadrat or quadrapod has two holes at top corners which precisely fit over pins
  - **D.** Has lasted >6 years
- Positioning: General Problem
  - **A.** Drilling holes is labor-intensive
  - **B.** Minimize number of holes to drill:
    - Construct PVC grid to use each time quadrats are censused
      - **a.** Grid could be 2 m on a side, thereby containing 4, 1 m^2 quadrats
      - **b.** Thus, one pin (and hole) at each corner of PVC grid is half the work of two holes for each 1 m^2 quadrat
    - Construct a "reference bar" (Witman)
      - **a.** Use 3 m long aluminum angle
      - **b.** Holes at each end of bar fit over alignment pins in rock
      - **c.** Drill holes in vertical aspect of angle bar at 50 cm intervals which receive bolts protruding from quadrat or quadrapod
      - **d.** Enables a strip of six 0.25 m^2 quadrats to be re-located with only two alignment pins
    - Insert one eyebolt (or any bolt with "polarity")
      - **a.** Place in a specified corner (i.e., lower right) of quadrat
      - **b.** Align quadrat so bottom side is parallel to eye of bolt
    - Use photos to relocate quadrats
      - **a.** Make large prints of initial and/or previous photos of quadrats
      - **b.** Laminate prints, take underwater to aid in relocation

- Labeling
  - **A.** Cow-ear tags
    - Modern Farm Co., 1825 Bighorn Ave., Cody, Wyoming 82414 (800) 443-4934
    - High visibility, durable plastic tags, great for marking permanent quadrats
    - Use described in: Witman, J.D. 1987. Subtidal coexistence: storms, grazing, mutualism, and the zonation of kelps and mussels. Ecol. Monogr. 57:167-187
  - **B.** Brass survey stakes epoxied into rock
  - **C.** Epoxy "blobs" with numbered tags (brass, stainless, plastic) embedded into blob

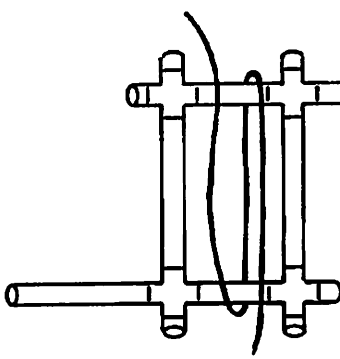
Transects

Use

- As a reference line to locate sampling areas
- As a band or strip transect
  - **A.** Swim along transect, count organisms within specified distance on either side of line (distance depends on organism to be counted)
  - **B.** Must use some sort of guide to delineate distance from line
    - PVC "wand"
    - Coat-hanger wire (small and dark-colored, won't disturb fishes)

Materials for Transects

- Fiberglass transect tapes (50,100,150m)
  - **A.** Available from forestry suppliers but are expensive
  - **B.** Tend to break, especially when used in heavy surge
  - **C.** Use tapes with minimal amounts of metal
  - **D.** If left overnight in an urchin "barrens," will be eaten by urchins
- "Homemade" Transects
  - **A.** Use 3/16" dacron or nylon line; polypropylene tends to float and is difficult to tie
  - **B.** Mark line at appropriate intervals with plastic shrink tubing (heat shrink) or niko-press sleeves (stamped with numbers)
  - **C.** If necessary, hammer lead sleeves on line for weighting (1 every 5 m)
- Permanent transects
  - **A.** Use leaded-core line attached to substratum with hardware drilled/epoxied into rock or embedded in marine cement
  - **B.** Secure to substratum in sections (so wayward anchors will rip out sections, not the entire line)
  - **C.** Generally, it is unwise to leave lines submerged between sampling periods as they will become fouled and probably will be snagged and lost
- Reels
  - **A.** Simplest consist of a flat "H"-shaped piece of plastic or wood
  - **B.** Make reel with PVC tubing; extensions for cranks



  - **C.** Better reels have hollow center in which to insert rod for rapid pay-out; cranks for rapid retrieval

References

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Sampling Irregular Surfaces with Quadrats

- Quadrats are essentially a two-dimensional technique and may not be appropriate for a highly structured system (i.e., coral reef)
- An irregular surface can greatly increase the area within a quadrat and densities may be over- or under-estimated
- Consult:

  A. Carleton, J.H., and P.W. Sammarco. 1987. Effects of substratum irregularity on success of coral settlement: quantification by comparative geomorphological techniques. Bull. Mar. Sci. 40:85-98.
    - Developed a 3-D profile gauge to analyze surface area, surface irregularity, average surface area, plate angle
    - Evaluated and compared 5 measures of surface irregularity (found vector dispersion to be most appropriate)

  B. Dahl, A. 1973. Surface area in ecological analysis: quantification of benthic coral reef algae. Mar. Biol. 23:239-249 (develops a surface-area index).

Sampling Irregular Surfaces with Transects

- Use small chain (link = 1.25 cm)
- Sufficiently heavy to drape over contours and stay in place

Census Methods for Fishes
(quantitative and qualitative)

General

- Must match census method to behavior of fishes (i.e., cryptic, schooling, attracted/repelled by divers) and whether single or multispecies surveys
- Must insure that visibility, water motion, time of day, and amount of time counting are constant from census to census or are taken into account in the analysis
- If interested in long-term monitoring (i.e., monthly changes), initially census fish over several consecutive days to determine if short-term variability is significant
- If necessary to estimate sizes of fish, consider a training program for yourself or assistants:
  - **A.** Use preserved fish of known sizes (DeMartini)
    - Attach fish to stake and insert stake into soft substratum along a transect
    - Divers calibrate their estimates by swimming several replicates of transect
  - **B.** Use PVC pipe of various lengths
    - Randomly thread pieces onto a transect
    - Divers calibrate their estimates by swimming transect and sorting estimates into a size-frequency distribution
    - Bell, J.D., G.J.S. Craik, D.A. Pollard, & B.C. Russell. 1985. Estimating length-frequency distributions of large reef fish underwater. Coral Reefs 4:41-44.
  - **C.** Use diving mask equipped with calibrated mask-bar
    - Count number of mask-bar units between snout and caudal fin when fish is parallel to mask
    - Swenson, W.A., W.P. Gobin, and T.D. Simonson. 1988. Calibrated mask-bar for underwater measurement of fish. North Amer. J. Fish. Manag. 8:382-385

Visual Transects
(Strip transects)

- Swim along transect, count fish within pre-defined "corridor"
- Since 1980, several studies have critically evaluated and developed visual survey methods (see references, below):
  - **A.** Limits of corridor should be carefully chosen and marked (using thin wire, etc.), as errors in judging distance can hopelessly bias results
  - **B.** Swimming speed should be standardized: highly mobile species overestimated at slow speeds, highly cryptic species largely overlooked at any speed
  - **C.** May need to use different methods for different groups in a multispecies census
  - **D.** Replicate transects
- Use tape recorder for efficiency (Cowen)
  - **A.** By not having to look up-and-down to record data, can keep better track of which fishes were counted
  - **B.** Can swim transect at a faster rate of speed, thus counting fishes before they react to diver

Cine/Video Transects

- Get a "permanent" record of transects
- Potential problems include: expense, equipment failure, less "visual" acuity than human eye, severe difficulties in estimating size,

and difficulties in identifying mobile and cryptic species

- Also can deploy video cameras on tripods for continuous and extended (up to 90 min) recording of fishes in an area (no diver interference)

Circular Plots

- At discrete points (randomly located), divers view (visual/video) all fish within a 360^0 arc and within a defined time period
- Generally require less time than transects, but results are more variable

Species-Time Relationships

- Rapid-Visual Technique (Jones and Thompson)
  - **A.** Within a given area, divers note presence of a species during successive and fixed, time intervals
  - **B.** Species counted only once and assigned a score according to the interval in which it was first noted
  - **C.** Assumes abundance of fishes correlated with time of occurance (i.e., most abundant species noted in first interval)
- Species accumulation curves (Kaufman and Ebersole)
  - **A.** Relatively quick method to compare fish populations in two or more areas
  - **B.** Start stopwatch and swim randomly through an area
  - **C.** Whenever a new species is noted, stop stopwatch and record time
  - **D.** Continue until rate of new additions equals some pre-defined limit (i.e., <2/10 min interval)
  - **E.** Plot cumulative number of species as a function of time

References

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Sampling Program for Soft Substrates

General

- Methods developed by Harper during an 8 yr study off Texas
- Techniques allow divers to sample all stations of one site in one day or less
- Up to 26 station-arrays were sampled monthly, each array <1 day; support vessel on-and-off station in 10 min or less

Specifics

Gear

- Sampling array (on surface-to-bottom line)
  - A. A small mushroom anchor separated from the samplers by about a 2 m interval
  - B. >3 small sampling devices (i.e.,Ekman grabs) at 1.5 m intervals, each with an elastic band
  - C. A swivel shackle with rolled-up lift bag attached
  - D. A large surface buoy
- A 1 liter VanDorn water sampler equipped with a back-pack type cylinder clamp is attached to a diver's cylinder
- A plastic jar for sediment collection

Procedure

- Vessel on station, line with attached anchor and samplers is payed out by tender
- When last sampler is underwater, divers grab line and enter water; weight of anchor helps pull entire array to bottom; tender keeps tension on line to keep samplers spread apart
- Line is cast loose when buoy goes over; vessel maintains station nearby
- On bottom, divers manually push samplers into substrate, trigger samplers, close jaws by hand if necessary, and secure vent flaps with elastic bands
- Diver with attached water sampler rolls to one side to get sampler close to bottom and triggers sampler
- Diver with plastic jar obtains superficial sediment sample
- Divers make visual observations of substratum
- Divers inflate lift bag to raise array
- Buoy line caught and array pulled aboard

Advantages

- Short on-station time; necessity where weather changes rapidly and large number of stations must be sampled
- Fine sediments are not swept away by pressure wave preceding a remote grab; divers can move samplers to an undisturbed area and collect
- Divers can make observations while collecting

Quantitative Photography

General

General plans for several framers or "quadrapods" for Nikonos underwater cameras are given in the following section; specific plans are in Appendix I. The framers are designed to hold the camera and 1-2 strobes to photograph a quadrat at the base of the quadrapod. Quadrapods minimize the amount of fumbling and maximize the number of photo-quadrats that can be taken on a dive because everything is pre-set. Typical uses of quadrapods include population surveys and monitoring fluctuations in the abundance of sessile organisms over time in permanent quadrats.

Abundance data can be obtained by projecting slides (of photo-quadrats) onto a sheet of random dots, counting the number of dots falling on a given species, and calculating the number of "hits" as a percent of the total yielding an estimate of percent cover (see: Sousa, W.P. 1979. Ecology 60:1225-1239). Use open circles (1-2 mm) instead of solid dots for ease of identification. The number of random dots is usually 100 although 200 is better statistically and more dots will decrease variance for some species. Each set of random dots should be used only four times (rotate the sheet to get four different configurations). A more accurate, but more time-consuming method to determine % cover is to use an electronic planimeter (MacTablet, Summagraphics Tablet, Numonics digitizer) connected to a computer or an image analyzer (i.e., Zeiss MOP-30), (Contact Witman for details).

In addition to abundance data, size-data are easily obtained by measuring organisms directly from the photo-quadrats. Obviously, a scale or rule must be placed along the edge of the photo-quadrat.

Sizes

- 0.5 m² (quadrapod area) (Witman, see Appendix I)
  - A. Uses Nikonos 15 mm lens
  - B. Designed to photograph spatial interactions among corals and other sessile inverts on Caribbean reefs
  - C. It is nearly the largest size that can be easily used; two divers are required to maneuver it against minor currents; one diver can handle the quadrapod if no currents are present
- 0.25 m² (quadrapod area) (Witman, see Appendix I)
  - A. Uses Nikonos 15 mm lens
  - B. Small enough for one diver to handle, yet quadrat area is large enough so that extensive areas can be covered
  - C. Witman has used two models: one with a 50x50 cm quadrat, the other reduces the camera-to-subject distance by utilizing a rectangular quadrat (60.3 x 41.5cm; same as film format)
  - D. Illumination provided by two Nikonos SB 102 strobes connected with a Nikonos double synch cord or use one strobe on slave
  - E. See: Witman, J.D. 1985. Refuges, biological disturbance, and rocky subtidal community structure in New England. Ecol. Monogr. 55:421-445.
- 0.25 m² (quadrat area) (Pratt)
  - A. Uses Nikonos 28 mm lens

B. Legs of quadrapod should measure 31 inches to photo a 53 x 48 cm quadrat (2544 cm²)

C. Lens should be set at 2 ft

- 0.05 m² (quadrat area) camera framer (Sebens)

A. Uses Nikonos 35 mm lens with #3 Hydro Photo Closeup lens

B. Designed for monitoring overgrowth interactions among relatively small sessile inverts on subtidal rock walls

C. Framer dimensions are: 18 x 25 cm quadrat (450 cm²), 47 cm legs, top frame is 15.5 x 11 cm

D. One strobe (Ikelite MV substrobe) is supported by an arm of aluminum angle that is attached to one of the legs just below the top frame; a mirror on the right side of the quadrat reflects some of the strobe light to provide more even illumination

E. See: Sebens, K.P. 1986. Spatial relationships among encrusting marine organisms in the New England subtidal zone. Ecol. Monogr. 56:73-96.

