VT EPSCoR Research on Adaptation to Climate Change (RACC) in the Lake Champlain Basin High School Program 2015-16

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Introduction:

Established in 2011, the VT EPSCoR CWDD is one of two centers funded by the National Science Foundation and created through the Research on Adaptation to Climate Change in the Lake Champlain Basin (RACC) award. RACC is focused on understanding the effects of changing climate on the Lake Champlain Basin and to develop adaptive management strategies for the Basin.

RACC builds transdisciplinary teams of social and natural scientists to study the Lake Champlain Basin as a coupled human and natural system affected by climate change. We combine collections of data on physical processes, governance, and land use with complex systems modeling. Models will enable scenario testing to help Basin managers and policy makers investigate how adaptive management can be designed and implemented to respond to climate change.

CWDD increases the Vermont Science-Technology-Engineering-Math (STEM) workforce in size and diversity through multiple approaches:

- Inspire diverse high school students and undergraduates to enter STEM careers by involving them directly in RACC research. Support the professional development of high school and middle school teachers through involving them in RACC research.

- Match high school teams, undergraduates and middle school teachers with RACC social and natural scientists, who will act as research mentors.
- Target support for girls and underrepresented minorities, veterans, economically disadvantaged high school students, and students with disabilities.
- Involve students from Puerto Rico, New York, Massachusetts, Maryland, Texas and other locations outside Vermont to bring a diverse pool of participants into the STEM pipeline.
- Cap off the year with at the VT EPSCoR Student Research Symposium where CWDD participants share research results and network with other STEM professionals.
- Support Native American and First Generation Vermont college students through scholarships to study STEM majors in Vermont.
- Enable the Governor's Institutes of Vermont (GIV) to reach out to every high school in Vermont with scholarships so that girls and economically disadvantaged students can attend the STEM summer institutes and Winter Weekends.
- Work with the Vermont Technology Council to connect undergraduates and small technology businesses that provide students with paid internships.

Research on Adaptation to Climate Change in the Lake Champlain Basin (RACC):

The RACC center is organized around an overarching theme with three research hypothesis-driven questions, involving a diversity of scientists and engineers from academia and the private sector that are integrated with public and private stakeholders, undergraduates, middle school teachers, and high school students and teachers. They will study climate change-driven impacts on hydrological processes and nutrient transport in the lake basin (Questions 1 and 2), and develop ecosystem assessment scenarios and models to inform the work of policymakers (Question 3 and Integrated Assessment Model (IAModel)).

Overarching Question**:** How will the interaction of climate change and land use alter hydrological processes and nutrient transport from the landscape, internal processing and eutrophic state within the lake and what are the implications for adaptive management strategies?

Question 1: What is the relative importance of endogenous in-lake processes (e.g., internal loading, ice cover, hydrodynamics) versus exogenous to-lake processes (e.g., land use change, snow/rain timing, storm frequency and intensity, land management) to lake eutrophication and algal blooms?

Question 2: Which alternative stable states can emerge in the watershed and lake resulting from non-linear dynamics of climate drivers, lake basin processes, social behavior, and policy decisions?

Question 3: In the face of uncertainties about alternate climate change, land use and lake response scenarios, how can adaptive management interventions (e.g., regulation, incentives, treaties) be designed, valued and implemented in the multi-jurisdictional Lake Champlain Basin?

For more information visit: www.uvm.edu/~epscor

2015-2016 High School Program:

The CWDD supports high school teams interested in engaging in RACC research as either Streams Project teams or Social Science Teams. This year will be the eighth year of the VT EPSCoR Streams Project. Each year, the project changes to align with the needs of the overall research program.

Goal: Increase the number and diversity of high school students interested in STEM careers.

Objectives:

- Students and teachers experience active research;
- Students and teachers develop scientific field and lab knowledge and skills;
- Students make connections with college science faculty, programs, and campuses.

Strategies:

- Train students and teachers in watershed ecology, climate change, systems thinking, and field and lab skills during residential training week.
- Task high school teams with collecting high quality data for the VT EPSCoR research project Research on Adaptation to Climate Change (RACC).
- Convene a Symposium for presentations of RACC research progress, an opportunity for students to experience presenting scientific research, and a venue for students to see where their efforts fit into the overall research program.

Streams Project High School Teams

2015-2016

Manual Contents

About this manual:

- Become familiar with it at the outset of your participation.
- Use the "Team Project" section in the back of the manual to keep track of your research
- Use this in conjunction with the Streams Project website (www.uvm.edu/epscor/highschool) which hosts a wealth of additional resources:
    - a searchable PDF of this manual
    - data analysis tutorials
    - mapping and site information
    - searchable database
    - links to useful websites
    - presentation and symposium information

Email cwdd@smcvt.edu if you need assistance. Your message will be directed to the appropriate staff member.

RACC
Research on Adaptation to Climate Change

REFERENCE MANUAL FOR HIGH SCHOOL TEAMS 2015-2016

Site and Habitat Assessment

Contents

Site and Habitat Assessment General Info

There are several components to site and habitat assessment:

- Determining your site's waypoints
- Naming your site
- Stream Site General Assessment
- Habitat Assessment

Site and Habitat Assessment Field Checklist

- Data sheets
- Pencils and permanent marker
- Camera
- GPS
- Topographic maps/aerial photos
- Meter tape
- Meter stick
- Plant identification keys

Site Selection Criteria

- Stream Project sites represent a variety of land uses ranging from predominantly urban and/or agricultural drainage areas to mostly forested drainage areas. Land use can be gauged by looking at the land use upstream and surrounding a potential monitoring site.

- Your stream sites should be 2nd or 3rd order streams.

- Avoid meanders, and look to areas with riffles.

- Make sure that you have easy access to the stream sites. They should be not too far from a road, and should not require crossing private property. If accessing your site requires parking or walking through private property, always contact the landowner to request permission to access the site.

- Initial site selection will take place during high school training week using Google Maps. Sites will be confirmed upon initial field visit.

Stream Site Naming Convention

It is very important that any site you will be sampling for the Streams Project has been given a stream site name with the naming convention outlined here. The following outlines the general procedure for getting your stream site names and using them:

1. First you will work with Streams Project staff to take a GPS waypoint at your stream site along with an elevation measurement in feet. See the next section for details on this.

2. Based on abbreviations for the larger watershed your stream site falls into, the name of your stream, and the elevation measurement gathered in the field, you will be given a stream site code with the following format:

   The stream site code for Munroe Brook in the Lake Champlain Direct HUC8 watershed, with a GPS field elevation reading of 500 feet will look like this:

   SITE CODE **= LCD_MuBr_500**

3. It is ***very important*** that you make sure to use your designated stream site codes on ***ALL*** your field data records, and on ***ALL*** samples you send to either the water quality or macroinvertebrate labs. You will also need this code to upload data into the Streams Project database. Please don't collect/send in samples before your stream site code has been created and emailed to you.

GPSing your Stream Site

Before you begin sampling, you will work with Streams Project staff to collect the latitudes and longitudes of your monitoring sites with a GPS unit. During this field visit you will also collect the elevations of your monitoring sites in feet.

This is an important first step to be taken before sampling begins for the following reasons:

- We need to know where your stream sites are so that we can get you site codes.

- We need to know the exact location of your sites in order to delineate your watersheds. You can use these delineations to explore your watersheds in the GoogleEarth activity mentioned in the next section. This delineation also allows us to provide you with environmental and land use information that you may use in your final analysis.

Watershed Exploration

The following is an activity to be completed with your team on your own time to help you get a better sense of where your sites are and what land drains into the streams you are monitoring.

Materials:

- Computer with GoogleEarth. If a computer with GoogleEarth is not available at your school, you can use Google Maps.
- Two GoogleEarth files (.kml) sent to you by Streams Project staff: one of your monitoring sites, the other of the area of land that drains to your monitoring site – its watershed.

Instructions:

1. Email cwdd@smcvt.edu to receive your .kml files
2. Open both .kml files in GoogleEarth.
3. Zoom in and out to explore the area of land delineated by the watershed file
4. Answer the following questions about your two watersheds:

A. What human activity if any can you identify in each watershed?
B. How does the land use activity differ between your two watersheds?
C. How might the land use you've identified affect the in-stream water quality?
D. Which monitoring site do you think would have better water quality? Why?

Field Method: Stream Site Assessment

Fill out the "Stream Site General Assessment Data Sheet" once for each of your stream sites. Consider revisiting the Site Assessment if you notice major changes in your stream site (for instance after Tropical Storm Irene in 2011).

Thalweg Measurement

The thalweg is an imagined line following the deepest part of a stream. Importantly, it is not a line down the center of the stream (although in an artificially modified stream, it may be). It typically represents the point where most water flows through each cross-section of river. Measuring the variability of the thalweg gives one indication of the diversity of habitats available to fish and other members of stream communities. A highly channelized stream for example, might have a thalweg of relatively uniform depth. A non-manipulated stream of intermediate gradient would be expected to have a series of pools and riffles and therefore a highly variable thalweg. Channelized streams typically sustain far less diverse communities than more natural streams that have pool/riffle sequences. Natural variability in thalweg can be expected as streams vary in gradient.

Measuring thalweg in your study streams provides a measure of habitat variability. If the thalweg is relatively constant then it indicates little variability in depth, and water velocity. Streams where thalweg varies little can also be expected to have fewer eddies, slack water habitats, riffles, snags, and large woody debris. We might reasonably anticipate reduced macroinvertebrate diversity and less in-stream processing of particles and nutrients in streams with invariant thalweg measurements.

If thalweg measurements are planned for the same day as any sampling (water or macroinvertebrate) then do the sampling first and complete the measurements later. If you have enough team members to do all of the work at the same time then it is important that sampling happens upstream of other activities.

Thalweg Measurement Procedure:

1. Lay a meter tape along the stream to identify 100 sites 1 meter apart. Measurements can start at your sampling site and proceed 50m upstream and 50m downstream. If it is unsafe to make the measurements as just described then take measurements from the nearest safe 100m stretch and explain in the notes section.

2. At each 1m interval, locate the deepest part of the stream cross section.

3. Use a meter stick to measure the depth at the deepest point
   It is worth noting that the deepest point will only sometimes be in the center of the stream and can shift unpredictably. This is an important concept to emphasize and one of the reasons that it is considered unsafe to venture onto frozen streams and rivers.

Stream Site General Assessment Data

2015-2016

STREAM NAME:	**TOWN:**
DATE:	**LATITUDE:**
TIME:	**LONGITUDE:**
STREAM GRADIENT (HIGH OR LOW):	**RIVER BASIN:**
SITE DESCRIPTION:	**INVESTIGATORS:**

Stream sketch: On your sketch, note features that affect stream habitat, such as: riffles, runs, pools, ditches, wetlands, dams, riprap, outfalls, tributaries, landscape features, logging paths, vegetation and roads.

Watershed features	Location of stream headwaters (Town name): Predominant Surrounding Landscape: (circle one) Forest Field/Pasture Agricultural Commercial Residential Industrial Other (If other, please specify) Local Watershed non-point pollution (circle one): No evidence Some potential sources Obvious sources Please explain:
Stream reach characteristics *Enter from GIS Assessment Report if not measurable in the field	Bank full width (meters): Reach length (meters): Channelized? ____ *Upstream Dam: if Yes, ____ km upstream from site Other modifications: Bridge: Within Reach: Yes or No * Upstream: Yes or No if yes, how far? ___m Culvert: Within Reach: Yes or No * Upstream: Yes or No if yes, how far? ___m Pipes: Within Reach: Yes or No *Upstream: Yes or No if yes, how far? ___m *Distance of site from tributary mouth/main river channel: _____ km
Riparian vegetation (within 18 meters)	Width of vegetated riparian zone (looking downstream): Left bank_____ m Right bank_____m (estimated or measured?) Indicate the dominant type and record the dominant species present (circle one): Trees Shrubs Grasses Herbaceous None Dominant species (if known):
Large woody debris	Abundance of LWD (# logs ≥ 10 cm diameter in stream reach): Length of reach measured:_____m
Aquatic vegetation	Indicate the dominant type and record the dominant species present (circle one): Rooted emergent rooted submergent floating algae attached algae rooted floating free floating Portion of the reach with aquatic vegetation: _____ %
Sediment substrate	Odors (circle one): Normal Sewage Petroleum Chemical Sulfur None Other: Oils (circle one): Absent Slight Moderate Profuse
Water quality in channel	**Circle all that apply:** Debris Obvious Pollution: Sludge, Sawdust, Paper Fiber, Sand, Silt, Sewage, Oily Sheen, Trash, Iron, Scum, None Water Clarity**:** Clear, Slightly Turbid, Moderately Turbid, Very Turbid Water Color**:** Clear, Green, Milky, Brown, Tannic (L M H), Gray, Metallic, Reddish Odors**:** None, Musty, Fishy, Sewage, Manure, Sulfur(eggs), Oily/gas

Local Land Use (within about ¼ mile of site; adjacent and upstream)
Check "1" if present, "2" if clearly having an impact on a stream.

0	1	2	
			Residential
☐	☐	☐	Single-family housing
☐	☐	☐	Multi-family housing
☐	☐	☐	Lawns
☐	☐	☐	Commercial/Institutional
0	**1**	**2**	**Roads, etc.**
☐	☐	☐	Paved roads or bridges
☐	☐	☐	Unpaved roads
0	**1**	**2**	**Construction underway on:**
☐	☐	☐	Housing development
☐	☐	☐	Commercial development
☐	☐	☐	Road bridge construction/repair
0	**1**	**2**	**Agricultural**
☐	☐	☐	Grazing Land
☐	☐	☐	Feeding lots or animal holding areas
☐	☐	☐	Cropland
☐	☐	☐	Inactive agricultural land/fields
0	**1**	**2**	**Recreation**
☐	☐	☐	Power boating
☐	☐	☐	Golfing
☐	☐	☐	Camping
☐	☐	☐	Swimming/fishing/canoeing
☐	☐	☐	Hiking/paths
0	**1**	**2**	**Other**
☐	☐	☐	Mining or gravel pits
☐	☐	☐	Logging
☐	☐	☐	Industry
☐	☐	☐	Oil and gas drilling
☐	☐	☐	Trash dump
☐	☐	☐	Landfill

Comments:

Thalweg Measurement **Site Code: _____** **Date: _____**

Upstream distance from starting point (m)	Thalweg depth (cm)
0	
1	
2	
3	
4	
5	
6	
7	
8	
9	
10	
11	
12	
13	
14	
15	
16	
17	
18	
19	
20	
21	
22	
23	
24	
25	

Upstream distance from starting point (m)	Thalweg depth (cm)
26	
27	
28	
29	
30	
31	
32	
33	
34	
35	
36	
37	
38	
39	
40	
41	
42	
43	
44	
45	
46	
47	
48	
49	
50	

Downstream distance from starting point (m)	Thalweg depth (cm)
0	
1	
2	
3	
4	
5	
6	
7	
8	
9	
10	
11	
12	
13	
14	
15	
16	
17	
18	
19	
20	
21	
22	
23	
24	
25	

Downstream distance from starting point (m)	Thalweg depth (cm)
26	
27	
28	
29	
30	
31	
32	
33	
34	
35	
36	
37	
38	
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40	
41	
42	
43	
44	
45	
46	
47	
48	
49	
50	

Comments:

Habitat Assessment Data Sheet (2015-2016)

STREAM NAME:	**SITE CODE:**
DATE/TIME:	**INVESTIGATORS:**
LATITUDE	**LONGITUDE:**
SITE DESCRIPTION:	**WEATHER CONDITIONS:**

	Habitat Parameter	Reference	Good	Fair	Poor
1	**Epifaunal Substrate/Cover**	Greater than 70% (50% for low gradient streams) of stream bed and lower banks covered with mix of substrates favorable for epifaunal colonization and fish cover; substrates include snags, submerged logs, undercut banks, and un-embedded cobbles and boulders (for high gradient)	40-70% (30-50% for low gradient streams) of stream bed and lower banks covered with a mix of substrates favorable for epifaunal colonization and fish cover	20-40% (10-30% for low gradient streams) of stream bed and lower banks covered with substrates favorable for epifaunal colonization and fish cover; few substrate types present	Less than 20% (10% for low gradient streams) of stream bed and lower banks covered with substrates favorable for epifaunal colonization and fish cover; few substrate types present
		20 19 18 17 16	**15 14 13 12 11**	**10 9 8 7 6**	**5 4 3 2 1**
2a	**Embeddedness (high gradient)**	Gravel, cobble, and boulder particles are 0-25% surrounded by fine sediment. Layering of cobble provides diversity of niche space.	Gravel, cobble, and boulder particles are 25-50% surrounded by fine sediment.	Gravel, cobble, and boulder particles are 50-75% surrounded by fine sediment. Little open space between particles.	Gravel, cobble, and boulder particles are more than 75% surrounded by fine sediment. Almost no open space between particles.
		20 19 18 17 16	**15 14 13 12 11**	**10 9 8 7 6**	**5 4 3 2 1**
2b	**Pool Substrate Characterization (low gradient)**	Characterization (low gradient) Mixture of substrate materials, with gravel and firm sand prevalent; root mats and submerged vegetation common.	Mixture of soft sand, mud, or clay; mud may be dominant; some root mats and submerged vegetation present.	All mud or clay or sand bottom; little or no root mat; no submerged vegetation.	Hard-pan clay or bedrock; no root mat or vegetation.
		20 19 18 17 16	**15 14 13 12 11**	**10 9 8 7 6**	**5 4 3 2 1**
3a	**Velocity/Depth Patterns (high gradient)**	All 4 velocity/depth patterns present: slow-deep, slow-shallow, fast-deep, fast-shallow. Slow is < 1 ft/s. (0.3 m/s), deep is > 1.5 ft/s (0.5 m/s).	Only 3 of the 4 patterns present (if fast-shallow is missing, score lower than if missing other regimes).	Only 2 of the 4 patterns present (if fast-shallow or slow-shallow are missing, score low).	Dominated by 1 velocity/ depth pattern (usually slow-deep).
		20 19 18 17 16	**15 14 13 12 11**	**10 9 8 7 6**	**5 4 3 2 1**
3b	**Pool Variability (low gradient)**	Even mix of large-shallow, large-deep, small-shallow, small-deep pools present.	Majority of pools large-deep; very few shallow.	Shallow pools much more prevalent than deep pools.	Majority of pools small-shallow or pools absent.
		20 19 18 17 16	**15 14 13 12 11**	**10 9 8 7 6**	**5 4 3 2 1**

4	**Sediment Deposition**	Little or no enlargement of mid-channel bars or point bars and < 5% (20% in low gradient streams) of the bottom affected by sediment deposition.	Some new increase in bar formation, mostly from gravel, sand or fine sediment; -30% (20-50% in low gradient streams) of the bottom affected; slight deposition in pools.	Moderate deposition of new gravel, sand or fine sediment on old and new bars; 30-50% (50-80% in low gradient streams) of the bottom affected; sediment deposits at obstructions, constrictions, and bends; moderate deposition of pools prevalent.	Heavy deposits of fine material, increased bar development; > 50% (80% in low gradient streams) of the bottom changing frequently; pools almost absent due to substantial sediment deposition.
		20 19 18 17 16	**15 14 13 12 11**	**10 9 8 7 6**	**5 4 3 2 1**
5	**Channel Flow Status**	Water reaches base of both lower banks, and <10% of channel bed substrate is exposed.	Water fills >75% of the available channel or <25% of channel bed substrate is exposed.	Water fills 25-75% of the available channel, and/or riffle substrates are mostly exposed.	Very little water in channel and mostly present as standing pools.
		20 19 18 17 16	**15 14 13 12 11**	**10 9 8 7 6**	**5 4 3 2 1**
6	**Channel Alteration**	Channelization in the form of dredging, straightening, berms or stream bank armoring absent; stream with natural pattern.	Some channel alterations present along 10-20% of segment, usually in areas of bridge abutments; evidence of past channelization, (greater than past 20 yrs.) may be present, but recent channelization is not present.	Channelization along 20-80% of stream segment; riprap or armoring present on both banks.	Over 80% of the stream segment channelized and disrupted. Instream habitat greatly altered or removed entirely.
		20 19 18 17 16	**15 14 13 12 11**	**10 9 8 7 6**	**5 4 3 2 1**
7a	**Frequency of Riffles/Steps (high gradient)**	Occurrence of riffles/steps relatively frequent; distance between riffles is 5-7 times (steps 3-5 times) stream width; variety of habitat is key. In streams where riffles/steps are continuous, presence of boulders or other large, natural obstruction is important.	Occurrence of riffles/steps infrequent; distance between riffles is 7-15 times (steps 5-15 times) stream width.	Occasional riffle/step or bend; bottom contours provide some habitat; distance between riffles/steps is 15 to 25 stream widths.	Generally all flat water or shallow riffles/steps; poor habitat; distance between riffles/steps is >25 stream widths. Mostly runs.
		20 19 18 17 16	**15 14 13 12 11**	**10 9 8 7 6**	**5 4 3 2 1**
7b	**Channel Sinuosity (low gradient)**	The bends in the stream increase the stream length 2.5 to 4 times longer than the straight down-valley length.	The bends in the stream increase the stream length 1.5 to 2.5 times longer than the straight down-valley length.	The bends in the stream increase the stream length 1 to 1.5 times longer than the straight down-valley length.	Channel straight; waterway has been channelized for a long distance.
		20 19 18 17 16	**15 14 13 12 11**	**10 9 8 7 6**	**5 4 3 2 1**

8	**Bank Stability (score each bank)** *Note: determine left or right side by facing downstream.*	Banks stable; evidence of erosion or bank failure absent or minimal; < 5% of bank affected.	Moderately stable; infrequent, small areas of erosion mostly re-vegetated. 5-30% of bank in segment (or reach) has areas of erosion.	Moderately unstable; 30-60% of bank in segment (or reach) has areas of erosion; high erosion potential from crumbling, un-vegetated banks during floods.	Unstable; many eroded areas; "raw" areas frequent along straight sections and bends; obvious bank sloughing; 60-100% of bank has erosional scars.
	Score _____ (LB)	**Left bank 10 9**	**8 7 6**	**5 4 3**	**2 1 0**
	Score _____ (RB)	**Right bank 10 9**	**8 7 6**	**5 4 3**	**2 1 0**
9	**Bank Vegetation (score each bank)** *Note: determine left or right side by facing downstream.*	More than 90% of the stream bank surfaces and immediate riparian zone covered by native vegetation, including trees, understory shrubs, or herbaceous vegetation; vegetative disruption through grazing or mowing minimal or not evident; almost all plants allowed to grow naturally.	70-90% of the stream bank surfaces covered by native vegetation, but one class of plants is not well-represented; disruption evident but not affecting full plant growth potential to any great extent; more than one-half of the potential plant stubble height remaining.	50-70% of the stream bank surfaces covered by vegetation; disruption obvious; patches of bare soil or closely cropped vegetation common; less than one-half of the potential plant stubble height remaining.	Less than 50% of the stream bank surfaces covered by vegetation; disruption of stream bank vegetation is very high; vegetation has been removed to 5 centimeters or less in average stubble height.
	Score _____ (LB)	**Left bank 10 9**	**8 7 6**	**5 4 3**	**2 1 0**
	Score _____ (RB)	**Right bank 10 9**	**8 7 6**	**5 4 3**	**2 1 0**
10	**Riparian Vegetative Zone Width (score each side of channel)** *Note: determine left or right side by facing downstream.*	Width of naturally vegetated riparian zone >100 ft.; human activities, (i.e., parking lots, roadbeds, clear-cuts, lawns, or crops) and grazing have not impacted zone.	Width of riparian zone 50 - 100 ft.; human activities and grazing have impacted zone only minimally.	Width of riparian zone 25 - 50 ft.; human activities and grazing have impacted zone a great deal.	Width of riparian zone < 25 feet: little or no riparian vegetation due to human activities.
	Score _____ (LB)	**Left bank 10 9**	**8 7 6**	**5 4 3**	**2 1 0**
	Score _____ (RB)	**Right bank 10 9**	**8 7 6**	**5 4 3**	**2 1 0**
	HABITAT SCORE	**Sum of score for all 10 categories for stream type (high or low gradient): _____ x 100 = 200**			

Comments:

5 HABITAT ASSESSMENT AND PHYSICOCHEMICAL PARAMETERS

An evaluation of habitat quality is critical to any assessment of ecological integrity and should be performed at each site at the time of the biological sampling. In general, habitat and biological diversity in rivers are closely linked (Raven et al. 1998). In the truest sense, "habitat" incorporates all aspects of physical and chemical constituents along with the biotic interactions. In these protocols, the definition of "habitat" is narrowed to the quality of the instream and riparian habitat that influences the structure and function of the aquatic community in a stream. The presence of an altered habitat structure is considered one of the major stressors of aquatic systems (Karr et al. 1986). The presence of a degraded habitat can sometimes obscure investigations on the effects of toxicity and/or pollution. The assessments performed by many water resource agencies include a general description of the site, a physical characterization and water quality assessment, and a visual assessment of instream and riparian habitat quality. Some states (e.g., Idaho DEQ and Illinois EPA) include quantitative measurements of physical parameters in their habitat assessment. Together these data provide an integrated picture of several of the factors influencing the biological condition of a stream system. These assessments are not as comprehensive as needed to adequately identify all causes of impact. However, additional investigation into hydrological modification of water courses and drainage patterns can be conducted, once impairment is noted.

The habitat quality evaluation can be accomplished by characterizing selected physicochemical parameters in conjunction with a systematic assessment of physical structure. Through this approach, key features can be rated or scored to provide a useful assessment of habitat quality.

5.1 PHYSICAL CHARACTERISTICS AND WATER QUALITY

Both physical characteristics and water quality parameters are pertinent to characterization of the stream habitat. An example of the data sheet used to characterize the physical characteristics and water quality of a site is shown in Appendix A. The information required includes measurements of physical characterization and water quality made routinely to supplement biological surveys.

