UNIVERSITY OF VERMONT

WATERSHED ALLIANCE

Stream Monitoring Program

Watershed Educator's Handbook
2007/2008

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*Appenidx K, MS Excel data workbooks, and the entire handbook (MS Word or pdf) is available for download at http://www.uvm.edu/~watershd

Stream Monitoring Program Description
UVM Watershed Alliance

Rubenstein Ecosystem Science Laboratory
3 College Street, Burlington, VT 05401

BETHANY HANNA Outreach and Education Coordinator
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INTRODUCTION

The University of Vermont (UVM) Watershed Alliance is a collaboration of UVM Rubenstein School for the Environment and Natural Resources (RSENR), UVM Extension, and Lake Champlain Sea Grant. The Watershed Alliance supports **statewide watershed education and water quality monitoring** in Vermont middle schools, high schools, alternative education programs, and youth groups.

The primary objective of the UVM Watershed Alliance (UVM WA) is to increase awareness and knowledge of watershed issues in Vermont youth. The Watershed Alliance provides curricula, equipment, and **Watershed Educators** to schools and youth groups participating in our program, as well as support and guidance to teachers in designing programs in which children learn about watersheds and related water issues.

Watershed Educators (WEs) are trained UVM undergraduate student interns studying and interested in water resources and environmental education. WEs conduct workshops in schools and other academic settings throughout Vermont. The workshops feature a **classroom component**, where we introduce watershed concepts to participants through our interactive watershed model, as well as a **field component**, where we take to the field for hands-on water quality monitoring exercises.

"Taking Action" is the final element of the Watershed Alliance program. Taking Action is student-led **community outreach component**. Students are excited to share their valuable watershed knowledge with the rest of their community. In the past, local newspapers including the Times Argus, the Rutland Herald, and the Burlington Free Press have featured classes involved in our program. Annually, students attend an Environmental Congress at UVM, where they have an opportunity to share their findings with other students from throughout the state. Other participants present their findings to their local planning commissions, school boards, watershed groups, parents, and to over 10,000 Vermonters via a television program. Students have even produced their own series of public service announcements.

"It's meaningful for the kids to feel a part of something larger than the school, and to know that their data will go somewhere."
-Brian Slopey
U-32 High School Science teacher

Schools

Students from across the State of Vermont have participated in Watershed Alliance programs. Please see Appendix A. for a comprehensive list of schools.

Watershed Educators

Each semester, UVM WA employs between five and eight students as interns. In fall 2003, six students from RSENR and one student from the College of Arts and Sciences worked as interns with UVM WA. All interns complete ten hours of training, including watershed model presentation, river study theory and design, benthic macroinvertebrate sampling and identification methodology, and chemical, physical and biological sampling methodology.

Students that serve as interns are generally sophomores, juniors or seniors who are interested in water resources and environmental education. Interns receive one credit (50hr commitment) upon completion of the program, which includes submitting a project portfolio for review.

In the past, several students have returned to the UVM WA for subsequent internships, and have refined their skills as educators, or completed activity/curriculum development projects. We have received much positive feedback from interns, who are excited to move into the larger Vermont community as service learners.

PROGRAM ELEMENTS

Teacher Consultation

Approximate time commitment: 1 hr phone/in-person meeting, planning time (varies)

- Teacher fills out and returns UVM WA application
- UVM WA Outreach and Education Coordinator (OEC) reviews program and watershed study; consultation on study design, available resources and integration into curriculum. OEC provides training in use of Vermont Monitors Database
- Teacher receives pro-bono copies of Testing the Waters, Living Waters, Healthy Water Healthy People (HWHP), and UVM WA curriculum.
- Watershed Educator contacts teacher to review logistics and specific needs

Classroom Component

Approximate time commitment: 1-1 ½ hrs model presentation, 1 hr study design and equipment introduction

**Students complete Pre-monitoring questionnaire (appendix B, pg 10) prior to watershed model presentation.

- Watershed Model: Two Watershed Educators and/or the Outreach and Education Coordinator give watershed model presentation (see appendix C, pg 15). Key concepts

introduced include definition of watershed, types and sources of pollution, effects of pollution on humans and ecosystem, difference between non-point and point source pollution, and introduction to best management practices (see appendix D, pg 19)

- Study Design and Equipment Demo: WEs introduce monitoring concepts (see Testing the Waters pp.1-4) and background information about chemical, physical, and biological parameters (see HWHP pp 5-19, 30-44, 49-52). WEs guide participants in formulating a monitoring question and design a river study to address the question (appendix E, pg 22) WEs introduce equipment (appendix F, pg 25) and participants practice with each piece/method. See also field data collection sheet (see Testing the Waters pp 197-201 and appendix F2, pg 26).

Field Study Component

Approximate time needed: 3-6+ hrs field study, 3+ hrs data compilation, analysis and interpretation

- Watershed Educators and classroom teacher(s) lead students on river study field trip. Typically, students visit 1-3 sites and spend at least two hrs at each site collecting samples and recording data. If using BMI streamside survey method, everything is done in the field (appendix G, pg 28). If using the intensive BMI analysis, samples are preserved and brought back to school lab for identification and quantification (appendix H, pg 35). E.coli samples brought to classroom lab for processing and incubation. Participants read E.coli results 24-28 hrs after processing (see Appendix I, pg 60 for Quanti-Tray and Coliscan methodologies)

- Classroom teacher assists students in data analysis and interpretation (see Testing the Waters pp169-184 and HWHP Testing Kit Manual appendix A).

- **Students complete Post-Monitoring questionnaire (appendix B, pg 12)

Community Outreach Component

Approximate time commitment: variable

- Students prepare community outreach project (see Testing the Waters pp.185-196). Past projects include presentations to local watershed organizations, production of a radio spot, presentations to peers and/or parents, participation in a congress with other student monitors, and a student led teaching/learning day with younger students. See appendix J (pg 65) for a list of project ideas and other useful information about community outreach. See appendix K. for list of watershed organizations.

Optional Components

Approximate time commitment: variable. Contact Bethany Hanna for more info.

- Benthic Macroinvertebrate Sampling Methodology: this short DVD produced by River Network is available for loan. Your students will learn appropriate sampling methodology, demonstrated by experts.

- Rubenstein Ecosystem Science Laboratory: verify field data at this state-of-the-art lab with professional scientists. Students will learn to use EPA methodology to analyze total phosphorus and E.coli. This visit can be combined with a trip through ECHO at the Leahy Center for Lake Champlain (right next-door!) for a full day of academics and fun at the Lake.

- Why Macroinvertebrates?: This PowerPoint presentation reviews benthic macroinverbrate ecology, biology, and functionality as an indicator of ecosystem health.

- Wetland Slide Show: created in conjunction with Matthew Witten of the New England Interstate Water Pollution Control Commission and the VT Department of Environmental Conservation, this slide show takes a tour through wetlands in Vermont while introducing wetland functions and values.

- Lake Champlain Basin Program (LCBP) Atlas activity: The Lake Champlain Basin Program Atlas is a collection of maps and information specific to the Basin. Explore this valuable educational resource with the LCBP Atlas Treasure Hunt (see appendix L, pg 67). The LCBP Atlas is available for purchase from the LCBP, and is available for use online at www.lcbp.org.

Stream Monitoring Program Learning Objectives

UVM Watershed Alliance

Rubenstein Ecosystem Science Laboratory
3 College Street, Burlington, VT 05401

BETHANY HANNA Outreach and Education Coordinator
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Participants in the Watershed Alliance education program will meet the following learning objectives:

Classroom Component

1. Accurately define the term "watershed" (watershed: an area of land that drains into a water body)
2. Know the names of the basin and sub-basin watersheds in which one lives (ie. Otter Creek sub-basin, Lake Champlain basin) and describe how they are "nested".
3. Compare and contrast point and nonpoint source pollution types
4. Name at least three sources of both PS and NPS pollution. Know that the Clean Water Act regulates point source pollution; NPS is unregulated.
5. Identify and describe several land use types (residential, industrial, agricultural, managed forest).
6. List the potential sources (and which land use they are specific to) of the following pollutants: manure/animal feces, human feces, oil/automotive pollutants, salt, fertilizers, pesticides, and soil.
7. Describe the environmental impacts of each of the pollutants.
8. List the largest threats to water quality/ecosystem health in Lake Champlain (or Connecticut river)
9. List at least one Best Management Practice (BMP) for each land use type.
10. List at least three BMPs homeowners (students themselves) can use to reduce their environmental impact.

Field Study Component

1. Use the scientific inquiry process and knowledge acquired in component one to design and conduct a cooperative river study as a class or group (formulate monitoring question, design river study, collect and analyze data, make conclusions)
2. Define reasons why scientists monitor benthic macroinvertebrates, E.coli, phosphorus, conductivity, dissolved oxygen, pH and temperature in streams.
3. Show competency using UVM WA stream monitoring equipment.
4. Demonstrate competency using at least one data analysis method.
5. Demonstrate ability to input data to online database.

Taking Action

1. Describe the significance of creating community connections and sharing knowledge.
2. Demonstrate a way(s) of sharing information with community.
3. List at least one local stakeholder interested in the river study.

UVM Watershed Alliance Participants Appendix A

Center for Tech—Essex
Justin Sorenson
3 Educational Drive
Essex, VT 05452
802.318.3210
jsorenson@ejhs.k12.vt.us
Lamoille (7), Winooski (8), Northern Lake Champlain Watersheds (5)
Chittenden County

Central Vermont Academy
Nathan Knowles
317 Vine Street
Barre, VT 05641
802.479.0868
natefalling@hotmail.com
Winooski Watershed (8)
Washington County

Champlain Valley Union HS
9th/10th grade
John Wulff and Gay Craig
369 CVU Road
Hinesburg, VT 05461
802.482.7160
gay@cvuhs.org
JohnW@cvuhs.org
LaPlatte (3) and Lewis Creek Watersheds (5)
Chittenden County

Colchester Middle School
Adam Ferris
425 Blakely Road
P.O. Box 30
Colchester, Vermont 05446
802.655.1772
FARRISA@colchester.k12.vt.us
Winooski (8), Northern Lake Champlain Watersheds (5)
Chittenden County

Concord Middle School
6th Grade
Sally Heiser
173 School Street
Concord, VT
802.695.2550
burkehollow@hotmail.com
Passumpsic Watershed (15)
Essex County

Craftsbury Academy
11th/12th grade
Rob Libby and Walt Gutzmann
Craftsbury Common, VT
802.586.2541
vtlibbies@yahoo.com
Lake Memphremagog Watershed (17)
Orleans County

Currier Memorial School
6th grade
Carrie Mauhs-Pugh
Danby, VT
802.293.5191
cmp@vermontel.net
Otter Creek Watershed (3)
Rutland County

Edmunds Middle School
Don Fox
275 Main Street
Burlington, 05401
802.864.2220
dfox@bsdvt.org
Winooski (8), Northern Lake Champlain Watersheds (5)
Chittenden County

Folsom Educational and Community Center
Teresa Robinson
75 North Main Street
South Hero, VT 05486
802.372.6600
twrobins2000@yahoo.com
Northern Lake Champlain Watershed (5)
Grand Isle County

The Gailer School
Kelly Miller
4066 Shelburne Road
Shelburne, VT 05842
802.985.1276
kmiller2@uvm.edu
Winooski (8), Northern Lake Champlain Watersheds (5)
Chittenden County

Hazen Union HS
Teal Church
126 Hazen Union Drive
Hardwick, VT 05843
802.472.6511
tealchurch@juno.com
Lamoille Watershed (7)
Caledonia County

Hartland Elementary
David Eastman, Michelle Burnette
97 Martinsville Road
Hartland, VT 05048
802.436.2255
deastman@hartland.k12.vt.us
mburnett@hartland.k12.vt.us
Ottauquechee-Black (10), Lower Connecticut (13)
Windsor County

Lamoille Middle School
Liam Callahan
736 Route 15 West
Hyde Park, VT 05655
802.888.4261
liamcallahan@yahoo.com
Lamoille Watershed (7)
Lamoille County