Standard Close-up Framers

- Standard Close-up Framers: any manufacturer's close-up framer can be used for quantitative photography by simply attaching (i.e., with super glue) a cm scale to side post of the framer

Additional References

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Video/Time-lapse Photography

- Use Super 8 mm time-lapse movie camera (Sebens)

A. Eumig or Minolta (Model XL-401), both with built-in intervalometer

B. Power source is 8-10, D-cell batteries wired in series, with a small flash wired to camera

C. Camera, battery pack, and flash are contained in one rectangular Ikelite housing (eliminates thru-housing connectors)

D. Tested to 65 m, runs for approximately 3 days

- Time-lapse video units (Witman and Sebens)

A. Available from Quest Marine Video, 505 Calle Sorpresso, San Clemente, CA 92672

B. Powered by a 21-20 amp/hr nicad battery pack with a programmable timer

C. Camera can be turned on and off 7 times/day

D. Witman and Sebens are using system to record seasonal variation in fish predation at a remote site (30 m depth, 110km offshore)

- Use Nikon F camera with motor drive

A. Place camera with 250 shot film-pack and a homemade intervolameter in an Ikelite housing

B. Witman, J.D. 1985. ("Sizes". 0.25 m², pg. 25)

- See also, Christie, H. 1983. Use of video in remote studies of rocky subtidal interactions. Sarsia 68:191-194.

Photomosaics

- Can generate a photostrip of a large area
- Need sufficient overlap to insure even transition from photo to photo
  - A. Use plumb bob to maintain precise height
  - B. To determine distance of bottom covered:

$$\frac{\text{focal length of lens}}{\text{film size}} = \frac{\text{height off bottom}}{\text{b (area of coverage)}}$$

  - C. If need 30% overlap, take 30% of "b"

Useful Catalogs for Housings

- Jay-Mar Engineering Services, 1910 Milan Pl., San Pedro, CA 90732 (housings for video cameras and recorders)
- Ikelite Underwater Systems, 50 West 33rd St., Box 58100, Indianapolis, IN 46208
- Aqua Vision Systems, Inc., 804 Deslauriers St., Montreal, Quebec, Canada H4N 1X1 (sold by Helix); makes Capsule 8, aluminum housing for Sony Handycam 8 mm video camera, with wide angle lens in front port (94^{0} view), pre-set focus with large depth-of- field (30 cm to infinity); but must point-and-shoot as can't see through lens
- Helix, 310 South Racine Ave., Chicago, IL 60607 (a good general source for UW photo gear)
- Hypertech Underwater Video, 750 East Sample Rd., Pompano, FL 33064
- Video Vault, 20803 #19 Stuebner Airline, Spring, TX 77379
- Image Devices International, 1825 NE 149 St., Miami, FL 33181 (for broadcast industry equipment)

Underwater Magnifying Devices

Diopters

- Place diopter 2 lenses in lower half of face plate, use as "bifocals" (Chess)

Magnifiers

- Mladenov, P.V. & I. Powell. 1986. A simple under- water magnifying device for the diving biologist. Bull. Mar. Sci. 38:558-561.
- An anodized aluminum model with high-quality optics and battery-operated illumination can be obtained from Underwater Optics, 3164 Robin Hood Dr., Nanaimo, B.C., V9T 1P1 Canada, (604) 721-8866 for $295; design is based on Mladenov and Powell's)
- Pratt, W. 1973. Macrosnooping. Skin Diver, Sept. pp. 38-39. (a simple underwater hand lens)

Microscope

- See Kennelly, S. and A. Underwood. 1984. Underwater microscopic sampling of a sublittoral kelp community. J. Exp. Mar. Biol. Ecol. 76:67-78.

Methods for Collecting Organisms

Permits

- Obtain collecting permits from appropriate agencies

Airlifts/Suction Devices

- Used to remove small mobile organisms from hard substrates, or buried organisms from soft substrates
- Range from large to small, with rigid or flexible pipes, and powered by a diver's air supply (via BC connection) or by a separate scuba tank
- See Appendix II for airlift designs
- Additional references/designs
  - **A.** Chess, J.R. 1978. An airlift sampling device for in situ collecting of biota from rocky substrata. MTS Journal 12:20-23.
  - **B.** Davies, I.J., and D.J. Ramsey. 1989. A diver-operated suction gun and collection bucket for sampling crayfish and other aquatic macroinvertebrates. Can. J. Fish. Aquat. Sci. 46:923-927.
  - **C.** Kennelly, S.J., and A.J. Underwood. 1985. Sampling of small invertebrates on natural hard substrata in a sublittoral kelp forest. J. Exp. Mar. Biol. Ecol. 89:55-68.
  - **D.** Stretch, J.J. 1985. Quantitative sampling of demersal zooplankton: re-entry and airlift dredge sample comparisons. J. Exp. Mar. Biol. Ecol. 91:125-136.
  - **E.** Tanner, C., M.W. Hawkes, and P.A. Lebednik. 1977. A hand-operated suction sampler for the collection of subtidal organisms. J. Fish. Res. Bd. Can. 34:1031-1034.
  - **F.** Tunberg, B. 1983. A simple, diver-operated suction sampler for quantitative sampling in shallow, sandy-bottoms. Ophelia 22:185-188.

Algal-associated Organisms

- On small algal species, collect entire plant in nytex bags with draw-strings
- On large kelps (entire plant or specific portions)
  - **A.** "Bag" plant with large (1 m diameter) plankton net, cinch tight, cut protruding blades/fronds
  - **B.** Coyer, J.A. 1984. The invertebrate assemblage associated with the giant kelp, *Macrocystis pyrifera* at Santa Catalina Island, California: a general description with emphasis on amphipods, copepods, mysids, and shrimps. Fish. Bull. U.S. 82:55-66.
- Use epifauna sampler
  - **A.** Resembles horizontal version of benthic grab-sampler
  - **B.** Position sampler, close jaws, cut protruding blades/fronds
  - **C.** Virnstein, R.W., & R.K. Howard. 1987. Motile epifauna of marine macrophytes in the Indian River lagoon, Florida. I. Comparisons among three species of seagrasses from adjacent beds. Bull. Mar. Sci. 41:1-12.

Macroalgae

- Use inexpensive burlap bags
- Thread small line through bags and attach halibut clip (or caliper clamp) to line
- Bags can be quickly and easily clamped to each other, another line, etc.

Corers for Organisms in Soft Substrates

Coffee-can Corers
(Chess)

- Cut both ends off standard coffee cans, retain plastic lid
- Glue fine-mesh nytex to one end
- To use, insert can (nytex end up), dig away sediments on one side and slip plastic lid under open end

Suction Corer
(Carroll)

- Use appropriate length of Schedule 80 PVC
- Sharpen one end, attach 3/8" plywood to other using screws and glue
- Attach rope handle to plywood with through-bolt; also drill small hole in plywood and insert small rubber stopper
- To use: remove rubber stopper, insert corer into substrata, replace rubber stopper, pull out corer and place into plastic bag

Water-Column Collecting

- Two divers can readily push a 1 m net stretched over a square (78 x 78 cm) PVC frame; a square frame allows for better definition of vertical stratification patterns
- One diver can push two small nets connected via a bar (to form a "dumbell" shape); diver holds the center bar while swimming
- Plankton nets also can be attached to diver-towing vehicles; see: Ennis, G.P. 1972. A diver-operated plankton collector. J. Fish. Res. Bd. Can. 29: 341-343.

Cod-end Attachment I
(Chess)

- Get a plastic jar with flexible and threaded plastic lid (white)
- Cut top out of lid and bottom out of jar
- Sew or glue threaded lid "ring" to cod end
- Tie jar "tube" into end of plankton net
- Attach cod-end to net by screwing lid "ring" onto jar "tube"
- After sample has been collected, unscrew cod-end and attach it to another, intact jar for temporary storage; the method reduces spillage, especially on a rocking boat

Cod-end Attachment II
(Cowen)

- Purchase plastic cod-end, threaded ring (from Sea Gear, General Oceanics, etc.)
- Attach to net with large hose-clamp
- Screw in any wide-mouth collecting jar

Larval Tube Traps

(Gaines, modified by Witman)

- Use plexiglass cylinder with height:diameter ratio >8:1; this ratio will create dead space above cylinder such that plankton sweeping across top will "fall in"
- Fill cylinder with 12% formalin dyed to enhance visibility, formalin and fixed larvae will stay in cylinder for at least a month
- Cap cylinders before moving, attach to substratum or rack
- Sampling efficiency must be determined across a range of flow speeds
- See: Butman, C.A. 1986. Sediment trap braces in turbulent flow: results from a laboratory flume study. J. Mar. Res. 44: 645-693.

Emergence Traps

- Used to capture nocturnally active zooplankton as they emerge from diurnal shelters (algae, sand)
- For designs and discussions, see:
  - **A.** Hobson, E.S., and J.R. Chess. 1979. Zooplankters that emerge from the lagoon floor at night at Kure and Midway Atolls, Hawaii. Fish. Bull., U.S. 77:275-280.
  - **B.** Rutzler, K., J.D. Ferraris, and R.J. Larson. 1980. A new plankton sampler for coral reefs. Mar. Ecol. 1:65-72.
  - **C.** Youngbluth, J.J. 1982. Sampling demersal zooplankton: a comparison of field collections using three different emergence traps. J. Exp. Mar. Biol. Ecol. 61:111-124.

Octopuses

(Ambrose)

- To remove from holes or shelters (alive)
  - **A.** Fill 60 cc syringes with bleach
  - **B.** Direct bleach into shelter
- To anaesthetize, slowly drip EtOH into seawater

Fishes

Poisons/Anaesthetics

- Observe local and/or national regulations; obtain all permits/licenses before use
- Assume all chemicals are toxic to humans, use care when handling both topside and underwater
- Application underwater
  - **A.** If possible, mix a dye with the chemical for greater precision in directing the chemical underwater
  - **B.** Plastic bags
    - Use Sears "Seal-N-Save" device (or equivalent) to seal chemical into plastic bags
    - Double-bag for added strength
    - Transport to collecting site, puncture bags, squeeze bag to direct chemical into local area
  - **C.** Large plastic syringes (Hartney)
    - Affix flexible tubing and clamp to large syringe (50-100 ml)
    - Remove clamp, direct desired amount of chemical into crack/crevice, re- clamp for additional use
  - **D.** Plastic squeeze bottles:
    - Not effective as chemical is diluted with sea water after each squeeze

E. Collapsible plastic water jugs (Robertson)
  - Fill with poison/anaesthetic
  - Affix an outboard engine gas hose to valve of jug with cable ties; insure that bulb is distal to jug
  - Use open-close valve of jug and squeeze bulb to direct chemical
  - One-way valve of bulb prevents dilution after each application

- Poisons:

  A. Rotenone (natural poison); see: Smith, C.L. 1973. Small rotenone stations: a tool for studying coral reef fish communities. Amer. Mus. Novit. 2512:1-21.

  B. Pronox Fish (synthetic)

  C. Chem-fish (synthetic)

- Anaesthetics

  A. In the US, there are no legally registered fish anaesthetics that allow for the release of fish or use of fish for food within 21 days of anaesthetization

  B. Efficiency of anaesthetics can vary with water chemistry, temperature, size and species of fish; thus, effective concentrations may require frequent modification

  C. For full discussion and list of vendors, see: Gilderhus, P.A., and L.L. Marking. 1987. Comparative efficacy of 16 anaesthetic chemicals on rainbow trout. North Amer. J. Fish. Managm. 7:288-292.

  D. Four chemicals that produce rapid anaesthesia and recovery (Gilderhus and Marking, 1987):
  - MS-222 (Tricaine)
    - a. Only chemical registered for use as fish anaesthetic in US; lacks mammalian safety data, thus fish must be held for 21 days before release or use as food
    - b. Margin between effective and toxic dosages is narrow
  - Quinaldine:
    - a. Dilute with 9 pts EtOH or methanol if want fish unharmed, otherwise dilute with acetone or isopropyl
    - b. Moring, J. 1987. Use of the anaesthetic quinaldine for handling Pacific coast intertidal fishes. Trans. Amer. Fish. Soc. 99:803-806.
  - Benzocaine:
    - a. Over-the-counter, dissolve in acetone
    - b. McErlean, A.J., & V.S. Kennedy. 1968. Comparison of some anaesthetic properties of benzocaine and MS-222. Trans. Amer. Fish. Soc. 97:496-498.
    - c. Wedemeyer, G. 1970. Stress of anaesthesia with MS-222 and benzocaine in rainbow trout (*Salmo gairdneri*). J. Fish. Res. Bd. Can. 27:909-914.
  - 2-phenoxyethonol
    - a. Relatively high concentrations needed
    - b. Slow solubility in cold water

  E. CO_2:
  - Adequately anaesthetizes small fish, but requires fish to be removed to holding facility for anaesthetization
  - Use gaseous CO_2 or from $NaHCO_2$ and HCl
  - Deserves consideration as leaves no residues and requires no permits or holding time
  - See also: Post, G. 1979. Carbonic acid anaesthesia for aquatic organisms. Prog. Fish. Cult. 41:142-144.

- Chemicals may leave residues:

  A. Engstrom-Heg, R. 1987. Persistence of rotenone in ponds. North Amer. J. Fish. Managm. 7:162.

  B. Jaap, W.C., and J. Wheaton. 1975. Observations on Florida reef corals treated with fish-collecting chemicals. Fla. Mar. Res. Publ. 10:1-18.

C. Thomson, D.A., and C.E. Lehner. 1976. Resilience of a rocky intertidal fish community in a physically unstable environment. J. Exp. Mar. Biol. Ecol. 22-1-29.