Physical characterization includes documentation of general land use, description of the stream origin and type, summary of the riparian vegetation features, and measurements of instream parameters such as width, depth, flow, and substrate. The water quality discussed in these protocols are *in situ* measurements of standard parameters that can be taken with a water quality instrument. These are generally instantaneous measurements taken at the time of the survey. Measurements of certain parameters, such as temperature, dissolved oxygen, and turbidity, can be taken over a diurnal cycle and will require instrumentation that can be left in place for extended periods or collects water samples at periodic intervals for measurement. In addition, water samples may be desired to be collected for selected chemical analysis. These chemical samples are transported to an analytical laboratory for processing. The combination of this information (physical characterization and water quality) will provide insight as to the ability of the stream to support a healthy aquatic community, and to the presence of chemical and non-chemical stressors to the stream ecosystem. Information requested in this section (Appendix A-1, Form 1) is standard

to many aquatic studies and allows for some comparison among sites. Additionally, conditions that may significantly affect aquatic biota are documented.

5.1.1 Header Information (Station Identifier)

The header information is identical on all data sheets and requires sufficient information to identify the station and location where the survey was conducted, date and time of survey, and the investigators responsible for the quality and integrity of the data. The stream name and river basin identify the watershed and tributary; the location of the station is described in the narrative to help identify access to the station for repeat visits. The rivermile (if applicable) and latitude/longitude are specific locational data for the station. The station number is a code assigned by the agency that will associate the sample and survey data with the station. The STORET number is assigned to each datapoint for inclusion in USEPA's STORET system. The stream class is a designation of the grouping of homogeneous characteristics from which assessments will be made. For instance, Ohio EPA uses ecoregions and size of stream, Florida DEP uses bioregions (aggregations of subecoregions), and Arizona DEQ uses elevation as a means to identify stream classes. Listing the agency and investigators assigns responsibility to the data collected from the station at a specific date and time. The reason for the survey is sometimes useful to an agency that conducts surveys for various programs and purposes.

5.1.2 Weather Conditions

Note the present weather conditions on the day of the survey and those immediately preceding the day of the survey. This information is important to interpret the effects of storm events on the sampling effort.

5.1.3 Site Location/Map

To complete this phase of the bioassessment, a photograph may be helpful in identifying station location and documenting habitat conditions. Any observations or data not requested but deemed important by the field observer should be recorded. A hand-drawn map is useful to illustrate major landmarks or features of the channel morphology or orientation, vegetative zones, buildings, etc. that might be used to aid in data interpretation.

5.1.4 Stream Characterization

Stream Subsystem: In regions where the perennial nature of streams is important, or where the tidal influence of streams will alter the structure and function of communities, this parameter should be noted.

Stream Type: Communities inhabiting coldwater streams are markedly different from those in warmwater streams, many states have established temperature criteria that differentiate these 2 stream types.

Stream Origin: Note the origination of the stream under study, if it is known. Examples are glacial, montane, swamp, and bog. As the size of the stream or river increases, a mixture of origins of tributaries is likely.

5.1.5 Watershed Features

Collecting this information usually requires some effort initially for a station. However, subsequent surveys will most likely not require an in-depth research of this information.

Predominant Surrounding Land Use Type: Document the prevalent land-use type in the catchment of the station (noting any other land uses in the area which, although not predominant, may potentially affect water quality). Land use maps should be consulted to accurately document this information.

Local Watershed Nonpoint Source Pollution: This item refers to problems and potential problems in the watershed. Nonpoint source pollution is defined as diffuse agricultural and urban runoff. Other compromising factors in a watershed that may affect water quality include feedlots, constructed wetlands, septic systems, dams and impoundments, mine seepage, etc.

Local Watershed Erosion: The existing or potential detachment of soil within the local watershed (the portion of the watershed or catchment that directly affects the stream reach or station under study) and its movement into the stream is noted. Erosion can be rated through visual observation of watershed and stream characteristics (note any turbidity observed during water quality assessment below).

5.1.6 Riparian Vegetation

An acceptable riparian zone includes a buffer strip of a minimum of 18 m (Barton et al. 1985) from the stream on either side. The acceptable width of the riparian zone may also be variable depending on the size of the stream. Streams over 4 m in width may require larger riparian zones. The vegetation within the riparian zone is documented here as the dominant type and species, if known.

5.1.7 Instream Features

Instream features are measured or evaluated in the sampling reach and catchment as appropriate.

Estimated Reach Length: Measure or estimate the length of the sampling reach. This information is important if reaches of variable length are surveyed and assessed.

Estimated Stream Width (in meters, m): Estimate the distance from bank to bank at a transect representative of the stream width in the reach. If variable widths, use an average to find that which is representative for the given reach.

Sampling Reach Area (m^2): Multiply the sampling reach length by the stream width to obtain a calculated surface area.

Estimated Stream Depth (m): Estimate the vertical distance from water surface to stream bottom at a representative depth (use instream habitat feature that is most common in reach) to obtain average depth.

Velocity: Measure the surface velocity in the thalweg of a representative run area. If measurement is not done, estimate the velocity as slow, moderate, or fast.

Canopy Cover: Note the general proportion of open to shaded area which best describes the amount of cover at the sampling reach or station. A densiometer may be used in place of visual estimation.

High Water Mark (m): Estimate the vertical distance from the bankfull margin of the stream bank to the peak overflow level, as indicated by debris hanging in riparian or floodplain vegetation, and deposition of silt or soil. In instances where bank overflow is rare, a high water mark may not be evident.

Proportion of Reach Represented by Stream Morphological Types: The proportion represented by riffles, runs, and pools should be noted to describe the morphological heterogeneity of the reach.

Channelized: Indicate whether or not the area around the sampling reach or station is channelized (e.g., straightening of stream, bridge abutments and road crossings, diversions, etc.).

Dam Present: Indicate the presence or absence of a dam upstream in the catchment or downstream of the sampling reach or station. If a dam is present, include specific information relating to alteration of flow.

5.1.8 Large Woody Debris

Large Woody Debris (LWD) density, defined and measured as described below, has been used in regional surveys (Shields et al. 1995) and intensive studies of degraded and restored streams (Shields et al. 1998). The method was developed for sand or sand-and-gravel bed streams in the Southeastern U.S. that are wadeable at baseflow, with water widths between 1 and 30 m (Cooper and Testa 1999).

Cooper and Testa's (1999) procedure involves measurements based on visual estimates taken by a wading observer. Only woody debris actually in contact with stream water is counted. Each woody debris formation with a surface area in the plane of the water surface >0.25 m^2 is recorded. The estimated length and width of each formation is recorded on a form or marked directly onto a stream reach drawing. Estimates are made to the nearest 0.5 m , and formations with length or width less than 0.5 m are not counted. Recorded length is maximum width in the direction perpendicular to the length. Maximum actual length and width of a limb, log, or accumulation are not considered.

If only a portion of the log/limb is in contact with the water, only that portion in contact is measured. Root wads and logs/limbs in the water margin are counted if they contact the water, and are arbitrarily given a width of 0.5 m Lone individual limbs and logs are included in the determination if their diameter is 10 cm or larger (Keller and Swanson 1979, Ward and Aumen 1986). Accumulations of smaller limbs and logs are included if the formation total length or width is 0.5 m or larger. Standing trees and stumps within the stream are also recorded if their length and width exceed 0.5 m.

The length and width of each LWD formation are then multiplied, and the resulting products are summed to give the aquatic habitat area directly influenced. This area is then divided by the water

surface area (km^2) within the sampled reach (obtained by multiplying the average water surface width by reach length) to obtain LWD density. Density values of 10^3 to 10^4 m^2/km^2 have been reported for channelized and incised streams and on the order of 10^5 m^2/km^2 for non-incised streams (Shields et al. 1995 and 1998). This density is not an expression of the volume of LWD, but rather a measure of LWD influence on velocity, depth, and cover.

5.1.9 Aquatic Vegetation

The general type and relative dominance of aquatic plants are documented in this section. Only an estimation of the extent of aquatic vegetation is made. Besides being an ecological assemblage that responds to perturbation, aquatic vegetation provides refugia and food for aquatic fauna. List the species of aquatic vegetation, if known.

5.1.10 Water Quality

Temperature (°C), Conductivity or "Specific Conductance" (µohms), Dissolved Oxygen (µg/L), pH, Turbidity: Measure and record values for each of the water quality parameters indicated, using the appropriate calibrated water quality instrument(s). Note the type of instrument and unit number used.

Water Odors: Note those odors described (or include any other odors not listed) that are associated with the water in the sampling area.

Water Surface Oils: Note the term that best describes the relative amount of any oils present on the water surface.

Turbidity: If turbidity is not measured directly, note the term which, based upon visual observation, best describes the amount of material suspended in the water column.

5.1.11 Sediment/Substrate

Sediment Odors: Disturb sediment in pool or other depositional areas and note any odors described (or include any other odors not listed) which are associated with sediment in the sampling reach.

Sediment Oils: Note the term which best describes the relative amount of any sediment oils observed in the sampling area.

Sediment Deposits: Note those deposits described (or include any other deposits not listed) that are present in the sampling reach. Also indicate whether the undersides of rocks not deeply embedded are black (which generally indicates low dissolved oxygen or anaerobic conditions).

Inorganic Substrate Components: Visually estimate the relative proportion of each of the 7 substrate/particle types listed that are present over the sampling reach.

Organic Substrate Components: Indicate relative abundance of each of the 3 substrate types listed.

5.2 A VISUAL-BASED HABITAT ASSESSMENT

Biological potential is limited by the quality of the physical habitat, forming the template within which biological communities develop (Southwood 1977). Thus, habitat assessment is defined as the evaluation of the structure of the surrounding physical habitat that influences the quality of the water resource and the condition of the resident aquatic community (Barbour et al. 1996a). For streams, an encompassing approach to assessing structure of the habitat includes an evaluation of the variety and quality of the substrate, channel morphology, bank structure, and riparian vegetation. Habitat parameters pertinent to the assessment of habitat quality include those that characterize the stream "micro scale" habitat (e.g., estimation of embeddeddness), the "macro scale" features (e.g., channel morphology), and the riparian and bank structure features that are most often influential in affecting the other parameters.

EQUIPMENT/SUPPLIES NEEDED FOR HABITAT ASSESSMENT AND PHYSICAL/WATER QUALITY CHARACTERIZATION

- Physical Characterization and Water Quality Field Data Sheet*
- Habitat Assessment Field Data Sheet*
- clipboard
- pencils or waterproof pens
- 35 mm camera (may be digital)
- video camera (optional)
- upstream/downstream “arrows” or signs for photographing and documenting sampling reaches
- Flow or velocity meter
- *In situ* water quality meters
- Global Positioning System (GPS) Unit

* It is helpful to copy field sheets onto water-resistant paper for use in wet weather conditions

Rosgen (1985, 1994) presented a stream and river classification system that is founded on the premise that dynamically-stable stream channels have a morphology that provides appropriate distribution of flow energy during storm events. Further, he identifies 8 major variables that affect the stability of channel morphology, but are not mutually independent: channel width, channel depth, flow velocity, discharge, channel slope, roughness of channel materials, sediment load and sediment particle size distribution. When streams have one of these characteristics altered, some of their capability to dissipate energy properly is lost (Leopold et al. 1964, Rosgen 1985) and will result in accelerated rates of channel erosion. Some of the habitat structural components that function to dissipate flow energy are:

- sinuosity
- roughness of bed and bank materials
- presence of point bars (slope is an important characteristic)
- vegetative conditions of stream banks and the riparian zone
- condition of the floodplain (accessibility from bank, overflow, and size are important characteristics).

Measurement of these parameters or characteristics serve to stratify and place streams into distinct classifications. However, none of these habitat classification techniques attempt to differentiate the quality of the habitat and the ability of the habitat to support the optimal biological condition of the

region. Much of our understanding of habitat relationships in streams has emerged from comparative studies that describe statistical relationships between habitat variables and abundance of biota (Hawkins et al. 1993). However, in response to the need to incorporate broader scale habitat assessments in water resource programs, 2 types of approaches for evaluating habitat structure have been developed. In the first, the Environmental Monitoring and Assessment Program (EMAP) of the USEPA and the National Water-Quality Assessment Program (NAWQA) of the USGS developed techniques that incorporate measurements of various features of the instream, channel, and bank morphology (Meader et al. 1993, Klemm and Lazorchak 1994). These techniques provide a relatively comprehensive characterization of the physical structure of the stream sampling reach and its surrounding floodplain. The second type was a more rapid and qualitative habitat assessment approach that was developed to describe the overall quality of the physical habitat (Ball 1982, Ohio EPA 1987, Plafkin et al. 1989, Barbour and Stribling 1991, 1994, Rankin 1991, 1995). In this document, the more rapid visual-based approach is described. A cursory overview of the more quantitative approaches to characterizing the physical structure of the habitat is provided.

The habitat assessment matrix developed for the Rapid Bioassessment Protocols (RBPs) in Plafkin et al. (1989) were originally based on the Stream Classification Guidelines for Wisconsin developed by Ball (1982) and "*Methods of Evaluating Stream, Riparian, and Biotic Conditions*" developed by Platts et al. (1983). Barbour and Stribling (1991, 1994) modified the habitat assessment approach originally developed for the RBPs to include additional assessment parameters for high gradient streams and a more appropriate parameter set for low gradient streams (Appendix A-1, Forms 2,3). All parameters are evaluated and rated on a numerical scale of 0 to 20 (highest) for each sampling reach. The ratings are then totaled and compared to a reference condition to provide a final habitat ranking. Scores increase as habitat quality increases. To ensure consistency in the evaluation procedure, descriptions of the physical parameters and relative criteria are included in the rating form.

The Environmental Agency of Great Britain (Environment Agency of England and Wales, Scottish Environment Protection Agency, and Environment and Heritage Service of Northern Ireland) have developed a River Habitat Survey (RHS) for characterizing the quality of their streams and rivers (Raven et al. 1998). The approach used in Great Britain is similar to the visual-based habitat assessment used in the US in that scores are assigned to ranges of conditions of various habitat parameters.

A biologist who is well versed in the ecology and zoogeography of the region can generally recognize optimal habitat structure as it relates to the biological community. The ability to accurately assess the quality of the physical habitat structure using a visual-based approach depends on several factors:

- the parameters selected to represent the various features of habitat structure need to be relevant and clearly defined

- a continuum of conditions for each parameter must exist that can be characterized from the optimum for the region or stream type under study to the poorest situation reflecting substantial alteration due to anthropogenic activities

- the judgement criteria for the attributes of each parameter should minimize subjectivity through either quantitative measurements or specific categorical choices

- the investigators are experienced in or adequately trained for stream assessments in the region under study (Hannaford et al. 1997)

- adequate documentation and ongoing training is maintained to evaluate and correct errors resulting in outliers and aberrant assessments.

Habitat evaluations are first made on instream habitat, followed by channel morphology, bank structural features, and riparian vegetation. Generally, a single, comprehensive assessment is made that incorporates features of the entire sampling reach as well as selected features of the catchment. Additional assessments may be made on neighboring reaches to provide a broader evaluation of habitat quality for the stream ecosystem. The actual habitat assessment process involves rating the 10 parameters as optimal, suboptimal, marginal, or poor based on the criteria included on the Habitat Assessment Field Data Sheets (Appendix A-1, Forms 2,3). Some state programs, such as Florida Department of Environmental Protection (DEP) (1996) and Mid-Atlantic Coastal Streams Workgroup (MACS) (1996) have adapted this approach using somewhat fewer and different parameters.

Reference conditions are used to scale the assessment to the "best attainable" situation. This approach is critical to the assessment because stream characteristics will vary dramatically across different regions (Barbour and Stribling 1991). The ratio between the score for the test station and the score for the reference condition provides a percent comparability measure for each station. The station of interest is then classified on the basis of its similarity to expected conditions (reference condition), and its apparent potential to support an acceptable level of biological health. Use of a percent comparability evaluation allows for regional and stream-size differences which affect flow or velocity, substrate, and channel morphology. Some regions are characterized by streams having a low channel gradient, such as coastal plains or prairie regions.

Other habitat assessment approaches or a more rigorously quantitative approach to measuring the habitat parameters may be used (See Klemm and Lazorchak 1994, Kaufmann and Robison 1997, Meader et al. 1993). However, holistic and rapid assessment of a wide variety of habitat attributes along with other types of data is critical if physical measurements are to be used to best advantage in interpreting biological data. A more detailed discussion of the relationship between habitat quality and biological condition is presented in Chapter 10.

A generic habitat assessment approach based on visual observation can be separated into 2 basic approaches—one designed for high-gradient streams and one designed for low-gradient streams. High-gradient or riffle/run prevalent streams are those in moderate to high gradient landscapes. Natural high-gradient streams have substrates primarily composed of coarse sediment particles (i.e., gravel or larger) or frequent coarse particulate aggregations along stream reaches. Low-gradient or glide/pool prevalent streams are those in low to moderate gradient landscapes. Natural low-gradient streams have substrates of fine sediment or infrequent aggregations of more coarse (gravel or larger) sediment particles along stream reaches. The entire sampling reach is evaluated for each parameter. Descriptions of each parameter and its relevance to instream biota are presented in the following discussion. Parameters that are used only for high-gradient prevalent streams are marked with an “a”; those for low-gradient dominant streams, a “b”. If a parameter is used for both stream types, it is not marked with a letter. A brief set of decision criteria is given

for each parameter corresponding to each of the 4 categories reflecting a continuum of conditions on the field sheet (optimal, suboptimal, marginal, and poor). Refer to Appendix A-1, Forms 2 and 3, for a complete field assessment guide.

PROCEDURE FOR PERFORMING HABITAT ASSESSMENT

1. Select the reach to be assessed. The habitat assessment is performed on the same 100 m reach (or other reach designation [e.g., 40 x stream wetted width]) from which the biological sampling is conducted. Some parameters require an observation of a broader section of the catchment than just the sampling reach.

2. Complete the station identification section of each field data sheet and habitat assessment form.

3. It is best for the investigators to obtain a close look at the habitat features to make an adequate assessment. If the physical and water quality characterization and habitat assessment are done before the biological sampling, care must be taken to avoid disturbing the sampling habitat.

4. Complete the **Physical Characterization and Water Quality Field Data Sheet**. Sketch a map of the sampling reach on the back of this form.

5. Complete the **Habitat Assessment Field Data Sheet**, in a team of 2 or more biologists, if possible, to come to a consensus on determination of quality. Those parameters to be evaluated on a scale greater than a sampling reach require traversing the stream corridor to the extent deemed necessary to assess the habitat feature. As a general rule-of-thumb, use 2 lengths of the sampling reach to assess these parameters.

QUALITY ASSURANCE PROCEDURES

1. Each biologist is to be trained in the visual-based habitat assessment technique for the applicable region or state.

2. The judgment criteria for each habitat parameter are calibrated for the stream classes under study. Some text modifications may be needed on a regional basis.

3. Periodic checks of assessment results are completed using pictures of the sampling reach and discussions among the biologists in the agency.

Parameters to be evaluated in sampling reach:

1 EPIFAUNAL SUBSTRATE/AVAILABLE COVER

high and low gradient streams

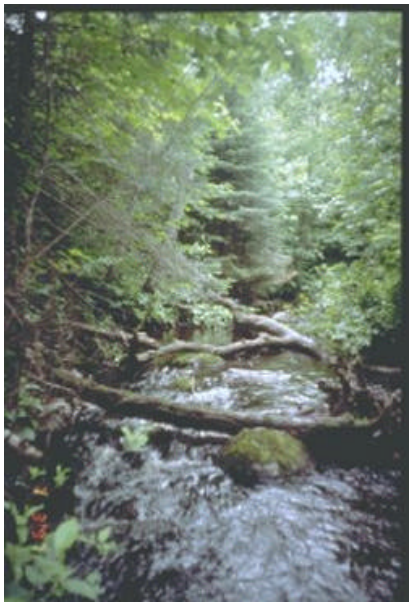
Includes the relative quantity and variety of natural structures in the stream, such as cobble (riffles), large rocks, fallen trees, logs and branches, and undercut banks, available as refugia, feeding, or sites for spawning and nursery functions of aquatic macrofauna. A wide variety and/or abundance of submerged structures in the stream provides macroinvertebrates and fish with a large number of niches, thus increasing habitat diversity. As variety and abundance of cover decreases, habitat structure becomes monotonous, diversity decreases, and the potential for recovery following disturbance decreases. Riffles and runs are critical for maintaining a variety and abundance of insects in most high-gradient streams and serving as spawning and feeding refugia for certain fish. The extent and quality of the riffle is an important factor in the support of a healthy biological condition in high-gradient streams. Riffles and runs offer a diversity of habitat through variety of particle size, and, in many small high-gradient streams, will provide the most stable habitat. Snags and submerged logs are among the most productive habitat structure for macroinvertebrate colonization and fish refugia in low-gradient streams. However, "new fall" will not yet be suitable for colonization.

Selected References

Wesche et al. 1985, Pearsons et al. 1992, Gorman 1988, Rankin 1991, Barbour and Stribling 1991, Plafkin et al. 1989, Platts et al. 1983, Osborne et al. 1991, Benke et al. 1984, Wallace et al. 1996, Ball 1982, MacDonald et al. 1991, Reice 1980, Clements 1987, Hawkins et al. 1982, Beechie and Sibley 1997.

Habitat Parameter	Condition Category			
	Optimal	**Suboptimal**	**Marginal**	**Poor**
1. Epifaunal Substrate/ Available Cover **(high and low gradient)**	Greater than 70% (50% for low gradient streams) of substrate favorable for epifaunal colonization and fish cover; mix of snags, submerged logs, undercut banks, cobble or other stable habitat and at stage to allow full colonization potential (i.e., logs/snags that are <u>not</u> new fall and <u>not</u> transient).	40-70% (30-50% for low gradient streams) mix of stable habitat; well-suited for full colonization potential; adequate habitat for maintenance of populations; presence of additional substrate in the form of newfall, but not yet prepared for colonization (may rate at high end of scale).	20-40% (10-30% for low gradient streams) mix of stable habitat; habitat availability less than desirable; substrate frequently disturbed or removed.	Less than 20% (10% for low gradient streams) stable habitat; lack of habitat is obvious; substrate unstable or lacking.
SCORE	20 19 18 17 16	15 14 13 12 11	10 9 8 7 6	5 4 3 2 1 0

1a. Epifaunal Substrate/Available Cover—High Gradient



Optimal Range

Poor Range

1b. Epifaunal Substrate/Available Cover—Low Gradient

Optimal Range (*Mary Kay Corazalla, U. of Minn.*)

Poor Range

2a EMBEDDEDNESS

high gradient streams

Refers to the extent to which rocks (gravel, cobble, and boulders) and snags are covered or sunken into the silt, sand, or mud of the stream bottom. Generally, as rocks become embedded, the surface area available to macroinvertebrates and fish (shelter, spawning, and egg incubation) is decreased. Embeddedness is a result of large-scale sediment movement and deposition, and is a parameter evaluated in the riffles and runs of high-gradient streams. The rating of this parameter may be variable depending on where the observations are taken. To avoid confusion with sediment deposition (another habitat parameter), observations of embeddedness should be taken in the upstream and central portions of riffles and cobble substrate areas.

Selected References

Ball 1982, Osborne et al. 1991, Barbour and Stribling 1991, Platts et al. 1983, MacDonald et al. 1991, Rankin 1991, Reice 1980, Clements 1987, Benke et al. 1984, Hawkins et al. 1982, Burton and Harvey 1990.

Habitat Parameter	Condition Category			
	Optimal	Suboptimal	Marginal	Poor
2.a Embeddedness (high gradient)	Gravel, cobble, and boulder particles are 0-25% surrounded by fine sediment. Layering of cobble provides diversity of niche space.	Gravel, cobble, and boulder particles are 25-50% surrounded by fine sediment.	Gravel, cobble, and boulder particles are 50-75% surrounded by fine sediment.	Gravel, cobble, and boulder particles are more than 75% surrounded by fine sediment.
SCORE	20 19 18 17 16	15 14 13 12 11	10 9 8 7 6	5 4 3 2 1 0

2a. Embeddedness—High Gradient

Optimal Range *(William Taft, MI DNR)*

Poor Range *(William Taft, MI DNR)*

2b POOL SUBSTRATE CHARACTERIZATION

low gradient streams

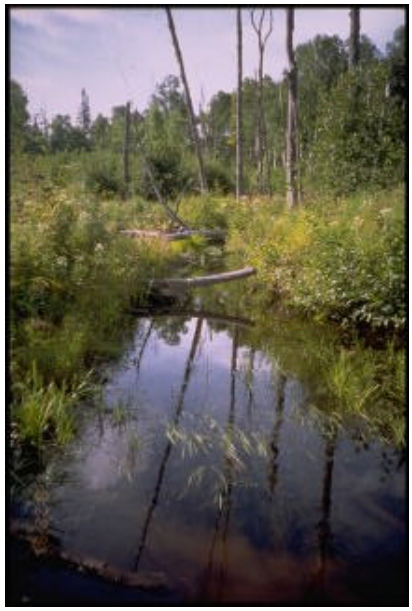
Evaluates the type and condition of bottom substrates found in pools. Firmer sediment types (e.g., gravel, sand) and rooted aquatic plants support a wider variety of organisms than a pool substrate dominated by mud or bedrock and no plants. In addition, a stream that has a uniform substrate in its pools will support far fewer types of organisms than a stream that has a variety of substrate types.

Selected References

Beschta and Platts 1986, U.S. EPA 1983.

Habitat Parameter	Condition Category			
	Optimal	Suboptimal	Marginal	Poor
2b. Pool Substrate Characterization (low gradient)	Mixture of substrate materials, with gravel and firm sand prevalent; root mats and submerged vegetation common.	Mixture of soft sand, mud, or clay; mud may be dominant; some root mats and submerged vegetation present.	All mud or clay or sand bottom; little or no root mat; no submerged vegetation.	Hard-pan clay or bedrock; no root mat or submerged vegetation.
SCORE	20 19 18 17 16	15 14 13 12 11	10 9 8 7 6	5 4 3 2 1 0

2b. Pool Substrate Characterization—Low Gradient



Optimal Range
(Mary Kay Corazalla, U. of Minn.)

Poor Range

3a VELOCITY/DEPTH COMBINATIONS

high gradient streams

Patterns of velocity and depth are included for high-gradient streams under this parameter as an important feature of habitat diversity. The best streams in most high-gradient regions will have all 4 patterns present: (1) slow-deep, (2) slow-shallow, (3) fast-deep, and (4) fast-shallow. The general guidelines are 0.5 m depth to separate shallow from deep, and 0.3 m/sec to separate fast from slow. The occurrence of these 4 patterns relates to the stream's ability to provide and maintain a stable aquatic environment.