Lamoille Union High School
9th/10th grade
Chris Tormey
736 VT Route 15 West
Hyde Park, VT 05655
888-4261
Lamoille Watershed (7)
Lamoille County

Mount Anthony Union HS
Dan Rosenthal
301 Park Street
Bennington, VT 05201
802.447.7511 (school)
drosenthal@svsu.org
Battenkill Watershed (1)
Bennington County

Mount Mansfield Union HS (VYCC)
Cara Butterly
1949 E. Main St.
Richmond, VT
802.434.3969 x144
cbutterly@vycc.org
Winooski Watershed (8)
Chittenden County

Northfield High School
10th&12th grade
Amy Urling
Northfield, VT 05663
UrlingA@WSSU.org
802.485.5751
Winooski Watershed (8)
Washington County

North Hero School
Karin Ames
6641 US Route 2
North Hero, VT 05474
802.372.8866
karinames@earthlink.net
Northern Lake Champlain Watershed (5)
Grand Isle County

Orwell Village School
Barbara Young
494 Main Street
Orwell, VT 05760
802.948.2871
byoung@shoreham.net
byoung@arsu.org
Southern Lake Champlain (4), Otter Creek-Little Otter Creek-Lewis Creek (3), Poultney-Mettawee (2)

Pine Ridge School
Paul Brown and Paula Verrastro
9505 Williston Road
Williston VT 05495
802.434.2161
Paul_Brown@antiochne.edu
Winooski Watershed (8)
Chittenden County

Poultney High School
7th Grade
Linda Nolan-Moore
154 East Main Street
Poultney, VT 05764
802.287.5861
linda.moore@rswsu.org
Poultney/Mettawee Watershed (2)
Rutland County

Roxbury Village School
6th grade
Diana Costello
Roxbury, VT 05669
802.485.7768
costello@madriver.com
Winooski Watershed (8)
Washington County

Rutland High School
9-12th grade
George Hooker
Rutland, VT
802.770.1107
ghooker@rutlandhs.k12.vt.us
Otter Creek Watershed (3)
Rutland County

St. Monica's School
Pamela Nadeau
79 Summer Street
Barre, VT 05641
802.476.5015
pamn@nh1.com
Winooski Watershed (8)
Washington County

Stafford Technical School
10th grade
Mark Skakel
Rutland, VT 05701
802.770.1033
mskakel@rutlandhs.k12.vt.us
Otter Creek Watershed (3)
Rutland County

SUCCESS School
9th/10th grade
Nathan Hensley
Rutland, VT
802.773.1906 x237
nhensley@rutlandhs.k12.vt.us
Otter Creek Watershed (3)
Rutland County

Union 32 High School
11th/12th grade
Brian Slopey
940 Gallison Hill Rd.
Montpelier, Vt. 05602
802.229.0321 ext. 1211
BRIANSLOP@aol.com
Winooski Watershed (8)
Washington County

The Walden School
Tammy Russell
135 Cahoon Farm Drive
W. Danville, VT 05873
802.563.3000
thnksnow@together.net
Passumpsic Watershed (15)
Caledonia County

Weathersfield Middle School
7th grade
Dave Lambert
PO Box 279
Ascutney, VT 05030
802.674.5400
lambert@vermontel.net
Lower Connecticut Watershed (13)
Windsor County

Websterville High School
11th/12th grade
Virginia Collins
Websterville, VT 05641
802.479.0141
al_vircollins@juno.com
Winooski Watershed (8)
Washington County

Williston Central School
Dick Farrell
195 Central School Drive
Williston, VT 05495
802.878.2762
quattm@wsdvt.org
Winooski Watershed (8)
Chittenden County

Winooski Middle School
Ben Durant
60 Normand Street
Winooski, VT 05404
802.655.3530
bendurant@gmail.com
Winooski Watershed (8)
Chittenden County

UVM Watershed Alliance

Pre-Monitoring Questionnaire

Please help us to evaluate our program.
Your responses are confidential.

It's OK if you don't know the answer but please guess!

School: _____

Birthday: (month/day only) _____/_____

1. Which of the following is a watershed?

_____ A structure that holds drinking water at a water treatment plant?
_____ An area of land that drains into a water body.
_____ A substance sprayed onto your driveway to increase its imperviousness and enable it to shed more water.
_____ A type of rain coat.

2. By 1970, many rivers in the US were fouled with raw sewage and industrial waste. What event/piece of legislation reversed this process of degradation and is considered by some to be the greatest environmental success story in the US?

_____

3. Which benthic macroinvertebrate is the most sensitive to water pollution and serves as an indicator of good water quality?

Dragonfly_____
Leech_____
Stonefly_____
Caddisfly_____
Midge_____

4. E.coli can occur naturally in our intestines without causing us to get sick.

   True _____
   False _____

5. According to the State of Vermont, which of the following e.coli test results would indicate it was SAFE to swim?

   80 colonies/100 mL _____ 150 colonies/100 mL_____
   100 colonies/100 mL_____ None of the above _____

6. Pollution in Lake Champlain is mainly the result of 'Point source' or 'Nonpoint source' pollution?

   Point _____ Nonpoint _____

UVM Watershed Alliance

Post-Monitoring Questionnaire

School: _____

Birthday: (month/day only) _____/_____

After learning about watersheds and water quality and going to the river, will you make changes in your everyday activities that may improve water quality?

_____Yes _____No

If Yes: what changes?_____

_____

_____

_____

What did you like best?

_____

_____

_____

_____

What did you like least?

_____

_____

_____

_____

Complete the following sentence: After spending a few days learning about watersheds and water quality monitoring, I learned……_____

_____

_____

_____

1. Which of the following is a watershed?

_____ A structure that holds drinking water at a water treatment plant?
_____ An area of land that drains into a water body.
_____ A substance sprayed onto your driveway to increase its imperviousness and enable it to shed more water.
_____ A type of rain coat.

2. By 1970, many rivers in the US were fouled with raw sewage and industrial waste. What event/piece of legislation reversed this process of degradation and is considered by some to be the greatest environmental success story in the US?

_____

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Leech_____
Stonefly_____
Caddisfly_____
Midge_____

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100 colonies/100 mL_____ None of the above _____

6. Pollution in Lake Champlain is mainly the result of 'Point source' or 'Nonpoint source' pollution?

Point _____ Nonpoint _____

Watershed Model Presentation Appendix C

Our watershed model is an interactive tabletop model produced by Enviroscape (www.enviroscapes.com). The 1-1 ½ hr presentation serves as an introduction to basic watershed concepts. There are two parts to the demonstration: "how water pollution occurs" and "preventing water pollution".

Below is an outline of the presentation. Please use this outline and the list of terms (see Appendix D.) to prepare your class for our visit. We use a pre and post program evaluation to assess learning. See also additional suggested evaluation methods and related activities.

How Water Pollution Occurs

1. **Audience awareness: POLLUTION**. What do you think of when you hear this word?
2. **Watershed Discussion: What is a watershed? What is a waterbody?** Everyone lives in a watershed.
   A. Discuss watershed scale and nested watersheds. Introduce model as a representative watershed.
   B. Discuss water cycle.

3. **Sources of Water Pollution: two types—point and non-point.**
   A. Discuss and demonstrate examples of point source
      i. industrial plant
      ii. sewage treatment plant and combined sewer overflows
      iii. storm drain
   B. Discuss non-point source pollution
      i. farm (erosion, fertilizers and pesticides, manure)
      ii. residential area (pet waste, septic systems, household chemicals, lawn/garden fertilizers and pesticides)
      iii. golf course (fertilizers and pesticides)
      iv. forest (erosion)
      v. roads (salt, automotive pollutants)
      vi. streambanks/lakeshore (erosion)
      vii. construction site (erosion)
   C. Demonstrate non-point sources using props (soil, fertilizers, pesticides, oil, salt, manure)

4. **Make it RAIN! Discuss runoff**.
   A. Why does runoff happen?
      i. Construction site: no vegetation or silt fencing to hold soil
      ii. Lawns and Golf Course: too much pesticides and/or fertilizers used, not absorbed by plants
      iii. Highways, Roads and Parking Lots: collect automotive pollutants and salt; impervious surfaces send pollutants to storm drain

iv. Stream banks and Lakeshore: lack of vegetation contributes to erosion
v. Forest Clearing: lack of vegetation, heavy equipment, and steep slope accelerate erosion
vi. Plowed fields: disturbed soils are vulnerable to erosion
vii. Crops: see lawn and golf course
viii. Manure: improper allocation of spread manure and lack of fences near streams

5. Why is pollution harmful?

A. Discuss the invisible components of runoff
   i. Nutrients: Manure (animal and human) and fertilizers may contribute excess nutrients (phosphorus and nitrogen) to the ecosystem resulting in eutrophication.
   ii. Toxic Substances: Toxins are poisonous substances (oil, pesticides, metals) and are harmful to animals and humans. Some toxins bioaccumulate (PCBs, mercury) and this has led to fish consumption advisories in many areas (including Lake Champlain).
   iii. Bacteria: Some species may cause diseases such as dysentery and typhoid fever. Bacteria can infect shellfish, which in turn can make consumers sick. Bacteria can also harm other aquatic organisms. E.coli is used as an indicator of fecal contaminator and indicates potential contamination of harmful bacteria. The VT state standard for recreational waters is less then 77 colonies E.coli/100 mL water.
   iv. Soil: erosion contributes excess sediment to water bodies which may affect recreational use of water, cause flooding, kill fish, destroy habitat, and disrupt reproduction habits. May also increase eutrophication due to phosphorus sorbed to soil particles.

6. Discuss home activities: how do we contribute to pollution in our daily lives?

A. Examples of how we pollute at home:
   i. Improper use and disposal of household chemicals
   ii. Excessive use of water
   iii. Failure to maintain septic systems
   iv. Pet wastes

7. Review, summary and questions.

A. Review and summarize point source (PS) and non-point source pollution (NPS).
   i. NPS contributes more than 50% of the total water pollution, and is the major source of pollution in Lake Champlain. Although it is such a large problem, NPS is unregulated, and pollution prevention (best management practices) is voluntary.
   ii. PS pollution has been regulated by the federal government since 1970 with the passage of the Clean Water Act (although there are non-compliance issues)

Preventing Water Pollution

1. **Prepare model**: drain lake, wipe off grime from first demonstration and refill lake.

2. **Check audience awareness:** ask for suggestions on how to prevent nonpoint source pollution and write them down.

3. **Introduce the phrase Best Management Practices (BMPs):** systems, activities and structures that can prevent nonpoint source pollution.

   A. BMPs are not 100% capable of eliminating pollution, but each one helps to reduce and prevent pollution.
   B. BMPs can be site and pollutant specific; therefore a sinlge BMP may not be effective with all pollutants found at a single location.

4. **Prepare and place the BMPs**
   A. Make fence, place along stream next to cows to illustrate the practice of preventing them from entering stream.
   B. Make a small berm out of clay and place along plowed field nest to lake.
   C. Place felt "grass" strips along edge of roadway next to lake, beside construction site, around lower half of clearcut forest.
   D. Place felt "wetland" on area to right of plowed farm field.
   E. Place manure containment bin on grass next to road on the farm.
   F. Make any other BMPs audience suggests

5. **Add "pollutants" to model as in previous demonstration**

6. **Demonstrate BMPs**
   A. Construction site: spray with rain, point out BMP (grass strip) and discuss. List other construction BMPs (silt fencing, straw bales).
   B. Lakeshore and streambanks: spray with rain. Point out BMP (grass strip) and discuss.
   C. Forest—see above. Other BMPs include selective cutting, erosion controls on logging roads
   D. Farm area: spray with rain. Notice how berm prevents soil from entering waterbody. Notice how wetland filters sediments, nutrients, pesticides. Other BMPs include contour plowing, conservation tilling, and vegetative filter strips. For crops, BMPs include appropriate use of fertilizers and pesticides, plant cover crops, rotate crops.
   E. Driveways and highways: spray with rain. Discuss grass strips. Other BMPS include permeable surfaces, good motorist habits including preventing oil leaks, recycling used oil.
   F. Cows: fencing cows out of the stream is a BMP, however it requires farmers to provide an alternative water source and shaded area, which can be costly.
   G. Manure containment: place manure in manure containment, spray with rain and observe how runoff is contained. Farmers still need to manage manure—

manure can be applied to the ground as a fertilizer (do not overapply or apply when ground is frozen).