Spears

- Use multi-prong pole spears or Hawaiian slings of appropriate size; small spears can effectively spear fish as small as 12 mm
- Design for very small fish (Chess)
  - A. Secure block of epoxy at tip of 1/4" or 1/8" stainless-steel shaft by tightly winding with stainless/copper wire
  - B. Insert prongs into epoxy: use stainless spring-steel wire for small prongs (inner wire from outboard control cables works well)
  - C. Attach loop of surgical tubing at opposite end of shaft, insert thumb through loop, and draw spear bow-and-arrow fashion

Airlifts

- Airlifts: can collect small fishes (cottids, gobies, blennies) from cracks/crevices

Slurp-guns

- Slurp-guns: effective for small fishes

"Mist" Gill Nets

(to catch fish alive)

- No float or lead lines
- Can be effective, but require constant monitoring to prevent fish death (by gilling or by predators)

Traps

- General
  - A. Can be effective, but some species may be trap-shy or trap-happy
  - B. Require regular monitoring
  - C. Fish may be injured or killed by decompression and/or temperature shock when trap is raised
- Smith, D., T. Kornfield, and B. Goodwin. 1981. Remotely operated trap for capture of territorial fishes. Prog. Fish Cult. 43:208-209.
- Simple trap design for small and/or yoy fishes
  - A. Cut neck from long-necked, plastic bottle (soda, mineral water, etc.)
  - B. Insert neck into body of bottle to make funnel entry, attach with marine seal
  - C. Cut bottom from bottle, replace with netting attached with marine seal

Underwater Hook-and-Line

- Use small fishing pole underwater, dangle baited hook in front of fish
- Wilson, T.C., 1981. An underwater fish tagging method. California Fish and Game. 67:47-50.

Electroshock

- Can be very effective, but is a logistical challenge
- Marine Fishes
  - A. Electrofishing unit in overhead vessel connected via 50 m lengths of multi-strand copper welding wire to stunning electrodes
  - B. Stunning field from 60 Hz, 24 v AC @ 115A delivered by two stainless grid electrodes (0.6 x 0.6 m) placed 2 m apart on bottom

C. When fish enter stunning field (0.6 x 0.6 x 2 m), diver (2-4 m outside stunning field) informs topside to activate units for 5-35 sec (depending on size of fish)

D. Divers collect stunned fish by hand; then tag, measure, and release in situ

E. See: Davis and Anderson 1989. ("Census methods for fishes." pg. 22)

- Freshwater Fishes

A. Backpack electrofisher placed in overhead vessel, connected via insulated cables to stunning electrodes

B. Electrodes modified into water-tight, 1m long probe with 2, 4 x 4 squares of wire screen positioned 15 cm apart @ distal end

C. Diver places probe close to fish and activates device: 1-2 sec sufficient for fish <100 mm within 20 cm of electrodes; larger fish must be within 10 cm

D. See: James, et al. 1987. Diver-operated electrofishing device. North Amer. J. Fish. Managm. 7:597-598.

General Curatorial Methods

Anaesthetization

- Need to relax specimens prior to fixation to avoid contraction or loss of appendages
- Types of anaesthetization include 7.5% $MgCl_2$, chilling, de-oxygenated water

Fixation

- Process stabilizes protein constituent of tissue
- Formalin is the most widely used fixative

A. Purchased as 37% solution of gaseous formaldehyde (37% formaldehyde = 100% formalin).

B. Ideal volume of formalin is 10x sample volume

C. Not a good preservative as will decompose to form acid; thus must be buffered (e.g., hexamine 200g/l)

D. Algae
  - Use 5% formalin (1 pt. 100% formalin: 19 pts. water)
  - Store fixed algae in black buckets to prevent pigment bleaching

E. Invertebrates
  - Use 5% formalin, inject larger forms
  - *Don't* use formalin as preservative for calcareous forms (e.g., fix in formalin, but transfer to non-formalin preservative)
  - *Do* use formalin as a fixative and preservative for high-water content organisms

F. Fishes
  - Use 10% formalin, inject or slit right side of abdominal cavity
  - Remove otoliths before fixing if needed for age determination

Preservation

- Process prevents autolysis and growth of bacteria and molds
- Alcohols are most widely used preservatives

A. Dilute alcohols with distilled water only; ethanol is better than isopropyl

B. Alcohols will bleach and dehydrate; shrinkage can be 40-70% (minimal for specimens with exoskeletons, maximal for soft-bodied forms).

C. Can reduce shrinkage by staged preservation (e.g. a few hours at 30%, then 50%) before final of 70%-80%

Labels

- Use plastic or treated linen paper
- Use carbon ink/typewriter ribbon or pencil (never ballpoint)

Resources

- Hereau, J.C., and A.L. Rice. 1983. Guidelines for marine biological reference collections. UNESCO Rpt. Mar. Sci. 22.
- Lincoln, R.J., and J.G. Sheals, 1979. Invertebrate animals. Collection and preservation. British Museum (Nat. Hist). Cambridge Univ. Press. 150 pp.
- Tsuda, R.T., and I.A. Abbott. 1985. Collection, handling, preservation, and logistics. pp. 67-86. In: Littler, M.M., and D.S. Littler (eds). 1985. Handbook of phycological methods. Ecological field methods: macroalgae. Cambridge Univ. Press. 617 pp.

Tagging Methods

General References

- Begon, M. 1979. Investigating animal abundance. Capture-recapture for biologists. Univ. Park Press, Baltimore. 104 pp.
- Emery, L., and R. Wydoski. 1987. Marking and tagging of aquatic animals: an indexed bibliography. U.S. Fish Wildlife Service Resource Publ. No. 165 (copies from U.S. Fish & Wildlife Service, Matomic Building, Rm. 148, Washington, D.C. 20240; Sup. of Doc. # I 49.66:1;65)
- Foster, M.S.,T.A. Dean, and L.E. Deysher. 1985. Subtidal techniques, pp. 189-232. In: Littler, M.M. and D.S. Littler (eds). 1985. Handbook of phycological methods. Ecological field methods: macroalgae. Cambridge Univ. Press. 617 pp.
- Stonehouse, B. (ed). 1978. Animal marking: recognition marking of animals in research. Univ. Park Press, Baltimore. 224 pp.

Algae

General Comments

- Algae usually tagged for growth and/or demographic studies; double tag all individuals
- For general review of algal demographics, see: Chapman, A.R.O. 1985. Demography. pp. 253-268. In: Littler, M.M., and D.S. Littler (eds). Handbook of phycological methods. Ecological field methods: macroalgae. Cambridge Univ. Press, 617 pp.

Types of Tags

- Plastic cable ties (tie wraps, zip-sticks)
  - A. Come in a variety of sizes and prices
  - B. Generally looped over or through various structures, usually with some sort of identification number
  - C. Effective identification numbers can be simple and inexpensive
    - Small pieces of milk carton or bleach bottle plastic
      - a. Attach Dymo label numbers
      - b. Etch numbers with soldering iron
      - c. Stencil numbers (stenciled numbers are best as they cannot be over grown or removed when brushed)
    - Small strips of Dymo tape (backing left on) with holes for cable tie
  - D. Cable ties may become brittle and break in near-freezing temperatures
- Bicycle handlebar tape, surveyors flagging
  - A. Can be tied around algae and will remain for weeks to 1-2 months (polyethylene more durable than vinyl)
  - B. Identification numbers can be stenciled or written with permanent felt markers, coded patterns of holes also can be used; label both ends of flagging.
  - C. Disadvantages
    - Can become brittle in water temperature <12° C
    - Fish tend to nibble on ends, accelerating erosion of tips and labels
- Bird bands: see Barilotti, D.C., & W. Silverthorne. 1972. A resource management study of *Gelidum robustum.* pp.255-261. In Nisizawa, K. (ed.). Proc. of the Seventh International Seaweed Symp. Univ of Tokyo Press, Tokyo.
- White plastic spoons (not clear spoons!)
  - A. Good for very small and/or fragile sporophytes attached on or near soft substrates
  - B. Write identification number on bowl of

spoon with felt-tip pen and/or attach a Dymo tape number; insert spoon handle into substratum

C. Highly visible and will remain in low energy areas for years, but can be "uprooted" by foraging bat rays or crabs seeking shelters

Methods to Measure Growth

- For large-bladed forms, punch holes in blade and monitor movement
- For small forms, stain with Calcofluor White (CW)
  - A. CW is a UV fluorescent molecule that binds to polysaccharides in cell wall of plants
  - B. Stain algal tissue by immersing in 0.01% (W/V) CW in seawater for 20 min., then rinse and return to field/lab aquaria
  - C. To measure growth: examine tissue with a fluorescent microscope; old tissue will fluoresce, new tissue (= growth since staining) will not
  - D. Waaland, S.D., and J.R. Waaland. 1975. Analysis of cell elongation in red algae by fluoresent labeling. Planta 126:127-138.

Small, Sessile Invertebrates

(barnacles, vermetid molluscs, cup corals)

- Place acetate sheets over substratum and organisms
  - A. Trace size and/or position with grease pencil
  - B. Creates a permanent record for survivorship studies, density, and distribution (nearest neighbor)
- Use photoquadrats (see "Quantitative Photography", pg. 25)

Soft-bodied Invertebrates

Garment tags, Floy tags,

(see also Fishes pg. 42)

- Have been used successfully on sea cucumbers; Muscat reports slight scarring after 1.5 yrs
- Have been used successfully on octopuses, but only if placed through the arm; placement through the web will last only a few days to weeks; see: Ambrose, R.F. 1982. Shelter utilization by *Octopus bimaculatus*. Mar. Ecol. Prog. Ser. 7:67-73.

Petersen Discs

(Ambrose)

- Two discs on either side of an octopus arm and connected by a pin will last several years
- Application requires removal and narcotization

Branding

- Brand can be formed from copper wire or tubing; either heat (from propane torch) or cold (liquid nitrogen) can be used
- Application requires removal and narcotization of organism and often results in inferior brands; negligible success with octopuses (Ambrose)

Vital stains

- Injection into octopuses
  - A. Requires brief restraining of octopus, but is quick and can be done in the field

B. Used with limited success, as mark is visible for only a few months (Ambrose)
C. See also: Kayes, R.J. 1974. The daily activity of *Octopus vulgaris* in a natural habitat. Mar. Behav. Physiol. 2:337-343.

- Painting onto sea anenomes

A. Lasts up to 12 mos with a single marking, longer with multiple markings
B. Use neutral red dye made into a thick paste
C. Remove anenome and chip of rock, blot column dry with paper towels
D. Apply dye thickly with paintbrush
E. See: Sebens, K.P. 1976. Individual marking of soft-bodied intertidal invertebrates in situ; a vital stain technique applied to the sea anenome *Anthropleura xanthrogrammica*. J. Fish Res. Bd. Can. 33:1407-1410.

Clam Siphons/Burrow Openings

- Metal rod with flagging tape

A. Place into substratum near siphons
B. Use different colored flagging on subsequent censuses
C. See: Breen, P.A., and T.L. Shields, 1983. Age and size structure in five populations of geoduc clams (*Panope generosa*) in B.C. Can. Tech. Rep. Fish Aquat. Sci. 1169. iv and 62 pp.

- Insert white plastic spoons (see also "Algae," pg. 36)
- PVC stakes (Engle)

A. Use suitable length of schedule 40 PVC
B. Apply stick-on numbers to each end of stake, cover with fiberglass resin
C. Insert near organism or burrow
D. When exposed portion becomes fouled, simply remove stake and invert
E. Stakes are non-descript, less likely to attract attention of "tourists"

Shelled molluscs

Paint/Ink

- Martex Tech-Pen and Tech-Pen Ink

A. For marking cohorts
B. Martex Corp., Box 681, Englewood, NJ 07631 (write for local dealer)
C. Ink avaliable in many colors; yellow, blue, green best for underwater
D. Apply small amount of ink to a dry and clean (perhaps sanded) surface; allow to air dry
E. Will last for 2-3 yrs

- India ink

A. For marking individuals
B. The surface should be sanded (using a Dremel tool or hand drill), both to clean as well as enhance contrast for ink
C. If more contrast is needed, apply a white background using typewriter correction fluid (non-water base)
D. Apply numbers with Rapidograph pen
E. Cover with super glue, fiberglass resin or a clear cement for protection (i.e., Dekophane Technical Cement, Rona Pearl Corp., Bayonne, NJ 07002)

- Fingernail polish: for marking cohorts, can be effective for short periods
- Dive-gear markers: for marking cohorts, can be effective for short periods

Pre-made Numbers
(for marking individuals)

- Wire markers (for telephone cables)

A. Available in most electrical supply stores
B. Numbers are adhesive and stick best to clean and dry surfaces
C. Coat with super glue, fiberglass resin or clear cement for protection

D. Can last for 2-3 yrs (Schmitt, Ambrose)

- Bee Tags (for marking honey bees)
  - A. Numbered plastic discs in five colors, ca. 2 mm diameter
  - B. Attach with Koppers epoxy or super glue (expensive, but fast)
  - C. Get from: Christian Graze KG, Postfach 2707, 7056 Weinstadt-Endersbach, FRG (ask for Opalith-Zeichenplattchen mit Kleben (bee tags with glue) @20.50 DM)
- Petersen Discs or stainless steel discs (for in situ marking of large gastropods)
  - A. Use plastic Petersen Discs or cut small (10 mm dia) discs from sheet of stainless steel and hand-punch numbers
  - B. Clean shell with Brillo pad
  - C. Apply a blob of Koppers epoxy or super glue
  - D. Smooth-out edges of epoxy/glue and affix disc
  - E. Insure that edges of discs are imbedded in epoxy/glue
- Stainless wire and numbered stainless discs
  - A. Used primarily for abalone
  - B. Thread wire through last two respiratory aperatures
  - C. Make sure that wire and discs are same alloy number to prevent electrolysis

Engraving

- Scribe number onto shell with an electric Dremel Tool (Division of Emerson Electric Co., 4915 21st St., Racine, WI 53406)

Tracking Movements of Motile Gastropods

- Use epoxy/super glue to attach dental floss (or equivalent) to cleaned shell so that floss will pay out as animal moves
- Attach spool of floss to a small wire loop and insure that it will freely pay out; track line for movement

Mass "Tagging" of Cohorts

- Hidu, H., and J.E. Hanks. 1968. Vital staining of mollusk shells with alizarin sodium monosulfonate. Proc. Natn. Shellfish. Assn. 58: 37-41.
- Trevelyan, G.A., and E.S. Chang. 1987. Light-induced shell pigmentation in post-larval *Mytilus edulis* and its use as a biological tag. Mar. Ecol. Prog. Ser. 39:137-144.