Selected References

Ball 1982, Brown and Brussock 1991, Gore and Judy 1981, Oswood and Barber 1982.

Habitat Parameter	Condition Category			
	Optimal	Suboptimal	Marginal	Poor
3a. Velocity/ Depth Regimes **(high gradient)**	All 4 velocity/depth regimes present (slow-deep, slow-shallow, fast-deep, fast-shallow). (slow is <0.3 m/s, deep is >0.5 m)	Only 3 of the 4 regimes present (if fast-shallow is missing, score lower than if missing other regimes).	Only 2 of the 4 habitat regimes present (if fast-shallow or slow-shallow are missing, score low).	Dominated by 1 velocity/ depth regime (usually slow-deep).
SCORE	20 19 18 17 16	15 14 13 12 11	10 9 8 7 6	5 4 3 2 1 0

3a. Velocity/Depth Regimes—High Gradient

Optimal Range *(Mary Kay Corazalla, U. of Minn.)*
(arrows emphasize different velocity/depth regimes)

Poor Range *(William Taft, MI DNR)*

3b POOL VARIABILITY

low gradient streams

Rates the overall mixture of pool types found in streams, according to size and depth. The 4 basic types of pools are large-shallow, large-deep, small-shallow, and small-deep. A stream with many pool types will support a wide variety of aquatic species. Rivers with low sinuosity (few bends) and monotonous pool characteristics do not have sufficient quantities and types of habitat to support a diverse aquatic community. General guidelines are any pool dimension (i.e., length, width, oblique) greater than half the cross-section of the stream for separating large from small and 1 m depth separating shallow and deep.

Selected References

Beschta and Platts 1986, USEPA 1983.

Habitat Parameter	Condition Category			
	Optimal	**Suboptimal**	**Marginal**	**Poor**
3b. Pool Variability (low gradient)	Even mix of large-shallow, large-deep, small-shallow, small-deep pools present.	Majority of pools large-deep; very few shallow.	Shallow pools much more prevalent than deep pools.	Majority of pools small-shallow or pools absent.
SCORE	20 19 18 17 16	15 14 13 12 11	10 9 8 7 6	5 4 3 2 1 0

3b. Pool Variability—Low Gradient

Optimal Range *(Peggy Morgan, FL DEP)*

Poor Range *(William Taft, MI DNR)*

4 SEDIMENT DEPOSITION

high and low gradient streams

Measures the amount of sediment that has accumulated in pools and the changes that have occurred to the stream bottom as a result of deposition. Deposition occurs from large-scale movement of sediment. Sediment deposition may cause the formation of islands, point bars (areas of increased deposition usually at the beginning of a meander that increase in size as the channel is diverted toward the outer bank) or shoals, or result in the filling of runs and pools. Usually deposition is evident in areas that are obstructed by natural or manmade debris and areas where the stream flow decreases, such as bends. High levels of sediment deposition are symptoms of an unstable and continually changing environment that becomes unsuitable for many organisms.

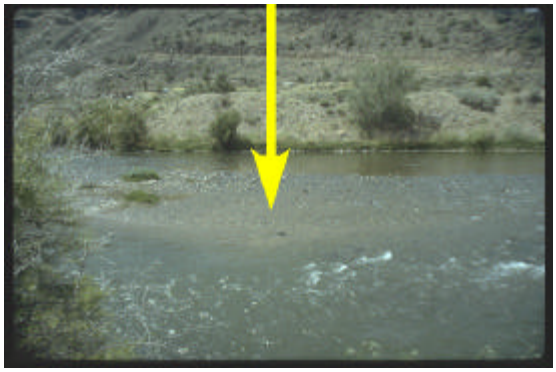
Selected References

MacDonald et al. 1991, Platts et al. 1983, Ball 1982, Armour et al. 1991, Barbour and Stribling 1991, Rosgen 1985.

Habitat Parameter	Condition Category																					
	Optimal					Suboptimal					Marginal					Poor					
4. Sediment Deposition **(high and low gradient)**	Little or no enlargement of islands or point bars and less than 5% (<20% for low-gradient streams) of the bottom affected by sediment deposition.					Some new increase in bar formation, mostly from gravel, sand or fine sediment; 5-30% (20-50% for low-gradient) of the bottom affected; slight deposition in pools.					Moderate deposition of new gravel, sand or fine sediment on old and new bars; 30-50% (50-80% for low-gradient) of the bottom affected; sediment deposits at obstructions, constrictions, and bends; moderate deposition of pools prevalent.					Heavy deposits of fine material, increased bar development; more than 50% (80% for low-gradient) of the bottom changing frequently; pools almost absent due to substantial sediment deposition.					
SCORE	20	19	18	17	16	15	14	13	12	11	10	9	8	7	6	5	4	3	2	1	0

4a. Sediment Deposition—High Gradient

Optimal Range



Poor Range
(arrow pointing to sediment deposition)

4b. Sediment Deposition—Low Gradient

Optimal Range

Poor Range
(arrows pointing to sediment deposition)

5 CHANNEL FLOW STATUS

high and low gradient streams

The degree to which the channel is filled with water. The flow status will change as the channel enlarges (e.g., aggrading stream beds with actively widening channels) or as flow decreases as a result of dams and other obstructions, diversions for irrigation, or drought. When water does not cover much of the streambed, the amount of suitable substrate for aquatic organisms is limited. In high-gradient streams, riffles and cobble substrate are exposed; in low-gradient streams, the decrease in water level exposes logs and snags, thereby reducing the areas of good habitat. Channel flow is especially useful for interpreting biological condition under abnormal or lowered flow conditions. This parameter becomes important when more than one biological index period is used for surveys or the timing of sampling is inconsistent among sites or annual periodicity.

Selected References

Rankin 1991, Rosgen 1985, Hupp and Simon 1986, MacDonald et al. 1991, Ball 1982, Hicks et al. 1991.

Habitat Parameter	Condition Category																					
	Optimal					Suboptimal					Marginal					Poor					
5. Channel Flow Status **(high and low gradient)**	Water reaches base of both lower banks, and minimal amount of channel substrate is exposed.					Water fills >75% of the available channel; or <25% of channel substrate is exposed.					Water fills 25-75% of the available channel, and/or riffle substrates are mostly exposed.					Very little water in channel and mostly present as standing pools.					
SCORE	20	19	18	17	16	15	14	13	12	11	10	9	8	7	6	5	4	3	2	1	0

5a. Channel Flow Status—High Gradient

Optimal Range

Poor Range
(arrow showing that water is not reaching both banks; leaving much of channel uncovered)

5b. Channel Flow Status—Low Gradient

Optimal Range

Poor Range *(James Stahl, IN DEM)*

Parameters to be evaluated broader than sampling reach:

6 CHANNEL ALTERATION

high and low gradient streams

Is a measure of large-scale changes in the shape of the stream channel. Many streams in urban and agricultural areas have been straightened, deepened, or diverted into concrete channels, often for flood control or irrigation purposes. Such streams have far fewer natural habitats for fish, macroinvertebrates, and plants than do naturally meandering streams. Channel alteration is present when artificial embankments, riprap, and other forms of artificial bank stabilization or structures are present; when the stream is very straight for significant distances; when dams and bridges are present; and when other such changes have occurred. Scouring is often associated with channel alteration.

Selected References

Barbour and Stribling 1991, Simon 1989a, b, Simon and Hupp 1987, Hupp and Simon 1986, Hupp 1992, Rosgen 1985, Rankin 1991, MacDonald et al. 1991.

Habitat Parameter	Condition Category			
	Optimal	Suboptimal	Marginal	Poor
6. Channel Alteration **(high and low gradient)**	Channelization or dredging absent or minimal; stream with normal pattern.	Some channelization present, usually in areas of bridge abutments; evidence of past channelization, i.e., dredging, (greater than past 20 yr) may be present, but recent channelization is not present.	Channelization may be extensive; embankments or shoring structures present on both banks; and 40 to 80% of stream reach channelized and disrupted.	Banks shored with gabion or cement; over 80% of the stream reach channelized and disrupted. Instream habitat greatly altered or removed entirely.
SCORE	20 19 18 17 16	15 14 13 12 11	10 9 8 7 6	5 4 3 2 1 0

6a. Channel Alteration—High Gradient

Optimal Range

Poor Range
(arrows emphasizing large-scale channel alterations)

6b. Channel Alteration—Low Gradient

Optimal Range

Poor Range *(John Maxted, DE DNREC)*

7a FREQUENCY OF RIFFLES (OR BENDS)

high gradient streams

Is a way to measure the sequence of riffles and thus the heterogeneity occurring in a stream. Riffles are a source of high-quality habitat and diverse fauna, therefore, an increased frequency of occurrence greatly enhances the diversity of the stream community. For high gradient streams where distinct riffles are uncommon, a run/bend ratio can be used as a measure of meandering or sinuosity (see 7b). A high degree of sinuosity provides for diverse habitat and fauna, and the stream is better able to handle surges when the stream fluctuates as a result of storms. The absorption of this energy by bends protects the stream from excessive erosion and flooding and provides refugia for benthic invertebrates and fish during storm events. To gain an appreciation of this parameter in some streams, a longer segment or reach than that designated for sampling should be incorporated into the evaluation. In some situations, this parameter may be rated from viewing accurate topographical maps. The "sequencing" pattern of the stream morphology is important in rating this parameter. In headwaters, riffles are usually continuous and the presence of cascades or boulders provides a form of sinuosity and enhances the structure of the stream. A stable channel is one that does not exhibit progressive changes in slope, shape, or dimensions, although short-term variations may occur during floods (Gordon et al. 1992).

Selected References

Hupp and Simon 1991, Brussock and Brown 1991, Platts et al. 1983, Rankin 1991, Rosgen 1985, 1994, 1996, Osborne and Hendricks 1983, Hughes and Omernik 1983, Cushman 1985, Bain and Boltz 1989, Gislason 1985, Hawkins et al. 1982, Statzner et al. 1988.

Habitat Parameter	Condition Category			
	Optimal	Suboptimal	Marginal	Poor
7a. Frequency of Riffles (or bends) **(high gradient)**	Occurrence of riffles relatively frequent; ratio of distance between riffles divided by width of the stream <7:1 (generally 5 to 7); variety of habitat is key. In streams where riffles are continuous, placement of boulders or other large, natural obstruction is important.	Occurrence of riffles infrequent; distance between riffles divided by the width of the stream is between 7 to 15.	Occasional riffle or bend; bottom contours provide some habitat; distance between riffles divided by the width of the stream is between 15 to 25.	Generally all flat water or shallow riffles; poor habitat; distance between riffles divided by the width of the stream is a ratio of >25.
SCORE	20 19 18 17 16	15 14 13 12 11	10 9 8 7 6	5 4 3 2 1 0

7a. Frequency of Riffles (or bends)—High Gradient

Optimal Range
(arrows showing frequency of riffles and bends)

Poor Range

7b

CHANNEL SINUOSITY

low gradient streams

Evaluates the meandering or sinuosity of the stream. A high degree of sinuosity provides for diverse habitat and fauna, and the stream is better able to handle surges when the stream fluctuates as a result of storms. The absorption of this energy by bends protects the stream from excessive erosion and flooding and provides refugia for benthic invertebrates and fish during storm events. To gain an appreciation of this parameter in low gradient streams, a longer segment or reach than that designated for sampling may be incorporated into the evaluation. In some situations, this parameter may be rated from viewing accurate topographical maps. The "sequencing" pattern of the stream morphology is important in rating this parameter. In "oxbow" streams of coastal areas and deltas, meanders are highly exaggerated and transient. Natural conditions in these streams are shifting channels and bends, and alteration is usually in the form of flow regulation and diversion. A stable channel is one that does not exhibit progressive changes in slope, shape, or dimensions, although short-term variations may occur during floods (Gordon et al. 1992).

Selected References

Hupp and Simon 1991, Brussock and Brown 1991, Platts et al. 1983, Rankin 1991, Rosgen 1985, 1994, 1996, Osborne and Hendricks 1983, Hughes and Omernik 1983, Cushman 1985, Bain and Boltz 1989, Gislason 1985, Hawkins et al. 1982, Statzner et al. 1988.

Habitat Parameter	Condition Category			
	Optimal	Suboptimal	Marginal	Poor
7b. Channel Sinuosity **(low gradient)**	The bends in the stream increase the stream length 3 to 4 times longer than if it was in a straight line. (Note - channel braiding is considered normal in coastal plains and other low-lying areas. This parameter is not easily rated in these areas.)	The bends in the stream increase the stream length 2 to 3 times longer than if it was in a straight line.	The bends in the stream increase the stream length 1 to 2 times longer than if it was in a straight line.	Channel straight; waterway has been channelized for a long distance.
SCORE	20 19 18 17 16	15 14 13 12 11	10 9 8 7 6	5 4 3 2 1 0

7b. Channel Sinuosity—Low Gradient

Optimal Range

Poor Range

8 BANK STABILITY (condition of banks)

high and low gradient streams

Measures whether the stream banks are eroded (or have the potential for erosion). Steep banks are more likely to collapse and suffer from erosion than are gently sloping banks, and are therefore considered to be unstable. Signs of erosion include crumbling, unvegetated banks, exposed tree roots, and exposed soil. Eroded banks indicate a problem of sediment movement and deposition, and suggest a scarcity of cover and organic input to streams. Each bank is evaluated separately and the cumulative score (right and left) is used for this parameter.

Selected References

Ball 1982, MacDonald et al. 1991, Armour et al. 1991, Barbour and Stribling 1991, Hupp and Simon 1986, 1991, Simon 1989a, Hupp 1992, Hicks et al. 1991, Osborne et al. 1991, Rosgen 1994, 1996.

Habitat Parameter	Condition Category											
	Optimal			Suboptimal			Marginal			Poor		
8. Bank Stability (score each bank) **Note: determine left or right side by facing downstream** **(high and low gradient)**	Banks stable; evidence of erosion or bank failure absent or minimal; little potential for future problems. <5% of bank affected.			Moderately stable; infrequent, small areas of erosion mostly healed over. 5-30% of bank in reach has areas of erosion.			Moderately unstable; 30-60% of bank in reach has areas of erosion; high erosion potential during floods.			Unstable; many eroded areas; "raw" areas frequent along straight sections and bends; obvious bank sloughing; 60-100% of bank has erosional scars.		
SCORE ___ (LB)	Left Bank	10	9	8	7	6	5	4	3	2	1	0
SCORE ___ (RB)	Right Bank	10	9	8	7	6	5	4	3	2	1	0

8a. Bank Stability (condition of banks)—High Gradient

Optimal Range
(arrow pointing to stable streambanks)

Poor Range *(MD Save Our Streams)*
(arrow highlighting unstable streambanks)

8b. Bank Stability (condition of banks)—Low Gradient

Optimal Range *(Peggy Morgan, FL DEP)*

Poor Range
(arrow highlighting unstable streambanks)

9 BANK VEGETATIVE PROTECTION

high and low gradient streams

Measures the amount of vegetative protection afforded to the stream bank and the near-stream portion of the riparian zone. The root systems of plants growing on stream banks help hold soil in place, thereby reducing the amount of erosion that is likely to occur. This parameter supplies information on the ability of the bank to resist erosion as well as some additional information on the uptake of nutrients by the plants, the control of instream scouring, and stream shading. Banks that have full, natural plant growth are better for fish and macroinvertebrates than are banks without vegetative protection or those shored up with concrete or riprap. This parameter is made more effective by defining the native vegetation for the region and stream type (i.e., shrubs, trees, etc.). In some regions, the introduction of exotics has virtually replaced all native vegetation. The value of exotic vegetation to the quality of the habitat structure and contribution to the stream ecosystem must be considered in this parameter. In areas of high grazing pressure from livestock or where residential and urban development activities disrupt the riparian zone, the growth of a natural plant community is impeded and can extend to the bank vegetative protection zone. Each bank is evaluated separately and the cumulative score (right and left) is used for this parameter.

Selected References

Platts et al. 1983, Hupp and Simon 1986, 1991, Simon and Hupp 1987, Ball 1982, Osborne et al. 1991, Rankin 1991, Barbour and Stribling 1991, MacDonald et al. 1991, Armour et al. 1991, Myers and Swanson 1991, Bauer and Burton 1993.

Habitat Parameter	Condition Category			
	Optimal	**Suboptimal**	**Marginal**	**Poor**
9. Vegetative Protection (score each bank) **Note: determine left or right side by facing downstream.** **(high and low gradient)**	More than 90% of the streambank surfaces and immediate riparian zones covered by native vegetation, including trees, understory shrubs, or nonwoody macrophytes; vegetative disruption through grazing or mowing minimal or not evident; almost all plants allowed to grow naturally.	70-90% of the streambank surfaces covered by native vegetation, but one class of plants is not well-represented; disruption evident but not affecting full plant growth potential to any great extent; more than one-half of the potential plant stubble height remaining.	50-70% of the streambank surfaces covered by vegetation; disruption obvious; patches of bare soil or closely cropped vegetation common; less than one-half of the potential plant stubble height remaining.	Less than 50% of the streambank surfaces covered by vegetation; disruption of streambank vegetation is very high; vegetation has been removed to 5 centimeters or less in average stubble height.
SCORE ___ (LB)	Left Bank 10 9	8 7 6	5 4 3	2 1 0
SCORE ___ (RB)	Right Bank 10 9	8 7 6	5 4 3	2 1 0

9a. Bank Vegetative Protection—High Gradient

Optimal Range
(arrow pointing to streambank with high level of vegetative cover)

Poor Range
(arrow pointing to streambank with almost no vegetative cover)

9b. Bank Vegetative Protection—Low Gradient

Optimal Range *(Peggy Morgan, FL DEP)*

Poor Range *(MD Save Our Streams)*
(arrow pointing to channelized streambank with no vegetative cover)

10 RIPARIAN VEGETATIVE ZONE WIDTH

high and low gradient streams

Measures the width of natural vegetation from the edge of the stream bank out through the riparian zone. The vegetative zone serves as a buffer to pollutants entering a stream from runoff, controls erosion, and provides habitat and nutrient input into the stream. A relatively undisturbed riparian zone supports a robust stream system; narrow riparian zones occur when roads, parking lots, fields, lawns, bare soil, rocks, or buildings are near the stream bank. Residential developments, urban centers, golf courses, and rangeland are the common causes of anthropogenic degradation of the riparian zone. Conversely, the presence of "old field" (i.e., a previously developed field not currently in use), paths, and walkways in an otherwise undisturbed riparian zone may be judged to be inconsequential to altering the riparian zone and may be given relatively high scores. For variable size streams, the specified width of a desirable riparian zone may also be variable and may be best determined by some multiple of stream width (e.g., 4 x wetted stream width). Each bank is evaluated separately and the cumulative score (right and left) is used for this parameter.

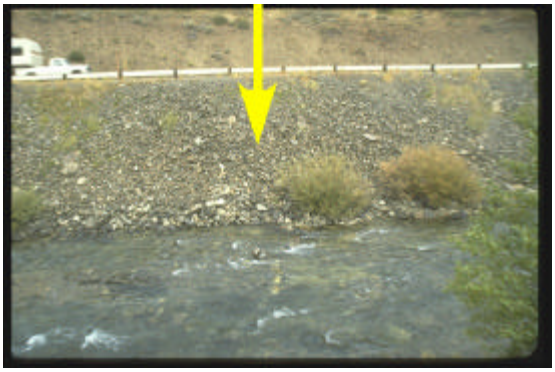
Selected References

Barton et al. 1985, Naiman et al. 1993, Hupp 1992, Gregory et al. 1991, Platts et al. 1983, Rankin 1991, Barbour and Stribling 1991, Bauer and Burton 1993.

Habitat Parameter	Condition Category											
	Optimal			Suboptimal			Marginal			Poor		
10. Riparian Vegetative Zone Width (score each bank riparian zone) **(high and low gradient)**	Width of riparian zone >18 meters; human activities (i.e., parking lots, roadbeds, clear-cuts, lawns, or crops) have not impacted zone.			Width of riparian zone 12-18 meters; human activities have impacted zone only minimally.			Width of riparian zone 6-12 meters; human activities have impacted zone a great deal.			Width of riparian zone <6 meters: little or no riparian vegetation due to human activities.		
SCORE ___ (LB)	Left Bank	10	9	8	7	6	5	4	3	2	1	0
SCORE ___ (RB)	Right Bank	10	9	8	7	6	5	4	3	2	1	0

10a. Riparian Vegetative Zone Width—High Gradient

Optimal Range
(arrow pointing out an undisturbed riparian zone)



Poor Range
(arrow pointing out lack of riparian zone)

10b. Riparian Vegetative Zone Width—Low Gradient

Optimal Range
(arrow emphasizing an undisturbed riparian zone)

Poor Range *(MD Save Our Streams)*
(arrow emphasizing lack of riparian zone)

5.3 ADDITIONS OF QUANTITATIVE MEASURES TO THE HABITAT ASSESSMENT

Kaufmann (1993) identified 7 general physical habitat attributes important in influencing stream ecology. These include:

- channel dimensions
- channel gradient
- channel substrate size and type
- habitat complexity and cover
- riparian vegetation cover and structure
- anthropogenic alterations
- channel-riparian interaction.

All of these attributes vary naturally, as do biological characteristics; thus expectations differ even in the absence of anthropogenic disturbances. Within a given physiographic-climatic region, stream drainage area and overall stream gradient are likely to be strong natural determinants of many aspects of stream habitat, because of their influence on discharge, flood stage, and stream power (the product of discharge times gradient). In addition, all of these attributes may be directly or indirectly altered by anthropogenic activities.

In Section 5.2, an approach is described whereby habitat quality is interpreted directly in the field by biologists while sampling the stream reach. This Level 1 approach is observational and requires only one person (although a team approach is recommended) and takes about 15 to 20 minutes per stream reach. This approach more quickly yields a habitat quality assessment. However, it depends upon the knowledge and experience of the field biologist to make the proper interpretation of observed of both the natural expectations (potentials) and the biological consequences (quality) that can be attributed to the observed physical attributes. Hannaford et al. (1997) found that training in habitat assessment was necessary to reduce the subjectivity in a visual-based approach. The authors also stated that training on different types of streams may be necessary to adequately prepare investigators.

The second conceptual approach described here confines observations to habitat characteristics themselves (whether they are quantitative or qualitative), then later ascribing quality scoring to these measurements as part of the data analysis process. Typically, this second type of habitat assessment approach employs more quantitative data collection, as exemplified by field methods described by Kaufmann and Robison (1997) for EMAP, Simonson et al. (1994), Meador et al. (1993) for NAWQA, and others cited by Gurtz and Muir (1994). These field approaches typically define a reach length proportional to stream width and employ transect measurements that are systematically spaced (Simonson et al. 1994, Kaufmann and Robison 1997) or spaced by judgement to be representative (Meador et al. 1993). They usually include measurement of substrate, channel and bank dimensions, riparian canopy cover, discharge, gradient, sinuosity, in-channel cover features, and counts of large woody debris and riparian human disturbances. They may employ systematic visual estimates of substrate embeddedness, fish cover features, habitat

types, and riparian vegetation structure. The time commitment in the field to these more quantitative habitat assessment methods is usually 1.5 to 3 hours with a crew of two people. Because of the greater amount of data collected, they also require more time for data summarization, analysis, and interpretation. On the other hand, the more quantitative methods and less ambiguous field parameters result in considerably greater precision. The USEPA applied both quantitative and visual-based (RBPs) methods in a stream survey undertaken over 4 years in the mid-Atlantic region of the Appalachian Mountains. An earlier version of the RBP techniques were applied on 301 streams with repeat visits to 29 streams; signal-to-noise ratios varied from 0.1 to 3.0 for the twelve RBP metrics and averaged (1.1 for the RBP total habitat quality score). The quantitative methods produced a higher level of precision; signal-to-noise ratios were typically between 10 and 50, and sometimes in excess of 100 for quantitative measurements of channel morphology, substrate, and canopy densiometer measurements made on a random subset of 186 streams with 27 repeat visits in the same survey. Similarly, semi-quantitative estimates of fish cover and riparian human disturbance estimates obtained from multiple, systematic visual observations of otherwise measurable features had signal:noise ratios from 5 to 50. Many riparian vegetation cover and structure metrics were moderately precise (signal:noise ranging from 2 to 30). Commonly used flow dependent measures (e.g., riffle/pool and width/depth ratios), and some visual riparian cover estimates were less precise, with signal:noise ratios more in the range of those observed for metrics of the EPA's RBP habitat score (<2).

The USEPA's EMAP habitat assessment field methods are presented as an option for a second level (II) of habitat assessment. These methods have been applied in numerous streams throughout the Mid-Atlantic region, the Midwest, Colorado, California, and the Pacific Northwest. Table 5-1 is a summary of these field methods; more detail is presented in the field manual by Kaufmann and Robison (1997).

Table 5-1. Components of EMAP physical habitat protocol.

Component	Description
1. Thalweg Profile	Measure maximum depth, classify habitat, determine presence of soft/small sediment at 10-15 equally spaced intervals between each of 11 channel cross-sections (100-150 along entire reach). Measure wetted width at 11 channel cross-sections and mid-way between cross-sections (21 measurements).
2. Woody Debris	Between each of the channel cross sections, tally large woody debris numbers within and above the bankfull channel according to size classes.
3. Channel and Riparian Cross-Sections	At 11 cross-section stations placed at equal intervals along reach length: • **Measure**: channel cross section dimensions, bank height, undercut, angle (with rod and clinometer); gradient (clinometer), sinuosity (compass backsite), riparian canopy cover (densiometer). • **Visually Estimate***: substrate size class and embeddedness; areal cover class and type (e.g., woody) of riparian vegetation in Canopy, Mid-Layer and Ground Cover; areal cover class of fish concealment features, aquatic macrophytes and filamentous algae. • **Observe & Record***: human disturbances and their proximity to the channel.
4. Discharge	In medium and large streams (defines later) measure water depth and velocity @ 0.6 depth (with electromagnetic or impeller-type flow meter) at 15 to 20 equally spaced intervals across one carefully chosen channel cross-section. In very small streams, measure discharge with a portable weir or time the filling of a bucket.