H. Lawns and Golf Course: spray with rain. BMPs include:
   i. Use pesticides and fertilizers sparingly and follow label instructions.
   ii. Use alternative fertilizers like compost or leave grass clippings on lawns. Have soil tested to find out exactly what grass needs.
   iii. Choose plants that are suited to the climate of your area to save on water, fertilizers and pesticides.
   iv. Do not dispose of grass clippings or leaves down storm drains or in streams.

I. Household Activities:
   i. Be a smart shopper; read labels and buy the least toxic product that will do the job. Buy biodegradable, recyclable products whenever possible.
   ii. Use household chemicals properly. Never burn or bury leftover chemicals. Never flush chemicals down drain or pour into storm drains. Check with local solid waste managers for proper disposal of household chemicals.
   iii. Clean up after pets
   iv. Use less water
   v. Maintain septic tank properly
   vi. Plant groundcover in your yard to prevent erosion
   vii. Don't litter. Recycle

Watershed Terms List Appendix D

*adapted from the Enviroscape User's Guide Glossary

- Acid Rain: Precipitation rendered (made) acidic by airborne pollutants. May contain toxic chemicals, such as mercury, that have escaped into the air from burning fossil fuels

- Algae/algae bloom: Green water plants; any of the large group of aquatic organisms that contain chlorophyll but lack special water carrying tissues. Through the process of photosynthesis, algae produce the majority of food and oxygen in water environments

- Bacteria: A large group of microscopic organisms of may different shapes, generally without chlorophyll. Some bacteria are helpful (as in a fermentation process), but certain species can cause diseases such as swimmer's itch, pneumonia or typhoid fever, among others.

- Best management practices (BMPs): Structural, nonstructural, and managerial techniques recognized to be the most effective and practical means to control nonpoint source pollutants, yet are compatible with the productive use of the resource to which they are applied.

- Clean Water Act (1972): the federal Clean Water Act has been public law since 1972. It requires the development of comprehensive programs for preventing, reducing, or eliminating the pollution of navigable waters and groundwaters and improving the sanitary condition of surface and underground waters.

- Combined sewer overflow (CSO): When stormwater systems are linked to sewer treatment systems, excessive stormwaters (rain events) added to the wastewater already in the system cause the excessive waters to bypass treatment and flow directly into rivers, streams and lakes.

- Erosion: the gradual wearing down of land by water, wind or melting snow. Soil is lost from streambanks, forests, hilly ground, lawns and farm fields.

- Eutrophication: the premature aging of a waterbody due to excessive nutrients and low oxygen levels. For example, phosphorus and nitrogen found in fertilizers and manure can cause sudden and excessive growth of algae and aquatic plants. When these plants die and decompose, dissolved oxygen is depleted.

- Fertilizers: chemical fertilizers applied by growers contain phosphorus and nitrogen. When excessively or improperly applied, chemical fertilizers enter waterbodies via runoff and contribute to eutrophication.

- Habitat: the physical environment or typical place within which a plant or animal naturally or normally lives and grows.

- Land use: the type of activities humans carry out on various area of land define land use (ie industrial, residential, agriculture).

- Benthic Macroinvertebrates (BMIs): bottom dwelling animals with no backbones that are visible to the unaided eye. BMIs collected from stream beds can be used as indicators of stream health.

- Nonpoint source (NPS): pollution that cannot be traced to a specific origin or starting point, but seems to flow from many different sources. NPS pollutants are generally carried off the land by stormwater (or melting snow) runoff. The commonly used categories for nonpoint sources are agriculture, forestry, urban, mining, construction, dams and channels, land disposal, and saltwater intrusion.

- Pesticide: an agent applied to crops, lawns, golf courses, or in household settings in order to manage pests (includes fungicides, herbicides, insecticides, rodenticides)

- pH: stands for "potential of hydrogen". A measure of the degree of the acidity or the alkalinity of a solution as measured on a scale ("pH scale") of 0 to 14. Levels outside the normal range (pH 6.5-8.0) aversely affects aquatic life.

- Phosphorus: a non-metallic element essential to life and found in fertilizers and animal waste. Excessive amounts introduced by runoff (and other ways) causes eutrophication in waterbodies.

- Point source: pollution discharged into waterbodies from specific, identifiable pipes or points, such as an industrial facility or municipal sewage treatment plant

- Riparian Buffers: vegetation (grasses, shrubs, and/or trees) directly adjacent to rivers, streams and lakes that traps water and associated pollution and prevents it from running off the land (acts as buffer); a BMP that helps prevent nonpoint source pollution.

- Riparian zone: the area where bodies of water meet upland areas which exhibits characteristics of both water and land areas.

- Runoff: that portion of precipitation that remains on the land until it ultimately reaches streams, rivers, lakes or other waterbodies; especially water

- Sediment: matter that settles to the bottom of a liquid; deposited by water, wind or glaciers

- Septic tank: a holding tank for collecting residential wastewaters. Used as an alternative to municipal sewer systems (esp. in rural areas). Wastewater collected in septic tanks must be treated before being released into the watershed.

- Temperature: altering stream temperatures due to industrial processes or removal of streamside vegetation negatively affects aquatic life (for example, raising stream temperatures affects trout, a cold-water species)

- Topography: the elevational profile of a land area. In the eastern US, where underground water flow is limited, watersheds can be delineated using topographical features.

- Wastewater treatment plant: sometimes synonymous with sewage treatment plant, but often an industrial treatment facility that processes the water to remove toxic and hazardous substances.

- Watershed: a region or area that may contain several rivers, streams, or lakes that ultimately drain to a particular watercourse or body of water.

Designing A River Study Appendix E

Why monitor?

In order to have a meaningful and successful monitoring program, it is essential to outline the reasons for it. Your study objective could be to increase overall awareness of the river, to develop basic monitoring skills, or to answer specific questions about the river.

If the objective of your program is to increase overall awareness of the river, it is useful to complete a physical survey of the river (see page 64 in *Testing the Waters*).

If your program objective is to help students learn basic stream monitoring skills, your program should include chemical monitoring (pH, alkalinity, conductivity, dissolved oxygen, phosphorus, nitrates), as well as biological (benthic macroinvertebrates, E.coli, coliform bacteria). A short physical survey may be included to provide students with a holistic consideration of the river.

I strongly encourage educators to involve students in the study design process. Student led projects often result in increased student participation and investment. Guide students in formulating specific questions about your river/watershed and design the study with a **question** in mind. The question is then the basis for investigation of the river(s) or stream(s) (inquiry-based study). Question formulation and study design can be done in collaboration with students, or by the lead educator(s) solely. Inquiry-based river studies are the most educational, useful, and fun type of study, and leads to a more "big picture" understanding of watersheds, the scientific process, and the roles we play as citizens and scientists.

Consider the existing uses of the river, and how water quality changes or degradation may affect these uses. For instance, the class may be interested in examining the water quality at a popular area swimming hole. The monitoring question could be: "Is this swimming hole safe for swimmers according to VT state law?". Students would monitor bacteria in the river, and compare the results to the state standard (less than 77 colonies E coli/100 ml water). Keep in mind that the current technology used to determine E. coli bacteria levels in streams requires 24 hours to process. Streams are constantly flowing, and the water that was sampled yesterday is long gone today. Therefore, the information about bacteria levels is already outdated when you receive it, and the numbers are used only as a very general guide to the recreational suitability of the river.

For help in designing your study, consult page 8 of *Testing the Waters*, and please contact Bethany at the Watershed Alliance.

Answer these questions, and you are on your way to a successful program!!!

1. Why are you monitoring?
2. What will you monitor?
3. What are your data goals?
4. Where will you monitor?

Adapted from Testing the Waters
Dates and Byrne, 1997. Testing the Waters: Chemical and Physical Vital Signs of a River. Kendall/Hunt Publishing Company, IA.

Possible Monitoring Questions

- Does this stream meet VT water quality standards?
- Does this stream provide appropriate wildlife habitat (birds, fish)?
- Is this stream negatively impacted by human development (leaking septic systems, erosion from construction, discharge from industry, farm runoff, removal of streamside buffers)?
- What sorts of benthic macroinvertebrates are found in this stream? Is the benthic macroinvertebrate community healthy in this stream?
- Is this stream safe for swimming right now? After a significant rain event?

Increase the Usefulness and Relevancy of your Data

It is important to remember the scope and limitations of projects when planning your monitoring program, and analyzing the data. In particular:

❖ Rivers are ephemeral. While one may find elevated levels of phosphorus one minute, levels may be normal the next. The same is true with coliform and E. coli data. It is difficult to make inferences about the water quality of a river without long-term, consistent data. Some ways to increase the usefulness of your data:

- ➢ From year to year, always monitor at the same spot in the river. This will provide baseline for analysis of long-term trends.
- ➢ If you have time, try scheduling your monitoring over an entire week. If possible, monitor in varying weather conditions. This may help illustrate pollution from runoff sources.

❖ Quality Control/Quality Assurance measures matter!! These measures help assure the validity and accuracy of the data we collect. See the UVM Watershed Alliance Quality Assurance/Quality Control Guidelines (following page)

- ➢ In order for the data to be useful, it needs to be collected in accordance with Watershed Alliance QA/QC guidelines. Our Watershed Educators have a working knowledge of the guidelines, and will assist in ensuring adherence to them.
- ➢ Access for data entry into the database will only be available to those classes led by trained facilitators and who pass all QA/QC standards.

Watershed Alliance Equipment Appendix F

We offer the following equipment for use in conjunction with our trained Watershed Educators. All equipment, including sample bottles, incubation trays, and petri dishes provided at no cost.

Water Quality Parameter	Equipment Available
Bacteria: Total Coliform and E.coli	Quanti-Tray method or Coliscan Easy-gel method
Phosphorus	Hach Pocket Colorimeter
Dissolved Oxygen	YSI digital meter or Hach Kit OX-2P titration method
Conductivity	YSI 85 digital meter
pH	LaMotte 5W-C digital meter or simple test strips
Temperature	YSI 85/LaMotte 5W-C digital meters or NIST certified alcohol thermometer
Benthic Macroinvertebrates	frame nets, sieves, identification keys, gloves, buckets, waders, forceps, jars, alcohol, trays, microscopes

QUANTI-TRAY vs COLISCAN METHOD FOR E. COLI QUANTIFICATION

The Quanti-Tray method is an EPA approved method for the quantification of E.coli. The method offers increased accuracy over the Coliscan method because there is no subjective identification of bacteria colonies required. On the other hand, using the Coliscan method is beneficial in demonstrating how to grow bacteria in petri dishes, and what these colonies of bacteria look like. Some teachers find it useful to do both methods and compare results.

HACH OX-2P DO KIT vs YSI DIGITAL METER

Similarly, some teachers find it useful to perform both the Hach kit and the YSI meter test for dissolved oxygen (provided there is sufficient time). The Hach kit test is significantly more involved, and requires that students where gloves and goggles, but is useful in demonstrating lab techniques and procedures. The YSI meter is quick, easy, and accurate, but provides a more limited opportunity for participation.