Sea Urchins

Penetrate Test

- Thread a stainless steel wire through a hollow needle which has been inserted through the test, attach identification beads (color-coded) through a tag, twist wire together (Nelson, B.V., and R.R. Vance. 1979. Diel foraging patterns of the sea urchin *Centrostephanus coronatus* as a predator avoidance strategy. Mar. Biol. 51:251-258.)
- Insert Floy tags into test, seat with silicone (Olsson, M., and G. Newton. 1979. A simple, rapid method for marking individual sea urchins. Calif. Fish Game 65:58-61.)
- Drill small hole (0.2 cm diameter) into test (but not into coelomic cavity) with a small finger drill

(0.5 cm), insert Floy tag just inside test wall; tags have lasted up to 81 days (Neill, J.B. 1987. A novel technique for tagging sea urchins. Bull. Mar. Sci. 44:92-94.)

- Additional references for test penetration
  - A. Ebert, T.A. 1965. A technique for the individual marking of sea urchins. Ecology 46:193-194.
  - B. Lees, D.C. 1968. Tagging subtidal echinoderms. Underwater Naturalist 5:16-19.

Penetrate Periproct

- Use small stainless steel fishing hooks
- For identification attach monofilament line with beads to hook
- Hook through periproct
- Fast and will last 3-4 days

Elastic Bands

- Fasten elastic bands (with attached tags) around ambitus of urchin; can last 84 days
- Dance, C. 1987. Patterns of activity of the sea urchin *Paracentrotus lividus* in the Bay of Port-Cros (Var, France, Mediterranean). Marine Ecology 8:131-142.

Aluminum Foil

- Crimp aluminum foil around long spines (will last 3-4 days on long-spined species)

Plastic Sleeve

- Slide small pieces of plastic over spine
- Use sleeve insulation from electrical wires and place over several spines; lasts 1-4 days
- Dance, 1987. Ibid.

Plastic Straws

- Push plastic straws over spines
- Will last up to five days on some species
- Mattison, J.E., J.D. Trent, A.L. Shanks, T.B. Akin, J.S. Pearse. 1977. Movement and feeding activity of red sea urchins (*Strongylocentrotus franciscanus*) adjacent to a kelp forest. Mar. Biol. 39:25-30.

Rubber Discs

- Rubber discs pushed over spines
- Works best on long-spined species
- Prepare thin rubber discs (i.e., from surgical gloves or thin cross-sections of 6 mm diameter surgical tubing) and number with India ink
- Stretch disc or tubing over end of plastic 1 ml pipette
- Place pipette (with attached disc or tubing) into a slightly larger-bore glass tube (5 mm)
- Place free tip of pipette over urchin spine; push disc or tubing down pipette and over spine using larger glass tube
- Requires less than 30 sec to tag, tags can last up to 12 mos
- Carpenter, R.C. 1984. Predator and population density control of homing

behavior in the Caribbean echinoid *Diadema antillarum*. Mar. Biol. 82:101-108.

Polypoxy Blobs
(Witman)

- Smear blobs of Polypoxy on spines and test (will last 3-4 days)

Spine Clipping
(Witman)

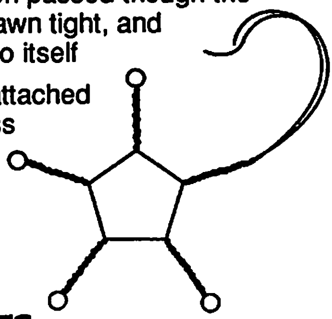
- Clip off 1-2 cm wide rows of spines with wire clippers
- Lasts for about 1 week with *S. droebachiensis*, but spines may be lost in other species

Vital Stains
(Briscoe, after Loosanoff,1937)

- Add 2 g Nile Blue Sulfate to 30 ml distilled water; boil to dissolve then dilute to 1 liter with seawater (don't re-use)
- Staining:
  - A. Shake excess water from urchin
  - B. Place urchin upside-down, partially into a beaker (filled with stain) to get patterns
  - C. Stain 30 sec to 1 min: patterns last 5-6 days; stain up to 6 weeks
  - D. Rinse urchin quickly with seawater after staining (to remove free stain)
- Reduce mortality by reducing stress to urchins as much as possible (e.g., avoid warm water, temperature changes, time out of water, etc.)

Wire Harness
(Cuenca, modified by Campbell and Briscoe)

- Use thin and inert wire (aluminum wire from Sargent-Welch, SPL wire, cat.# S85105-00-G)
- Bend wire into a pentagon with wire twists and loops at points of pentagon and a long "tail" from one of the twists
- Oral side of urchin placed in pentagon (pentagon should be just large enough to clear rasping motion of Aristotle's lantern), then wire twists are bent up and around test
- Tail is then passed though the loops, drawn tight, and secured to itself
- A tag is attached to harness



Sea Stars

Wire Harness

- Wire harness described for urchins should work for the "inflexible"stars, but not for the agile stars; simply bend wire twists between arms

Vital stains

- Need to remove animal for marking and subsequent identification
- Campbell, D.B. 1984. Foraging

movements of the sea star *Asterias forbesi* in Narragansett Bay, Rhode Island USA. Mar. Behav. Physiol. 11:185-198. (used Nile blue sulfate)

- Feder, H.M. 1955. The use of vital stains in marking starfish. Science 85:412.

External Tags

- Floy Tags
  - A. Insert into interradius
  - B. Can last up to 35 mos
  - C. Birkeland, C.T. 1974. Interactions between a seastar and seven of its predators. Ecol. Monog. 44:211-232.
- Spaghetti Tags
  - A. Tags held by nylon monofilament loop inserted through base of ray
  - B. Lasts at least 1 year
  - C. Dayton, P.K., G.A. Robilliard, R.T. Paine and L.B. Dayton. 1974. Biological accomodation in the benthic community at McMurdo Sound, Antartica. Ecol. Monogr. 44:105-128.
  - D. Savy, S. 1987. Activity pattern of the sea star *Marthasterias glacialis* in Port-Cros Bay (France, Mediterranean coast). Marine Ecology 8:97-106.

Engraver

- Use engraver to scar aboral surface

Grease Pencils

(Foster)

- Grease pencils: good for short-term marks on some species

Crustaceans

- Punch holes into uropods; will last through several molts
- Insert garment or Floy tag in epimeral suture; will remain when animal molts
- Apply Tech-Pen ink or Polypoxy to carapace; short-term, will last until animal molts
- Cooper, R. 1970. Retention of marks and their effects on growth, behavior, and immigrations of the American lobster, *Homarus americanus*. Trans. Amer. Fish. Soc. 99:409-417.

Fishes

General References

- For review of tags and tagging, see:
  - A. Jakobsson, J. 1970. On fish tags and tagging. Oceanogr. Mar. Biol. Ann. Rev. 8:457-499.
  - B. Wydoski, R., and L. Emery. 1983. Tagging and marking. pp. 215-237. In: Nielsen, L.A. and D. L. Johnson (eds). Fisheries Techniques. Amer. Fish. Soc., Bethesda, MD
- Numerous papers evaluate the effectiveness of various tag types (primarily on salmonids); check Zoological Record; Pices; Marking Techniques

External

- General comments
  - A. Location of tag usually through head or dorsal musculature, sometimes through caudal peduncle
  - B. Often affect behavior: fishes with external bead tags spend inordinate amounts of time soliciting cleaning
  - C. Also prone to "snag" on complex substrata

D. Often rapidly overgrown with algae, reducing effectivness

E. If use external tags, test to insure that behavior and susceptibility to predators/ disease are not compromised

- Types

A. Floy tags

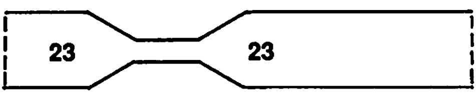
- Floy FD-68B tags are popular
- Floy Tag & Mfg., Inc., Box 85357 Seattle, WA 98145-1357

B. Garment tags

- Often available in local garment district
- Pennant of tag can be stamped, dymo-taped, or dyed with Rit dyes
- Also can remove pennant, string beads onto shaft, and melt top of shaft to hold beads

C. Surveyors Flagging Tape

- Cut broad-based notch in length of tape



- Number both sides of notch with marking pens
- Thread long end of tape into large needle
- Push needle through dorsal musculature under dorsal fin until notch is in musculature; trim tag
- Tagging can be done in situ, requires <2 min/fish; tags last up to 2 yrs
- Blumer, L.S. 1984. Simple, inexpensive method of tagging Ictalurid fishes for individual identification. Prog. Fish Cult. 46:152-154.

D. Monofilament and beads

- Use a stout needle to thread monofilament line into dorsal musculature just under 1st dorsal spine or through caudal peduncle above lateral line
- String colored beads on line for i.d. (get beads from hobby store); visual resolution of green and blue is difficult and >4-5 beads is difficult to distinguish and record
- Loop and tie line so that it is flush against spine; a drop of cyanacrylate glue on knot will make knot permanent
- Can also thread line through fish so have portion on each side, string beads, melt tip of line into a blob to hold beads
- See: Langley, R.G., & J.W. Driscoll. 1989. Caudal peduncle tag used for identification of individual small fishes. Prog. Fish. Cult. 51:55-57. (less damage and longer lasting)

E. Small plastic chips (see: Thompson, K.W., L.A. Knight, & N.C. Parker. 1986. Color-coded fluorescent plastic chips for marking small fishes. Copeia (2): 544-545.)

F. Fluorescein Dye-releasing Tag

- For visual tracking, attach tag to fish, follow dye trail (up to 5 hrs)
- Leaman, B.M. 1976. An inexpensive tag for short-term visual tracking studies. J. Fish. Res. Bd. Can. 33:1628-1629.

- Tagging Guns

A. Use Dennison Mark II tagging gun with small-bore needle

B. Again, check local garment district for availability

C. Can be used for garment and Floy tags

D. Can be used in situ, rinse metal parts with WD-40 after use

- Tagging fish underwater after collection

A. Collect underwater (see: Methods for Collecting Organisms. "Fishes," pg. 33), then use tagging gun or flagging tape technique

B. References:

- Wilson, T.C. 1981. An underwater fish

tagging method. Calif. Fish Game. 67:47-50. (uses small fishing poles)

- Gitschlag, G.R. 1986. A collapsible trap for underwater fish tagging. Bull. Mar. Sci. 39:719-722.

- Tagging fish underwater without collection

A. Primarily for larger fish
- Hand-spear modified to hold hollow needle which accepts Floy dart-tag
- Tag remains in fish after penetration
- Mathews and Bell. 1979. A simple method for tagging fish underwater. Calif. Fish Game 65:113-117.

B. Cowen has tagged California sheepshead from 0.5-1.2 kg in size and observed tagged individuals for up to 1.5 yrs

C. Be aware that for some fishes, "spear-tagging" may result in subsequent aversion to diver/observation

Mutilation

- Fin clipping

A. Variable regeneration times

B. Use pelvic fins, not fins important in locomotion

C. Can be done in situ

- Cold Branding

A. Must capture and remove fish from water

B. Use brand chilled with liquid nitrogen, avoid branding lateral line

C. Brand often not visible in fishes that have irregular color patterns and/or can change intensity of background color

D. Raymond, H.L. 1974. Marking fishes and invertebrates. I. State of the art of fish branding. MFR Paper 1067. Mar. Fish. Rev. 36:1-6; and II. Brand size and configuration in relation to long-term retention on steelhead trout and chinook salmon. MFR Paper 1068. Mar. Fish. Rev. 36:7-10.

- Subcutaneous Injections

A. Inject acrylic paint or alcian blue subdermally, can be done underwater

B. Lasts about 1 yr; effective primarily for lightly-pigmented and small-scaled species

C. Thresher, R.E. & A.M. Gronell. 1978. Subcutaneous tagging of small reef fishes. Copeia 1978(2):352-353.

D. Hill, J., and G.D. Grossman. 1987. Effects of subcutaneous marking on stream fishes. Copeia 1987(2):492-495.

- Sub-scale injections (Sikkel, after VandenBarge and R. Warner)

A. For fish with large scales, can be done in situ: capture and immobilize with net

B. Use 1 or 3 cc syringe and 25 G needle

C. Inject solution of alcian blue (Sigma) under scale and into scale pocket, taking care not to lift scale

D. Will fade after a year, but is recognizable up to 2 yrs

Telemetry

- 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. 74:998-999.
- Ralston, S.L. and M.H. Horn. 1986. High tide movements of the temperate herbivorous fish *Cebidichthys violaceus* (Girard) as determined by ultrasonic telemetry. J. Exp. Mar. Biol. Ecol. 98:35-50.
- Schramm, H.L., and D.J. Black. 1984. Anaesthesia and surgical procedures for implanting radio transmitters into grass carp. Prog. Fish Cult. 46:185-190.
- Stasko, A.B., and D.G. Pincock. 1977. Review of underwater telemetry, with emphasis on ultrasonic techniques. J. Fish. Res. Bd. Can. 34:1261-1285.
- Winter, J.D. 1983. Underwater biotelemetry, pp 371-393. In: Nielsen, L.A. and D.L. Johnson (eds). Fisheries techniques. Amer. Fish. Soc., Bethesda, MD. 1-468 pp.