* Substrate size class and embeddedness are estimated, and depth is measured for 55 particles taken at 5 equally-spaced points on each of 11 cross-sections. The cross-section is defined by laying the surveyor's rod or tape to span the wetted channel. Woody

debris is tallied over the distance between each cross-section and the next cross-section upstream. Riparian vegetation and human disturbances are observed 5 m upstream and 5 m downstream from the cross section station. They extend shoreward 10 m from left and right banks. Fish cover types, aquatic macrophytes, and algae are observed within channel 5 m upstream and 5 m downstream from the cross section stations. These boundaries for visual observations are estimated by eye.

Table 5-2 lists the physical habitat metrics that can be derived from applying these field methods. Once these habitat metrics are calculated from the available physical habitat data, an assessment would be obtained from comparing these metric values to those of known reference sites. A strong deviation from the reference expectations would indicate a habitat alteration of the particular parameter. The close connectivity of the various attributes would most likely result in an impact on multiple metrics if habitat alteration was occurring. The actual process for interpreting a habitat assessment using this approach is still under development.

Table 5-2. Example of habitat metrics that can be calculated from the EMAP physical habitat data.

Channel mean width and depth
Channel volume and Residual Pool volume
Mean channel slope and sinuosity
Channel incision, bankfull dimensions, and bank characteristics
Substrate mean diameter, % fines, % embeddedness
Substrate stability
Fish concealment features (areal cover of various types, e.g., undercut banks, brush)
Large woody debris (volume and number of pieces per 100 m)
Channel habitat types (e.g., % of reach composed of pools, riffles, etc.)
Canopy cover
Riparian vegetation structure and complexity
Riparian disturbance measure (proximity-weighted tally of human disturbances)

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Water Quality

Contents

Water Quality Assessment General Information

There are several components to the Water Quality Assessment:

- Discharge or flow
- pH
- Total Suspended Solids (TSS) and nutrients

Use the "Water Quality Assessment Data" sheet to record your data. The data sheet and field labels that you need to take with you in the field are embedded throughout this section and there are extra copies in the inside back pocket of your manual. The data that you collect and record in the field on your "Water Quality Assessment Data Sheet" should be entered online as soon as possible after your site visit.

As always, before entering the field, consider safety, check supplies and equipment lists, and check weather and directions. Be sure to let someone know where you are going and when you expect to return. Do not perform field work alone.

Field Equipment Checklist

General Equipment

- Waders
- Spray bottles with disinfectant solution and rinse water
- Pencils and permanent marker
- Water Quality Assessment Data sheets and other field sheets
- Clipboard
- Insect repellent/sunscreen
- Sunglasses
- Camera
- First Aid kit
- Cell phone

Water Samples

- 4 bottles for Nutrients/TP: labeled with site code, date, rep (or blank), and TP
- 3 bottles for TSS: labeled with site code, date, rep, and TSS
- Cooler with ice
- Extra bottles and labels

Physical parameters

- Meter tape
- Meter stick
- Water temperature/pH meter
- Stopwatch
- Tennis ball

Sensor Stewarding

- Small screwdriver
- Sponge for cleaning

Water Quality Monitoring Background Information

What is water quality monitoring?

For our purposes, we define water quality monitoring as the sampling and analysis of water constituents and conditions. These may include:

- Introduced pollutants, such as pesticides, metals, and oil
- Constituents found naturally in water that can nevertheless be affected by human sources, such as dissolved oxygen, bacteria, and nutrients

The magnitude of their effects can be influenced by properties such as pH and temperature. For example, temperature influences the quantity of dissolved oxygen that water is able to contain, and pH affects the toxicity of ammonia.

What part of water quality does the RACC Streams Project monitor?

The RACC Streams Project collects water quality data on temperature, pH, phosphorus and nitrogen (important nutrients for primary production), and total suspended solids. We also collect information on macroinvertebrate communities, which are longer-term indicators of water quality, whereas water samples (i.e. temperature, pH, phosphorus, nitrogen, TSS) are a snapshot of water quality at a specific sampling moment.

The data that the RACC Streams Project collects are used for the purposes detailed below as well as the individual research projects of high school teams and undergraduate interns.

Who monitors water quality?

Local and national water quality professionals, volunteers, and researchers, have been monitoring water quality conditions for many years. In fact, until the past decade or so (when biological monitoring protocols were developed and began to take hold), water quality monitoring was generally considered the primary way of identifying water pollution problems. Today, professional water quality specialists and volunteer program coordinators alike are moving toward approaches that combine chemical, physical, and biological monitoring methods to achieve the best picture of water quality conditions.

Government agencies have searchable water quality databases. Below are a few of the larger publicly-available databases:

- USEPA Storage and retrieval data base (STORET):
  - www.epa.gov/storet/
- US Geological Survey (USGS): Fixed monitoring stations for hydrology and water quality monitoring
  - waterdata.usgs.gov/nwis/qw
- National Oceanic and Atmospheric Administration (NOAA):
  - Sea Grant Program (Lake Champlain & Great Lakes)
  - National Status and Trends Program
  - www.research.noaa.gov/oceans/t_hydrology.html
- State Agencies
  - VT Department of Environmental Conservation: www.anr.state.vt.us/dec/dec.htm
  - VT Department of Fish & Wildlife: www.vtfishandwildlife.com
- University research reports and journal articles

Why monitor water quality?

Water quality monitoring can be used for many purposes:

- *To identify whether waters are meeting designated uses.* All states have established specific criteria (limits on pollutants) identifying what concentrations of chemical pollutants are allowable in their waters. When chemical pollutants exceed maximum or minimum allowable concentrations, waters may no longer be able to support the beneficial uses such as fishing, swimming, and drinking for which they have been designated. Designated uses and the specific criteria that protect them (along with anti-degradation statements say waters should not be allowed to deteriorate below existing or anticipated uses) together form water quality standards. State water quality professionals assess water quality by comparing the concentrations of chemical pollutants found in streams to the criteria in the state's standards, and so judge whether streams are meeting their designated uses.

  Water quality monitoring, however, might be inadequate for determining whether aquatic life uses are being met in a stream. While some constituents (such as dissolved oxygen and temperature) are important to maintaining healthy fish and aquatic insect populations, other factors, such as the physical structure of the stream and the condition of the habitat, play an equal or greater role. Biological monitoring methods are generally better suited to determining whether aquatic life is supported.

- *To identify specific pollutants and sources of pollution.* Water quality monitoring helps link sources of pollution to a stream quality problem because it identifies specific problem pollutants. Since certain activities tend to generate certain pollutants (e.g., bacteria and nutrients are more likely to come from an animal feedlot than an automotive repair shop), a tentative link might be made that would warrant further investigation or monitoring.

- *To determine trends.* Chemical constituents that are properly monitored (i.e., consistent time of day and on a regular basis, using consistent methods) can be analyzed for trends over time.

- *To screen for impairment.* Finding excessive levels of one or more chemical constituents can serve as an early warning "screen" of potential pollution problems.

Table 1. Sources of water quality degradation and the types of pollutants associated with each source.

Source	Common Associated Chemical Pollutants
Cropland	Turbidity, phosphorus, nitrates, temperature, total solids
Forestry harvest	Turbidity, temperature, total solids
Grazing land	Fecal bacteria, turbidity, phosphorus, nitrates, temperature
Industrial discharge	Temperature, conductivity, total solids, toxics, pH
Mining	pH, alkalinity, total dissolved solids
Septic systems	Fecal bacteria (i.e., Escherichia coli, enterococcus), nitrates, phosphorus, dissolved oxygen/biochemical oxygen demand, conductivity, temperature
Sewage treatment plants	Dissolved oxygen and biochemical oxygen demand, turbidity, conductivity, phosphorus, nitrates, fecal bacteria, temperature, total solids, pH
Construction	Turbidity, temperature, dissolved oxygen and biochemical oxygen demand, total solids, and toxics
Urban runoff	Turbidity, phosphorus, nitrates, temperature, conductivity, dissolved oxygen and biochemical oxygen demand

In-Stream Measurements

Stream Flow, Stage & Discharge

What is stream flow and why is it important?

Stream flow, or discharge, is the **volume** of water that moves over a designated point over a fixed period of time. It is often expressed as cubic feet per second (ft^3/sec).

The flow of a stream is directly related to the amount of water moving off the watershed into the stream channel. It is affected by weather, increasing during rainstorms and decreasing during dry periods. It also changes during different seasons of the year, decreasing during the summer months when evaporation rates are high and shoreline vegetation is actively growing and removing water from the ground. August and September are usually the months of lowest flow for most streams and rivers in most of the country.

Water withdrawals for irrigation purposes can seriously deplete water flow, as can industrial water withdrawals. Dams used for electric power generation, particularly facilities designed to produce power during periods of peak need, often block the flow of a stream and later release it in a surge.

Flow is a function of water volume and velocity. It is important because of its impact on water quality and on the living organisms and habitats in the stream. Large, swiftly flowing rivers can receive pollution discharges and be little affected, whereas small streams have less capacity to dilute and degrade wastes.

Stream velocity, which increases as the volume of the water in the stream increases, determines the kinds of organisms that can live in the stream (some need fast-flowing areas; others need quiet pools). It also affects the amount of silt and sediment carried by the stream. Sediment introduced to quiet, slow-flowing streams will settle quickly to the stream bottom. Fast moving streams will keep sediment suspended longer in the water column. Lastly, fast-moving streams generally have higher levels of dissolved oxygen than slow streams because they are better aerated.

This section describes one method for estimating flow in a specific area or reach of a stream. It is adapted from techniques used by several monitoring programs and uses a float (an object such as an orange, ping-pong ball, pine cone, etc.) to measure stream velocity. Calculating flow involves solving an equation that examines the relationship among several variables including stream cross-sectional area, stream length, and water velocity. One way to measure flow is to solve the following equation:

Flow = ALC / T, *where:*

A = Average cross-sectional area of the stream (stream width multiplied by average water depth)

L = Length of the stream reach measured (usually 20 ft.)

C = A coefficient or correction factor (0.8 for rocky-bottom streams or 0.9 for muddy-bottom streams). This allows you to correct for the fact that water at the surface travels faster than near the stream bottom due to resistance from gravel, cobble, etc. Multiplying the surface velocity by a correction coefficient decreases the value and gives a better measure of the stream's overall velocity

T = Time, in seconds, for the float to travel the length of L

The flow of a stream has significant effect on habitat characteristics for residing plants and animals. Rates of stream flow are typically increased during times of high rainfall and decreased during dry

periods. Variability also occurs due to seasonal changes. Higher summer temperatures are associated with higher evaporation and transpiration rates, decreasing the water levels and stream flow. Riparian vegetation is also typically at its peak photosynthetic rate during this time. This increases plants' need for water and decreases the rate of stream flow.

Human action also has a great impact on the stream flow of a channel. Irrigation for agriculture and industry is the main source of stream water depletion found in the greater Vermont area. These factors can easily disrupt the flow of streams with dry periods followed by periods of high stream flow rates. Consistency of stream flow is important for the habitat quality, and consequently, the organisms living in those habitats. Pollution and sediment deposits that can contaminate stream habitats are likely to occur from non-point sources such as run off from neighboring treatment facilities and residential areas. Large streams typically have the ability to filter these pollutants through vegetation and fast stream flow rates; however, smaller streams do not filter pollutants as easily due to shallow depths, slow stream flow rates, and lower water levels.

What is stage?

Stream stage is the height, in feet, of the water surface from an established point, usually the stream bottom. There are a variety of ways stage can be measured. The U.S. Geological Survey (USGS) maintains thousands of stations throughout the country which, among many parameters, measure stage through the use of a stilling well or bubbler system. In a stilling well setup, a float or pressure sensor is suspended inside the well and an electronic data recorder logs stage readings. In other cases, a bubbler system determines stage by measuring the pressure required to send a flow of gas through a tube and out a fixed location in the stream. The deeper the water is, the more pressure is needed to push the gas out.

Stage is a key factor affecting stream discharge. Discharge is equal to a stream's velocity times its cross-sectional area, and area is equal to width multiplied by stage. Thus, stage is a stream property that impacts and is impacted by human activity. Changes in stage occur naturally throughout the seasons and with precipitation events, but can also be caused by human-induced erosion and can lead to damage of manmade structures, such as bridges and roads.

References

Adopt-A-Stream Foundation. *Field Guide: Watershed Inventory and Stream Monitoring Methods,* by Tom Murdoch and Martha Cheo. 1996. Everett, WA.

Mitchell, M.K., and W. Stapp. *Field Manual for Water Quality Monitoring.* 5th Edition. Thompson Shore Printers.

Missouri Stream Teams. *Volunteer Water Quality Monitoring.* Missouri Department of Natural Resources, P.O. Box 176, Jefferson City, MO 65102.

Temperature

Why is temperature important?

The rates of biological and chemical processes depend on temperature. Aquatic organisms from microbes to fish are dependent on certain temperature ranges for their optimal health. Optimal temperatures for fish depend on the species: some survive best in colder water, whereas others prefer warmer water. Benthic macroinvertebrates are also sensitive to temperature and will move in the stream to find their optimal temperature. If temperatures are outside this optimal range for a prolonged period of time, organisms are stressed and can die. Temperature is measured in degrees Fahrenheit (F) or degrees Celsius (C).

For fish, there are two kinds of limiting temperatures: the maximum temperature for short exposures and a weekly average temperature that varies according to the time of year and the life cycle stage of the fish species. Reproductive stages (spawning and embryo development) are the most sensitive stages. Table 2 provides temperature criteria for some species.

Temperature affects the oxygen content of the water (oxygen levels become lower as temperature increases), the rate of photosynthesis by aquatic plants, the metabolic rates of aquatic organisms, and the sensitivity of organisms to toxic wastes, parasites, and diseases.

Causes of temperature change include weather, removal of shading, stream bank vegetation, impoundments (a body of water confined by a barrier, such as a dam), discharge of cooling water, urban storm water, and groundwater inflows to the stream.

Sampling and Equipment Considerations

Temperature in a stream will vary with width and depth. It can be significantly different in the shaded portion of the water on a sunny day. In a small stream, the temperature will be relatively constant as long as the stream is uniformly in sun or shade. In a large stream, temperature can vary considerably with width and depth regardless of shade. If it is safe to do so, temperature measurements should be collected at varying depths and across the surface of the stream to obtain vertical and horizontal temperature profiles. This can be done at each site at least once to determine the necessity of collecting a profile during each sampling visit. Temperature should be measured at the same place every time.

Temperature is measured in the stream with a thermometer or a meter or sensor. Alcohol-filled thermometers are preferred over mercury-filled because they are less hazardous if broken. Armored thermometers for field use can withstand more abuse than unprotected glass thermometers and are worth the additional expense. Meters for other tests, such as pH (acidity) or dissolved oxygen, also measure temperature and can be used instead of a thermometer. For the purposes of this research we will use an iButton temperature sensor to measure temperature continuously.

Table 2: Maximum average temperatures for growth and short-term maximum temperatures for selected fish (°C and ° F). *(Brungs and Jones 1977)*

Species	Max. weekly average temp. for growth (juveniles)	Max. temp. for survival of short exposure (juveniles)	Max. weekly average temp. for spawning [a]	Max. temp. for embryo spawning [b]
Atlantic salmon	20 °C (68 °F)	23 °C (73 °F)	5 °C (41 °F)	11 °C (52 °F)
Bluegill	32 °C (90 °F)	35 °C (95 °F)	25 °C (77 °F)	34 °C (93 °F)
Brook trout	19 °C (66 °F)	24 °C (75 °F)	9 °C (48 °F)	13 °C (55 °F)
Common carp	---	---	21 °C (70 °F)	33 °C (91 °F)
Channel catfish	32 °C (90 °F)	35 °C (95 °F)	27 °C (81 °F)	29 °C (84 °F)
Largemouth bass	32 °C (90 °F)	34 °C (93 °F)	21 °C (70 °F)	27 °C (81 °F)
Rainbow trout	19 °C (66 °F)	24 °C (75 °F)	9 °C (48 °F)	13 °C (55 °F)
Smallmouth bass	29 °C (84 °F)	---	17 °C (63 °F)	23 °C (73 °F)
Sockeye salmon	18 °C (64 °F)	22 °C (72 °F)	10 °C (50 °F)	13 °C (55 °F)

a - Optimum or mean of the range of spawning temperatures reported for the species
b - Upper temperature for successful incubation and hatching reported for the species
c - Upper temperature for spawning

pH

What Is pH and why is it important?

pH is a term used to indicate the alkalinity or acidity of a substance as ranked on a scale from 1.0 to 14.0. Acidity increases as the pH gets lower. Fig. 5.9 presents the pH of some common liquids.

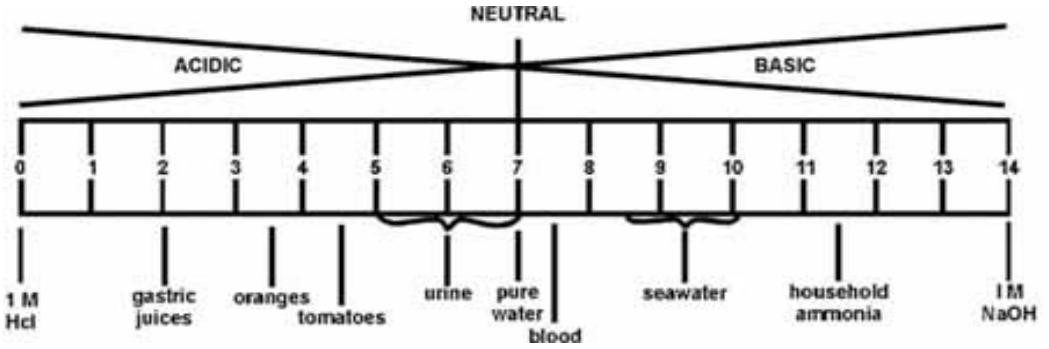

Figure 1: pH of selected liquids

pH affects many chemical and biological processes in the water. For example, different organisms flourish within different ranges of pH. A large variety of aquatic animals prefer a range of 6.5-8.0. pH outside this range reduces the diversity in the stream because it stresses the physiological systems of most organisms and can reduce reproduction. Low pH can also allow toxic elements and compounds to become mobile and "available" for uptake by aquatic plants and animals. This can produce conditions that are toxic to aquatic life, particularly to sensitive species like trout. Changes in acidity can be caused by atmospheric deposition (acid rain), surrounding rock, and certain wastewater discharges.

The pH scale measures the logarithmic concentration of hydrogen (H+) and hydroxide (OH-) ions, which make up water (H+ + OH- = H2O). When both types of ions are in equal concentration, the pH is 7.0 or neutral. Below 7.0, the water is acidic (there are more hydrogen ions than hydroxide ions). When the pH is above 7.0, the water is alkaline, or basic (there are more hydroxide ions than hydrogen ions). Since the scale is logarithmic, a drop in the pH by 1.0 unit is equivalent to a 10-fold increase in acidity. So, a water sample with a pH of 5.0 is 10 times as acidic as one with a pH of 6.0, and pH 4.0 is 100 times as acidic as pH 6.0.

Analytical and equipment considerations

pH can be analyzed in the field or in the lab. If it is analyzed in the lab, you must measure the pH within 2 hours of the sample collection. This is because the pH will change due to the carbon dioxide from the air dissolving in the water, which will bring the pH toward 7.

pH Meters

A pH meter measures the electric potential (millivolts) across an electrode when immersed in water. This electric potential is a function of the hydrogen ion activity in the sample. Therefore, pH meters can display results in either millivolts (mV) or pH units.

A pH meter consists of a *potentiometer,* which measures electric current; a glass electrode, which senses the electric potential where it meets the water sample; a reference electrode, which provides a constant electric potential; and a temperature compensating device, which adjusts the readings according to the temperature of the sample (since pH varies with temperature). The reference and glass electrodes are frequently combined into a single probe called a combination electrode.

The most important part of the pH meter is the electrode. Follow the manufacturer's instructions for proper maintenance. Infrequently used or improperly maintained electrodes are subject to corrosion, which makes them highly inaccurate.

Phosphorus

Why is phosphorus important?

Phosphorus is an essential nutrient for the plants and animals that make up the aquatic food web. Since phosphorus is the nutrient in short supply in most fresh waters, even a modest increase in phosphorus can, under the right conditions, set off a whole chain of undesirable events in a stream including accelerated plant growth, algae blooms, low dissolved oxygen, and the death of certain fish, invertebrates, and other aquatic animals.

There are many sources of phosphorus, both natural and human. These include soil and rocks, wastewater treatment plants, runoff from fertilized lawns and cropland, failing septic systems, runoff from animal manure storage areas, disturbed land areas, drained wetlands, water treatment, and commercial cleaning preparations.

Forms of phosphorus

Phosphorus has a complicated story. Pure, "elemental" phosphorus (P) is rare. In nature, phosphorus usually exists as part of a phosphate molecule (PO_4). Phosphorus in aquatic systems occurs as organic phosphate and inorganic phosphate. Organic phosphate consists of a phosphate molecule associated with a carbon-based molecule, as in plant or animal tissue. Phosphate that is not associated with organic material is inorganic. Inorganic phosphorus is the form required by plants. Animals can use either organic or inorganic phosphate.

Both organic and inorganic phosphorus can either be dissolved in the water or suspended (attached to particles in the water column).

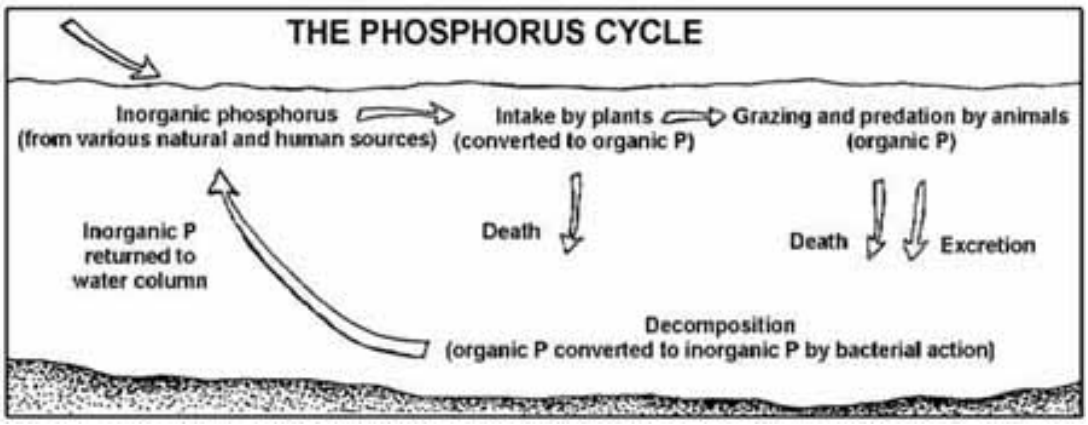

Figure 2: The Phosphorus Cycle: Phosphorus changes form as it cycles through the aquatic environment.

Phosphorus cycles through the environment, changing form as it does so (Fig. 2). Aquatic plants take in dissolved inorganic phosphorus and convert it to organic phosphorus as it becomes part of their tissues. Animals get the organic phosphorus they need by eating either aquatic plants, other animals, or decomposing plant and animal material.

As plants and animals excrete wastes or die, the organic phosphorus they contain sinks to the bottom, where bacterial decomposition converts it back to inorganic phosphorus, both dissolved

and attached to particles. This inorganic phosphorus gets back into the water column when the bottom is stirred up by animals, human activity, chemical interactions, or water currents. Then it is taken up by plants and the cycle begins again.

In a stream system, the phosphorus cycle tends to move phosphorus downstream as the current carries decomposing plant and animal tissue and dissolved phosphorus. It becomes stationary only when it is taken up by plants or is bound to particles that settle to the bottom of pools.

In the field of water quality chemistry, phosphorus is described using several terms. Some of these terms are chemistry based (referring to chemically based compounds), and others are methods-based (they describe what is measured by a particular method).

The term "orthophosphate" is a chemistry-based term that refers to the phosphate molecule all by itself. "Reactive phosphorus" is a corresponding method-based term that describes what you are actually measuring when you perform the test for orthophosphate. Because the lab procedure isn't quite perfect, you get mostly orthophosphate but you also get a small fraction of some other forms.

More complex inorganic phosphate compounds are referred to as "condensed phosphates" or "polyphosphates." The method-based term for these forms is "acid hydrolyzable."

Monitoring phosphorus

Monitoring phosphorus is challenging because it involves measuring very low concentrations down to 0.01 milligram per liter (mg/L) or even lower. Even such very low concentrations of phosphorus can have a dramatic impact on streams. Less sensitive methods should be used only to identify serious problem areas.

While there are many tests for phosphorus, only four are likely to be performed by volunteer monitors.

1. The *total orthophosphate* test is largely a measure of orthophosphate. Because the sample is not filtered, the procedure measures both dissolved and suspended orthophosphate. The EPA-approved method for measuring total orthophosphate is known as the ascorbic acid method. Briefly, a reagent (either liquid or powder) containing ascorbic acid and ammonium molybdate reacts with orthophosphate in the sample to form a blue compound. The intensity of the blue color is directly proportional to the amount of orthophosphate in the water.
2. **The *total phosphorus* test measures all the forms of phosphorus in the sample (orthophosphate, condensed phosphate, and organic phosphate). This is accomplished by first "digesting" (heating and acidifying) the sample to convert all the other forms to orthophosphate. Then the orthophosphate is measured by the ascorbic acid method. Because the sample is not filtered, the procedure measures both dissolved and suspended orthophosphate. <u>The Streams Project uses this method.</u>**
3. The *dissolved phosphorus* test measures that fraction of the total phosphorus which is in solution in the water (as opposed to being attached to suspended particles). It is determined by first filtering the sample, then analyzing the filtered sample for total phosphorus.
4. *Insoluble phosphorus* is calculated by subtracting the dissolved phosphorus result from the total phosphorus result.

All these tests have one thing in common: they all depend on measuring orthophosphate. The total orthophosphate test measures the orthophosphate that is already present in the sample. The others measure that which is already present and that which is formed when the other forms of phosphorus are converted to orthophosphate by digestion.