UVM Watershed Alliance Site Name:_____

Data Collection Sheet

***use one sheet per sampling date per site*

Date _____ Name(s)_____

School name_____ Watershed Address:_____

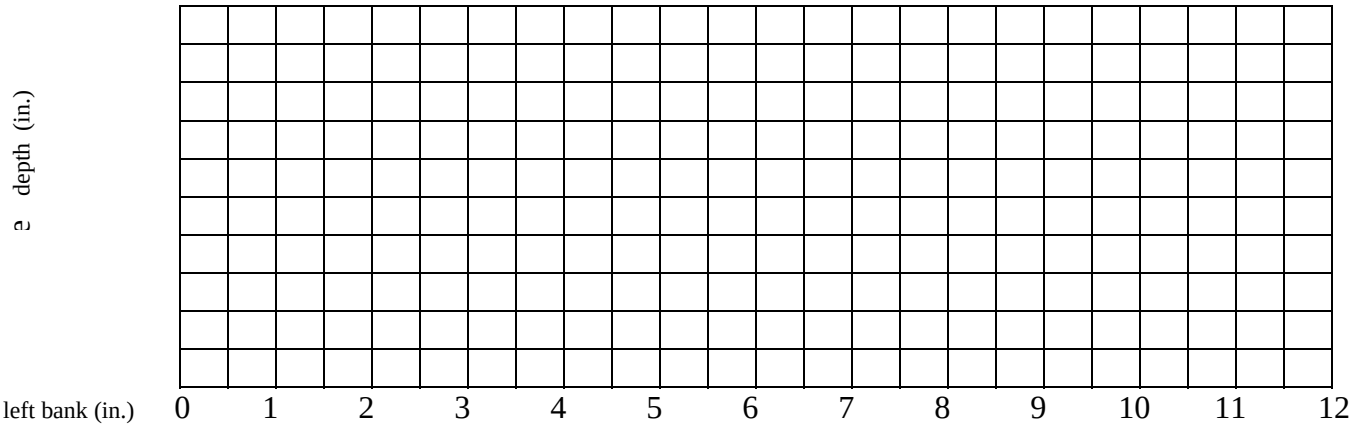
Weather past 24 hours:_____
Weather now:_____

Observations:_____
_____
_____

Instructions: Repeat each test twice in a fast section, then twice in a slow section, then average. Make sure you record **UNITS!!!!**

Parameter **Trial #**	Phosphorus	DO	**Conductivity**	pH	Turbidity	Velocity
Fast— 1						
2						
average						
Slow—1						
2						
average						

Cross-sectional area of Stream: in order to calculate discharge, you will need to make an estimate of the cross-sectional area of the river. Measure the depth of the river (in inches) at 6 inch intervals and record below.



**ON BACK: sketch the riverbank and corridor and indicate where your sample site is (note fast/slow sample collection spots)

Bacteria Sampling and Results Datasheet

***use one sheet per sampling date*

When collecting bacteria samples, follow instructions given by the UVM WA Watershed Educator. Remember: always collect sample in flowing water, and don't contaminate samples by touching inside of sampling jar, cap, or processing supplies!

Date_____

Sampling team names:_____

Processing team names:_____

Site #	Sample #	Total Coliform (in colonies bacteria/100mL water)	Total E.coli (in colonies bacteria/100mL water)	Analysis method used*

*Note: analysis method refers to the method you used to quantify the bacteria present in the sample (either Coliscan or QuantiTray methods)

Summary Table

Site #	average Total Coliform (colonies/100mL)	average E.coli (colonies/100mL)

Suggested Field and Lab methods for UVM Watershed Alliance

Geoff Dates
1/10/03

Method 4.1: *A Streamside Survey*

In this method, all work is done in the field. This involves collecting the sample using a net, sorting and identification of major groups (mostly orders, a few families, some classes) of benthic macroinvertebrates, and assessment of primary habitat characteristics. Approximately 0.28 square meters of the stream bottom are sampled. The relative abundance and richness of the each major group is determined, a field sheet is filled out, and the organisms are returned to the stream.

The Method At a Glance

Habitat Sampled:	Riffle Bottom
Sampling Device:	Net (Metal Frame or Sieve)
Level of Effort:	Not standardized
Quantitative:	No
# Samples	1 sample from 1 fast and 1 slow area in the riffle

Step 1: Assemble Collection and Field Processing Equipment and Supplies

Essential Items

		Purpose
☐	Directions to Collection Site	To locate collection sites
☐	Site Numbers for Each Site	To identify site on field sheet
☐	Streamside Survey Field Sheets (1 per site)	To record field data
o	Collection Net (Metal Frame or Sieve Net 500-600 micron mesh)	To catch organisms during collection
o	Arm-length gloves	To protect hands during collection
o	5-gallon bucket	To "swirl" the sample
o	Forceps	To handle critters
o	Squirt bottle	To back flush critters from sieve

o White Tray	To hold critters for sorting
o Ice cube trays (2)	To hold critters during identification
o Hand lens or magnifying glass	To help with identification
o Simple picture key	To help with identification
o Float (orange or grapefruit)	To estimate current velocity
o Stop watch	To time float
o Cloth tape measure (50 or 100 feet)	To measure segment length, width, and or marked string

Optional Items	**Purpose**
o Waders or high boots	To keep your feet dry
o Clipboard	To hold field sheet

Step 2: Follow directions to the first site and find the riffle you will be sampling.

Step 3: Measure a 200' segment that contains the riffle. Draw a map of the river that includes this segment on the "site sketch" sheet.

Step 4: Fill in the top half of the Collection Field Sheet.

Step 5: Scope the Situation - identify your collection spots.

Step 6: Get into position: Wade to the downstream side of the first spot and choose the specific place you will put the net.

Step 7: Position the net: ***Holder:*** Position the net on the cobbles on the bottom, opening facing upstream, and stand to one side, downstream of the net. ***Sampler:*** position yourself upstream of the net opening and off to one side.

Step 8: Dislodge the critters ***Sampler:*** Within a roughly square area upstream of the net, dislodge the critters by picking up individual cobbles from the bottom and rubbing them off with your hands to dislodge the critters so that they are carried into the net. Continue until all cobbles in the rectangular area in front of the net have been cleaned of critters. Then dig into the bottom to dislodge burrowing critters.

Step 9: Lift the net out of the water. ***Holder:*** Lift the net out of the water and bring the sample to shore for processing.

Notes: For A Sieve Net:

1. When you are finished disturbing the bottom, remove the cobbles used to anchor the net and rub them off so that any clinging critters are washed into the net.
2. Remove the net carefully from the water so as not to lose any critters: *Holder:* grab the top of the net handles. *Sampler:* grab the bottom of the net handles and the bottom edge of the net.
3. Together, lift the net out of the water with a forward scooping motion.
4. Bring the two handles together and roll the net around them.

Step 10: Bring the sample to shore and transfer the contents of the net to a bucket to separate the organisms from the debris by "swirling" the sample.

Step 11: Swirl the Sample. This procedure uses gravity to separate the organisms from the heavy debris.

1) Place the sample into a 5-gallon bucket about 1/3 full of water.
2) Use a swirling motion to create a whirlpool in the bucket. Organic debris and critters will get caught in the whirlpool.
3) Pour the water and organic debris into a #30 sieve, leaving the rocks, gravel and sand behind on the bottom of the bucket.
4) Fill about 1/3 of the bucket with water and repeat the swirling 15-20 times or until all that's left in the bucket is rocks, gravel and sand and the water you pour of has no debris in it.
5) Go back to step 10.

Step 12: Transfer the organisms to a white tray for sorting and identification. Turn the sieve upside down over the tray and tap it several times to empty the contents onto the tray. Squirt a small amount of water over the bottom of the sieve to flush the organisms onto the tray using a squirt bottle. Cover the bottom of the tray with about 1" of water.

Step 13: Pick and sort the organisms into the ice cube trays. Use the magnifier, tweezers, and the picture key, pick a representative of each type of organism present in the tray and place each in a separate compartment in the ice cube tray with water. Try to pick the largest specimen for each type. Look carefully! Many of these critters are quite small. Don't worry about identifying them at this point. Use obvious physical differences (other than size) to sort them. Examples include

- ✓ Presence or absence of legs
- ✓ Presence or absence and location of gills
- ✓ Presence or absence of "tails"
- ✓ Presence or absence and location of prolegs (e.g. at the end of the abdomen, on each abdominal segment)
- ✓ Unusual appendages
- ✓ Overall body shape (e.g. worm-like, segmented, round)
- ✓ A clearly visible head capsule
- ✓ Type of movement (swimming versus crawling)
- ✓ Color and pattern (to some extent)

Many of these characteristics are described in the picture key included with this manual ("A Simple Picture Key: Major Groups of Benthic Macroinvertebrates Commonly Found In Freshwater New England Streams").

Step 14: Identify the major group of each type of organism in the ice cube tray. Follow the instructions in the picture key ("A Simple Picture Key: Major Groups of Benthic Macroinvertebrates ") to identify the major group to which each organism belongs.

The major groups are (Sidebar?):

Order Ephemeroptera	(mayflies)
Order Plecoptera	(stoneflies)
Order Trichoptera	(caddisflies)
Order Diptera	
Family Chironomidae	(midges)
Family Tipulidae	(craneflies)
Other Families	(blackflies, horseflies, etc.)
Order Odonata	(dragonflies & damselflies)

Order Megaloptera	(fishflies, dobsonflies)
Order Coleoptera	(beetles)
Order Amphipoda	(scuds)
Order Isopoda	(sowbugs)
Order Decapoda	(crayfish)
Class Gastropoda	(snails)
Class Pelecypoda	(clams)
Class Oligochaeta	(bristle worms)
Class Hirudinea	(leeches)

1. Label each compartment as you identify the critter. Place any critters you cannot identify into a separate compartment labeled "unknown."
2. Sort the critters so that there is only one compartment for each major group. Some critters within the same major group look quite different. You may discover that critters that you placed in separate compartments are actually in the same major group. If so, put them in the same compartment for that major group. So, for some major groups, you may end up with several critters in the same compartment.
3. You should end up with at least one representative of each major group that you found in your sample in the ice cube tray. Some compartments may have several different types within a major group.

Step 15: Estimate the relative abundance of each major group and place the appropriate letter code next to that major group on the "Identification" part of the Streamside Survey Field Sheet.

1. To estimate relative abundance, look at the organisms you left in the white tray.
2. For each organism identified, estimate its relative abundance in the white tray.
3. Place one of the following letter codes in the "Relative Abundance" column next to the appropriate major group which best characterizes the relative abundance:

   N = (none in sample)

   R = (rare, very few in sample)

   C = (common, many in sample)

D = (dominant, most abundant group)

4. For the unknown organisms, place the appropriate letter code next to the "unknown" space.

Step 15: Estimate the richness of each major group and record it next to that major group on the "Identification" part of the Streamside Survey Field Sheet.

1. Look in each compartment of the ice cube tray.
   - For those compartments with only one critter, record a "1" in the "richness" next to the appropriate major group.
   - For those compartments with more than one critter (each should be a different type), record the number of critters in the "richness" column next to the appropriate major group.
2. Add the richness values for the first column (mayflies, stoneflies and caddis flies). Write this total in the box for "EPT Richness." Add the totals in the other two columns and write the totals in the spaces next to "Total Richness 1" and "Total Richness 2." Add these three totals and fill in the "Total Richness" Box.

Step 16: Estimate the richness of each major group and record it next to that major group on the "Identification" part of the Streamside Survey Field Sheet.

Look at the white tray. Estimate the total number of critters in the tray and check the box in the "Estimated Total Abundance" section that comes closest to your estimate. Don't worry about this too much – it's only an estimate.

Step 17: Fill in "Habitat Characteristics" section of the Streamside Survey Field Sheet.

1. **River Bottom Composition:** What is the stream bottom made of? Look at the stream bottom of the entire 50' segment and estimate and record the percentage of the bottom which is composed of the different materials listed.
2. **Embeddedness:** Embeddedness is the percent surface area of larger particles (boulder, rubble or gravel) surrounded or covered by sand or silt. Pick up a few rocks from the stream bed. The bottom of the rock will usually be lighter in color than the rest. This lighter area was embedded. Estimate the percentage of each of several rocks that was embedded and average the results.

3. **Current Velocity:** The velocity (how fast the water is moving) will be measured at two 10 foot sections: one in fast current and one in slow current. At each section, two people measure off ten feet, standing in the stream and holding the tape or string above the water between them. The upstream person should stand facing one of the banks and hold the float, 3/4 submerged in the water to his or her front. Avoid placing the float in the "eddy" caused by your legs. Release the float when the timer tells you to. The timer will measure the number of seconds the float takes to travel the 10 feet. Do this twice in each section and record the result. Try to pick an open path where the float will not encounter rocks or other stream obstructions.

Step 17: Place all organisms back into the stream.