Exclosures/Inclosures

Cages

General Comments

- Widely used in order to exclude or include various organisms/effects
- Cages will foul rapidly and must be cleaned regularly
- Caging experiments must be carefully designed so as to minimize or eliminate changes due to the cage itself; consult the following:
  - **A.** Hulberg, L.W. & J.S. Oliver. 1980. Caging manipulations in marine soft-bottom communities: importance of animal interactions or sedimentary habitat modifications. Can. J. Fish. Aquat. Sci. 37:1130-1139.
  - **B.** Schmidt, G.H. & G.F. Warner. 1984. Effects of caging on the development of a sessile epifaunal community. Mar. Ecol. Prog. Ser. 15:251-263.
  - **C.** Stocker, L.J. 1986. Artifactual effects of caging on the recruitment and survivorship of a subtidal colonial invertebrate. Mar. Ecol. Prog. Ser. 34:305-307.
  - **D.** Virnstein, R.W. 1978. Predator caging experiments in soft sediments: caution advised. pp. 261-273. In: M.L. Wiley (ed). Estuarine interactions. Academic Press, New York.

Quick-release Cages

- For an effective quick-release cage design, see: Thayer, C.W. 1985. Quick-release cages and repetitive censusing of sessile epifauna. J. Exp. Mar. Biol. Ecol. 94:251-257.

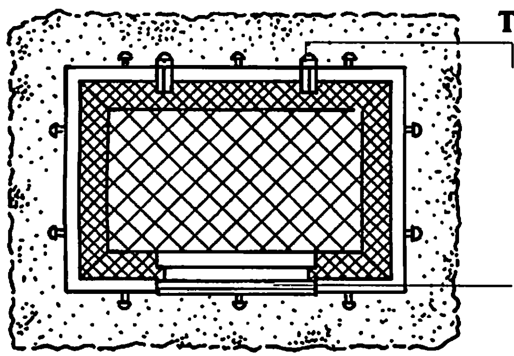
Caging Materials

- Vexar (Made byDuPont)
  - **A.** Widely used plastic mesh of various sizes
  - **B.** Stretched over frames made of PVC pipes (Schedule 40) and attached with cable ties
  - **C.** Small sizes and gauges are available in local garden shops, larger and heavier material must be ordered
- Hardware cloth
  - **A.** Stainless steel is best, held to substrata by stainless steel screws with same alloy number
  - **B.** Galvanized will rust
- Chicken wire/stucco wire: avoid as solder induces electrolysis, resulting in a pile of wire pieces

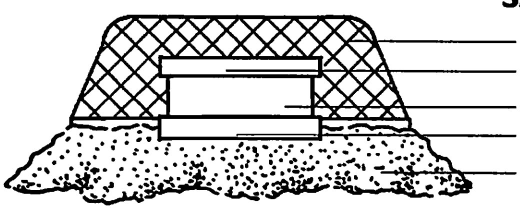
Attachment of Vexar Cages to Rock Surfaces
(Sebens)

- Uses aluminum angle frame embedded in quick-setting underwater concrete or epoxy
- Top cage frame has velcro hinge and "mates" with bottom frame; secured by cable ties (see figure on page 46)

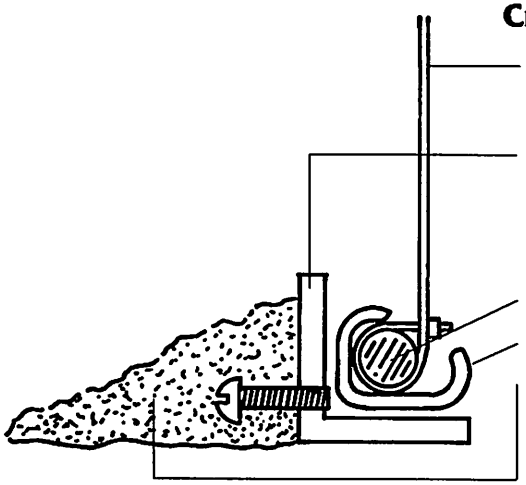
Topview



Sideview (showing hinge)



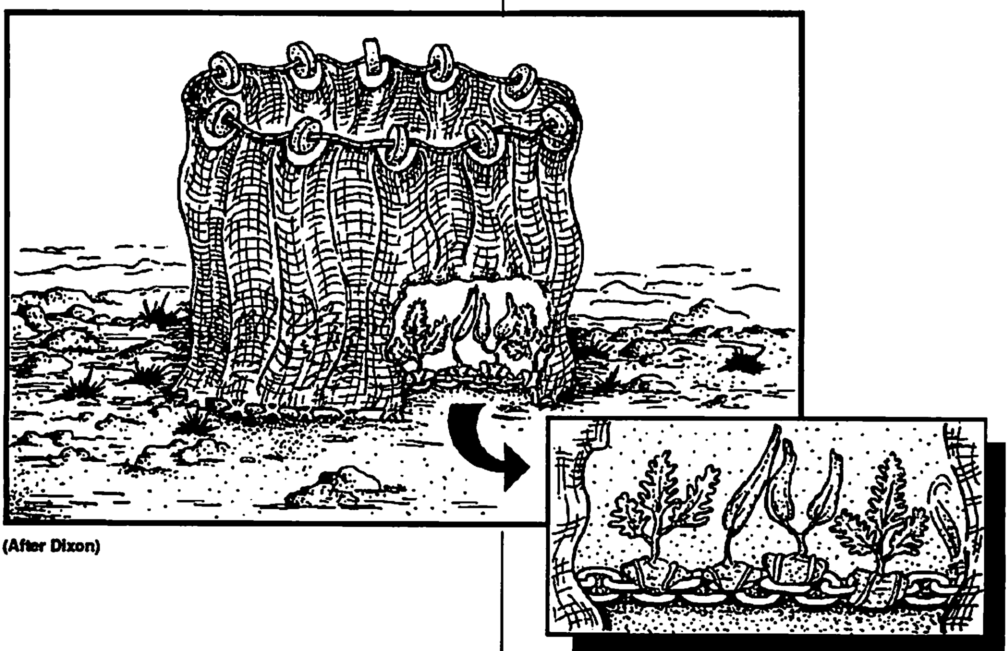
Crossview



Large Cages (>1 m)

(Schroeter and Dixon)

- Cut length of chain equal to desired circumference of cage
- Secure chain to bottom of fishing net (approximately 5 mm mesh)
- Cork-line of net elevates cage
- Secure chain to substratum with T-shaped spikes of re-bar or eyebolts and marine cement
- As netting is flexible, cage will withstand severe surge, although may need to "purse" cage opening in severe surge, as net can be forced onto substratum and "collect" organisms
- Use largest floats practical
  - A. To counter the effects of fouling (which will weigh down cage)
  - B. To minimize "bending" due to surge/current and subsequent snagging on substratum



(After Dixon)

"Cage-less" Designs

Astroturf

- When placed around an area, will effectively keep snails in or out
- Fouls fast, thus requires regular cleaning

Tangle-Foot

(Witman)

- A sticky chemical originally designed as an insect trap
- Will prevent chitons and limpets from entering or leaving an area
- The Tangle-Foot Company, Grand Rapids, MI 49504

Anti-fouling Paint/Copper Plates

- Excludes limpets in the intertidal, may also work in the subtidal
- See: Foster, M.S., and W.P. Sousa. 1985. Succession. pp. 269-290. In: Littler, M.M., and D.S. Littler (eds). Handbook of phycological methods. Ecological field methods: macroalgae. Cambridge Univ. Press 617 pp.

Colonial Sea Anenomes as "Natural" Barriers

(Laur)

- Remove pieces of substrate (mussel shells, rocks) with sea anenomes attached (*Corynactis* on Pacific coast)
- Attach pieces to substrata with epoxy so as to form borders of 1-2" width
- Benthic invertebrates (snails, urchins, etc.) are effectively deterred by the anenome's cnidocytes (fish are not deterred, thus the differential effects of fish browsing can be tested)
- Laur seldom had to remove an invertebrate for a period of at least two weeks, indeed, urchins often formed a "front" at the border

Manipulation of Substrata and Organisms

Materials/Techniques

- Materials for artificial substrates; techniques for clearing natural substrates
- Artificial substrates (i.e., concrete, plexiglass, tiles, PVC) can easily be epoxied onto natural substrates (use Koppers)
- Natural substrates can be cleared with scrapers and wire brushes, pneumatic hammers, or water-soluble chemicals
- For details, see:
  - **A.** Foster and Sousa, 1985 (Exclosures/ Inclosures. pg. 45 "Cageless Designs")
  - **B.** Harriott, V.M., and D.A. Fisk. 1987. A comparison of settlement plate types for experiments on the recruitment of scleractinian corals. Mar. Ecol. Prog. Ser. 37:201-208. (found significant variability between 8 plate types)
- Another design for settling plates:
  - **A.** Use heavy black plastic, with strips of vinyl floor tiles along top and bottom
  - **B.** Hang from docks, etc.

Attachment of Sessile Organisms

- Attachment of sessile organisms to natural or artificial substrata (to manipulate spatial patterns and densities)
  - **A.** Remove organism and associated "chip" of substrate
    - Attach with epoxy (Koppers or Sea Goin')
    - Works well for:
      - a. Encrusting algae (see, Paine, R.T. 1984. Ecological determinism in the competition for space. Ecology 65:1339-1348.)
      - b. Sea anenomes (see Exclosures/ Inclosures. pg.48 "Cageless Designs")
      - c. Solitary cup corals (Ambrose, Coyer)
  - **B.** Glue small patches of velcro onto organism and substratum (Witman)
    - Works well for small bivalves and any organism with hard exoskeleton
    - Also used to examine predator response to prey

Tether Weakly Mobile Organisms

(I.e., crabs, molluscs)

- Use super glue to attach monofilament line to carapace/shell; may need to smooth part of shell with Dremel tool
- Secure free end to substratum with hooks, etc.
- Heck, K.L., and T.A. Thoman. 1981. Experiments on predator-prey interactions in vegetated aquatic habitats. J. Exp. Mar. Biol. Ecol. 53:125-134.

Outplanting Gametophytes and Transplanting Sporophytes

For detailed discussion, see:

- Foster et al 1985. Subtidal techniques, pp.189-232. In: Littler, M.M. and D.S. Littler (eds.). 1985. Ecological Field Methods: Macroalgae. Cambridge University Press. 617 pp.

- Deysher, L.E., and T.A. Dean. 1986. In situ recruitment of sporophytes of the giant kelp *Macrocystis pyrifera*: effects of physical factors. J. Exp. Mar. Biol. Ecol. 103:41-63.

Transplant Newly Recruited Algae

(Schroeter and Dixon)

- Collect various sporophytes which have settled on small cobbles (<15 cm, longest dimension)
- Use cable ties to affix cobbles to 1-2 m lengths of chain
- For identification, cable-tie numbered tags to each cobble
- Transplanting is quick and easy, plants will stay in one area in all but most severe surge
- See diagram for "Exclosures/ Inclosures. Large Cages," (pg. 47)

Transplant Small Sporophytes

(e.g., *Fucus*)

- Insert steel nail through one end of stainless steel fishing swivel; embed nail-end of swivel in marine cement, leaving other end exposed
- Wrap small piece of surgical tubing around stipe just above holdfast; secure tubing and a cotter ring with an aluminum bird band
- Attach cotter ring to exposed end of fishing swivel
- See: Van Alstyne, K.L. 1988. Herbivore grazing increases polyphenolic defences in the intertidal brown alga, *Fucus distichus*. Ecology 69:655-663.

Transplant Small Sporophytes

(e.g., *Codium*)

- Twist open strands of "soft" line (e.g., nylon)
- Insert holdfast amongst strands, allow line to recoil
- Anchor attached line to substratum

Small Artificial Reefs

(Barilotti, Dean, Schroeter, Dixon)

- Transplant adult kelps onto re-bar embedded in cement bags (See "Site Markers. General Location," pg. 13)
- Use bicycle inner tubes as rubber bands to attach plants
- Stake bags to substratum and pin bags to each other to create reef (use sharpened re-bar with T's)
- Schroeter and Dixon made a 10 x 1 x 0.5 m reef at a depth of 50 ft which is still intact after 5 yrs

Artificial Substrates Mimicking Biological Species

- Fishing line (hydroids), artificial grapes (tunicates), vial caps (dead barnacles): see Dean, T.A. 1981. Structural aspects of sessile in- vertebrates as organizing forces in an estuarine fouling community. J. Exp. Mar. Bio. Ecol. 53:163-180.

- Nylon fish net (erect bryozoans): see Russ, G.R. 1980. Effects of predation by fishes, competition, and structural complexity of the substratum on the establishment of a marine epifaunal community. J. Exp. Mar. Biol. Ecol. 42:55-69.
- PVC pipes (erect coral): see Adey, W.H., and J.M. Vassar. 1975. Colonization, succession and growth rates of tropical crustose coralline algae (Rhodophyta, Cryptonemiales). Phycologia 14:55-69.
- Plastic straws (worm tubes): see Woodin, S.A. 1978. Refuges, distrubance, and community structure: a marine soft bottom example. Ecology 59:274-284.
- Artificial seagrass or algae
  - **A.** Use "Olefern", a fibrous polypropylene strip
  - **B.** See:
    - Bell, J.D., A.S. Steffe, and M. Westoby. 1985. Artificial seagrass: how useful is it for field experiments on fish and macroinvertebrates? J. Exp. Mar. Biol. Ecol. 90:171-177.
    - Harlin, M.M. 1973. "Obligate" algal epiphyte: *Smithora naiadum* grown on a synthetic substrate. J.Phycol. 9:230-232.
  - **C.** Plastic strips (4 mil) also work well, can last 3+ years in field
- Artificial kelp: Schroeter, Dixon, and Kastendieck have used custom-made plastic models of giant kelp

Making Models of Fish

- Helfman, G.F. 1983. Resin-coated fishes: a simple model technique for in situ studies of fish behavior. Copeia. 1983. (2):547-549.
- Hicks, D. 1966. Reproduction of biological specimens in neoprene latex. Museum News, Technical Supplement. 15:45-50.