Sampling and equipment considerations

Monitoring phosphorus involves two basic steps:

- Collecting a water sample
- Analyzing it in the field or lab for one of the types of phosphorus described above

Sample Containers

Sample containers made of either some form of plastic or Pyrex glass are acceptable to EPA. Because phosphorus molecules have a tendency to "adsorb" (attach) to the inside surface of sample containers, if containers are to be reused they must be acid-washed to remove adsorbed phosphorus. Therefore, the container must be able to withstand repeated contact with hydrochloric acid. Plastic containers either high-density polyethylene or polypropylene might be preferable to glass from a practical standpoint because they will better withstand breakage. Some programs use disposable, sterile, plastic Whirl-pak® bags. The size of the container will depend on the sample amount needed for the phosphorus analysis method you choose and the amount needed for other analyses you intend to perform.

Dedicated Lab Glassware

All containers that will hold water samples or come into contact with reagents used in this test must be dedicated. That is, they should not be used for other tests. This is to eliminate the possibility that reagents containing phosphorus will contaminate the lab glassware. All lab glassware and containers that contain water for phosphorus analysis should be acid-washed. The only form of phosphorus this manual recommends for field analysis is total orthophosphate, which uses the ascorbic acid method on an untreated sample. Analysis of any of the other forms requires adding potentially hazardous reagents, heating the sample to boiling, and using too much time and too much equipment to be practical. In addition, analysis for other forms of phosphorus is prone to errors and inaccuracies in a field situation. Pretreatment and analysis for these other forms should be handled in a laboratory.

Ascorbic Acid Method

In the ascorbic acid method, a mix of reagents including sulfuric acid, potassium antimonyl tartrate, ammonium molybdate, and ascorbic acid added to the water sample. This colors the sample blue in direct proportion to the amount of orthophosphate in the sample. A spectrophotometer, a specialized laboratory instrument, measures the amount of light absorbed or transmitted at a wavelength of 700 - 880 nanometers. This measurement is called “absorbance” or “transmittance”.

We translate that absorbance into a meaningful phosphorus concentration (ug/L) by referencing a standard curve. The standard curve is a series of standard solutions with known concentrations of phosphorus. Those standards are the basis of our standard curve, which provides us with a reference for measuring unknown samples collected from the field.

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Most of this information was retrieved in May 2010
from: http://www.epa.gov/volunteer/stream/index.html However, some of this information was edited by EPSCoR Streams Project staff.

Note on analysis from Johnson State College Lab:
Samples were digested in 10ml aliquots as per Standard methods 4500-P J. TP was analyzed using the ascorbic acid colorimetry (AQ2 method EPA-118-A Rev. 5).

Nitrogen

Nitrogen is an essential element for life, just like phosphorus. It is essential for plant growth and in many systems it can be a limiting factor (which is why we fertilize our gardens!). However, an excess of nitrogen can cause significant water quality problems. An abundance of nitrogen can accelerate eutrophication, causing drastic increases in aquatic plant growth and harmful algae blooms. Some other impacts of excess of nitrogen you may have heard of include acid rain and increased ozone in the atmosphere.

Nitrogen occurs naturally and is found in fertilizers, waste water, runoff, and from the combustion of fossil fuels. It is more mobile in the environment than phosphorus (phosphorus is usually bound to soil particles). Nitrogen is found in several different forms in both terrestrial and aquatic ecosystems. These forms include ammonia (NH_3), nitrates (NO_3), and nitrites (NO_2). When it's in the water it is commonly in nitrate or ammonia and in abundance causes excessive plant and algae growth, leading to eutrophication.

Due to the abundant plant growth, excess nitrates can cause hypoxia (low levels of dissolved oxygen) and can become toxic to warm-blooded animals at higher concentrations (10 mg/L) or higher) under certain conditions. The natural level of ammonia or nitrate in surface water is typically low (less than 1 mg/L); in the effluent of wastewater treatment plants, it can range up to 30 mg/L.

The Nitrogen Cycle

Plants need nitrogen to grow, which they mainly obtain from organic matter in soil, provided in the form of animal and plant decay. This type of nitrogen in the decayed organic matter is unavailable to plants. Microorganisms in the soil convert the organic forms of nitrogen into inorganic forms that plants can use. Then when these plants die, they recontribute the nitrogen back into the cycle. Other major contributors of nitrogen to the landscape are from agriculture fertilizers and waste.

The other part of the nitrogen cycle is atmospheric in the form of N_2. It occurs naturally as 78% of the atmosphere. Precipitation brings down atmospheric nitrogen. Additionally, legumes (soybeans, alfalfa, clover) are also able to convert atmospheric nitrogen into plant usable nitrogen.

Sampling Considerations

Nitrates from land sources end up in rivers and streams more quickly than other nutrients like phosphorus. This is because they dissolve in water more readily than phosphates, which have an attraction for soil particles. As a result, nitrates serve as a better indicator of the possibility of a source of sewage or manure pollution during dry weather.

References

http://extension.missouri.edu/p/WQ252
http://water.epa.gov/type/rsl/monitoring/vms57.cfm

Note on analysis from Johnson State College Lab:

Samples were digested in 10ml aliquots as per Standard methods 4500-P J. TN was analyzed using A Copper/Cadmium Reduction followed by Sulfanilamide-NEDD colorimetry (AQ2 method EPA-127-A Rev. 6).

Total Suspended Solids (TSS)

What are solids and why are they important?

Total solids are dissolved solids plus suspended and settleable solids in water. In stream water, dissolved solids consist of calcium, chlorides, nitrate, phosphorus, iron, sulfur, and other ions particles that will pass through a filter with pores of around 2 microns (0.002 cm) in size. Suspended solids include silt and clay particles, plankton, algae, fine organic debris, and other particulate matter. These are particles that will not pass through a 2-micron filter.

The concentration of total dissolved solids affects the water balance in the cells of aquatic organisms. An organism placed in water with a very low level of solids, such as distilled water, will swell up because water will tend to move into its cells, which have a higher concentration of solids. An organism placed in water with a high concentration of solids will shrink somewhat because the water in its cells will tend to move out. This will in turn affect the organism's ability to maintain the proper cell density, making it difficult to keep its position in the water column. It might float up or sink down to a depth to which it is not adapted, and it might not survive.

Total suspended solids (TSS) are a common and useful measure of water quality in streams. Higher concentrations of suspended solids can often mean higher concentrations of bacteria, nutrients, pesticides, and metals in the water. These pollutants may attach to the sediment particles on the land and be carried into water bodies with storm water. In the water, the pollutants may be released from the sediment or travel farther downstream.

High concentrations of suspended solids can also cause problems for industrial use, and affect the efficiency of wastewater treatment plants, because the solids may clog or scout pipes and machinery.

Total suspended solids also affect water clarity. Higher suspended solids decrease the passage of light through water, thereby slowing photosynthesis by aquatic plants. Water will heat up more rapidly and hold more heat; this, in turn, might adversely affect aquatic life that has adapted to a lower temperature regime.

Sources of solids include industrial discharges, sewage, fertilizers, road runoff, and soil erosion. Total solids are measured in milligrams per liter (mg/L).

Sampling and equipment considerations

Total suspended solids are important to measure in areas where there are discharges from sewage treatment plants, industrial plants, or extensive crop irrigation. In particular, streams and rivers in arid regions where water is scarce and evaporation is high tend to have higher concentrations of solids and are more readily affected by human introduction of solids from land use activities. TSS measurements can be useful as an indicator of the effects of runoff from construction, agricultural practices, logging activities, sewage treatment plant discharges, and other sources. **As with turbidity, concentrations often increase sharply during rainfall, especially in developed watersheds**. They can also rise sharply during dry weather if earth-disturbing activities are occurring in or near the stream without erosion control practices in place. Regular monitoring of TSS can help detect trends that might indicate increasing erosion in developing watersheds. Total suspended solids are related closely to stream flow and velocity and should be correlated with these factors. Any change in TSS over time should be measured at the same site at the same flow.

Summary: How does RACC measure Total Suspended Solids (TSS)?

Total suspended solids are measured by weighing the amount of solids present in a known volume of sample. This is done by filtering a known volume of water sample through a pre-weighed filter. The residue retained on the filter is dried in an oven at 103 to 105° C until the weight of the filter no longer changes. The increase in weight of the filter represents the total suspended solids. Since the residue is so light in weight, the lab will need a balance that is sensitive to weights in the range of 0.0001 gram. Balances of this type are called analytical balances, and they are expensive (around $3,000). The technique requires that the filters be kept in a desiccator, which is a sealed glass container that contains material that absorbs moisture and ensures that the weighing is not biased by water condensing on the filter. Some desiccants change color to indicate moisture content.

The measurement of total solids cannot be done in the field. Samples must be collected using clean glass or plastic bottles or Whirl-pak® bags and taken to a laboratory where the test can be run.

References

APHA. 1992. *Standard methods for the examination of water and wastewater.* 18th ed. American Public Health Association, Washington, DC.

Water Quality Assessment Data Sheet

2015-2016

Stream Name:	Site Code:
Latitude/Longitude:	Date/Time:
Site Description:	Investigators:

Weather conditions:

Now		**Past 24 hours**
___	Storm	___
___	Rain (steady)	___
___	Showers (intermittent)	___
___	Clear/sunny	___
___	% cloud cover	___

Has there been heavy rain in the last 7 days?
Air temperature (°C):

Instream Features:

Parameter	**Field Measurement**
Water temperature	°C
Water pH	
Stream depth	m
Discharge (calculated on separate sheet)	m^3/s
Canopy cover	%
Obvious pollution	Yes or No Describe:

Comments:

DATA FORM FOR CALCULATING FLOW

Solving the equation: $\text{Flow} = \frac{A\,L\,C}{T}$

Where:

A = Average cross-sectional area of the stream. L = Length of the stream reach measured (usually 6.5 meters). C = A coefficient or correction factor (0.8 for rocky-bottom streams or 0.9 for muddy-bottom streams). T = Time, in seconds, for the float to travel the length of L.

A: Average Cross-Sectional Area

Transect #1 (upstream)

Interval width (meters)	Depth (meters)
A to B = ____	____ (at B)
B to C = ____	____ (at C)
C to D = ____	____ (at D)
D to E = ____	____ (shoreline)
Totals [ ]	[ ] ÷ 4

= Avg. depth [ ] m

Cross-sectional area of Transect #1

= Total width (m) X Avg. depth (m)

[ ] X [ ] = [ ] m^2

Transect #2 (downstream)

Interval width (meters)	Depth (meters)
A to B = ____	____ (at B)
B to C = ____	____ (at C)
C to D = ____	____ (at D)
D to E = ____	____ (shoreline)
Totals [ ]	[ ] ÷ 4

= Avg. depth [ ] m

Cross-sectional area of Transect #2

= Total width (m) X Avg. depth (m)

[ ] X [ ] = [ ] m^2

(Cross-sectional area of Transect #1 + Cross-sectional area of Transect #2) ÷ 2 = Average Cross-sectional area

A = ([ ] m^2 + [ ] m^2) ÷ 2 = [ ] m^2

L: Length of Stream Reach

[ ] m

C: Coefficient

[ ]

T: Travel Time

	Travel Time of Float (sec.)
Trial #1	____
Trial #2	____
Trial #3	____
Total	[ ] ÷ 3

= Avg. time [ ] sec.

Flow = $\frac{A\,L\,C}{T}$ = $\frac{[\ ]\ [\ ]\ [\ ]}{[\ ]}$ = [ ] **m^3/sec.**

Field Method:

Flow & Discharge

I. Introduction

Discharge, or stream flow, is the volume of water that moves over a designated point over a fixed period of time. The rate of discharge is expressed in cubic meters per second (m^3/sec) and is calculated using measurements of stream width, depth, and velocity.

A stream's discharge is directly affected by factors such as precipitation, riparian vegetation, and surrounding land use. The volume and velocity of the water directly impacts water quality and a stream's ability to support macroinvertebrate life.

The average cross-sectional area of a stream reach is determined by measuring the total stream width and stream depth along two transects. Stream velocity is determined by measuring the time it takes a tennis ball to travel the length of a stream reach. Stream flow is calculated from these variables according to the equation Flow = (A x L x C) / T.

II. Equipment and Materials

a. Meter tape
b. Meter stick
c. Stopwatch
d. Tennis ball
e. Waders
f. Data entry form
g. Clipboard and pencil

III. Average Cross-Sectional Area

a. Transect #1
   i. Using your measuring tape, designate the upstream end of your stream reach by stretching the measuring tape across the stream perpendicular to the stream banks.
   ii. Measure the width of the stream from wetted edge to wetted edge. Record as the Total Width in meters under Transect #1 (Upstream) on the data entry form.
   iii. Divide the Total Width into four equal intervals. Record as the Interval Width from Point A to B, Point B to C….D to E.
      1. Example: Total Width=12 meters→Intervals= 3 meters
   ii. Measure the water depth at each interval point (see Figure 3) and record as the depth in meters. Interval E is the shoreline, so its depth may be 0 meters.
   iii. Follow the calculations on the data entry form to determine the Cross-Sectional Area of Transect #1.
b. Designate Reach Length
   i. Using your measuring tape, measure 6 meters (~20 feet) downstream from Transect #1. Record as the Length of Stream Reach (L) in meters.
   ii. The downstream end will be Transect #2.
c. Transect #2
   i. Follow the steps provided above for Transect #1 and record all values under Transect #2 (Downstream) on the data entry form.
   ii. Calculate the Cross-Sectional Area of Transect #2.
d. Following the directions on the data entry form to calculate the Average Cross-Sectional Area (A) of the stream reach

V. Travel Time

a. Position one researcher above Transect #1 with the tennis ball. A second researcher should be positioned below Transect #2 ready to catch the tennis ball as it travels downstream.
b. The upstream researcher should drop the tennis ball slightly upstream of Transect #1. Position the ball so that it will travel along the fastest current.
c. Using a stopwatch, begin timing when the tennis ball passes Transect #1 and stop timing when the ball completely passes Transect #2. The downstream researcher should catch the tennis ball.
d. Record the time as the Travel Time (T) in seconds.
e. Repeat steps a-d for a total of at least three trials.
f. Calculate the Average Travel Time (T) in seconds.

VI. Specify the Coefficient or the Correction Factor (C)

a. Enter 0.8 for rocky-bottom streams or 0.9 for sandy-bottom streams

VII. Calculate Discharge

a. Flow = (A x L x C)/T

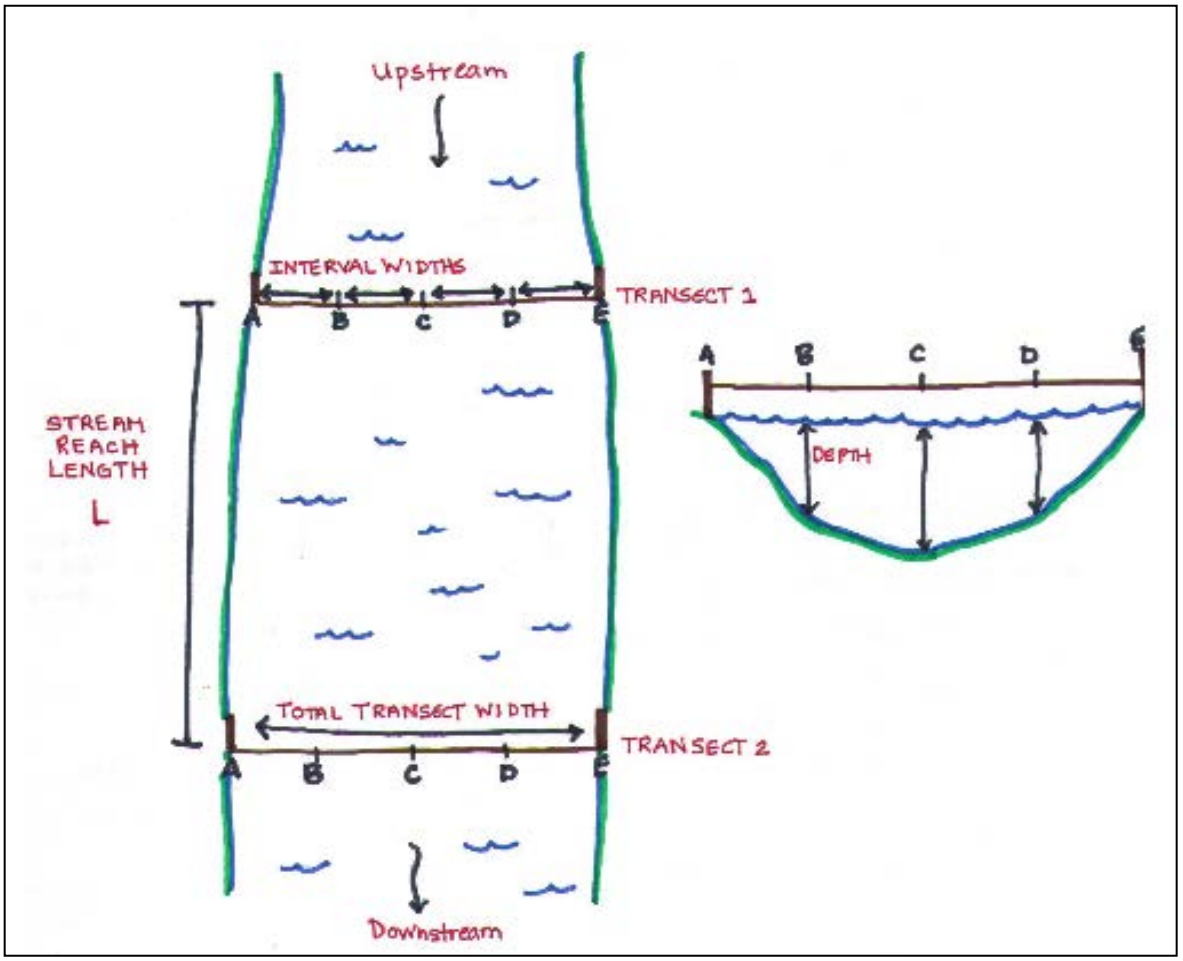


Figure 3: Discharge Diagram

Field Method:

pH and Temperature

The pH meter must be calibrated before every stream visit (or weekly, depending). They are calibrated using two pH buffer solutions (7 and 4). The buffer solutions should be at room temperature when you calibrate the meter. Because buffer pH values change with temperature, the meter must have a built-in temperature sensor that automatically standardizes the pH when the meter is calibrated.

Calibration Procedure:

1. Turn on the meter by pressing the On/Off button.
2. Press and hold the On/Off/Cal button until the "OFF" message becomes "CAL," release the button.
3. At this point, the meter can be immersed in the buffer 7 solution (yellow colored).
4. When buffer 7 has been accepted by the meter, the meter will display, "pH 4.01 Use."
5. Rinse the pH meter with tap or stream water.
6. At this point, the meter can be immersed in the buffer 4 solution (pink colored).
7. When the buffer 4 has been accepted by the meter, the meter will display, "Ok 2."

The meter is now calibrated and ready for use!

In general, sample away from the stream bank in the main current. The outside curve of the stream is often a good place to sample since the main current tends to hug this bank. In shallow stretches, wade into the center current carefully to measure temperature. If wading to the center current is not possible, reach out from the shore as far as **safely** possible.

Taking a Measurement:

1. Place the pH meter probe into at least 4 inches below the surface of the stream. Allow the meter to become acclimated to the stream.
2. When the readings have stabilized at a constant reading (about 1 minute), record the pH and temperature readings on the Water Quality Assessment Data.
3. To store the meter, add tap or stream water to the pH meter cap and store it standing up.

References

Brungs, W.S. and B.R. Jones. 1977. T*emperature Criteria for Freshwater Fish: Protocols and Procedures.* EPA-600/3-77-061. Environ. Research Lab, Ecological Resources Service, U.S. Environmental Protection Agency, Office of Research and Development, Duluth, MN.

APHA. 1992. *Standard methods for the examination of water and wastewater.* 18th ed. American Public Health Association, Washington, DC. River Watch Network. 1992. Total alkalinity and pH field and laboratory procedures (based on University of Massachusetts Acid Rain Monitoring Project). July 1.

Field Method:

Water Samples for TSS & Nutrients/Phosphorus

I. Introduction

Water quality samples are taken for nutrients and total suspended solids at stream sites every 2 to 3 weeks. Samples are collected in analyte-specific screw-top sample bottles. Following collection, the samples are kept cold and delivered to the Saint Michael's College Water Quality Laboratory.

II. Equipment and Materials

a. Waders
b. Labels and Permanent Marker
c. Bottle of Phosphorus blank water
d. Cooler
e. Ice packs or ice
f. Sample bottles: 4 Total Phosphorus bottles per site (125mL) and 3 TSS bottles (1L) per site

Figure 4: 4 Nutrients/Total Phosphorus bottles (125mL)

Figure 5: 3 Total Suspended Solids bottles (1L)

III. Prior to Departure

a. Label all bottles according to the Water Quality Field Labels protocol with the site code, date, analyte and replicate number
b. Confirm that you have all the equipment and materials listed above

IV. Prior to Sample Collection: Field Blank

a. Before taking stream samples, a field blank must be taken for each set of total phosphorus samples.
b. Following the same field method you use to collect the water samples (being careful not to touch the inside of the cap, etc.), fill the phosphorus blank bottle with total phosphorus blank water, leaving approximately 1/2 inch of air space. See Figures 6 and 7.

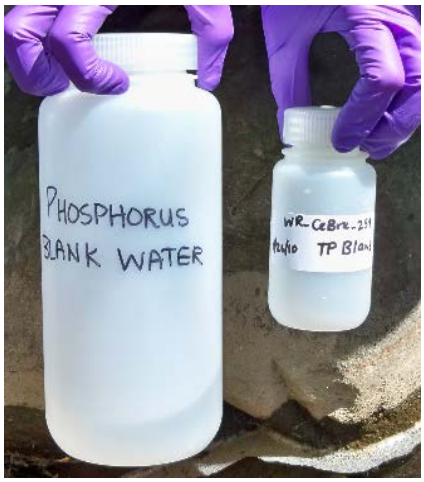


Figure 6: Phosphorous blank bottles

Figure 7: Leave ½ inch of air space

V. Sample Collection

a. Enter the water downstream from your sampling site and move away from the stream bank into the main current. Avoid disturbing the sediment as much as possible.
b. Stand facing upstream.

c. Carefully remove the cap from the bottle just before sampling. Avoid touching the inside of the bottle or cap. If you accidentally touch the inside of the bottle, use another one.
d. Hold the bottle near its base and plunge it (opening downward) below the water surface. Collect a water sample 8 to 12 inches beneath the surface or mid-way between the surface and the bottom if the stream reach is shallow.
e. Turn the bottle underwater into the current. In slow-moving stream reaches, push the bottle underneath the surface and in the upstream direction.

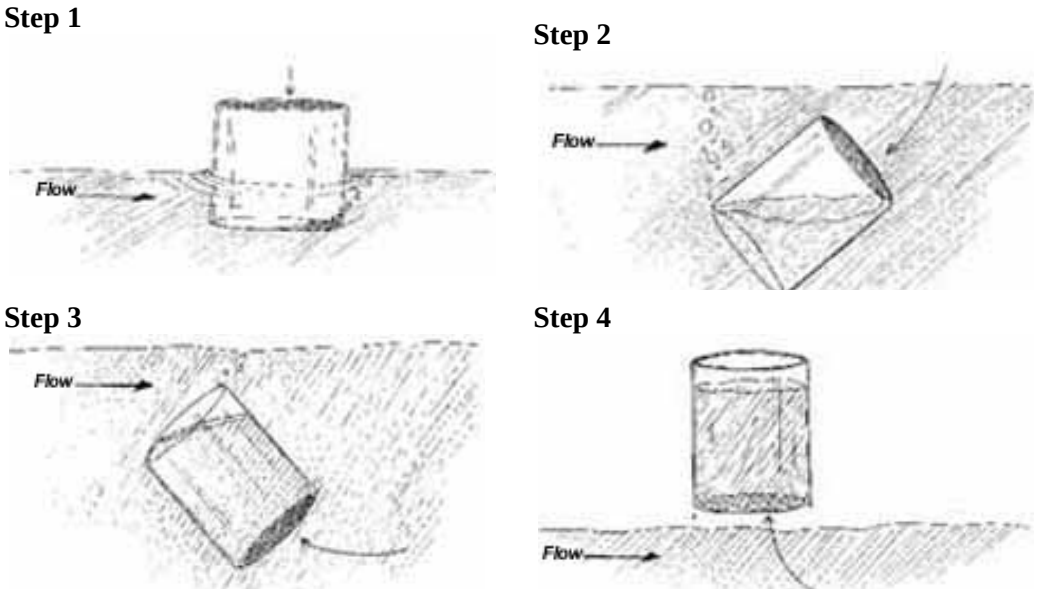


Figure 8: Sample Collection – how to fill water sample bottles

f. If necessary, adjust the volume of sample by gently pouring water out of the bottle
    i. Total Phosphorus samples: see Figure 8 for filling directions
    ii. TSS samples: fill completely
g. Make sure to complete the above steps (in Section V) for three total phosphorus and three TSS bottles at each site.
h. Place all samples, including blanks, in a cooler on ice for transport back to school or to the SMC Water Quality Laboratory

Figure 9: Cooler for Transporting Samples back from field sites

Water Quality Field Labels

*Please use labels provided to you by the VT EPSCoR staff

Site Code
Date
Analyte and Replicate Number

Analyte Abbreviations:
Phosphorus: **TP**
Total Suspended Solids: **TSS**

Total Phosphorus Samples

WR_CeBrk_259
4/26/2010
TP 1 of 3

Total Suspended Solids

WR_CeBrk_259
4/26/2010
TSS 1 of 3

Shipping Water Quality Samples to St Mike's WQ Lab

I. Introduction

Following sample collection, water quality samples are kept cold and shipped to the Water Quality Laboratory for analysis via UPS or USPS. Samples are to be shipped the next day after the sampling day.

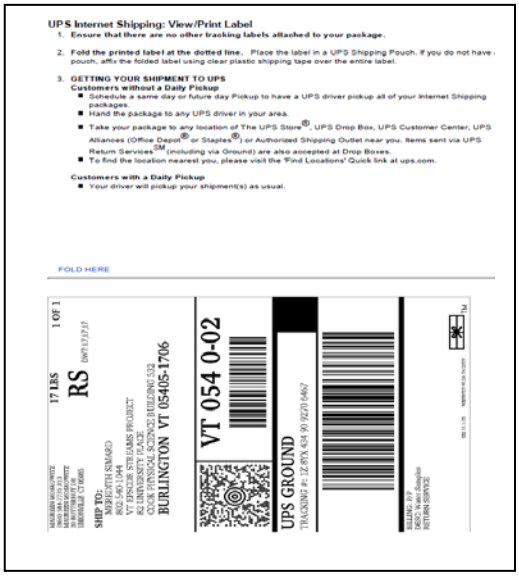
REMEMBER: Phosphorus samples must be stored in the freezer and TSS samples must be stored in the refrigerator before shipping. Phosphorus samples should be completely frozen before shipping. TSS samples cannot be frozen.