Step 18: Assess the results:

This is a simple assessment of impairment based on the relative abundance and richness of the sample you collected.

Look at your field sheet in the "Relative Abundance" and "Richness" columns. Compare the results with the following table to determine if your results show that the site is "seriously impaired" or "not or slightly impaired." If it doesn't fall into either of these categories, you can classify it as moderately impaired.

Level of Impairment	Relative Abundance	Richness
Seriously Impaired	Mayflies and stonfliies not present *and* sample is dominated by worms, leeches, midges, sowbugs, scuds, clams, or snails	Total Richness <8 EPT Richness <4
Not impaired or slightly impaired	Sample is dominated by mayflies stoneflies or caddis flies If caddisflies dominate then mayflies or stoneflies are common	Total Richness >12 EPT Richness >8

Intensive Sampling Method Appendix H

Collection Method A2: Collecting a simple semi-quantitative sample using a net

Collecting benthic macroinvertebrates with a net requires teams of at least two people – three per team is best. Two people can do the collection. The other can fill out the collection field sheet, and assist with sample processing and preservation.

The Method At a Glance

Habitat Sampled	Riffle Bottom
Sampling Device	Net (Metal Frame or Sieve)
Level of Effort	Standardized by area
Quantitative?	Semi: Area specified but not delineated by the sampling device
# and Type of Samples	1 – 3 simple replicates from fast and slow areas of the riffle

The following steps should be carried out by two people: one to hold the net (the net "holder") and the other to dislodge critters (the "sampler"). Note that the procedure described below can be done with any collection net: D-frame, rectangular frame, or sieve. Special instructions that apply to the sieve net are noted when needed.

Step 1: Assemble Collection and Field Processing Equipment and Supplies

Essential Items	Purpose
o Directions to Collection Sites	To locate collection sites
o Site Numbers for Each Site	For labeling samples
o Collection Field Sheets	For permanent record of collection
o Collection Net (500-600 mm mesh): 18" X 8" rectangular metal frame net or 1 meter square sieve	To catch dislodged organisms during collection
o Arm-length Gloves	To protect hands during collection
o Sieve Bucket (#30 mesh)	To help collect and transfer samples
o Soft, Nylon Bristle Brush	To scrub critters off rocks
o Forceps	To pick critters off net and sieve bucket
o 1 qt. Mason Jars w/ Rubber Seal Lids, nalgene bottles, or zip-lock bags (1 for	To hold preserved samples

each replicate)

o 90% de-natured Ethyl Alcohol	To preserve samples
o Labeling Tape and Pencils	To label sample containers
o Float (orange or grapefruit)	To measure current velocity
o Stop Watch	To measure current velocity
o Tape Measure (50' minimum) or 50' string marked in 10' sections	To measure segment length, width, and velocity

Optional Items

	Purpose
o Waders or high boots	To keep your feet dry
o USGS Topographic Map	To help locate collection sites
o Clipboard	To hold field sheet
o 5-gallon Bucket	All purpose container
o Life Jackets	For safety in deeper water

Step 2: Follow directions to the first site and find the riffle you will be sampling.

Step 3: Measure a 200' segment that contains the riffles you will sample. Draw a map of the river that includes this segment on the "site sketch" sheet.

Step 4: Fill in the top half of the Collection Field Sheet.

Step 5: Scope the Situation - identify your collection spots. Following are general guidelines:

- ✓ Aim for fast and slow places with cobble bottoms – these are rocks 2-10" in diameter.
- ✓ Riffle should be long enough to accommodate three replicate samples.
- ✓ Sample in the main channel, unless the water is too deep. In any case, avoid areas along the margins that may be dry at low flows.
- ✓ Sample spots that are 4 – 18" deep
- ✓ If you are collecting replicates, try to sample from a single riffle at similar depth and bottom type.

- ✓ Ideal sampling locations consist of rocks 5 to 10 cm in diameter sitting on top of pebbles.
- ✓ Avoid the transition zone from riffle to pool.
- ✓ Avoid bottom materials dominated by rocks larger than 50 cm in diameter..
- ✓ Avoid bridges and other large human-made structural features If unavoidable, sample at least 50 meters upstream of a bridge and 200 meters (more would be better) downstream of a bridge.

Step 6: Get into position: Wade to the downstream side of the first spot and choose the specific place you will put the net.

Step 7: Position the net on the bottom. ***Holder:*** Position the net on the cobbles on the bottom, opening facing upstream, and stand to one side, downstream of the net. ***Sampler:*** position yourself upstream of the net opening and off to one side.

> **Note: For A Sieve Net**
> Since this net is 1-meter long and does not have a rigid bottom, it may be hard to properly seat and anchor the net on the bottom. You may wish to use a few cobbles to secure and anchor the sieve to the bottom. Be sure to use cobbles that fall within the collection area.

Step 8: Dislodge the Critters ***Sampler:*** Wear heavy-duty gloves!! Work within an area upstream of the net opening as wide as the net and 2/3 the width.[1] Pick up all the individual cobbles from the bottom and rub them off with your hands, or with a brush, to dislodge the critters so that they are carried into the net. Inspect each cobble to be sure there are no hangers on. When the cobbles are done, set them outside the collection area and gently dig into the soft bottom (if any) to dislodge burrowing critters.

Step 9: Lift the net out of the Water. ***Holder:*** When you've finished at this spot, carefully lift the net out of the river and prepare to process the sample.

[1] For example, if your net is 1-meter wide, collect from an area about 2/3-meter upstream of the net opening.

Note: For A Sieve Net:

1. When you are finished disturbing the bottom, remove the cobbles used to anchor the net and rub them off so that any clinging critters are washed into the net.
2. Remove the net carefully from the water so as not to lose any critters: *Holder:* grab the top of the net handles. *Sampler:* grab the bottom of the net handles and the bottom edge of the net.
3. Together, lift the net out of the water with a forward scooping motion.
4. Bring the two handles together and roll the net around them.

Step 10: Bring the Sample to Shore For Processing

Step 11: Inspect the Sample.

If your sample has a lot of sand and gravel, you may want to use the "swirling" technique to separate it out. See sidebar.

If the sample is "clean" – there is not a lot of stuff other than bug - go to the next step.

Step 12: Bring the net and the sieve bucket to a shallow area of quiet water.

Step 13: Transfer the contents of the net to the sample container. Use either a bucket or a sieve bucket to do this.

Using a Sieve Bucket

1. Gather the sample material into one corner of the net.
2. Grab that corner of the net from the bottom, holding the sample material in a clump, and turn the net inside out into the sieve bucket. Try to knock any sample remaining on the net into the bucket, or back flush the net into the sieve bucket using a jar of water.
3. Transfer the contents of the sieve bucket to a sample jar.
4. Place the sieve bucket in shallow water so that water comes up through the bottom screen and moves the sample around.
5. Move or shake the bucket to get the sample into the bottom of the bucket below the spout.
6. Move the sample to the spout and position the jar under it. Place the net under the jar to catch any organisms that miss the jar. Scrape the sample into the jar.

Using 5-gallon buckets

1. Rinse the net thoroughly into the bucket. If necessary, pick any clinging organisms from the net by hand and put them in the bucket. At this point, all of the sample should be off the net and in the bucket.
2. Look through the material in the bucket and immediately return any fish, amphibians, or reptiles to the stream.
3. Carefully remove large pieces of debris (leaves, twigs, and rocks) from the sample.
4. While holding the material over the bucket, use the forceps, spray bottle, and your hands to pick, rub, and rinse the leaves, twigs, and rocks to remove any attached organisms.
5. Use a magnifying lens and forceps to find and remove small organisms clinging to the debris. When you are satisfied that the material is clean, discard it back into the stream.
6. Carefully pour the contents of the bucket through the net (or a sieve) into a second bucket (which has not yet been used, should be completely empty). The sample will wind up in the net or sieve. The water will wind up in the second bucket.
7. Use your spray bottle, forceps, sugar scoop, and gloved hands to remove all the material from bucket #1 onto the net or sieve. Carefully remove large pieces of debris (leaves, twigs, and rocks) from the sample.
8. As a final check, repeat the process above, but this time, pour bucket #2 over the net or sieve, into bucket #1. Transfer any organisms on the net or sieve into the jar.
9. Fill the jar with 90% ethyl alcohol so that all material is submerged in alcohol. Put the lid tightly back onto the jar and gently turn the jar upside down two or three times to distribute the alcohol and remove air bubbles.

Step 14: Label the sample container. Complete the sampling station ID tag. Be sure to use a pencil, not a pen, because the ink will run in the alcohol. The tag should include your station number, the stream, location (e.g., upstream from a road crossing), date, time, and the names of the members of the collecting crew. Place the ID tag into the sample

container, writing side facing out, so that identification can be seen clearly.

Swirling the Sample (Optional) Sidebar?

This procedure uses gravity to separate the organisms from the heavy debris.

1) Place the sample into a 5-gallon bucket about 1/3 full of water.
2) Use a swirling motion to create a whirlpool in the bucket. Organic debris and critters will get caught in the whirlpool.
3) Pour the water and organic debris into a #30 sieve, leaving the rocks, gravel and sand behind on the bottom of the bucket.
4) Fill about 1/3 of the bucket with water and repeat the swirling 15-20 times until all that's left in the bucket is rocks, gravel and sand and the water you pour of has no debris in it.
5) Go back to step 11.

Replicates: Repeat the collection process until you've got the required number of replicates.

Lab Processing Method 2.1: Picking and Sorting A 100 Critter-1/4 Sub-sample

This section describes the laboratory procedures for picking and sorting 1/4 of the field sample and 100 critter Sub-sample. It is designed so that you can stop after picking the tray and rough sorting into major groups. At some other time, you can do the identification and data analyses.

The Method At a Glance

Sample Type	Whole Field Sample (detritus included)
Sample Preservation:	90% Ethyl Alcohol (before adding to Sample) or Comparable
Sub-sample Type	Random: Minimum of 1/4 of the sample and 100 organisms.
Sub-sampling method	Remove an unbiased, random representative sub-sample by picking critters from a gridded sub-sampling tray with 12 4x4 inch squares.
Minimum % of sample picked	25%

Preparation

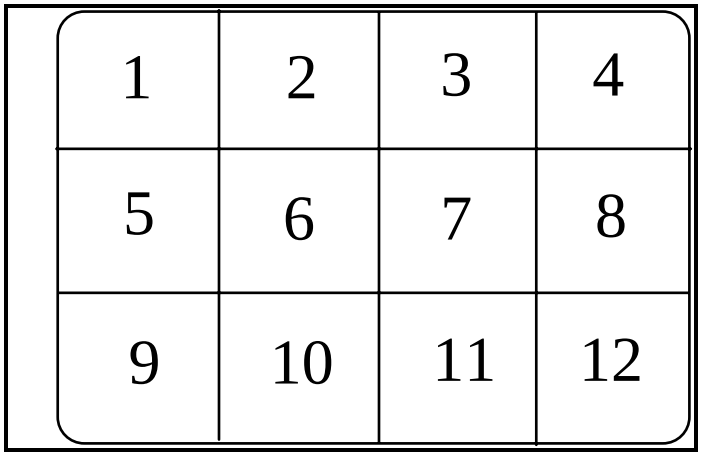
Assemble the following equipment and supplies:

	Essential Items:	**Purpose:**
o	#30 Sieve (1)	To strain the alcohol from the sample
o	Labeling Tape & Pencils	To label sample in petri plates and storage vials
o	Small Vials (3-4 per sample)	To store sorted and/or identified samples
o	Lighted Magnifiers (1 per work station)	To get a close-up view of the sample in the tray
o	One Shallow (1" deep) White Tray	To hold sample while picking sub-sample
o	Four 4-Compartment Petri Plates	To hold sorted organisms during picking
o	Two Forceps - fine tipped	To pick and manipulate critters
o	Wash bottle w/90% Ethyl Alcohol (1 per work station)	To preserve organisms

o	Sample Processing Record (1 per replicate)	To record picking
o	Random # generator or 12 pieces of paper numbered 1 – 12	To randomly select a square in the tray for picking.
o	Tally counter (optional)	To keep track of the critter count

Sidebar: Making a sub-sampling tray.