Manipulations of Abiotic Factors in the Field

Light

- Shade benthic algae with layers of Vexar or equivalent stretched on frames and suspended so as to not restrict algal movement (figure 1)
  - **A.** Denley, E.J., and P.K. Dayton. 1985. Competition among macroalgae. pp. 512-530. In: Littler, M.M., and D.S. Littler (eds). Handbook of phycological methods. Ecological field methods: macroalgae. Cambridge Univ. Press, 617 pp.
  - **B.** Shades need to be cleaned regularly
- Increase light by:
  - **A.** Removing algal canopy
  - **B.** Transplant to a shallower depth
  - **C.** Suspend at various levels in the water column, using PVC trays (figure 2)
  - **D.** See: Foster et al,1985., Subtidal Techniques, pp. 189-232. Ibid.

Nutrients

- Use fertilizer in porous containers
- Use Osmocote fertilizer in nylon-mesh covered trays (pellets covered with release-controlling polymer film)
  - A. Be aware that PO_4 in Osmocote pellets is insoluable in seawater, thus will get different NO_3/PO_4 ratios (Zimmerman)
- See Foster et al, 1985. Ibid.

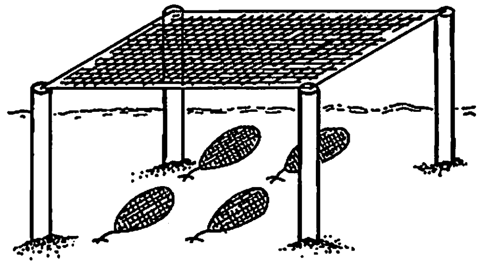


figure 1

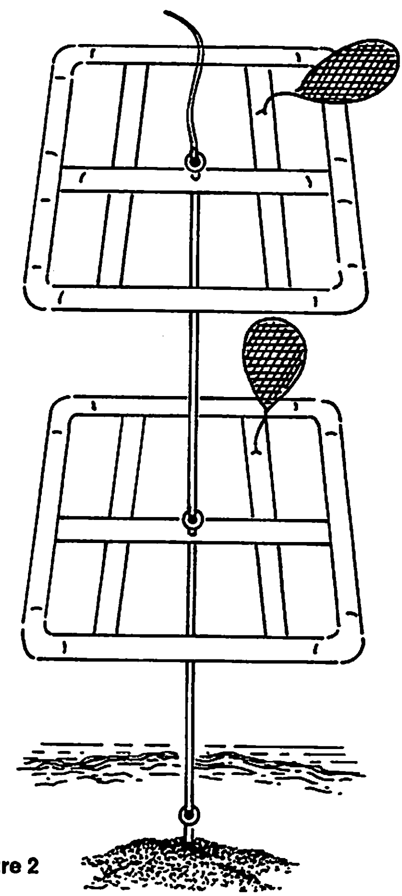


figure 2

Light Measurement

Resources

- For an excellent review of physics and measurement of light in the subtidal, see: Ramus, J. 1985. Light. pp. 33-52. In: Littler, M.M., and D.S. Littler (eds). Handbook of phycological methods. Ecological field methods: macroalgae. Cambridge Univ. Press, 617 pp.

Radiometry

- Measures amount of radiant energy flux (total irradiance) over a desired time interval
- Generally, need a hemispherical collector for benthic algae; spherical for phytoplankton
- Popular instruments:
  - **A.** Integrating Quantum Scalar Irradiance Meter (Biospherical Instruments, Inc., 4901 Morema Blvd., #1003, San Diego, CA 92117)
  - **B.** Li-Cor Model LI-185A Quantum Meter with diver-held sensor on a 50 m cable (LI-COR, Inc. 4421 Superior St., Lincoln, NB 68504)
  - **C.** Photographic light meters in housings: good for quick-and-dirty, relative light readings

Spectral Distribution

- Measures spectral irradiance or specific wavelengths using spectro-radiometer
- Until recently, most instruments were custom-made; popular "mass-produced" instruments now available, but are expensive ($15,000-$25,000)
  - **A.** Biospherical Instruments MER 1000
  - **B.** Li-Cor LI-1800 UW

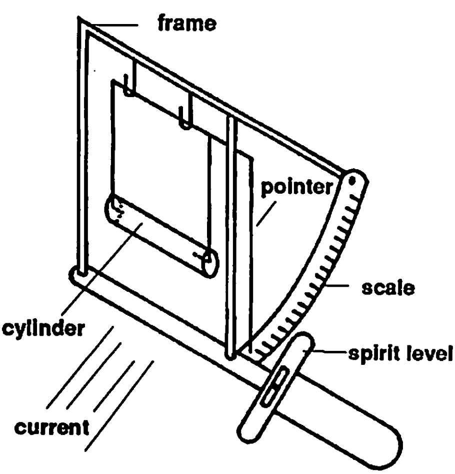
Water Motion Measurement

General References

- For an excellent review of the physics of water motion, as well as pros and cons of various measuring devices (primarily intertidal), see:
  - **A.** Denny, M.W. 1988. Biology and the mechanics of the wave-swept environment. Princeton Univ. Press. 329 pp. (See Chap 19)
  - **B.** Denny, M.W. 1985. Water motion. pp. 7-32. In: Littler, M.M., and D.S. Litter (eds). Handbook of phycological methods. Ecological field methods: macroalgae. Cambridge Univ. Press, 617 pp.
- For a supporting discussion of subtidal measurements, see: Foster et al, 1985. Subtidal Techniques. Ibid.

Current Direction and Velocity, Subtidal

- Simply time drift rate of plankton or bits of algae across a known distance



- Video-tape particle paths
  - **A.** Set camera on macro setting
  - **B.** Use side illumination
  - **C.** Measure velocity on VCR monitor
    - Determine number of frames required to traverse known distance
    - One frame usually = 1/30 sec
- Home-made current meter (Warner)
  - **A.** Suspend a solid PVC cylinder from a wire frame so that cylinder is free to swing with the current
  - **B.** A pointer attached to the cylinder moves over a scale attached to the frame
  - **C.** Frame is attached to handle on which a spirit level is mounted to ensure a "0" reading
  - **D.** Calibrate with a flume or equivalent; response is reasonably linear up to 25 cm/sec
  - **E.** For qualitative data only
- Home-made flow probes
  - **A.** Construct thermistor flow probe according to design in: LaBarbera, M., and S. Vogel. 1976. An inexpensive thermistor flowmeter for aquatic biology. Limnol. Oceanogr. 21:750-756.
  - **B.** Calibrate flowmeter in flowtank before use; place in Ikelite housing
  - **C.** Used by Patterson and Sebens; contact for latest improvements
- Electromagnetic meters or mechanical devices with propellers; see Denny 1988 and Foster et al, 1985, for specifics on types and vendors; consider also:
  - **A.** General Oceanics Digital Flowmeter
    - Large rotor for low velocities
    - General Oceanics Inc., 1295 N.W. 163rd St., Miami, FL 33169 (details in Data Sheet #101-678)

B. Interocean Current Meter (Model S4)

- An excellent, programmable electronic meter, but expensive
- Interocean Systems, Inc., 3540 Aero Court, San Diego, CA 92123-1799

C. Marsh-McBurney

- Electromagnetic device, with deck-side meter and submersible probe
- Popular, cost approx. $5,000
- 8595 Grovemont Circle, Gaithersburg, MD 20760, (301) 869-4700

Total Water Motion, Subtidal

- Measure how fast a substance dissolves
- High levels of suspended solids will confound results, as will measure both motion and scour
- Expect high variability
- Types

A. Plaster-of-Paris "clog-cards"

- Make experimental and control blobs in a plastic ice tray, allow to set for 4 wks, dry in oven to insure that both are same weight and shape (trim from bottom)
- Glue to plastic cards, place in field and lab aquarium (=control) with same field temperature, but devoid of any water motion
- After period of time, collect, dry in oven, then weigh experimental and control
- See:

a. Doty, J.E., and M.S. Doty. 1973. Abrasion in the measurement of water motion with the clod-card technique. Bull. Southern California Acad. Sci. 72:40-41.

b. Doty, M.S. 1971. Measurement of water movement in reference to benthic algal growth. Botanica Marina 14: 32-35.

c. Muus, B.J. 1968. A field method for measuring "exposure" by means of plaster balls. Sarsia 34:61-68.

C. Candy (Denny)

- For measurements on a 1-3 min scale
- Use Lifesavers or Velamints
- Use experimental and control as above

Maximum Force Measurement

- Maximum force dynamometer measures hydrodynamic shear forces striking a test object
- Developed by Denny for intertidal, modified by Witman for shallow (12 m) subtidal
- Test object bolted to a "slider" device which moves in direction of force and to a distance proportional to force
- Deflection recorded on smoked glass slide in the intertidal design
- Deflection in subtidal design:

A. Gold-plated bathythermograph slide

B. Slide inked with magic marker (lasts 3 days)

- In subtidal, slider may become immobile after 3 weeks, as mechanism is clogged with sediment
- See:

A. Denny, M.W. 1983. A simple device for recording the maximum force exerted on intertidal organisms. Limnol. Oceangr. 28:1269-1274.

B. Witman, J.D. 1987. (see: Quadrats and Transects. Quadrats, Permanent Quadrats, pg.18)

continued on page 56

Sedimentation Measurement

- Measure amount of sediment that accumulates in sediment tubes or traps
- Use a minimum height:diameter ratio of 2:3; see:

A. Gardner, W. 1980a. Sediment trap dynamics and calibration: a laboratory evaluation. J. Mar. Res. 38:17-39.

B. Gardner, W. 1980b. Field assessment of sediment traps. J. Mar. Res. 38:41-52.

C. Butman, C.A. 1986. Sediment trap biases in turbulent flow: results from a laboratory flume study. J. Mar. Res. 44:645-693.

D. Butman, C.A., W.D. Grant and K.D. Stolzenbach. 1986. Predictions of sediment trap biases in turbulent flows: a theoretical analysis based on observations from the literature. J. Mar. Res. 44: 601-644.

E. Gulliksen, B. 1982. Sedimentation close to a near vertical rocky wall in Balsfjorden, northern Norway. Sarsia. 67: 21-27.

Maximum Force Measurement

continued from page 55

Surge

- A float, line, and protractor can be used to get a relative estimate
- Hold protractor along bottom, parallel to surge, read angle of deflection, calculate values
- See:

A. Foster et al. 1985. Subtidal Techniques, pp. 189-232. In: Littler, M.M. and D.S. Littler (eds). 1985. Ecological field methods: Macroalgae. Cambridge University Press. 617 pp.

Special Applications

Blue-Water Diving

- Heine, J.N. (ed). 1986. Blue water diving guidelines. Cal. Sea Grant Coll. Pub. No. T-CSGCP-014. (Available from Calif. Sea Grant College Program, Univ. of Calif., A-032, La Jolla, CA 92093)

Diving in Polluted Water

- Colwell, R.R. (ed). 1982. Microbial hazards of diving in polluted waters. Maryland Sea Grant Pub. No. UM-SG-TS-82-01. (Available from Sea Grant Program, Univ. of Maryland, 1224 H.J. Patterson Hall, College Park, MD 20742)

Diving Sled Design

(for rapid reconnaissance and mapping)

- Sigl, W., V. VonRad, H.J. Oeltzschner, K. Braune, and F. Fabricius. 1969. Diving sled: a tool to increase the efficiency of underwater mapping by scuba divers. Mar. Geol. 7:357-363.

Dive Tables and Theory

(USN, Huggins, DCIEM, Buhlmann (Swiss), RNPL (British))

- Huggins, K. 1987. Microprocessor applications to multi-level air decompression problems. Mich. Sea Grant College Prog. MICHU-SG-87-201 (available from Mich. Sea Grant Publications, 2200 Bonisteel Blvd., Ann Arbor, MI 48109; $5)

Nitrox Diving

- Hamilton, R.W., D.J. Crosson, and A.W. Hulbert (eds.) 1989. Workshop on enriched air nitrox diving. National Undersea Research Program Research Report 89-1. 153 pp. (Available from NURP-NOAA 6010 Executive Blvd., Rm. 805, Rockville, MD 20852)

AAUS Publications

- Many topics covered in special sessions of annual meetings
  - A. Special Session on Coldwater Diving Proceedings. 1989. M.A. Lang and C.T. Mitchell (eds.)
  - B. AAUS Dive Computer Workshop Proceedings. 1989. M.A. Lang and R.W. Hamilton (eds.)
  - C. Biomechanics of Safe Ascents Workshop. 1990. Proceedings. M.A. Lang and G.H. Egstrom (eds.)
- Available from:

  American Academy of Underwater Sciences, 947 Newhall St., Costa Mesa, CA 92627

Customizing Scuba Gear

Surgical Tubing

- Surgical tubing: use in place of straps (i.e., knife straps, ankle weights) for quick attachment/ removal

Wide Rubber Straps

- When used as shoulder straps on backpacks, will eliminate chafing on wet suit (which happens when nylon webbing becomes stiff)
- Can also be used as weight belts

Gloves

- Gloves: tight-fitting, heavy-duty rubber gloves normally used for dish-washing or cleaning (Playtex, Bluettes) provide warmth, protection, and excellent dexterity

Attachment for Gear

- Insert "loop" made of brass rod into 7-10 lb hip weights
- Can easily clip-off catch bags, slates, etc.