II. Equipment

a. Printed UPS or USPS Return Label
b. ThermoSafe Fisher insulated shipping box
c. Frozen cold packs
d. Water quality samples

III. Print UPS or USPS Return Label

a. Before your sampling day, notify **Janel** (email cwdd@smcvt.edu or call 802-654-3271) when you plan to sample so she can be ready to receive and process the samples in the lab. Let her know what you will be shipping (how many water or soil samples) and if possible, provide her with an estimate of the final weight in pounds. She will email you a prepaid return ground shipping label as a PDF attachment.
b. Open and print the label:



IV. Packing Samples

a. You have received 1-3 ThermoSafe insulated shipping boxes

b. Immediately following sample collection, place your TP bottles in the freezer and your TSS bottles in the fridge.
c. The next morning, pack your water quality samples inside the shipping box. Leave the white cooler inside the cardboard box. Add 6 freezer packs to keep the samples cold during transport.

d. Put the lid on the cooler. Close and seal the cardboard box with tape.
e. Tape the printed shipping label to the top of the box, covering any previously used labels.

V. Ship Samples

a. Bring your packed box to your local UPS store or USPS office. Follow this link to find a store that is closest to you: http://www.ups.com/dropoff?loc=en_US

VI. Confirm Shipment

a. Send Janel an email at cwdd@smcvt.edu to let her know that the samples have been shipped.
b. Enter the information from your Water Quality Assessment into the Streams Project Database.

Sensor Stewarding

Contents

The iButton Temperature Sensor

General Information

As described earlier in this section, scientists are interested in studying the temperature of bodies of water for many reasons. The RACC research program is interested in temperature data because of its effects on aquatic organisms such as microbes, fish, and benthic macroinvertebrates which are adapted to surviving within certain temperature ranges.

What is a Thermochron iButton?
The Thermochron iButton (Figure 1 below), is a durable tool which measures and records temperature data. As the user, you define the rate at which the device records temperature. The iButton can store up to 2048 temperature values taken at equal time intervals ranging from 1- 255 minutes. It can take up to 10 minutes for an iButton to register a change in temperature.

The iButton is not waterproof.
Please keep it out of the stream environment unless in its protective capsule casing.

Figure 1. Thermochron iButton

The temperature sensor will need to be checked for general care and maintenance. The Tasks include:

Performed once around time of first visit:
Task 1: Download the One-Wire Viewer software and program the iButton for a mission
Task 2: Install the iButton housing in the stream

Performed every 2-3 Weeks:
Task 3: Visit the site and check on the iButton, fill out a Sensor Field Data Sheet

Last Site Visit:
Task 4: Remove the iButton and housing from the streambed
Task 5: Download the iButton data to your computer
Task 6: Depending on your team's status, you may be asked to return the iButton to the stream to collect temperatures over the winter! Please be in touch with Janel.

For Additional Help:
Visit http://www.maxim-ic.com/app-notes/index.mvp/id/3358 and scroll down to the section that says "Device Function Viewers." Here you will find detailed instructions that explain how to navigate and use the Thermochron viewer, how to start new missions, and how to view and download data.

For more help, please visit: http://www.maxim-ic.com/app-notes/index.mvp/id/5335

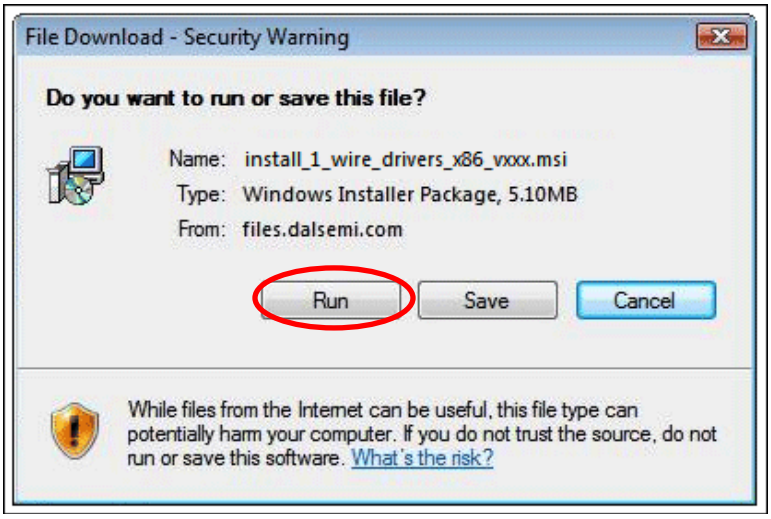
Task 1: Downloading iButton Software

You must first download the iButton software (called One-Wire Viewer) and install it onto your office computer. With this software, you will be able to manage and download data from the iButton. This is a free download, but is not available for Mac users.

1. Direct your web browser to:
   http://www.maxim-ic.com/products/ibutton/software/tmex/download_drivers.cfm

   Select your appropriate operating system. Also select 32 or 64-bit (you can check this by clicking on the link on the page, "Is my computer running a 32-bit or 64-bit operating system?"). If you have problems, you may need to download the latest version of Java. Go to http://java.com/en/ to install Java or check for Java update version.

2. After clicking "Download," click "Run." In the window (shown below) that asks, "Do you want to run or save this file?" Select "Run."



3. After this, a window will appear (shown below), asking if you would like to run the software, click "Run".

4. The **One-WireViewer Setup Wizard** will pop up, follow the prompts below
   a. Select "Next"
   b. Accept the license agreement and select "Next"

c. Choose a destination folder (default should be acceptable), select “Next”
d. Finally, click “Install”
e. One-WireViewer will install and notify you of a successful install.

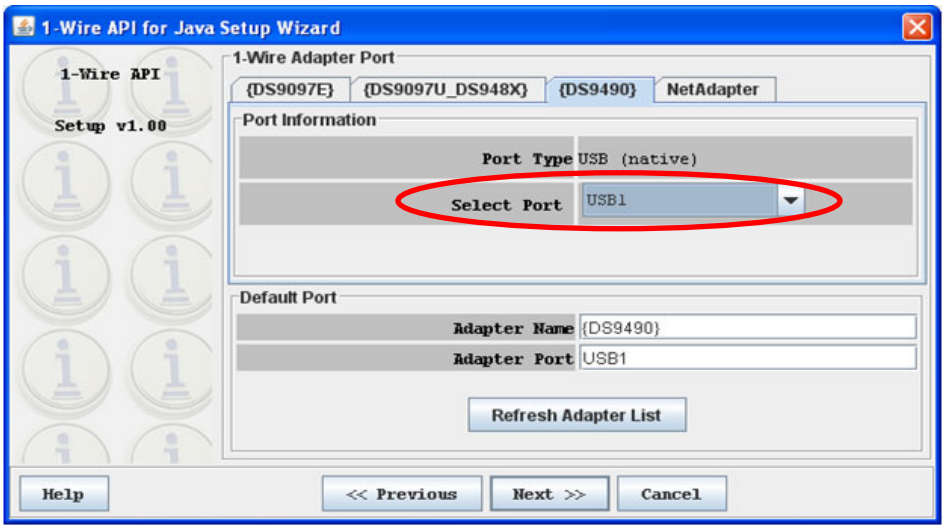
5. Once you have installed the software, you are ready to plug your USB iButton® adapter into the computer.

   Insert the USB, without the iButton inside, to the computer, as shown below:

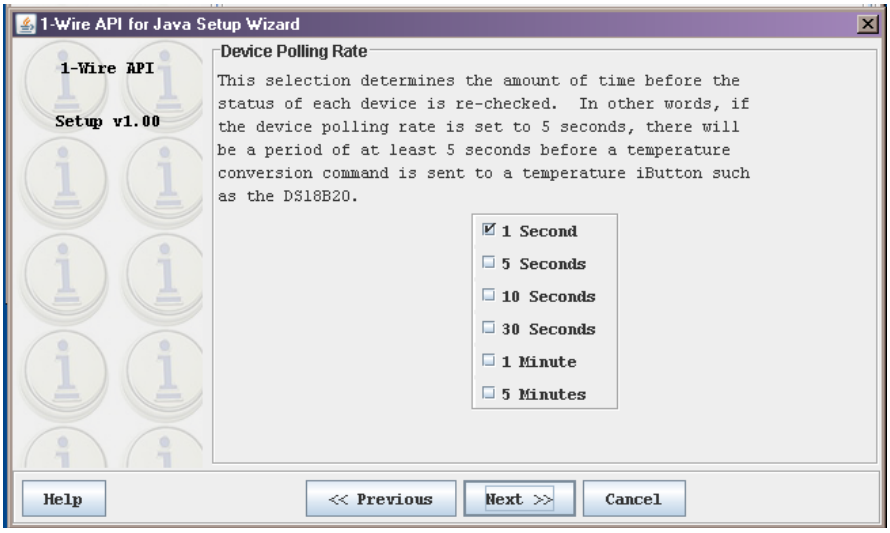
6. Once this has been inserted, Windows will identify the USB adaptor as new hardware and automatically install the driver software. This should only take a few moments, click “Close,” or “Finish,” when complete.

7. Once this is complete:
   a. Go to the “Start” menu
   b. Click on “Programs”
   c. Scroll to the file that says "1-Wire Drivers x86"
   d. Click on OneWireViewer.exe

8. When the One-Wire Viewer is run for the very first time, it will launch the **1-Wire API for Java Setup Wizard**. The initial screen (see below) asks for the type of adapter and port. In order to decide which port to select from the “Select Port” drop down menu, look at where you plugged the iButton adapter into, if it is the top or first USB port, select USB1, if it is the second or second from the top, select USB2, etc.



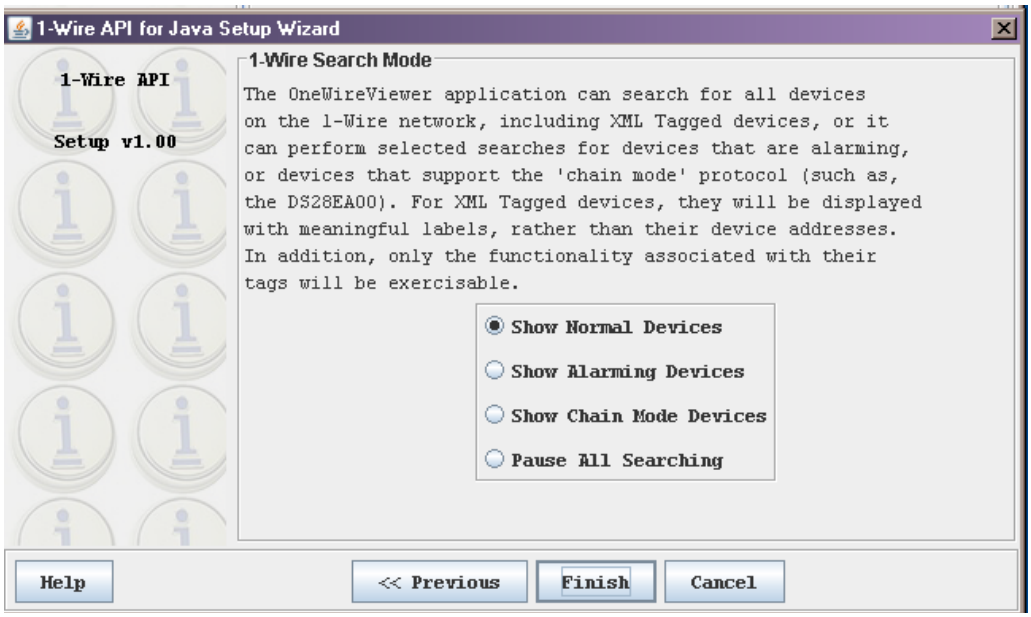
9. Click “Next,” if the port selection was successful, you will see the screen below:



If instead you see an error message “1-Wire Net not available,” please visit http://www.maxim-ic.com/app-notes/index.mvp/id/5057#port-selection for help.

Keep the default polling rating, and then click “Next.”

10. The following screen will appear:

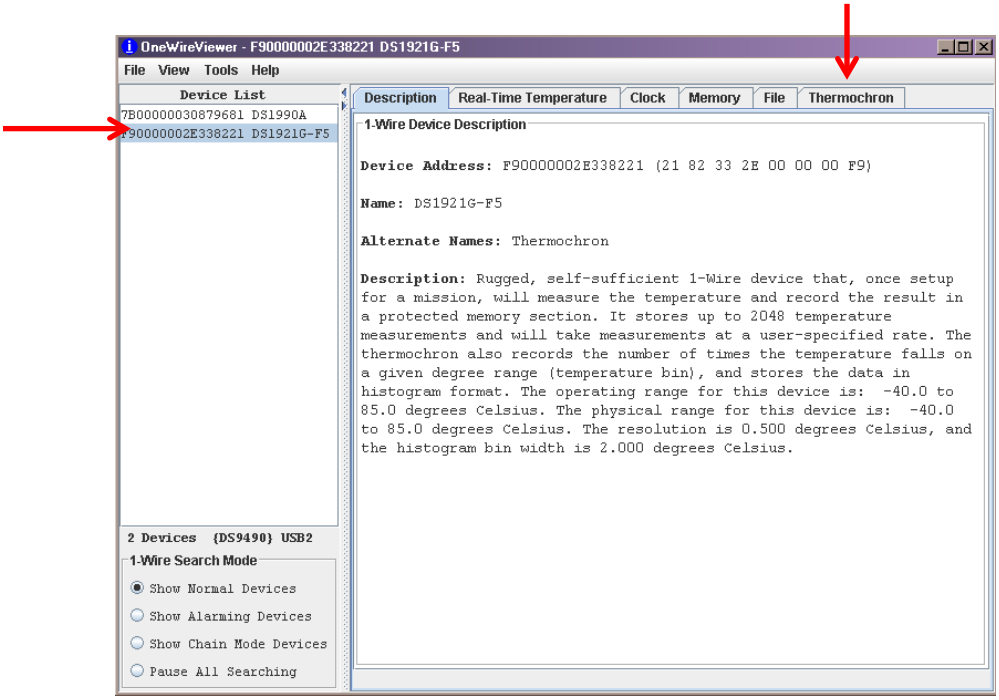


Keep the “Show Normal Devices” selection and click “Finish.”

11. You may now insert the iButton into the back section of USB iButton adapter, insert it so that the flat side of the iButton is facing the engraved (i) part of the adapter, or it will not be registered by the computer.

USB iButton Adapter

12. The following window will appear, click on the second device listed, DS1921G-FG, (horizontal red arrow shown below).

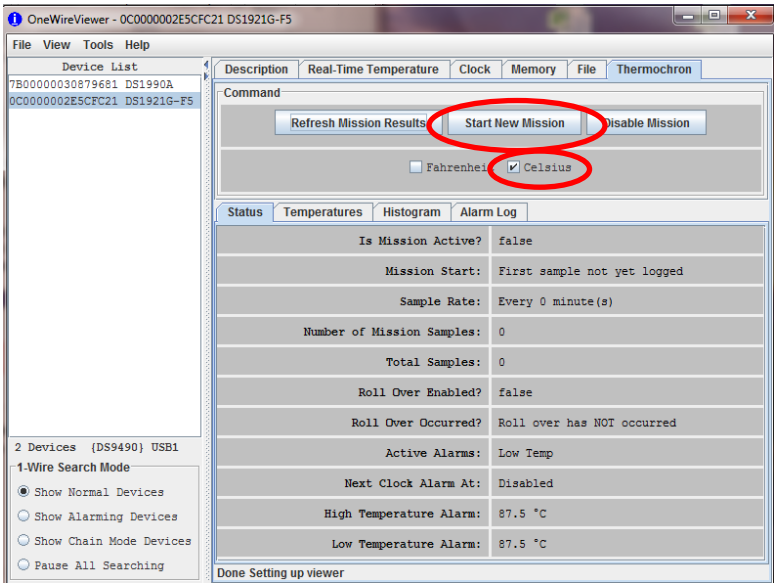


13. Click on the “Thermochron” tab (vertical red arrow above) to program the iButton and to view/download temperature data.

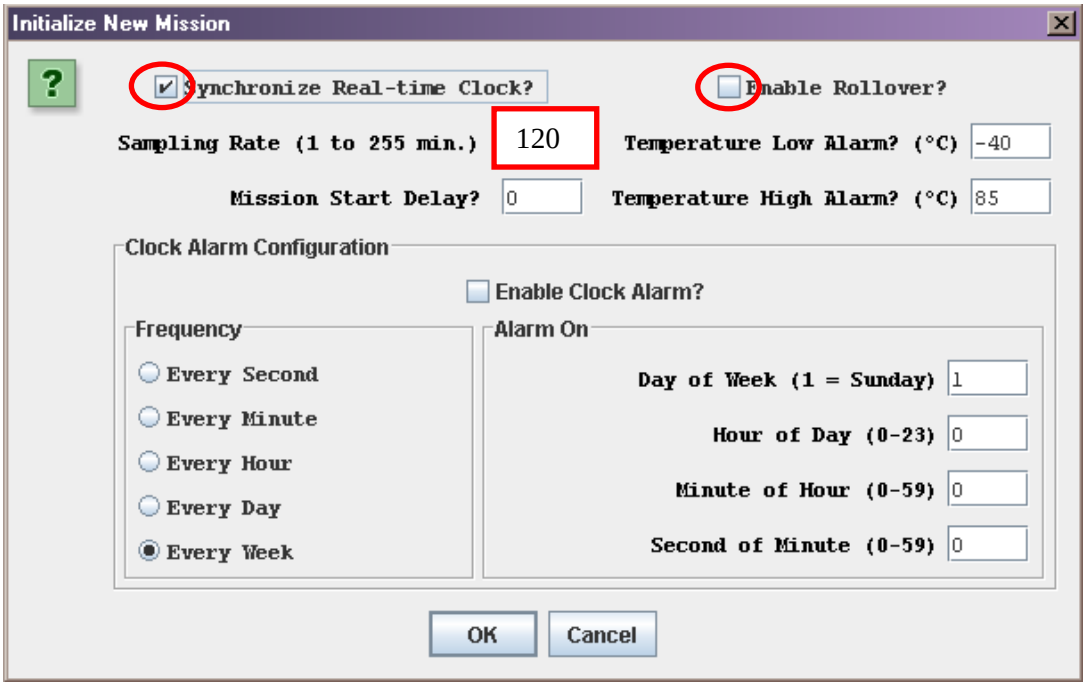
Program the iButton for a Mission

Before deployment of the iButton, you will have to program it first to tell it how often you wish it to take a temperature reading. This is done with the iButton inserted into the USB Adaptor, with the One-Wire Viewer software.

1. With the iButton plugged into the computer through the USB Adaptor, open up One Wire Viewer. Within the "Thermochron" tab, be sure temperature is being taken in degrees Celsius, and then click on the "Start New Mission" button, as shown below.



2. As shown below, ENABLE the "Synchronize Real-time Clock" and ENABLE rollover. Enabling this choice on your iButton, once it reaches maximum storage capacity, causes the device to overwrite data from the beginning of the mission to allow for new data to be stored. Since you will be collecting data frequently enough, this will not be a problem. **Be sure that the Sampling Rate is set at 120 minutes (or to sample every 2 hours)**.



3. If you like, you may choose to set a delay start to your mission until the time you believe you will enter your iButton into the stream. Enter delay time in minutes. Make note of this on your first Sensor Field Data Sheet.

4. Leave the Clock Alarm Configuration settings as they are.

5. Click "OK" to start the mission.

Removal from USB Adaptor:

Once programming/communication with the iButton is complete, close the program, eject the device, and unplug the USB from the computer, then insert the straight edge of a paper clip into the side of the adapter, pushing towards the iButton in order to discharge the device from the adapter.

To eject iButton from USB adapter

iButton Capsule:

This capsule protects the device from environmental risks such as moisture, pressure, and solvents. The stainless steel screws on the top of this capsule allow the iButton to be mounted to a cable, enabling the user to secure it in a stream environment. Once your iButton is secured in the capsule, it is ready for use. A small screwdriver has been provided to ensure it is always tight.

iButton Capsule

Task 2: Install the iButton Housing

As explained earlier in this section, water temperature varies within a stream environment. Therefore, it is important to install the iButton in an appropriate sampling location where representative readings will be made.

Equipment and Materials

a. iButton
b. iButton capsule
c. Ziploc bag for storage and transport
d. Rebar with attached PVC housing
e. Small screw driver
f. Hammer (ideally a mallet or post pounder)
g. Work gloves
h. Eye protection

1. Upon arriving at your site, scope out the area upstream and downstream to determine an ideal location. Choose a spot in or near a riffle where the water will be well mixed and not stagnant in a pool. There is a tendency of pools and shallow water to be warmer than the majority of the stream. Also, sections with lots of cobbles or gravel where there is no bedrock, fine sediment or clay, is best. You may even consider a spot behind a large boulder that can act as a landmark and protection. Finally, make sure that the logger will be submerged throughout the monitoring period (below your best low water/August estimate), and away from human activity (i.e. do not place the temperature logger in a popular fishing spot).

2. Be sure to thoroughly document where the logger is placed (in the comments section of your first Sensor Field Data sheet) to ensure its retrieval. Consider taking pictures of the site, noting date and time of placement, and describing the install location to help you relocate the sensor.

3. Once a spot in the stream has been chosen, put on work gloves and eye protection, and hammer the rebar or nail stake into the streambed. The rebar should be sunk about 3 feet deep and feel secure.

4. Place your iButton in the capsule and ensure the cap is tight. Check the housing and make sure it is secure to the rebar. The iButton housing is meant to keep the capsule with iButton underwater and protected from rocks and debris. Also, it may be helpful to tie flagging on either side of the bank across from the installation so that it is easy for you to find in the future.

Task 3: Site Visits

Every two to three weeks, visit the stream site to check the iButton housing. Be sure to completely fill out the Sensor Field Data sheet. Note that you'll need to fill in the date and time of when you program the iButton, deploy it in the stream, and remove it at the end of the season.

It is important to record dates and times of placement and removal of iButtons on the field sheet so that temperature logged outside of the stream can be deleted at a later time.

1. Navigate to your site and carefully wade out to your installation.

2. Pull the PVC housing above the water's surface (housing will still be connected to rebar).

3. Hold a kitty litter tray underneath the PVC housing (in case the iButton capsule falls out). Unscrew the housing and inspect the inside. Ensure that the iButton capsule is within the PVC housing and clean the housing of any sand or debris that may have clogged up inside.

4. Replace the iButton capsule in the housing and re-submerge the housing in the stream.

5. Ensure the Sensor Field Data Sheet is filled out.

Task 4: Retrieving the iButton

You have set the temperature sensor to record every two hours, which allows for almost 6 months of data. Therefore, to record a full year of data, we must remove and download the data at least once to ensure there is enough memory space. It is ideal to remove the temperature sensor before the stream freezes-over and before the water has become cold enough that it is unsafe to continue monitoring. As winter approaches, keep an eye on the weather and use your best judgment for deciding when to retrieve the sensor (possibly in late November).

Depending on your team's status, you may be asked to return the iButton after Task 5 (described below). Please in touch with Janel for details (email jroberge@smcvt.edu or call 802-654-3271).

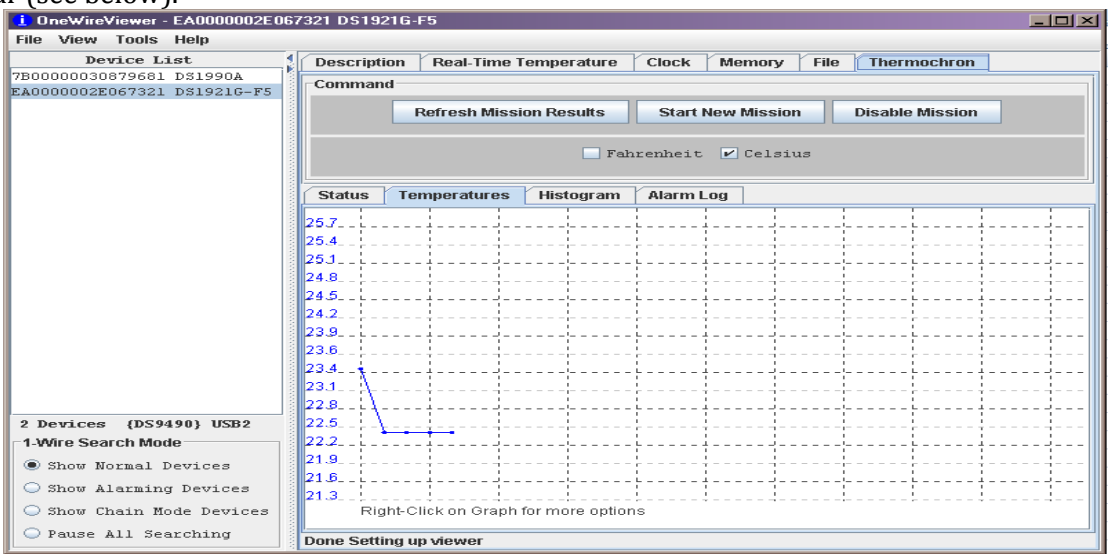
Task 5: Back at the Office

Once you have retrieved the iButton, return to the office to your host computer where the One-WireViewer has been installed.

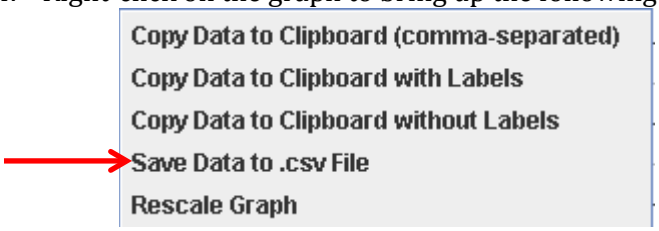
1. Plug the iButton into the USB adapter, plug it into the computer and again go to: Programs > 1-Wire Drivers x86 > 1-WireViewer.exe.