Mark a grid with 12 squares on the bottom of the white trays as shown below, and number each square. Use a permanent magic marker or grease pencil.



Tip:

Organize a team of people at each work station. one person to pick the tray and another person to sort the organisms into tentative major groups and record the information on the Sample Processing Record.

Step 1: Rinse and prepare the sample

1. Pour the preserved field sample (alcohol and debris) into #30 sieve and rinse off the preservative in a sink.
2. Rinse and visually examine large material – rocks, twigs and leaves. Pick any small clinging organisms, such as midges, off these materials and place the organisms in the sieve.
3. Discard the material.

Sidebar Tip: *Swirling the Sample:*

If your sample jar has a lot of gravel and sand in it (more than half the jar), you might want to "swirl" it to remove this bulky material in order to make picking easier.

1) Pour the preserved field sample (alcohol and debris) into #30 sieve.
2) Dump the sample from the sieve into a 5-gallon bucket (not the sieve bucket).
3) Fill the bucket about half full of water.
4) Swirl the contents of the bucket. You'll notice that the lighter material (including the critters) tends to come to the top.
5) Pour off the water, and the material floating in it, into the sieve, leaving the sand and gravel in the bucket.
6) Repeat steps 3 - 5 until, when you pour off the water, no lighter material comes out of the bucket. You may need to repeat this 15 to 20 times to get all the lighter material out of the sand and gravel.
7) Continue with step 3 below and discard the sand and gravel in the bucket.

Step 2: Transfer the sample to a gridded tray

1. Turn the sieve upside down over the tray and tap it several times to empty the contents onto the tray.
2. Squirt a small amount of water over the bottom of the sieve to flush the organisms onto the tray using a sink sprayer or squirt bottle.
3. Cover the bottom of the tray with about 1/4" of water. Evenly distribute the sample (including the detritus) over all the squares in the tray.

Step 3: Mark several petri plates with the site and replicate number Mark the *bottom* of each petri plate with the site and replicate number of the sample you're processing. Use labeling tape and pencil (the alcohol will dissolve most inks).

Step 4: Select a random starting square on the gridded tray. Use a random number generator (computer or printed) or pick one of 12 pieces of paper (each with a number from 1 to 12) out of a container to identify a starting square.

Step 5: Pick all the organisms from the starting square and place in the petri plates in alcohol. Use the lighted magnifier and the forceps to systematically pick all the organisms out of the first square.

- Be sure to look for very small organisms as well as the larger ones.
- Turn over rocks, leaves and twigs and look for organisms that may be stuck to these materials.
- Pull apart clumps of algae to find organisms that may be tangled there.
- Any organism which is lying over a line separating two squares is considered to be in the square containing its head or (if it's headless) the majority of its body.

Hint: Using two forceps makes this step easier.

Step 6: Rough sort as you pick As each organism is removed from the square, rough sort it into one of the compartments of a marked petri dish with other similar organisms.

- Keep the macroinvertebrates covered with alcohol.
- Don't worry about identifying the organisms at this point. Just use the obvious physical differences among the organisms (e.g. overall body shape, # of tails, etc.), Use the dissecting scope to help you see these differences, but don't worry about precision -- you'll identify them later.

Hint: It's easier to see the organisms if you place the petri plates on top of a plain white sheet of paper.

Step 7: Have someone check that there are no critters left in the square when you are finished.

Step 8: When you've finished picking the starting square, mark the corresponding square on the "Sample Processing Record" lab sheet with an "x."

Step 9: Select another random square and continue picking and sorting until you've done at least 1/4 of the squares (3 in a 12-square grid) and over 100 organisms.

1. As you finish picking each square, mark the corresponding square on the "Sample Processing Record" lab sheet with an "x."

2. When you've picked 3 squares and have over 100 organisms, you may stop.
3. *If not, you must continue picking one square at a time until you've picked over 100 organisms or the entire tray, whichever comes first.*
4. *Be sure to pick all the organisms from the last square, even if you pick well over 100 organisms.*

Note: It's possible that you may pick the entire tray and still not have 100 organisms.

Step 10: Pick rare organisms. When you have finished picking the squares. Quickly scan the un-picked part of the sample for any types of critters you did not pick from the selected squares. Pick *one* of each type.

The purpose of this step is to find rare critters that may not have been in your subsample. Missing these would underestimate the diversity of your sample. Yet picking only one of each will not substantially affect the total number of critters picked and your estimate of abundance. For now, keep these in a separate compartment of the petri plate.

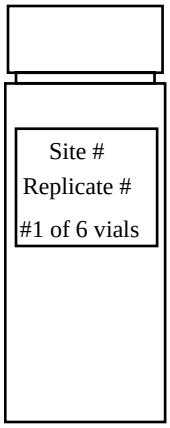
Step 11: Fill in the "Picking and Initial Sorting" section of the Macroinvertebrate Sample Processing Record

- Be sure to fill in the number of squares you picked on this sheet!
- Fill in the number of rare critters that you picked from outside of your selected squares.
- Note: If you wish to identify the organisms at this point, proceed to SectionB: Lab Identification.
- Otherwise, continue with step 11 below.

Step 12:Transfer the contents of the petri plate to labeled capped vials filled with alcohol

1. Unless you are going to identify the critters in this session, transfer the contents of each compartment of the petri plate to its own vial. The contents of several compartments can be combined if there are only a few organisms or if you think they'll be easy to re-sort later.

2. Until you have positively identified them, place all rare critters that you picked from outside the selected sub-sampled squares in a separate vial labeled "rare."
3. Fill each vial completely with 90% ethyl alcohol and cap tightly. Using labeling tape and a pencil, label each vial like this:



An inside label is recommended as well as an exterior label.

Step 13: Repeat steps 1-10 for each replicate and fill out the Macroinvertebrate Sample Processing Record

Step 14: Clean up and save some organisms for a reference collection If you wish, pick out specimens that representing each major group or family from the sample left in the tray (if any) and save in a properly labeled vial to be identified for a reference collection.

Note: be sure to note on the Macroinvertebrate Sample Processing Record any critters you remove for your reference collection!

Sidebar: About Reference (Voucher) Collections

A reference collection is a set of preserved organisms that are properly identified and labeled. It can be used for a number of things:

Aid to identification: Drawings in keys are intended to clarify the body parts you use to identify the critters. But, they are 2-dimensional and don't always exactly look like the organism in front of

you. Real specimens, especially if they are in good shape, can be placed beside the unknown organism.

Record of the types of organisms you've collected: This is sometimes called a "voucher" collection. As a quality control measure, your voucher collection can be used to verify that you've identified the critters in your sample correctly.

Teaching/training tool. Reference collections can be used to train people how to properly identify the critters.

A few suggestions:

- Try to have examples of the critters at different life stages, since your samples certainly will. so you can see how the body parts develop.
- Keep the specimens covered in alcohol. Dried, shriveled critters are no help!
- Try not to handle delicate specimens, like some of the mayflies, to much. Appendages break off easily.
- Make sure that each specimen, when it's removed from the vial, is transferred to a labeled container and doesn't get mixed in with samples.
- Keep adding fresh specimens as they become available.
- You can buy reference collections from: [Check NABS] You also may be able to get them donated by an entomology lab.

3) Lab Methods for Identifying Samples

This section lists two methods for identifying the critters in your picked and sorted samples. The key to the whole benthic macroinvertebrate monitoring effort is to correctly identify the organisms in the sample.

However, this is a very forgiving step in the process. Because you are saving your samples, you can always go back and correct identification errors, pick more of the sample (if you've saved it), and answer questions that may come up about your sample processing. In fact, this is one of the main advantages of preserving your samples.

In this section, we describe two methods for identifying the critters in your sample:

Identification Method 3.1: Identification of major groups (level 1)

Identification Method 3.2: Identification of families (level 2)

As the names imply, the main difference between them is that in the first you are identifying major groups (mostly orders) of macroinvertebrates. In the second, you are identifying families. Which one you pick depends entirely upon your data quality goals, expectations about the level of sensitivity you need, and your resources.

Here's another advantage to working with preserved samples: some people start out identifying major groups and then do the families if needed. If you are uncertain, start with method 3.1 and see how it goes.

Identification Method 3.1: Identification of Major Groups

This section describes the laboratory procedures for identifying major groups of benthic macroinvertebrates. To do this, "keys" are used. Keys are guides to the identification of plants or animals. They arrange macroinvertebrates' characteristics in a way that leads you in a logical manner to place each organism in its correct grouping or "taxon," (order or family, for example).

We include two keys with this manual:

1) "*A Simple Picture Key: Major Groups of Benthic Macroinvertebrates Commonly Found In Freshwater New England Streams*" is used as you would a field guide, by finding the picture of the organism that looks most like the one you're trying to identify and checking to see that it has certain characteristics.
2) The other ("*Key To The Freshwater Macroinvertebrate Fauna of New England*") is a "dichotomous key" which uses "couplets" – two mutually exclusive statements regarding a physical characteristic of the organism – and illustrations that highlight the characteristic being examined.

Instructions for using both keys are contained in the keys themselves.

Step 1: Set up work stations

Set up one or more work stations, each with the following:

Essential Items:	**Purpose:**
o Labeling Tape & Pencils	To label sample in petri plates and storage vials
o Small Vials (3-4 per sample)	To store sorted and/or identified samples
o Dissecting scope: at least 40 power (1 per work station)	To magnify critters for identification
o 4-Compartment Petri Plates (4 per work station))	To hold sorted organisms during picking and identification
o Forceps - fine tipped (2 per work station)	To pick and manipulate critters during picking and identification
o Wash bottles w/90% Ethyl Alcohol (1 per work station)	To preserve organisms
o Taxonomy Keys & References	To identify organisms
o Plain white paper	To place under petri plates in scope or on table for contrast
o Sample Processing Record (1 per replicate)	Filled out, to record # of squares picked
o Identification Lab Sheets (1 per sample)	To record identification

* Locate the sample processing record that goes with the sample you're going to identify. There should be one record for each replicate sample. Check to see that the number of squares picked is noted.

Step 2: Mark several petri plates with the site and replicate number

Mark the *bottom* of each petri plate with the site and replicate number of the sample you're processing. Use labeling tape and pencil.

Step 3: Fill in the top of the Benthic Macroinvertebrate Identification Sheet - Level 1 (Form 6)

Fill in the site #, river/stream, date sampled, your name, and the date of the identification at the top of the form. Also fill in the # of squares picked from the tray boxes at the bottom for each replicate. Note that one identification sheet is used per site for all 3 replicates.

Step 4: Sort the sample into the compartments of the petri plates

If the sample has been sorted, place the contents of each vial into its own compartment. If it hasn't been sorted, rough sort, following steps in the previous section.

Step 5: Use the picture key to identify the major group of the organisms in each of the compartments

Place the petri plate on the platform of the dissecting scope. Focus the dissecting scope so you can see the whole organism. *Hint: It's easier to see body characteristics if the organisms are lit from above against a white background.*

Follow the instructions in the picture key ("A Simple Picture Key: Major Groups of Benthic Macroinvertebrates Commonly Found In Freshwater New England Streams") to identify the major groups.

The major groups are:

Order Ephemeroptera	(mayflies)
Order Plecoptera	(stoneflies)
Order Trichoptera	(caddisflies)
Order Diptera	
Family Chironomidae	(midges)
Family Tipulidae	(craneflies)

Other Families	(blackflies, horseflies, etc.)
Order Odonata	(dragonflies & damselflies)
Order Megaloptera	(fishflies, dobsonflies)
Order Coleoptera	(beetles)
Order Amphipoda	(scuds)
Order Isopoda	(sowbugs)
Order Decapoda	(crayfish)
Class Gastropoda	(snails)
Class Pelecypoda	(clams)
Class Oligochaeta	(bristle worms)
Class Hirudinea	(leeches)

Move any organisms that don't belong in that compartment to another compartment.

Step 6: Place any organisms you cannot identify into an "unknown" compartment

Place any organisms that you cannot identify using the picture key into a separate petri plate compartment. Don't worry about these for now. In the next step, you'll identify these using another key.