Wet Suit

Getting into a "skin-inside" wet suit... comfortably

- Place a small amount of baby shampoo into a plastic detergent bottle and fill with water
- Squirt diluted shampoo into arms and legs of wet suit, then rub together to form sudsy film
- Slip (literally) into wet suit

Dry Suit

- Getting in and out of a dry suit with long hair: put nylon panty hose over head, then pull neck seal on or off

Weight Belts/weights

- "No stress" weight belt (Carroll)
  - A. Attach existing weight belt to a wide and thick piece of rubber mat; increasing area will relieve lower back pain
  - B. Drill holes into weights and attach to rubber mat with brass or stainless steel through-bolts
- Sew small pockets onto wetsuit, insert small weights
- Ankle weights
  - A. Purchase ankle weights (variety available) or attach large fishing weights to surgical tubing, slip over ankle
  - B. Will get some of weight off back, also good for reducing movement of air to boot portion of a dry suit
    - Widely used
    - Not heavy enough to sink you once primary belt has been ditched

Knee Pads

- Cut portion of legs from an old wet suit
- Cut portion of automobile tire inner-tube

Pockets

- Pockets on outer thigh of wet suit: pleated and with drain hole, can carry lots of things which get lost or entangled in a standard net bag

Hot Packs for Divers

- Super-saturated solution of sodium acetate, induce precipitation and subsequent exothermic reaction by mechanical shock
- Place pak next to stomach, releases heat for 1 hr
- Can get quite warm, so place pack between vest or "T" shirt and wet suit jacket
- To re-use, simply boil pack to dissolve precipitate
- For catalog, contact: Prism Technologies, 2560 North Elston, Chicago, IL 60647

Useful Catalogs for Supplies

Resources

- Forestry Suppliers, Inc., 205 W. Rankin St., Box 8397, Jackson, MS 39204-0397, (800) 647-5368 (call or write for free catalog)
- Chicopee, 70 Clarkesville Hwy., Box 70, Cornelia, GA 30531 (carries screening of various types)
- Nichols Net & Twine Co., Inc., RR 3, Bend Road, East St. Louis, IL 62201
- National Brand and Tag Co., Box 430, Newport, KY 41072 (for #'ed brass tags of all sizes)
- M & E Marine Supply, Box 601, Camden, NJ 08101 (commercial diving supply catalog)
- "Guide to Scientific Instruments" (published yearly as a supplement to *Science*)
- "Buyers Guide/Directory" (published yearly by Sea Technology)
- Sterling Net and Twine Co., Inc., 18 Label St., Montclair, NJ 07042 (for Vexar ™)

Appendix I

Quadrapod Design

(Witman, Lavoie)

General

- Plans are for 0.5 m² quadrapod
- Modify plans for 0.25 m² quadrapod (see pp. 66-67)

Materials

- Type of aluminum to order:
  - A. 1 x 1 x 1/8" angle
  - B. 5/8 x 5/8 x 1/8" angle
  - C. 1.5 x 0.5 x 1/8" channel
- Quadrapod is constructed of 1 x 1 x 1/8" angle with lengths according to plans. Order a lot. The camera bar and the cross pieces that support the camera bar are made from channel stock; supports for the data panel from 5/8" angle

Quadrapod Base

- Build the quadrapod base by cutting the 1" angle to make a rectangle of 58.31 x 85.75 cm (inner edge dimensions)
- Cut a piece of inch angle ("a" on diagram) so that it defines the sides of the 0.5 m² quadrat (85.75 cm) and supports the data panel 10 cm in from the end of the quadrat
- Fasten everything with stainless machine bolts (weld the frame only after you have shot a test roll)
- Attach 5/8" angle to the inner side of "a" so that it forms a shelf "b" to bolt on the data panel. Repeat on other side of data panel.

Quadrapod Top

- Build the quadrapod top. The plans are designed so that the side legs come straight up from the quadrat base. You could design it so that the legs angle in towards the center (as in the 0.25 m² quadrapod) which would decrease the size of the rectangle forming the quadrapod top. However, the 0.5 m² quadrapod will be stronger and easier to construct if the legs come straight up.
  - A. Cut inch angle to create a rectangle with a length equal to the length of the base (95.75 cm) and a width of 25 cm
  - B. Cut two channel cross-pieces to form a rectangle 26 cm wide around the center of the top frame
  - C. Bolt the cross-pieces in place with the flat side up, giving the camera bar a large flat surface to slide back-and-forth on when adjusting the camera position
  - D. Return to details for camera bar attachment once the basic frame has been constructed, but first, mill a groove at least 10 cm long in the center of each channel cross-piece. Width of groove depends on the size bolt used for bar attachment (1/4" stainless in original design)

Attach Top to Base

- The hardest part of construction is the attachment of the legs to the top and base. The camera has to be suspended and centered 58 cm above the bottom quadrat. At this height the 15 mm lens should cover the 58 x 86 cm rectangle with a few cms of spare room around the sides. The frame is designed so that the 0.5 m^2 area is enclosed in a rectangle in the same proportions as the format of a 35 mm negative (where length = 1.47 x width). This enables the camera-to-subject distance (58 cm) to be minimized, resulting in better detail in the photos.

A. Cut two pieces of inch angle 58 cm long and attach them perpendicular to the quadrat base at a point 14 cm in from the end of the base "c"

B. With C-clamps and vertical supports, clamp the top onto the 58 cm verticals. Adjust so that the center of the top rectangle is directly over the center of the quadrapod base (an important step, as the lens must be centered)

C. Cut inch angle approximately 61 cm long to form the other side-support "d," but measure it in place; bottom angle will be 72°

D. Bolt legs to form frame

Camera bar and attachment

- Machine away one wall of the 1.5 inch channel so that the base plate of the Nikonos fits in press-tight
- Do this so that the camera is centered in the middle of the channel length (at least 70 cm long, but depends on how you attach the strobe mounts to the channel; strobes should angle at approximately 45°)
- Drill a hole for connector (2 cm wide)
- Machine the channel down so that the channel thickness is 0.075 inch (important, as it determines the contact of the connector to the camera)
- Cut two pieces of inch angle 1.5" wide so that the camera bar can be attached to the quadrapod top
- Bolt-diameter should correspond to the diameter of the milled groove in the channel so that the camera position can be adjusted
- Place camera on the bar (held in place by the strobe connector and the walls of the channel) and slide it into position so that the center of the 15 mm lens is perpendicular to the center of the 58 x 85 cm quadrat base

Area Covered by Nikonos 15mm Lens at Different Camera-to-Subject Distances (Hulbert)

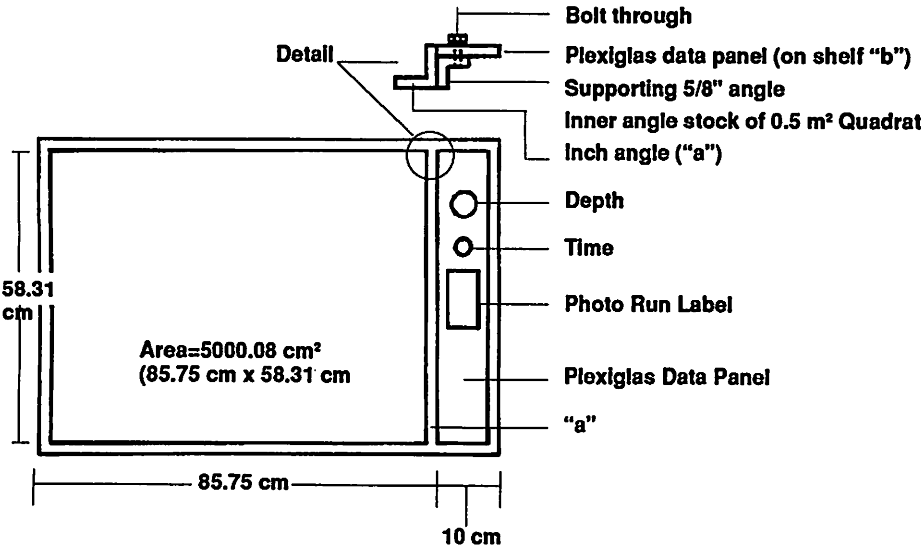
A. Data were obtained by photographing the bottom of a pool at a series of camera-to-subject distances

B. Data provide a useful first-approximation for designing new camera framers for the 15 mm lens

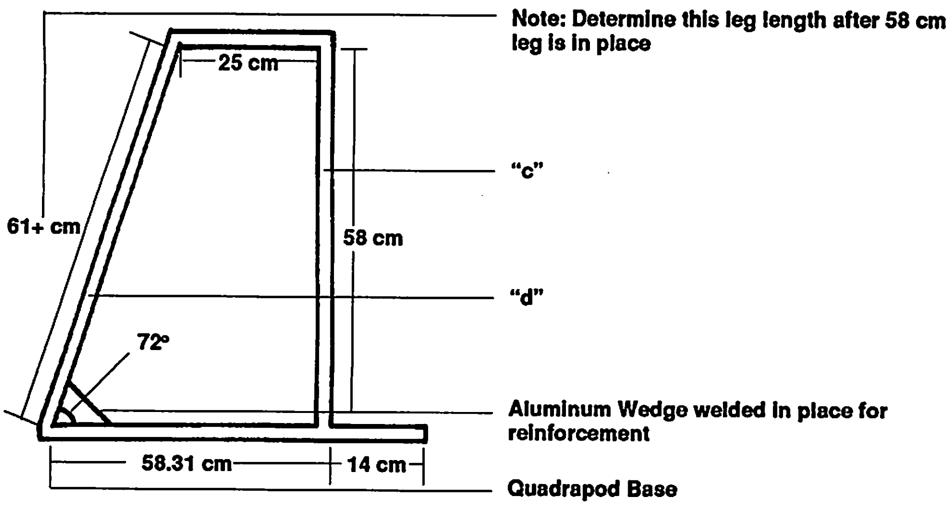
Camera-Subject Distance (cm)	Side A (cm)	Side B (cm)	Area (cm^2)	Proportion of meter sq
10.0	20.0	13.6	272.0	0.0272
20.0	40.0	27.2	1088.0	0.1088
25.0	50.0	34.0	1700.0	0.1700
30.0	60.0	40.8	2448.0	0.2448
32.5	65.0	44.2	2873.0	0.2873
36.0	70.0	47.6	3332.0	0.3332
40.0	80.0	54.4	4352.0	0.4352
45.0	90.0	61.2	5508.0	0.5508
50.0	100.0	68.0	6800.0	0.6800
55.0	110.0	74.8	8228.0	0.8228
60.0	120.0	81.6	9792.0	0.9792
62.5	125.0	85.0	10625.0	1.0625
65.0	130.0	88.4	11492.0	1.1492
70.0	140.0	95.2	13328.0	1.3328
80.0	160.0	108.8	17408.0	1.7408
90.0	180.0	122.4	22032.0	2.2032
100.0	200.0	136.0	27200.0	2.7200
110.0	220.0	149.6	32912.0	3.2912
120.0	240.0	163.2	39168.0	3.9168