2. Select the second device (your iButton®) from the device list, then select the "Thermochron" tab from the tabs on the right.

3. Next select the "Temperatures" tab within the Thermochron window and a graph should appear (see below).



4. Right-click on the graph to bring up the following widow:



5. Choose the "Save Data to .csv File" option and title the file with the stream site code and date (Ex: "LCD_BrtltBrk_139.csv"). Email the .csv file to Janel Roberge at jroberge@smcvt.edu with the subject, "High School temp."

6. This entire .csv file represents all the temperature readings taken since you started the mission at the start of the field season. Refer to the Data Analysis section of this manual for QCing your temperature data.

7. After you complete the final download, place the two iButtons, capsules, and USB adaptor in a Ziploc bag for storage. Store the equipment in a safe place until the Symposium in the spring.

References:

Mauger, S. 2008. Water temperature data logger protocol for Cook Inlet salmon streams. Cook Inletkeeper, Homer, Alaska. 10 p.

http://www.maxim-ic.com/

Task 6: The Return

Depending on the status of your team and which streams you are sampling, you may be asked to return the iButton to the stream to continue collecting temperature data over the winter. Please be in contact with Janel (jroberge@smcvt.edu). If you will be participating in the Streams Project next year, please follow the instructions from above, Task 1 steps 3-8, for programing the iButton again for deployment. These will be retrieved by the technician in the spring!

If you do not need to return the iButton, try and remove the installation from the stream. If you are unable to remove the rebar, remove the PVC housing and cable from the rebar using a screwdriver to undo the hoseclamps.

The Stage Sensor

General Information

As described in the Water Quality section of this manual, there are many methods for measuring stage, or water level, of fresh water streams. For the purpose of this research, the use of a water level data logger is ideal. Water level data loggers are battery-powered devices that measure stage at regular intervals and/or during or after storm events. RACC uses a data logger called the HOBO U20.

Figure 2. The HOBO Water Level Logger, U20 Model
Image courtesy of http://www.onsetcomp.com/products/data-loggers/u20-001-01

General Care – The Stage Sensor is a delicate and expensive piece of equipment, please take care not to bash or drop it. Upon site visits, ensure that the PVC housing is still intact, that is has not been knocked over from a storm event, and that the inside of the PVC pipe is free of debris. You may have to clean the pressure port (shown above) of algae buildup. If you have any questions or concerns, contact Janel at 802-654-3271.

What is a HOBO Water Level Data Logger and how does it work?
A HOBO Water Level Data Logger, or stage sensor, is programmed to collect data at user-defined time intervals or under set conditions, such as during storm events. To obtain stage information, the sensor takes a reading of water pressure and uses this reading to calculate water depth. The sensor stores the stage data until it is reconnected to a computer or a waterproof data shuttle shown below in Figure 3.

Figure 3. The HOBO U-Shuttle
Image courtesy of http://www.onsetcomp.com/products/communications/u-dtw-1

The HOBO sensor detects both atmospheric and water pressure. To calculate stage from a pressure reading, we are interested only in the amount of water pressure a HOBO sensor is experiencing. Therefore, a second HOBO sensor is installed above ground to record the atmospheric (or barometric) pressure at the site. We can then subtract the atmospheric pressure from the underwater HOBO sensor's pressure reading. Thus, we can determine the true water pressure and calculate an accurate stage reading.

How is a HOBO Water Level Data Logger installed?

The ideal spot in a stream for the sensor to be placed is in a pool below a riffle and where sediment and debris are unlikely to collect. A stilling well, made of PVC pipe, is installed in the stream to protect the sensor from strong currents and debris. The well is vented to allow pressure equilibrium between the environments inside and outside of the pipe. The sensor is suspended from the lid of the well so that the sensor is always underwater but not in danger of being buried by silt. For calibration purposes, a staff gauge is also installed on site to be used as a point of reference.

What is a Waterproof Data Shuttle and how does it work?

The Waterproof Data Shuttle is used to download data in the field from the HOBO sensor and transfer the data back to a host computer in the office (called offloading). It is like storing data on a thumb drive. It is also used to launch the logger to set the sampling program and sychronize the date and time. The shuttle uses infrared light to transfer data, allowing the logger to remain sealed and completely waterproof.

What can go wrong with a Water Level Data Logger?

Several environmental factors can interfere with a sensor's ability to collect accurate data. Shock from being dropped; biological growth inside the sensor's nose cone or on the sensor itself; and exposure to solvents can decrease the sensor's functionality. Care and regular maintenance are required to extend a sensor's useful life.

I. In the Field: Stewarding the Stage Sensor

CWDD staff will install two stage sensors at one of your stream sites – one in the stream and one in the air recording barometric pressure. You will be expected to check on the status of the sensors every 2-3 weeks when you visit your site for your routine water quality assessments.

1. Locate the stage installation in the stream. If it is safe, wade out to it and remove the sensor from the protective PVC housing.

2. Inspect the sensor for damage and wipe it clean with the provided sponge as best you can.

3. Remove any debris that may have jammed up against the housing.

4. If necessary, clean out the inside of the housing of any sand.

5. Return the sensor to the protective PVC housing.

6. Follow the prompts on the Sensor Field Data Sheet on measuring the depths at the sensor including total stream depth, depth from stream bottom to sensor, and depth from sensor to the surface of the water.

7. Locate and visually inspect the barometric pressure stage sensor for any damage or disturbance.

8. Make notes on your Sensor Field Data Sheet.

II. Removing the Stage Sensors at Season's End

The stage sensors are able to withstand cold temperatures; however for our purposes it is ideal to remove the stage sensors before the stream freezes over when the water has become cold enough that it is unsafe to continue monitoring. As winter approaches, keep an eye on the weather and use your best judgment for deciding when to retrieve the sensor (possibly in late November).

Carefully place the sensor back in its storage box for transport back to your school. If you can, remove the installation from the stream, you may need extra tools for this depending on your installation.

Store the stage sensor in a safe place (like a locked desk drawer) until returning it at the Symposium in the spring. Notify Janel (email jroberge@smcvt.edu or call 802.654.3271) about the storage location of the sensor.

Sensor Field Data Sheet

2015-2016

Stream Name:	Site Code:
Investigators:	Date:

Temperature Sensor

A. Only applicable at deployment visit:

Serial Number	
Time Interval	120 minutes
Initial Program Launch Date and Time	
Deployment Date and Time	
Retrieval Date and Time	
Name of .csv data file	

B. Field Notes performed every 2-3 weeks:

Sign of damage or disturbance? Is the sensor buried in sand or covered with debris or fouled with algae?

_____

Is the sensor dewatered?

_____

Stage Sensor

Serial Numbers	
Total Depth at Sensor	
Depth from Stream Bottom to Sensor	
Depth from Stream Surface to Sensor	

Sign of damage or disturbance? Is the sensor buried in sand or covered with debris or fouled with algae?

_____

Is the sensor dewatered?

_____

Other Comments: (map, problems, observations, etc.)

Macroinvertebrates

Contents

Macroinvertebrates General Info

There are several components to assessing macroinvertebrate communities in streams:

- Discharge or flow (recorded on Water Quality Assessment Data Sheet from Water Quality Section)
- Substrate (recorded on Macroinvertebrate Habitat Data Sheet)
- Macroinvertebrate Identification (recorded on Macroinvertebrate Data Sheet)

Take all the components listed above each time you sample for macroinvertebrates. You will sample for macroinvertebrates once at each of your two stream sites.

Use the "Macroinvertebrate Habitat Data Sheet" and the "Macroinvertebrate Data Sheet." Also, complete the "Water Quality Assessment Data Sheet" each time you visit and sample the stream.

The data sheet and field labels that you need to take macroinvertebrate samples are at the end of this section of your manual. You should enter the data that you collect and record in the field on your "Water Quality Assessment Data Sheet" and "Macroinvertebrate Habitat Data Sheet" online **as soon as possible** after your field visit. You may enter the "Macroinvertebrate Data Sheet" online whenever you are ready to do so.

If you wish, you may choose to sample for macroinvertebrates a second time, such as a week after a major storm event has occurred. This will provide you with two sampling dates for each stream. Your first sampling date acts as a "baseline" sample and your second sampling date could be used to assess the impact of the storm event on the macroinvertebrate community.

Macroinvertebrate Equipment Field Checklist

General Equipment

- Waders
- Spray bottles with disinfectant solution and water
- Gloves
- Pencils and permanent marker
- Datasheets (one for each site)
- Clipboard
- Insect repellent/sunscreen
- Sunglasses
- Camera
- First Aid kit
- Cell phone
- Meter stick
- Water temperature/pH meter

Bug collection

- Kick net
- Kitty litter tray
- #30 sieve
- 4 Whirl-paks® for each site + extras
- 2 large mason jars per site
- Ethanol
- Field forceps
- Plastic spoon

Macroinvertebrate Communities in Wadeable Streams

All macroinvertebrate samples are collected during the late-Summer, early-fall index period, from September to mid-October unless otherwise discussed. A field crew selects a representative riffle section in the steam reach to be sampled. Physical characteristics recorded at each selected site include: stream width, depth, water velocity, water temperature, weather conditions, substrate composition, substrate embeddedness, canopy cover, stream bank condition, and immediate upstream land use. All data are entered onto a field sheet with appropriate site and sampling event identifiers, along with additional comments that may be applicable to the site evaluation. This sampling protocol is based on methods used by the Vermont Department of Environmental Conservation.

Complete a "Macroinvertebrate Habitat Data Sheet" for each macroinvertebrate sample you collect.

Field Method:

Macroinvertebrate Sampling

Macroinvertebrate samples are collected using an 18 inch wide x 9 inch high rectangular frame net with a 500 micron mesh size.

1) One person operates the net while the other person operates the stopwatch.

2) Place the net in the riffle, being sure the base of the net is firmly set against the stream bottom and there is water flowing into the net.

3) Using your hands, disturb an area immediately upstream of the net (square area, 18" x 18"), ensuring that all pieces of substrate are moved and rubbed clean of attached organisms and flow into the net opening. After scrubbing the larger substrates, disturb any underlying gravel to an approximate depth of 10 cm. This typically takes about 30 seconds but it is more important to complete the procedure than to exactly time 30 seconds.

4) Turn the contents of the net inside out into the kitty litter tray with lots of rinse water taken from the stream.

5) Rinse and scrub large gravel of remaining organisms and remove it from the net along with leaves and sticks. Any material adhered to gravel, leaves, and sticks is likely to contain macroinvertebrates, so be thorough.

6) Transfer the contents of the kitty litter tray into a #30 (=600 µm) sieve to remove small particles and water from the sample.

7) Using forceps and a plastic spoon, transfer contents of sieve into a Whirl-pak® and fill approximately half the Whirl-pak® with 100% ethanol, being sure to cover the entire sample but also leave enough room to close the bag. Be sure the Whirl-pak® contains a paper label with the following information: Stream name @ road name, town/state/country, site code, replicate number, month-day-year, collector name/School name. The label should be inside the bag in the ethanol – for that reason, **do not use pen or ink-jet printouts**. If a sample occupies more than one bag then label each part of the sample with the same sample number and write *1 of 4; 2 of 4…* etc. **Do not rely on sharpie markings on the outside of the bag. Leaking ethanol removes all traces and the sample becomes useless**.

8) Turn the net inside out and rinse thoroughly to remove debris. Use the net inside out for the next sample. The act of sampling will further rinse the outside of the net (and it will become the inside for the following sample).

9) Moving up-stream, repeat steps 1-8 at 3 additional locations within the riffle *representing a range of velocities and substrate types* characteristic of that riffle, being careful to avoid areas that have been previously disturbed. The total active sampling time should roughly equal 2 minutes for the sampling site (approximately 30 seconds at each location). Do not mix the four replicates – they should be maintained as separate samples through all field and lab procedures.

10) You will end up with 4 separate replicates from each site. Store 4 Whirl-paks® in quart-size mason jar (or as many as are needed) until ready to process. This "composite" sampling methodology effectively collects samples representative of the entire macroinvertebrate community of that riffle.

Field Method:

Modified Pebble Count of Riffle Habitat

This method is used to describe the substrate particle size classes within the "riffle" habitat of high gradient stream types that are targeted by the VTDEC for macroinvertebrate community assessments. The method is based on the more rigorous technique developed by Wolman (1954) to describe coarse river bed materials, and modifications of this technique developed by the Forest Service developed to describe the channel bed materials within stream reaches (Bevenger and King 1995, and Harrelson, et al 1994).

Riffle Pebble count Procedure:

1. A minimum of 100 particles are to be recorded on a tally sheet.

2. Diagonal transects across the stream are paced off until a minimum 100 count is reached. Transects begin at the lower end of the wetted portion of the stream bed within the macroinvertebrate sampling section or riffle. A pebble is selected as described below every two paces in larger streams > 20m across, or every pace in smaller streams <20m across.

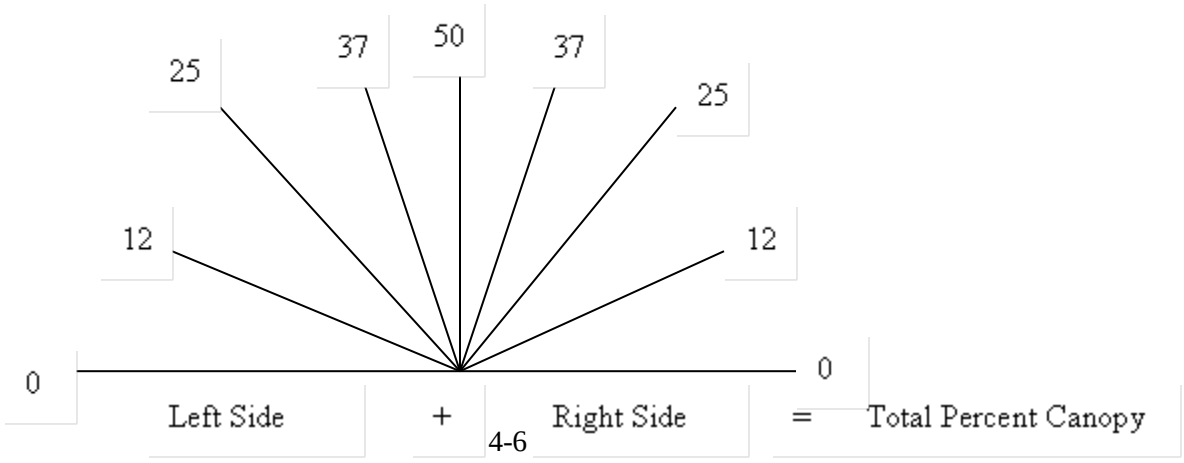
3. Averting (closing) one's eyes, a bit of substrate (e.g., a pebble) is selected by touching the bottom with one's index finger. You may also consider using a meter stick to 'choose' a type of substrate (this is an extremely useful way to be unbiased in your selection and also to avoid any potentially dangerous bending and reaching in the stream). The randomly selected substrate is then placed in a particle size category. Size categories were initially based on the Wentworth's size classes, which were then lumped into larger biologically based size classes used by the VTDEC to describe substrate composition. The VTDEC size categories are: Sand **<2mm** (.08"), Gravel **2-16mm**(.08-2.5"), Course Gravel **16-64mm** (.63-2.5"), Cobble **64-256mm** (2.5-10.1"), Boulder **>256mm** (>10.1").

4. Size categories are determined by using a gravelometer, essentially a metal plate with squares of the above size classes cut out, or by estimation. The size category is called out to a recorder, who keeps track of the tally until the minimum of 100 particles is reached. If this occurs in the middle of a transect, that transect is completed even if it means going over the 100 mark.

Percent Canopy Measurement Guidance

Stand in the center of the stream/river and extend both of your arms straight out creating a 180-degree angle. Observing the overhead canopy cover, start to lift your arms up from the straight out position slowly towards your head. Stop when each arm is in alignment with the overhead canopy. Then estimate the angle of your left and right arm using the figure below for guidance. Combine the percent canopy values from the left and right side to obtain the total percent canopy.

Percent Canopy Calculator



Laboratory Method:

Identifying, Preserving, & Counting Macroinvertebrates

1) Pour contents of one individual sample (usually one Whirl-pak®) into a bucket. Add water and gently pour organic matter into a #30 sieve to remove all excess ethanol. Continue to add water and swirl sample until organic matter has been removed and sand remains in bucket. This will be processed later.

2) Spread organic matter evenly over a tray that is divided into 12-squares (the provided 'cafeteria tray'). Add a small amount of water to the tray to allow the sample to be evenly spread, but not so much as to cause the macroinvertebrates to float freely around the tray.

3) Randomly choose a number between 1 and 12, which will correspond to a square on your tray (use 12-sided dice or this excel formula: =int(rand()*12)+1). Use dominoes to separate this square and the next 2 consecutive squares from the rest of the sample. This will represent one quarter of your sample. Alternatively, generate a series of random numbers and work through the list until you've reached 3 squares or 75 insects. Pick all organisms from the selected sections with the aid of a 2x magnifier while keeping a tally. Completely pick each of the 3 squares (i.e., do not leave any insects remaining). For bugs which are not intact, only tally the heads, not other body parts which are found.

4) After the 3 squares have been completely picked either take a break and check the area again later for bugs which have been missed, or have someone else check your sample. If after this time the minimum number of 75 organisms from the sample has not been reached, pick additional grids on the tray to reach that number. **Record the total number of grids (squares) that were picked** so that sample density or relative abundance can be calculated.

5) Sort animals into major groups, and preserve in 75% EtOH with 1% glycerin (provided).

6) Using the keys provided, identify each individual to genus/species (depends on reference collection) except for the Chironomidae and Oligochaeta which will not be identified beyond the family and subclass level, respectively.

7) Store identified insects in a plastic vial with label indicating information on the site name, replicate sample number, date, identification (**in pencil or laser printed**).

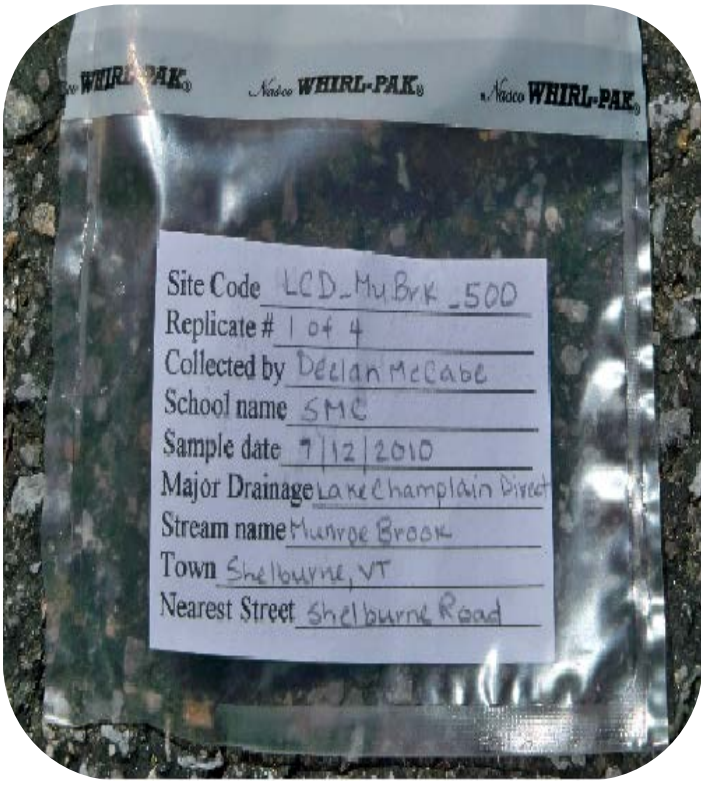
8) Keep a record of all identification data on your Macroinvertebrate Data sheet and enter the data online.

Macroinvertebrate Field Labels

- Please use labels provided by the Streams Staff

- Labels should be complete **IN PENCIL** and placed **INSIDE** the Whirl-pak®. Labels filled out with markers will be erased by the ethanol!

Site Code
Replicate Number
Collectors
School Name
Sample Date
Major Drainage
Stream Name
Town
Nearest Street



Macroinvertebrate Habitat Data Sheet

2015-2016

Stream Name:	Site Code:
Latitude/Longitude:	Date/Time:
Site Description:	Investigators:

Sample Collection	Time spent collecting each replicate sample (should be 30 seconds): 1) 2) 3) 4) Comments:
Field Measurements	Air Temp: ___°C Water temp: ___°C pH: _____ Velocity (m/s) (at mid-point): _____ Bank full width (m): ____ Wetted width (m): _____ Depths where samples collected(m): 1) 2) 3) 4) Canopy cover: 100 90 80 70 60 50 40 30 20 10 0 %

PEBBLE COUNT

Particle	Millimeters	Transect 1 (100 pebbles)	Total #	Item %
Clay/Silt/Sand	< 0.004-2.0			
Gravel	2.0-16			
Coarse gravel	16-64			
Cobble	64-256			
Boulder	> 256			
Bedrock				
		Totals:		

Comments:

Macroinvertebrate Data Sheet

2015-2016

Sample ID Number (yy/mm/dd/Rep #): ___________________

Site Code: ___________________

Watershed: _______________ Town: __________ Location: ______________

Picked by: _________________ Date Picked: ____________

Number Bugs Found: __________ Number Squares Picked: ______________

Sorted By: ______________ Date Sorted: ____________

IDed by: _______________ Date IDed: ______________

Date	Initials	Class	Order	Family	Notes	#

Uploading Data

Contents

Uploading Field Data

High school teams are responsible for uploading all of their data into the online database with the exception of the lab results for Total Suspended Solids (TSS) and Nutrients (TP/TN), and the sensor (stage and temperature) data. The lab data will be added to the online database by CWDD staff once it has been analyzed in the lab. You will email sensor data to CWDD staff (See the Water Quality section for these instructions).

The data that you are expected to upload to the online database correspond to the following data sheets:

Field Data Sheets:

- Site Assessment
- Habitat Assessment Data
- Water Quality Assessment Data
- Macroinvertebrate Habitat Data

Lab Data Sheets:

- Macroinvertebrate Data Sheet: Taxonomic information on the macroinvertebrates collected at your stream sites (Instructions in next section)

To keep on top of entering the data, ***enter your data within a month of collecting the data*** in the field. Ideally, you will enter field data soon after each site visit. It is important to have all data entered in a timely manner so that teams can have the data available to them when they begin their research projects. By January, CWDD staff will begin to check in with teams who do not have all of their data entered.

To enter data into the online database, go to the following website for the first three items listed above: **PLEASE CONTACT CWDD@SMCVT.EDU FOR THIS LINK**

Once you are at the web page:

1. Select your stream site code under "Stream/Site Name." You'll notice three boxes appear with the following titles: Site Assessment Data, Water Quality Assessment Data, and Macroinvertebrate data. These correspond to the three field sheets mentioned above. The dates in each box represent the collection date of previous entries for this site.
2. **To add** a new entry, select the "New" button for the data you would like to enter. Fill out the electronic sheet which corresponds to your field sheet for the respective data. Click "submit" when you have finished.
3. **To edit** an already submitted field sheet, select the date of the entry in the box and then select the "Edit" button. Make the necessary changes and then click "submit."
4. **To view** a field sheet that has already been submitted, select "View." Click the "Site Summary" page to go back to the initial view.

Uploading the data to the online database is an important step in growing our Streams Project database and making it available to participants and other interested parties. The recommendation for monthly uploading will help ensure we keep the flow of data coming!

If you have any questions about uploading data, please contact cwdd@smcvt.edu .

Uploading Macroinvertebrate Data

Additionally, you are expected to enter the following taxonomic data on the macroinvertebrates collected at your stream sites. For a reminder on how to upload macroinvertebrate data, a video tutorial is available:

PLEASE CONTACT CWDD@SMCVT.EDU FOR THIS LINK

To enter your macroinvertebrate taxonomic data, please use the following web page:

PLEASE CONTACT CWDD@SMCVT.EDU FOR THIS LINK

Once you are at the web page titled "Streams Macroinvertebrate Input Form":

1. **Select your** stream site code under "Stream/Site Name", select your school or organization under "School/Organization", and select "Replicate Number".
2. **Fill in** the information "Number of Squares Picked", "Sample ID Number", "Sorted By", "Date Sorted", "Time & Date Collected", "IDed by", "Number of Bugs", and "Date IDed" from the macroinvertebrate label and Macroinvertebrate Data Sheet sheets.
3. **To add** insect macroinvertebrate counts for your replicates, check the "Insect" box. Then select the proper "Order" and "Family", and finally enter the number of insects under "Count." To add a new insect species click the down arrow to the left of "Count."
4. **To add** non-insect macroinvertebrate counts for your replicates, uncheck the "Insect" box. Then select the proper "Phylum", "Class", "Order" and "Family", and finally enter the number of insects under "Count." To add a new macroinvertebrate species click the down arrow to the left of "Count."
5. **Check and review** data and information entered into the Macroinvertebrate Input Form.
6. **Click "submit" when you have finished.** You must click Submit for the data to be uploaded.

Uploading the data to the online database is an important step in growing our Streams Project database and making it available to participants and other interested parties. The recommendation for monthly uploading will help ensure we keep the flow of data coming!

If you have any questions about uploading data, please contact cwdd@smcvt.edu

Data Analysis and Presentations

Contents

Data Analysis Overview

You should begin thinking about and preparing your poster or oral presentation for the VT EPSCoR Student Research Symposium in April as soon as possible. The basis of your poster or presentation will be an analysis of the data you have gathered during the past year and/or historical data (from the Streams Project online database, or other sources).

The Streams Project has created a **data analysis tutorial** to help guide you through the process of exploring and asking more in-depth analysis questions about your dataset. This should be your primary guide for beginning your data analysis, but the VT EPSCoR CWDD staff members are always available to help you along the way. Some modules are Streams Project –specific, while others are useful to anyone interested in analyzing data.