Step 7: Use the dichotomous keys to identify "unknown" organisms

Use one or both of the two dichotomous keys to identify the unknown organisms:

- "Key To The Freshwater Macroinvertebrate Fauna of New England."
- "Aquatic Diptera Immatures" (Figure 16.1 excerpted from "Aquatic Entomology by W. Patrick McCafferty).

If you are still not sure about a particular organism, have a more experienced person assist you or preserve it in a vial until someone is available at a later date.

Step 8: Count and record the number of organisms in each major group on the Benthic Macroinvertebrate Identification Sheet - Level 1 (Form 6)

1. When you're certain of the identity of the organisms in all the compartments (except for those in the unknown compartment), count the total number of individuals ("density") in each major group.

2. Record your count in the 'D' column under the appropriate replicate # on the Benthic Macroinvertebrate Identification Sheet - Level 1 (Form 6).

Step 9: Estimate the number of different families of organisms in each major group and record on the Benthic Macroinvertebrate Identification Sheet - Level 1

Estimate the number of perceived families ("richness") within each major group. For many of the organisms, the picture key included with this manual will enable you to actually identify the families. For the mayflies, stoneflies, and caddisflies, damselflies, and dragonflies, use the differences in body characteristics identified in the diagrammatic keys in McCafferty's Aquatic Entomology to differentiate the organisms. Note that it is not necessary to actually identify the families, just check for certain key distinguishing traits, some of which are listed below:

Mayflies: the location, shape and texture of the gills; the presence or absence of "tusks" on the head; the presence or absence of filtering hairs on the front legs; the size, shape, and orientation of the head; and the length of the antennae.

Stoneflies: presence or absence of gills at the base of the legs; presence or absence of gills underneath the first two abdominal segments; whether the wingpads are parallel or divergent, the shape of the body; color pattern; the shape of the thorax.

Caddisflies: presence or absence of "shields" on each segment of the thorax; the size and shape of the anal prolegs; shape of the body; whether antennae are visible, presence or absence of a case; presence or absence of humps on the top and/or sides of the first abdominal segment.

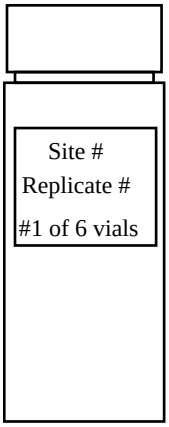
Don't worry about the clams, snails, crayfish, worms, sowbugs, scuds and other non-insects. Pay attention to obvious differences only.

Note: Size is not a distinguishing characteristic, nor is color.

Record your estimate in the 'R' column under the appropriate replicate # on the Benthic Macroinvertebrate Identification Sheet - Level 1 (Form 6).

Step 10: Store the sorted organisms in capped vials with alcohol.

Transfer the contents of each compartment of the petri plate to its own vial. The contents of several compartments can be combined if there are only a few organisms or if you think they'll be easy to re-sort later. Fill each vial completely with 90% ethyl alcohol and cap tightly. Using labeling tape and a pencil, label each vial like this:



Place all unidentified organisms into a separate vial. A more experienced person can identify them at a later date.

Step 11: Fill in the Level 1 - Major Group Identification box on the Macroinvertebrate Sample Processing Record (Form 5)

Fill in a check, the date, and your name in the Level 1 - Major Group Identification box on the Macroinvertebrate Sample Processing Record for this replicate.

Step 12: Repeat steps 1-11 for each replicate and record on the Benthic Macroinvertebrate Identification Sheet - Level 1 (Form 6)

Identify the organisms in each replicate sample and record the density and richness in the appropriate column of the Benthic Macroinvertebrate Identification Sheet - Level 1 (Form 6).

Identification Method 3.2: Identifying the Families for Selected Major Groups in the Sub-sample

This section describes the laboratory procedures for identifying benthic macroinvertebrate families for selected major groups in the sub-sample, after you've identified the major groups. This requires training and experience in macroinvertebrate taxonomy. We recommend that an aquatic biologist, entomologist, or someone familiar with macroinvertebrate taxonomy be present during the identification or subsequently verifies the identification.

Step 1: Identify the Major Groups in the sample (optional)

Identifying the major groups will make it easier to sort the organisms for family identification (see the previous section for instructions on how to do this). However, you can skip this step if you wish.

Step 2: Set up work stations

Set up one or more work stations, each with the following:

Essential Items:	**Purpose:**
o Labeling Tape & Pencils	To label sample in petri plates and storage vials
o Small Vials (3-4 per sample)	To store sorted and/or identified samples
o Dissecting scope: at least 40 power (1 per work station)	To magnify critters for identification
o 4-Compartment Petri Plates (4 per work station))	To hold sorted organisms during picking and identification
o Forceps - fine tipped (2 per work station)	To pick and manipulate critters during picking and identification
o Wash bottles w/90% Ethyl Alcohol (1 per work station)	To preserve organisms
o Taxonomy Keys & References	To identify organisms
o Plain white paper	To place under petri plates in scope or on table for contrast
o Sample Processing Record (1 per replicate)	Filled out, to record # of squares picked
o Identification Lab Sheets (1 per sample)	To record identification

* Locate the sample processing record that goes with the sample you're going to identify. There should be one record for each replicate sample. Check to see that the number of squares picked is noted.

Step 3: Mark each petri plate with the site and replicate number

Mark the *bottom* of each petri plate with the site and replicate number of the sample you're processing. Use labeling tape and pencil.

Step 4: Fill in the top of the Benthic Macroinvertebrate Identification Sheet - _Level 2 (Form 7)

Fill in the site #, river/stream, date sampled, your name, and the date of the identification at the top of the form. Be sure to fill in the # of squares picked from the tray boxes at the bottom for each replicate. Note that one identification sheet is used per site -- 3 replicates.

Step 5: Sort the sample into the compartments of the petri plates

If the sample has been sorted, place the contents of each vial into its own compartment. If it hasn't been sorted, rough sort, following step 8 in the previous section.

Step 6: Use the "Simple Picture Key" to identify the families of the organisms in each of the compartments for the orders Diptera, Coleoptera, and Megaloptera

Place the petri plate on the platform of the dissecting scope. Focus the dissecting scope so you can see the whole organism. Hint: It's easier to see body characteristics if the organisms are lit from above against a white background.

Follow the instructions in the picture key ("A Simple Picture Key: Major Groups of Benthic Macroinvertebrates Commonly Found In Freshwater New England Streams") to identify the families.

Move any organisms that don't belong in that compartment to another.

Step 7: Place any organisms you cannot identify into an "unknown" compartment

Place any organisms that you cannot identify using the picture key into a separate petri plate compartment. Don't worry about these for now. In the next step, you'll identify these using another key.

Step 8: Use the family level dichotomous key in "Aquatic Entomology (enclosed) to identify "unknown" organisms and the families in the orders Ephemeroptera, Plecoptera, and Trichoptera

If you are still not sure about a particular organism, try another key or have a more experienced person assist you. If no one is available to help you, preserve it in a vial until someone is available at a later date.

Step 9: Count and record the number of organisms in each family on the Benthic Macroinvertebrate Identification Sheet - Level 2 (Form 7)

When you're certain of the identity of the organisms in all of the compartments (except for those in the unknown compartment), count the total number of individuals ("density") in each family.

Record your count in the 'D' column under the appropriate replicate # on the Benthic Macroinvertebrate Identification Sheet (Families).

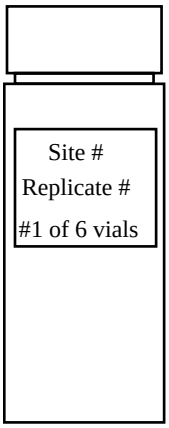
Step 10: Identify the functional feeding group for each family

Identify the functional feeding group (shredders, scrapers/grazers, filtering collectors, gathering collectors, and predators2) for each family. Generalized functional feeding groups for each family are listed on the lab sheet. However, you should verify these suggested groups by using Cummins & Wilzbach's Field Procedures for Analysis of Functional Feeding Groups of Stream Macroinvertebrates to determine the functional feeding group for each family. Note that since we are not identifying the organisms in the field, some of the couplets won't apply. You will need to consult with an aquatic biologist to verify the suggested groups and to identify the functional feeding group of a family that has more than one possible group. Circle the appropriate functional feeding group on the form.

Step 11: Store the sorted organisms in capped vials with alcohol

Transfer the contents of each compartment of the petri plate to its own vial. The contents of several compartments can be combined if there are only a few organisms or if you think they'll be easy to re-sort later. Fill each vial completely with 90% ethyl alcohol and cap tightly. Using labeling tape and a pencil, label each vial like this:

2 See Chapter II "A Closer Look At Benthic Macroinertebrates" for a description of these groups.



Place all unidentified organisms into a separate vial. A more experienced person can identify them at a later date.

Step 12: Fill in the Level 2 - Family Identification box on the Macroinvertebrate Sample Processing Record (Form 5)

Fill in a check, the date, and your name in the Level 2 - Family Identification box on the Macroinvertebrate Sample Processing Record for this replicate.

Step 13: Repeat steps 1-11 for each replicate and record on the Benthic Macroinvertebrate Identification Sheet - Level 2 (Form 7)

Identify the organisms in each replicate sample and record the density and richness in the appropriate column of the Benthic Macroinvertebrate Identification Sheet (Families).

Quality Assurance

The main quality assurance challenge is to make sure that all the organisms are correctly identified. This is particularly important when family identification is involved. To assure this, two measures are recommended:

1) **Voucher Collection:** All processed samples should be saved for later verification by RWN or project staff, the state aquatic biologist (if available) or other professionals. Samples should be stored in labeled vials filled with 90% ethyl alcohol with seals that prevent the alcohol from evaporating. Samples should be checked every few months and the alcohol replenished, if needed.
2) **Reference Collection:** Examples of each family or major group found should be positively identified by an experienced person. These examples should be stored in vials with a label that correctly identifies the organism. This collection is used to compare with the unknown critters from your river samples to help you identify them.

3) **Archive:** For various reasons, you may wish to save the picked and/or unpicked art of your samples, in addition to the critters themselves. There are a number of reasons you might want to do this:
   - **Picked Debris**: As a quality check, you may want an outside professional to go through your picked debris to see if you've missed any critters.
   - **Unpicked Debris:** If you sub-sampled, you will have left over debris (sand, twigs, etc.) containing critters. You may want to save this material for future processing, for example if your lab processing changes to require picking the whole sample.

Suggested Keys

There are many keys to macroinvertebrates. Below is a list of suggested keys to help you identify major groups and families. Some are easier to use than others and each key has its own unique approach. For this reason, it's generally a good idea to have several keys to help you identify the organisms:

Major Groups:

* Dates, Geoff, A Simple Picture Key: Major Groups of Benthic Macroinvertebrates Commonly Found In Freshwater New England Streams, River Watch Network, Montpelier, VT. January 1993.
* Fiske, Steve and Byrne, Jack, Key to the Freshwater Macroinvertebrate Fauna of New England. River Watch Network, Montpelier, VT. March 1988.
* Lehmkuhl, Dennis M., How to Know the Aquatic Insects. William C. Brown Publishers, Dubuque Iowa.
* McCafferty, W. Patrick, Aquatic Entomology: the Fisherman's and Ecologists' Illustrated Guide to Insects and Their Relatives. Jones and Bartlett Publishers, Inc. Boston, MA

Families:

* Cummins, K. and Wilzbach, M. Field Procedures for Analysis of Functional Feeding Groups of Stream Macroinvertebrates.

* Lehmkuhl, Dennis M., How to Know the Aquatic Insects. William C. Brown Publishers, Dubuque Iowa. 1979.

* McCafferty, W. Patrick, Aquatic Entomology: the Fisherman's and Ecologists' Illustrated Guide to Insects and Their Relatives. Jones and Bartlett Publishers, Inc. Boston, MA

* Merritt, R. and Cummins, K. An Introduction to the Aquatic Insects of North America (Second Edition). Kendall / Hunt Publishing Company, Dubuque Iowa. 1984.