0.5 m² Quadrapod

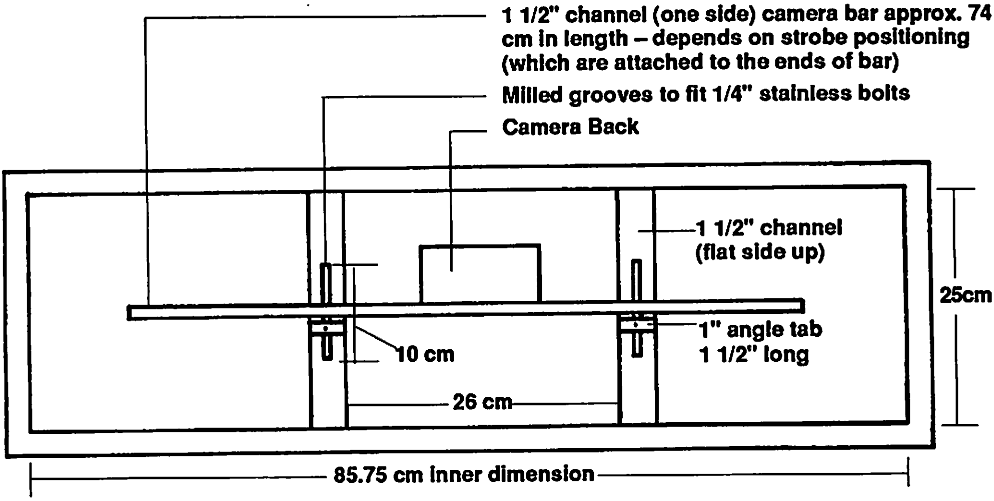
0.5 m² Quadrapod Base



Sideview of 0.5 m² Quadrapod

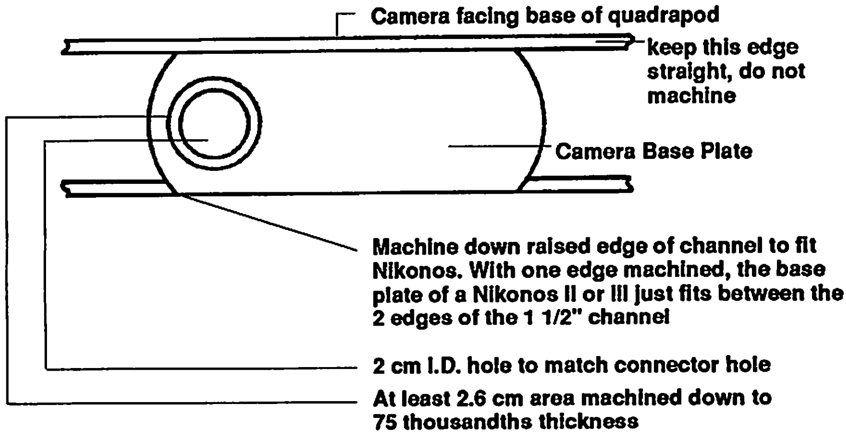


0.5 m² Quadrapod Top

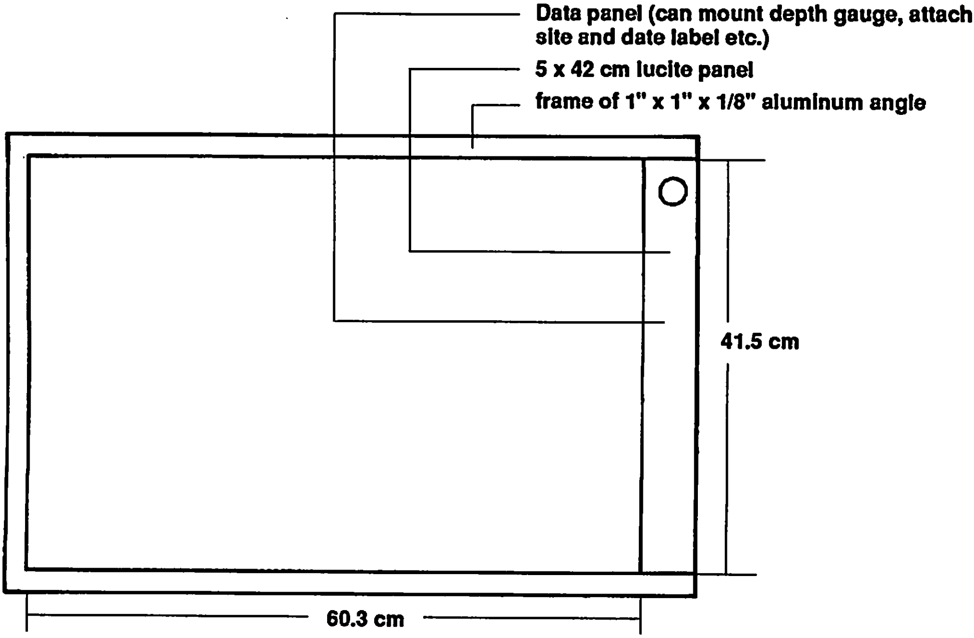


Camera Bar and Attachment

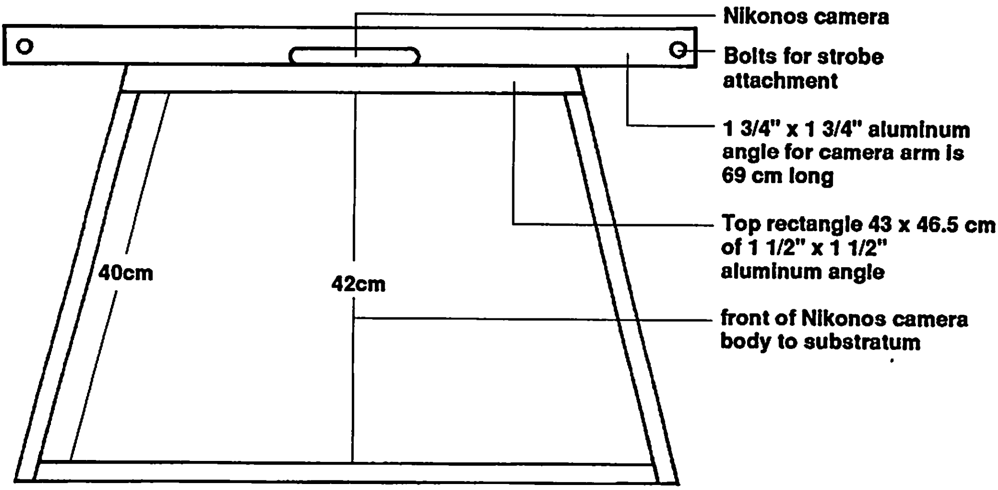
(Detail of site of camera attachment on 1 1/2" channel bar)

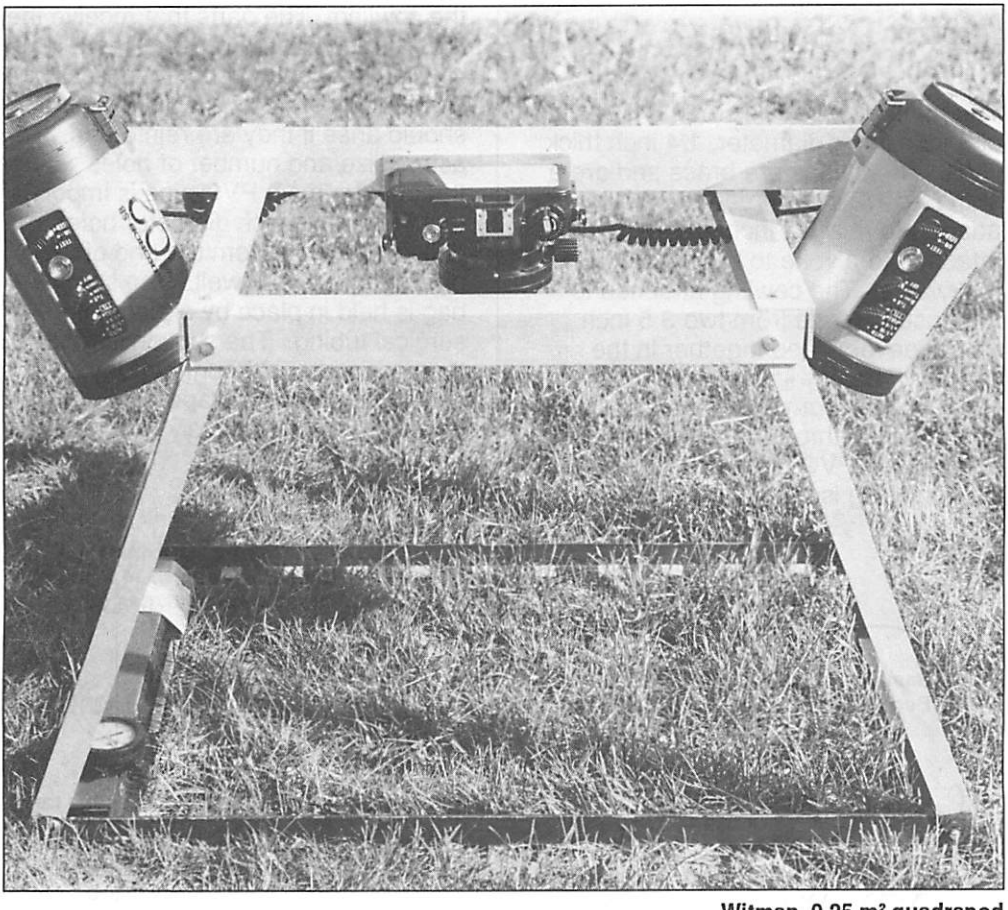


0.25 m² Quadrapod Base



Sideview of 0.25 m² Quadrapod





Witman, 0.25 m² quadrapod

Appendix II

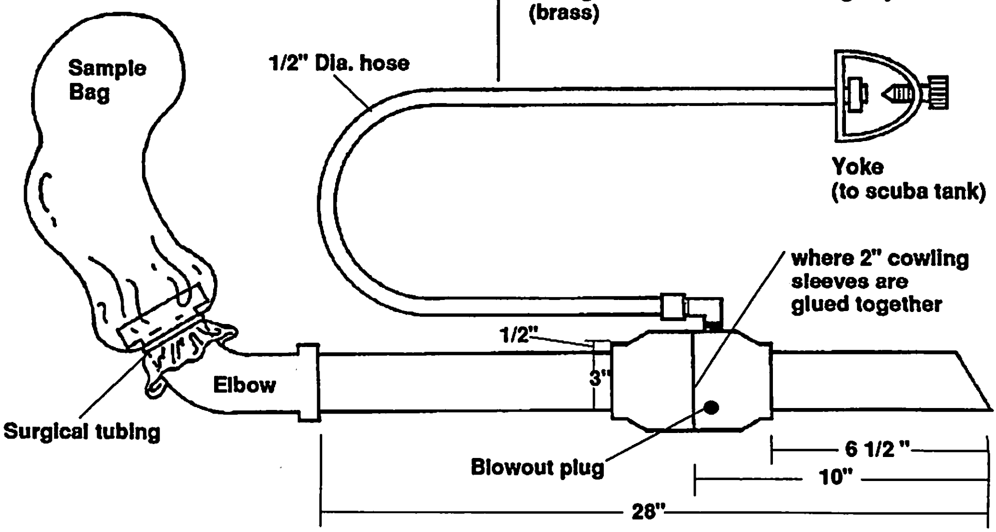
Airlift Design I

(Witman, after Harris)

The main pipe consists of a 3 ft section of 3 inch inner diameter, 1/4 inch thick PVC. The fittings are brass and are a standard 1/2 inch threaded elbow and adaptor to fit a 1/2 inch rubber hose attached to a yoke to fit on a SCUBA tank valve. The cowling attached to the hose is made from two 3.5 inch PVC sleeves glued together in the middle. The cowling creates an air space for the compressed air to enter and circulate through the 1/8 inch holes in the PVC pipe wall. A plastic blowout-plug is glued into the side of the cowling. The parts that receive the most stress are the brass elbow/ cowling connection and the rubber hose/yoke connection. No problem should arise if they are reinforced. The actual size and number of holes through the main PVC pipe is important. Eight 1/8 inch diameter holes drilled 10 inches from the end of the pipe seem to work well. The sample bag is held in place by a piece of surgical tubing. The suction is more than adequate, although a putty knife should be used to scrape the substratum while simultaneously vacuuming the area with the airlift.

Airlift:

Overall length: 36"
Pipe: 3" I Dia. PVC (3 1/2" OD) 1/4" thick
Hose: >1/2" Dia. rubber approx. 4 ft. long
Fittings: 1/2" brass elbow Std. gas yoke (brass)

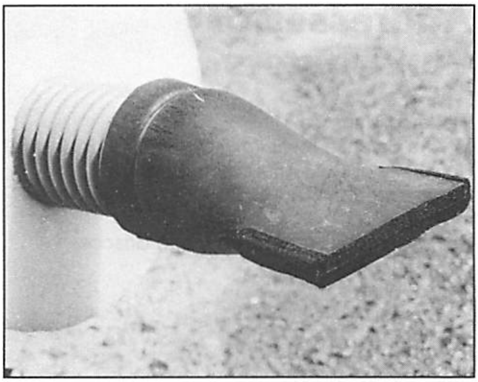


Distance to position of holes thru pipe wall: 8 evenly spaced holes 1/8" diameter, 10 inches from end of pipe

Airlift Design II

(Chess)

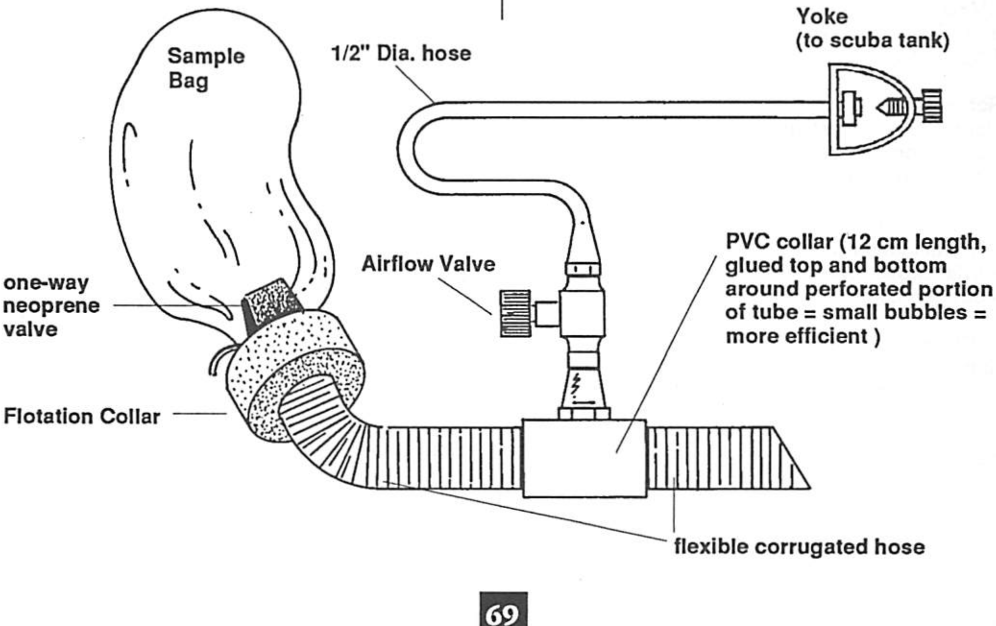
- This design differs from "Airlift Design I" (Witman) by:
  A. Airflow is controlled with a valve placed adjacent to the cowling
  B. A flexible corrugated hose (swimming pool cleaning hose) is used instead of rigid PVC (hose will require floatation collar)
  C. A one-way "valve" made of neoprene rubber prevents sample from falling down hose when airflow is stopped
  D. See: Chess, J.R., 1978. Some procedures for assessing organisms associated with rocky substrata. pp.25-28. In: Lipovsky, S., and C.A. Simenstad (eds). Gulshop 1978. Washington Sea Grant Publications, University of Washington, Seattle WA.



Close-up of "valve" inside sample bag.

Airlift:

Overall length: variable
Flotation Collar: cork
Hose: swimming pool cleaning hose, (3 cm length, 38 mm diameter)
Fittings: std. gas yoke



Appendix III

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