* Peckarsky, Barbara L., et al, Freshwater Macroinvertebrates of Northeastern North America. Cornell University Press. 1990

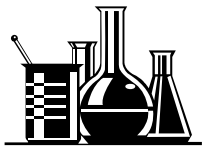
* Pennak, Robert W., Freshwater Invertebrates of the United States, 3rd Edition, Wiley-interscience Publication. 1987

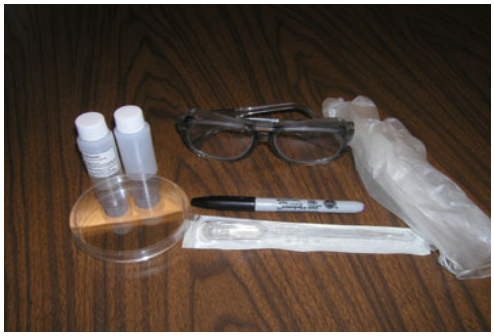
* Smith, Douglas G., Key to the Freshwater Macroinvertebrates of Massachusetts, University of Massachusetts, Amherst MA 1991.

* Wiggins, G.B. Larvae of the North American Caddisfly Genera (Trichoptera), University of Toronto Press, Toronto Canada. 1977.

Coliscan Easygel Protocol Appendix I

What You Need:





- 1 Petri Dish
- 1 100 ml, small plastic bottle
- 1 ml dropper
- 1 bottle of Coliscan
- 1 waterproof marker
- Scotch or masking tape
- Safety Glasses
- Gloves
- Laminated Color Guide

ALSO DON'T FORGET

NO SANDALS

Here is what you need to do:

1. Put on safety glasses and gloves.

2. Label the top of the petri dish using a permanent marker. Use very small letters around the edges so that you don't block the view of the bacterial growth.

Make sure to include the location of the site(use abbreviations if possible), date, time, and your initials. Also, include the number of ml used. Check with your teacher for this number*.

3. Collect a water sample in the small, plastic, collection bottle.

4. Before continuing, be sure the Coliscan is completely thawed and in liquid form.

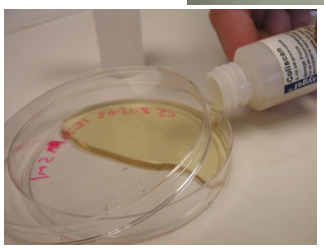
5. Open the 1 mL dropper's plastic package near the bulb of the dropper, making sure to only handle the bulb.

6. Open the containers of the water sample and Coliscan. Place the caps of both containers top down.

7. Using the 1.0 mL dropper, transfer 1.0-5.0 mL of water from the collection bottle to the bottle of Coliscan.

8. Tightly twist the caps back on both containers.

9. Gently swirl the mixture containing both the Coliscan and water sample.



10. Open the petri dish just enough to pour the contents of the mixture into the dish, then immediately put the lid back on the dish.

11. Gently swirl the petri dish until it is entirely covered with liquid (be careful not to splash over the side or on the lid).

12. Leave the dish undisturbed until the mixture becomes a solid gel(approximately 10 minutes). Once the gel is solid, tape the lid of the petri dish down.

13. Incubate at 35° for 24 hours or at room temperature for 48 hours.

14. Inspect the dishes using the Coliscan Easygel Colony Color Guide. Count purple colonies and consult with your teacher to determine the number by which to multiply.

15. Do the following prior to disposal in normal trash:
    - Place about 5 mL (about 1 teaspoon) of straight bleach onto the surface of the medium of each plate. Allow to sit at least 5 minutes. Place in water tight bag and discard in trash.

* If you use 1.0 mL, you will multiply the end result by 100 to get colonies bacteria/100mL water. If you use 5.0 mL, you will multiply by 20 to get colonies/100mL.

E.coli Quanti-Tray Testing Procedure

- Insert thermometer through grommet located at top center of incubator to a position half an inch above middle shelf.
- Plug in incubator and turn dial to ~ 4.75. Incubator is ready when stable at 35°C (~ 15 minutes).

- Turn sealer on. The amber light should illuminate. Allow sealer to warm up and green ready light to come on (up to 10 minutes).

- Attach input shelf to sealer.
- Place an empty Quanti-Tray Rubber Insert on input shelf with large cut out facing away from the sealer.
- Obtain water sample to 100 ml line in sterile vessel.
- Aim Colilert snap pack into 100 ml vessel.

- Open Colilert at score line and add contents to water sample.
- Cap (being careful not to touch the inside of the cap or jar) and shake until dissolved.

- Label back of Quanti-Tray with name, date and location and follow steps 1-5 (below) to fill Quanti-Tray with water sample:

- Place sealed Quanti-Tray in rubber insert, making sure that the tray is properly seated in the rubber insert with each well in its corresponding rubber insert hole.

- Slide the rubber insert with Quanti-Tray into the sealer until the motor grabs the rubber insert and begins to draw it into the sealer.

- In approximately 15 seconds, the Quanti-Tray will be sealed and partially ejected. Remove from the sealer.

- Turn sealer off.

- Once Quanti-Tray is sealed it is ready to be placed in incubator for 24 hours at 35°C.

Note: Results are valid at 24-28 hours.

Result Interpretation

- Look for fluorescence by holding the UV light 5 inches away from the Quanti-Tray in a dark environment. Be sure to hold the light away from your eyes and toward the sample.

- Count the number of small and large wells that fluoresce separately and use the 'Idexx Quanti-Tray/2000 MPN Table' to determine the 'Most Probable Number'.

Appearance	Result
Less yellow than the Comparator	Negative for total coliforms and E.coli
Yellow equal to or greater than the comparator	Positive for total coliforms
Yellow and fluorescence equal to or greater than the comparator	Positive for E.coli

Disposal of Quanti-Trays

Since E.coli will continue to grow if not disposed of properly, all Quanti-Trays must be returned to UVM.

COMMUNITY OUTREACH—Taking Action Appendix J

The Watershed Alliance requires that all groups participating in our watershed education program complete a community outreach project (called "Taking Action"). Community outreach is an essential part of the Watershed Alliance program because it integrates the important information the students learned into a service-learning component.

Students are proud to share the results of their hard work with parents, peers, community members, and local officials. Students learn about civic participation and the importance of community involvement. In addition, community members are more readily influenced by students in their own community than by outside "experts" presenting the same information.

The outcomes of the Watershed Alliance watershed education program are multiplied many times by the effort of the students, who by far outnumber the educators enlisted with us. This "multiplication effect" is essential to our efforts as educators in strengthening the environmental knowledge base and positive community involvement in Vermont.

Therefore, student led community outreach projects are an effective way to spread important information about watersheds.

Ideas for **Taking Action** projects:

- Presentation of data/projects to a local conservation board/commission/district, town planners, local watershed organization, parents, peers, students in the same school, and/or students in a neighboring school
- Educational outreach and mentoring—hands on learning day with younger students
- Create a slide show (powerpoint or photo) for presentation
- Create a video
- Write/produce a play
- Produce a radio ad and air it on local radio station
- Hold an informational poster campaign in school/community
- Hold a community watershed forum with speakers and open communication session
- Hold a watershed speaker series followed by presentation of student data/project
- Hold a watershed science fair

These are just a few ideas. The UVM WA recommends you hold a brainstorming session to come up with student-led project ideas. Generally, it is a good idea to decide which community outreach project you will be completing at the same time as the study deign is finalized, as the design may be affected.

It is useful to alert the local press to the river study. Many of our past participants have been featured in newspaper articles. Contact your local newspaper (press contact list included on following page) to inform them of your river study and Taking Action project.

Examples of Past/On-Going **Taking Action** Projects:

- Gay Craig and her Champlain Valley Union students (9/10th grade) present an annual report to the Lewis Creek Association (LCA). The students work closely with LCA to ensure that the data collected is useful to them.

- Websterville Baptist Christian School students, led by teacher Virginia Collins, produced an educational radio ad about their Winooski River study.

- During a Watershed Alliance project, students in a local school discovered E.Coli bacteria in the school water, and presented the data to town officials. This led to a "boil water" notice for the entire town, and the repair of the problem.

- Students at Danby's Currier Memorial School, led by Carrie Mauhs-Pugh, are working with local farmers on an on-going yearly study of a stream near the farm. The students plan to present the findings to the farmers along with suggested Best Management Practices.

- Ninth and tenth grade students at the SUCCESS School (an alternative school in Rutland) worked in conjunction with the VT Department of Environmental Conservation and the Rutland Natural Resource Conservation District on a study of the Moon Brook. The Moon Brook, located in Rutland city, is included on the state's 303 (d) list of impaired waters. The Upper Otter Creek Watershed Council (UOCWC--a group of local officials, experts, watershed organizations, and concerned citizens formed to address issues in the upper Otter Creek watershed) is interested in the data the students collected. Students plan to present their study at an UOCWC meeting.

- At Union -32 High School in Montpelier, Brian Slopey's twelfth grade Environmental Science students acted as educators and mentors to students from the middle school. Slopey's students designed hands-on projects that engaged the younger students as they taught them about watersheds and water quality. Slopey praised the students and the project, noting that his students benefited from the experience as much as the middle school students.

Lake Champlain Basin Atlas Treasure Hunt Appendix L

Northern Cartographic created the Lake Champlain Basin Program Atlas for the Lake Champlain Basin Program. The Atlas contains 40 different maps of the Lake Champlain Basin, and provides information about many relevant topics. This activity is an introduction to using the atlas and to the information it contains. The atlas is also online at www.lcbp.org.

Getting Started
Double click on My Computer, then on the CD drive, then on the **Index.html** icon.
This is the front page of the Atlas. Now click anywhere on the screen to open the Atlas…

Introduction to the Atlas
First, click on the Using the Atlas link in the second paragraph. Read the directions.
Click on the Map Index link.
Name three topics from the Issues in the Basin menu_____

_____

_____

What four topics start with the same letter in the Nature of the Basin menu?

_____

_____

_____

_____

What is the most interesting topic to you and/or your group in the Socio-Economic menu?

_____

Climate
From the map index page, highlight **Climate** under **Nature of the Basin** and click GO. Read the text.

Estimate the average annual precipitation in your area_____
(Hint: open up both the political boundaries map and the climate map. To do this in Microsoft Explorer, click on File, then New…Window. In new window, go to map index and find Political Boundaries map)

Is it higher, lower or the same as areas directly adjacent to the lake?_____

BONUS**Why is precipitation higher/lower/the same in your area?_____

_____

_____

Which Watershed Do I Live in?

From the Map Index page, highlight the link to the **Sub-basin and Tributaries** watershed in which you live (under **Nature of the Basin** menu) and click GO. Read the text. *If you are not sure which sub basin you live in, ask your teacher*.

How many hectares are drained by your watershed?_____

BONUS**How many acres are in a hectare?_____

Find one watershed group located in your Sub basin (hint—click on the Learn More link)

_____

Water Quality Monitoring
From the Map Index page, highlight the **water quality monitoring** page under the **Issues in the Basin** menu and click GO. Read the text.

Locate the two biological (first map) monitoring sites closest to the outlet of the Otter Creek.
What is being monitored there?_____

Who is collecting the data?_____

In your internet browser, go to GOOGLE www.google.com. Type in the name of the organism(s) being monitored.
Tell me more about the organism(s)_____

Bonus**Why do you think they are being studied?_____

_____

_____

_____

Transportation
From the Map Index page, choose the **Transportation** link under the **Socio-Economic** menu and click GO. Read the text.

Why were waterways (rivers and lakes) so important for transportation in the pre-settlement and early settlement periods?_____

_____

_____

You are on a mission to reach the Atlantic Ocean by canoe. Assuming you had very strong arms and plenty of food and water on board, could you get there from your town with out leaving the water (except to go around dams, etc)? (hint: see Lake Champlain Region map and Rivers and Streams map)_____

Outline your travel path—tell me which water bodies you would travel on, and for approximately how many miles_____

_____

_____

_____

_____

_____

_____

_____

If you traveled 10 miles per day, how many days would it take you?_____

For those with really strong arms, how many days would it take if you paddled 15 miles per

day?_____