The tutorials can be found on the website here:

http://www.uvm.edu/~epscor/new02/?q=node/1027

The first link on the page that says "Complete Tutorial Series - All Modules" will open a PDF with all of the modules compiled into one document. The subsequent links are for accessing modules individually. The following is a list of the individual modules and what they cover:

- Module 1: What is science?
- Module 2: Understanding Streams Project Data
- Module 3: Refining and Retrieving Data
- Module 4: Data Exploration
- Module 5: Statistical Analysis
- Module 6: Summarizing Results and Drawing Conclusions

In this tutorial, statistical analysis is demonstrated using Microsoft Excel. Within each module, look for the "WATCH VIDEO" icon that looks like this:

These videos help you visualize a number of procedures outlined in the tutorial. **NOTE: To be able to watch the videos, download the QuickTime Player, if it is not already on your computer: http://www.apple.com/quicktime/download/

Viewing and Downloading Streams Project Data

To view or download data in the Streams Project's database, go to the following location website:

www.uvm.edu/epscor/redir/streamsprojectdata

Once you are at the web page:

1. Select the stream sites for which you'd like data. If you'd like data from multiple sites, hold down the "Ctrl" button in between selections. If you'd like data for all the streams sites, select the first stream site, hold down the "Shift" button, and the select the last stream site in the list.

2. Select the report that represents the type of data you are interested in under "Available Reports."

3. Select the date range for which you'd like data.

4. Once you've made these selections click the "Generate Report" button.

5. You can view the data available for these criteria on the webpage that appears. If you click on the heading of a data field in the table, a little box will pop up describing the data contained in that field.

6. To download the data seen here, click the "Export to Excel File" text above the table and save the file on your local computer.

An explanation of the data in the database, and a description of how to download data from this web page can also be found in **Module 3: Refining and Retrieving Data** of the Data Analysis Tutorial. The link to this module can be found here:

http://www.uvm.edu/~epscor/new02/?q=node/1027

Temperature Data Analysis

Typical base flow temperatures in small streams show daily fluctuations that remain fairly consistent or change gradually over time. Because water is thermally stable, stream temperatures change far less rapidly than air temperatures so thermal peaks and troughs tend to lag behind peaks and troughs in air temperature. Average stream water temperatures follow seasonal weather patterns. Because of shading, we generally expect forested streams to be cooler and less variable in temperature than urban streams.

High water events modify these base flow patterns. How exactly the temperature is modified depends on the timing of the storm and the conditions in the watershed. We might expect the runoff from a parking lot to increase stream water temperatures. Runoff from forested land would not have the same effect and may even have opposite effects. Storms associated with cold fronts would obviously have different effects than summer thunderstorms.

Before we use the temperature data for any analysis, follow these quality control steps to ensure your data is viable.

Removing Extraneous Data
Your first step will be to remove any air temperature measurements that may have been recorded at the beginning (when you first programmed your iButton) and at the end (when you removed your sensor) of your field season. Refer to your Sensor Field Data Sheets for the dates and times of your program start, deployment, and removal from the stream. Remove any measurements that represent temperatures taken before or after deployment.

Departures from Normal Conditions:
Next, you will need to check to see if the changes you observe are reasonable for the air temperatures and storm events that have occurred in your area. Assuming that your temperature sensor is working normally, you should expect regular daily fluctuations unless storms occur. It will be important to review your temperature data, while this can be a tedious task, it is necessary to find any errors that may have occurred. Use the table below and follow these basic guidelines when reviewing your data:

- Flag temperature records that are outside the range of -1°C and 30°C
- Question values if there was a rate of change greater than 3°C per hour or a daily mean change of greater than 3°C between two successive days
- Flag any outliers that are in the upper and lower 5th percentiles of an overall distribution for a day

Table 1. The following is a short list of symptoms, potential causes, and cures:

Symptom	Potential Cause	Solution
Constant temperature	Sensor malfunction Sensor at spring-fed source	Replace sensor Use thermometer to confirm reading and inform CWDD staff
Sensor tracks air temperature	Sensor not submerged	Dig a deeper hole in benthos or place probe in pool
Intermittent increases in variability	Intermittent low water exposing sensor Intermittent storms, particularly in urban sites	Check nearby stream gage to see if low water or high water makes more sense. Dig sensor deeper if low water is the problem. Storm-caused variability = real data

High-temperature spikes at installation and removal	Air temperature recorded during handling; expect this in most data sets	Do not include in graphing or data summaries; still valuable as a test of sensor function: compare with NOAA records
Gradual decrease in temperature through Fall	Normal seasonal cooling of stream	Record precious data!
Intermittent decrease in temperature and reduction in variability	Storm events, particularly in forested streams	Confirm that rain occurred using USGS gage data for a close stream

Access Past Air Temperature Data:
To compare your temperature sensor data to air temperatures, go to NOAA's National Weather Service web site to download nearby air temperatures.

http://www.nws.noaa.gov/climate/index.php?wfo=btv

Under #2, you will see a list of locations; choose the town nearest to your stream site and enter the date of interest (select "Archived Data," for access to the past three months). For older data, navigate to this webpage: **http://cdo.ncdc.noaa.gov/qclcd/QCLCD?prior=N**

Copy the minimum and maximum temperatures over to an excel grid one day at a time and compare to your temperature sensor data. Flag and do not include any data that does not fall within this range.

Access Past Stream Flow Data:
You can also access some water temperature data on the Vermont USGS website:

http://waterdata.usgs.gov/vt/nwis/rt

The following stations provide water temperature data, view them by selecting them on the map or choose them from a list under, "Statewide Streamflow Real-Time Table." Adjust the date range to match your time of interest and hit "go."

04284751 WINOOSKI R @ US2, BLW STEVENS BR, NR MONTPELIER VT

04294500 LAKE CHAMPLAIN AT BURLINGTON, VT

04294140 ROCK RIVER NEAR HIGHGATE CENTER, VT

Macroinvertebrate Data - Community Analysis

The following Calculation of Metrics document is a resource on calculating metrics that describe the macroinvertebrate communities found in the streams you are examining through your analysis. The document details the meaning and application of eight commonly used metrics to describe macroinvertebrate communities.

These calculations will help you interpret your macroinvertebrate taxonomic data and is a necessary first step before further analysis as described in the Data Analysis Tutorials. For the Streams Project, we're investigating differences among streams of varying surrounding land use, so metrics such as taxa richness (taxa = species, or lowest level of identification for your samples, i.e., family, genus), composition, and functional feeding groups of your samples are relevant to consider.

You do not have to use all of these metrics, so choose one of particular interest or relevance to your study question. We are happy to help along the way, but please use this resource to get you started!

Note: Refer to the following spreadsheet online for determining the biotic index values and functional feeding group designations for you macroinvertebrate identifications. The majority of the biotic index values are assigned to the genus level (there are some that are family level) and therefore are not helpful for your analysis. We recommend the following indices: total sample richness; EPT richness; mayfly richness; stonefly richness; % EPT; % mayflies; % dominant taxon.

The document can be found online at: **http://epscor.w3.uvm.edu/2/node/1027**

Additionally, a classification of the taxa by functional feeding groups can be found in Appendix A of the University of Minnesota "Guide to the Aquatic Invertebrates of the Upper Midwest", which is included in your macroinvertebrate ID binder.

Calculation of Metrics

1. Density- Is the relative abundance of animals in a sample.

Calculation: Number of animals in subsample / proportion of sample processed.

Example : 300 animals picked / 0.25 (or one quarter of sample picked) = 1200 animals/sample

2. Richness- Species richness is the number of species in a sample unit.

Calculation: Richness is the total number of distinct taxa identified in a sample. Note immature larva identified to family or genus are not considered a distinct new taxa if a genus or species identification is determined within its group.

Example :

Taxon	# orgs Rep 1	# orgs Rep 2
Ephemerellidae Ephemerella sp	2	0
Ephemerellidae Ephemerella dorothea	3	4
Ephemerellidae Ephemerella invaria	0	2
Richness =	1	2
Mean Richness =	1.5	

3. EPT Index- The EPT index is a subset of the above richness measure. It is the number of species in the sample in the generally more environmentally sensitive orders Ephemeroptera, Plecoptera, and Trichoptera.

Calculation: The number of distinct taxa identified in a sample from the insect orders Ephemeroptera, Plecoptera, Trichoptera. Note same rules apply as above for richness in determining number of distinct taxa.

4. EPT/EPT & Chironomidae - Is a measure of the ratio of the abundance of the intolerant EPT orders to the generally tolerant Diptera family Chironomidae.

Calculation: The number (abundance) of animals from the orders Ephemeroptera, Trichoptera and Plecoptera, divided by the above plus the number of Chironomidae.

5. % Oligochaeta - Is a measure of the percent of the macroinvertebrate community made up of the Order Oligochaeta.

Calculation: The number (abundance) of Oligochaeta divided by the total number of animals in sample.

6. Percent Model Affinity of Orders (PMA-O) - Is a measure of order level similarity to a model based on the reference streams Novak and Bode (1992).

Calculation: Determine the percent composition for each major group - Coleoptera, Diptera, Ephemeroptera, Plecoptera, Trichoptera, Oligochaeta, Other. Compare to the "Model" for the appropriate stream community (see below), then add up the lower of the two values for each of the groups (assessment site vs Model), this is the PMA-O for the assessment site.

PMA-O = min (X_a or X_r)

Where: X_a = the percent composition of order X from the assessment site;

X_r = the percent composition of order X from the appropriate reference condition;

Example:

Percent Composition Major Grps	Assessment Site % Comp	Model for MMC (Medium Mt)
Coleoptera	20	**6**
Diptera	55	**18**
Ephemeroptera	**10**	34
Plecoptera	**2**	8
Trichoptera	**3**	33
Oligocheata	10	**0.5**
Other	**0**	0.5
PMA-Orders =	39.5 rounded = 40.0	

7. Hilsenhoff Biotic Index - BI (0-10) - Is a measure of the macroinvertebrate assemblage tolerance toward organic (nutrient) enrichment (Hilsenhoff, 1987). In many ways this index is both an indicator taxa metric and functional group metric, since those taxa which become more dominant in moderately enriched streams are those which are taking advantage of shifts in the available food base in the stream.

Calculation : Multiply the number of individuals of a taxon by its assigned tolerance value, see VTDEC BI values, modified from Hilsenhoff 1987, and Bode 1996. Total all these products, and divide by the total number of individuals <u>of each taxon assigned a tolerance value</u>. This is the Bio Index value.

$$HBI = \sum_{i=1}^{i} \frac{n_i a_i}{N}$$

Where: n is the number of individuals of the "i"th taxon;

a is the index value of that taxon;

N is the total number of individuals in the sample assigned a Bio Index Value

Example :

Taxon	Count	BI Tolerance Value	Subtotal Ct × BI
Ephemerllidae imm	(10)	NA	NA
Ephemerella sp	**10**	4	**40**
Ephemerella needhami	**10**	1	**10**
Plecoptera Leuctridae imm	**20**	0	**0**
Diptera Cricotopus bisinctus	**5**	6	**30**
Trichoptera Symphitopsyche alhedra	**10**	3	**30**
Trichoptera Symphitopsyche sp	**5**	5	**25**
Totals	***60***		***145***
Site Bio Index Value	145/60 = ***<u>2.42</u>***		

8. Pinkham–Pearson Coefficient of Similarity – Functional Groups – (PPCS-F) - Is a measure of functional feeding group similarity to a model based on the reference streams. It is similar in concept to the **PMA-O** in that a site is compared to a model of the composition of the functional feeding groups as opposed to order level taxonomic changes. Also the Pinkham Pearson Coefficient of Similarity (Pinkham1976) was used as the similarity index. By replacing functional feeding groups with families, the formula can easily be recalculated to yield a Pinkham Pearson Coefficient of Similarity – Families (PPCS-Fam). This would provide a family-level comparison between a pair of sites.

Calculation: At the assessment site determine the percent composition of the six major functional groups (Collector Gatherer, Collector Filterer, Predator, Shredder-Detritus, Shredder-Herbivore, Scraper) as assigned by VTDEC after Merrit and Cummins 1996, Bode 1996. For each functional group determine the product (min/max) between the assessment site vs the Model for the stream community sampled. Add these products and divide by six (# of functional groups). This is the PPCS- F.

$$\text{PPCS-F} = 1/k \sum_{i=1}^{k} \frac{\min(x_i a, x_i b)}{\max(x_i a, x_i b)}$$

Where: k = the number of comparisons between stations (6)

x_i = the number of individuals in functional group i

a, b = site a, site b

Example :

Functional Group	Assessment Site % Comp	"Model" for MMC	Product (min/max)
Collector Gatherer	68	32	0.47
Collector Filterer	10	30	0.33
Predator	2	13	0.15
Shredder - Detritus	0	4	0.00
Shredder - Herbaceous	16	1	0.06
Scraper	2	13	0.15
PPCS-F =			**0.19**

Presenting Your Data: VT EPSCoR Student Research Symposium

All participants of the RACC High School program commit to presenting their research findings at the annual Vermont EPSCoR Student Research Symposium. A symposium is a great way for researchers to present and discuss their work and it provides an important channel for the exchange of information between researchers. At the Vermont EPSCoR Student Research Symposium, participants have the option to choose whether they present their research through a poster or an oral presentation. Both are great ways to share your work!

Posters versus Oral Presentations

Although it can be challenging to present a year's worth of work in 10 minutes, oral presentations can be a rewarding experience because you are the only one front of an audience whose attention you know you have. Oral presentations are brief and consequently the presentation must be clearly and succinctly presented.

Posters are a visual presentation of information that is understandable to the viewer without verbal explanation. Poster presenters have the opportunity to share their work with one person at a time, over an extended period of time. This allows the presenter to describe and discuss their research in greater detail than would be possible in an oral presentation to significantly more people, and allows for dialogue with poster viewers.

Posters

A research or academic poster provides a means of communicating your research at a conference or research symposium. Posters printed by Vermont EPSCoR are 3' x 4' (or 36'' x 48"), horizontally or vertically aligned. Upload your final poster file when registering for the symposium by the deadline announced in early March. The CWDD will print and set up your poster at the symposium.

How to Create a Poster Using PowerPoint

For many, this is the first time creating a research poster. Here are some tips for making an informative and attractive research poster:

1. Open PowerPoint
2. Click the 'Design' menu/tab at the top of the screen and select 'Page Setup'
    i. Change the dimensions of the slide from the default setting to: Width=48, Height=36 (for a horizontal poster), or Width=36, Height=48 (for a vertical poster). This is an important **FIRST** step – if you change the dimensions after putting content on the slide, you will have to re-format all text boxes, graphs, tables, photos, etc.
3. Critical poster elements:
    i. Title, Author(s) and affiliation(s)
    ii. Abstract/Summary (*optional*)
    iii. Introduction/Background: a brief but important overview to secure the viewer's attention
    iv. Materials and Methods: a brief description of the processes and procedures used, photos (*optional*) should be >300dpi
    v. Results: outcomes, findings and data displayed through text, tables, graphs, photos, etc.
        - Bulleted lists (rather than paragraphs) may help the reader understand the most important findings
        - Tables, graphs and photos should have captions. Graphs should have a legend, avoid 3-D graphs as they are hard to interpret
    vi. Discussion/Conclusions: summary or discussion of the significance and relevance of the results, identify possible future research
    vii. References
    viii. Acknowledgements
    ix. Please include the following text somewhere on the poster: Funding provided by NSF Grant EPS-1101317
4. Upload final poster file when registering for the symposium

Tips:

A. Use the "Designing Conference Posters" website to get ideas on poster layout and to download poster templates: http://colinpurrington.com/tips/academic/posterdesign
B. Choose a background and text color scheme. No need to go crazy: a white/light poster with black/dark text is often much easier to read than a multi-colored poster. Use cool/muted colors, solid colors, a color gradient, etc.

C. Lettering can make a difference in how easy-to-read your poster is. Here are some suggestions:
    - Title: at least 72 pt., bold preferred
    - Section Headings: at least 48 pt., bold preferred
    - Body Text: at least 24 pt.
    - Avoid using all capital letters
    - Use sans serif (Arial) for titles & headings
    - Use serif (Times New Roman) for body text
    - Use bulleted lists where possible instead of paragraphs
    - Use *italics* instead of underlining
    - White or light colored lettering is hard to read on a dark background when printed. Use black lettering instead on a light colored background
D. Logos: Do not forget to include the logos for the organization(s) that helped make the research possible!
    - Funding source: The National Science Foundation's (NSF) logo can be used by recipients of NSF support for the sole purpose of acknowledging that support: https://www.nsf.gov/policies/logos.jsp. Please include the following text somewhere on the poster: Funding provided by NSF Grant EPS-1101317
    - VT EPSCoR, RACC, CWDD and others if they were important contributors. Logos are available on the "Resources" website: http://www.uvm.edu/~epscor/new02/?q=node/900
    - Your school logo!

Example posters from the 2015 VT EPSCoR Student Research Symposium:

http://epscor.w3.uvm.edu/2/node/2635

Oral Presentations

A research talk provides a means of communicating your research at a conference or research symposium. Oral presentations at the VT EPSCoR Student Research Symposium are limited to 10 minutes: 8 minutes to present your research, 2 minutes for the audience to ask questions. Presenters often use the general rule of "1 slide per minute"; however the number of slides needed varies based on the complexity of the content of the slides. Upload your final PowerPoint file when registering for the symposium by the deadline announced in early March or bring the file to the symposium on a USB drive. The CWDD will provide the computer, screen, podium, microphone and laser pointer for your use.

Oral Presentation Structure (*suggested*):

- Title, Author(s), Affiliation (1 slide)
- Outline, *optional* (1 slide): overview of the structure of your talk, some speakers prefer to put this at the bottom of their title slide, audiences like predictability
- Introduction/Background
    - Motivation and problem statement (1-2 slides): Why should anyone care? Most researchers overestimate how much the audience knows about the problem they are addressing
    - Related Work (0-1 slides)
    - Methods (1 slide): Cover quickly in short talks
- Results (4-6 slides): Present key results and key insights. This is the main body of the talk. Its structure varies greatly as a function of the research conducted. Do not superficially cover all results; cover key result well. Do not just present numbers; interpret them to give insights. Do not put up large tables of numbers as your audience will not have time to take in that much information at once
- Discussion/Conclusions (1 slide): summary or discussion of the significance and relevance of the results, identify possible future research
- References
- Acknowledgements
- Please include the following text somewhere on your slides: Funding provided by NSF Grant EPS-1101317

Logos: Do not forget to include the logos for the organization(s) that helped make the research possible!

- Funding source: The National Science Foundation's (NSF) logo can be used by recipients of NSF support for the sole purpose of acknowledging that support: https://www.nsf.gov/policies/logos.jsp. Please include the following text somewhere on your slides: Funding provided by NSF Grant EPS-1101317
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- Your school logo!

Example posters from the 2015 VT EPSCoR Student Research Symposium:

http://epscor.w3.uvm.edu/2/node/2635

Resources

RACC High School Resources: http://www.uvm.edu/~epscor/new02/?q=node/900

- Includes links to datasets available online, including:

  Data and Data Analysis

  - VT Department of Environmental Conservation Lake Champlain Long Term Monitoring
  - VT Department of Environmental Conservation Volunteer Monitoring
  - USGS Stream Gauge Data
  - Vermont Water Quality Data
  - NOAA Quality Controlled Local Climatological Data
  - VT EPSCoR Data Analysis Tutorials
  - Data Analysis in Excel

- Helpful hints on posters and oral presentations

- High resolution logos to include on your poster, etc.

Data Analysis Videos by Dr. Declan McCabe:

https://www.youtube.com/watch?v=3QByp-83ceQ

- Gets you started on data analysis. When you compare two groups, will it be significant or not?

http://www.uvm.edu/~epscor/new02/?q=node/1237

- Walks you through how to find different data sources online, how to groom and present your data using Excel, and how to use PowerPoint to create a presentation

Video Tutorials

http://www.uvm.edu/~epscor/new02/?q=node/1686

- Uploading Macroinvertebrate Data
- Programming the iButton Temperature Sensor
- Downloading Data from the iButton Temperature Sensor

Field Safety

Contents

First Aid Kit

When working in the field, it is important to be prepared for emergencies. Although you will not be traveling far from your car when you visit your field sites for the VT Streams Project, accidents may still happen. Therefore, a well-stocked first aid kit is an important thing to have. Carry a first aid kit with you to your site or keep one in the car. You may purchase a pre-made kit at the store, or you may make your own using the recommended list of items below as a reference. Whichever you chose, it is important to include any personal items such as medications and emergency phone numbers. Check the kit regularly and replace any used or out-of-date items.

- Adhesive bandages (assorted sizes)
- Antibiotic ointment
- Antiseptic wipes
- Instant cold compress
- Hydrocortisone ointment
- Scissors
- Sterile gauze pads (assorted sizes)
- Butterfly bandages
- Tweezers
- Prescription medications (asthma inhalers, Epipen)
- Emergency phone numbers
- Charged cell phone

Didmyo Fact Sheet

Didymosphenia geminate, commonly known as "Rock Snot" or "Didymo," is an aggressive freshwater alga that has undergone a recent large expansion in range. It has the potential to form nuisance blooms during which it can form mats several inches thick by attaching itself to streambeds by stalks that form a thick brown mat on rocks, plants, and other aquatic surfaces. The thick growth reduces the quantity and quality of aquatic habitat.

Didymo was detected in rivers of Vermont, New York, and New Hampshire during the summers of 2006 and 2007. Because the factors that cause Didymo to undergo rapid growth are unknown and there is no known method of eradication, it is important to prevent the spread of these algae to uninhabited streams. Therefore, ***we disinfect all waders and equipment when traveling between streams***. In order to prevent the spread of didymo to other regions waders should not be transported and used in different regions or countries.

Follow the link for a detailed description of Didymo by the Vermont Department of Environment Conservation Water Quality Division:

http://www.anr.state.vt.us/dec//waterq/lakes/htm/ans/lp_didymo.htm#how_can_I_disinfect

Disinfecting Waders

We have supplied your team with concentrated Quaternary Ammonium Disinfectant (Quat solution) to kill and prevent the spread of nuisance biological agents such as Didymo. This procedure is adapted from the Vermont Agency of Natural Resources method for equipment disinfection.

ATTENTION: Quat is a highly basic solution. Protective gloves MUST be worn when handling the concentrated solution. Once diluted with water, it is safe to handle.

To prepare a 2.5% solution:

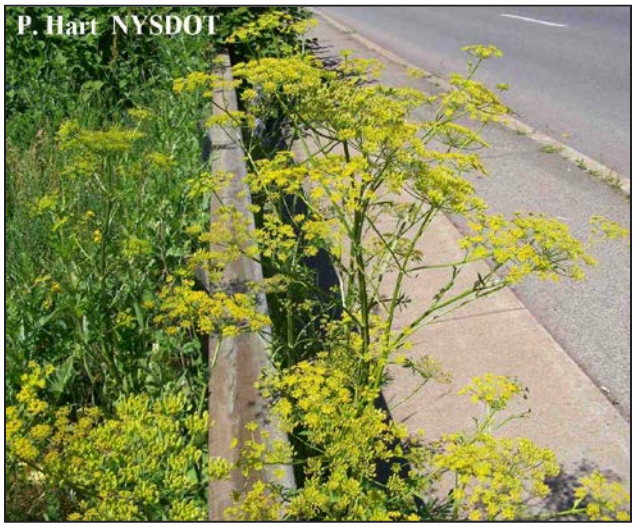
- Add 25mL of concentrated Quat to a spray bottle. Dilute to 1L. (For 500mL of solution, add 12.5mL of concentrated Quat and dilute with water to 500mL.) **Quat solutions should be replaced every 2 – 3 days to remain effective, so prepare only as much as is necessary for a site visit.**

- Fill the second spray bottle with water.

- When exiting the stream following sampling, spray waders and other equipment thoroughly with the 2.5% Quat solution. Let sit for ~2 minutes. Spray with the water to rinse.

Field Precautions

Poison Parsnip



- **Location**: Predominately found on the sides of highways and fields throughout Vermont.

- **Appearance**: The plants typically grow 3-6 feet tall and resemble Queen Anne's Lace, but the flowers are yellow instead of white.

- **Danger**:
  - The plant contains a high concentration of furocoumarin chemicals
  - The plant's juices may be transferred to your skin if you brush against the flower tops or broken leaves or stems
  - When the juices on the skin are exposed to ultraviolet light on both sunny and cloudy days the furocoumarin chemicals bind with nuclear DNA and cell membranes.
  - **This process destroys cells and skin tissue, causing severe burns in which the skin to reddens and blisters**

- **Protecting Yourself:**
  - Avoid exposure to the plant by choosing stream sites or access areas free from poison parsnip
  - If unavoidable, wear long sleeve shirts, pants (or your waders!), and gloves to prevent direct contact with your skin
  - Rinse and wash all clothing items and skin surfaces immediately following possible exposure. Keep exposed skin out of sunlight.

Poison Ivy

Poison ivy in spring.

Image © Jonathan Sachs 2002

Avoid poison ivy

Preventing contact with poison ivy

•Do not touch or handle any part of the plant

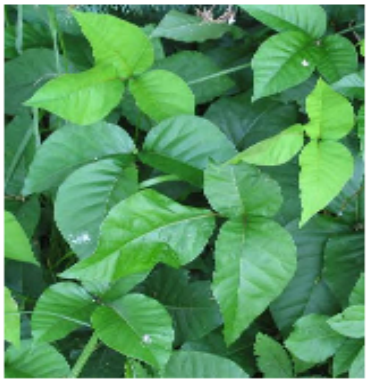
•Remove and wash shoes or clothing that has contacted poison ivy. Wash your hands immediately with soap and water

Preventative treatment Modified From http://poisoncontrol.uchc.edu

•If you have touched poison ivy, avoid spreading the oils to other body parts and wash the affected skin with soap and water within 15 minutes

•Use a nail brush to clean under finger nails

•Swab with rubbing alcohol after washing



Poison ivy in summer.

www.kentuckycrosswords.com

Myths Vs Facts: Fact #1: this fact list is modified from www.zanfel.com

Myth: Scratching poison ivy blisters will spread the rash.
Fact: Fluids from blisters will not spread the rash. Before blisters form, the rash can only be spread by unbound urushiol. Scratching of blisters can cause bacterial infection.

Myth: Poison ivy rash is "contagious."
Fact: The rash is a reaction to urushiol. The rash cannot pass from person to person after the urushiol binds to skin.

Myth: After the first time, I can't get poison ivy again.
Fact: Not everyone reacts to poison ivy upon first or subsequent exposures, people generally become more sensitized with each contact and may react more severely to subsequent exposures.

Myth: Once allergic, always allergic to poison ivy.
Fact: A person's sensitivity changes over time, even from season to season. People who were sensitive to poison ivy as children may not be allergic as adults.

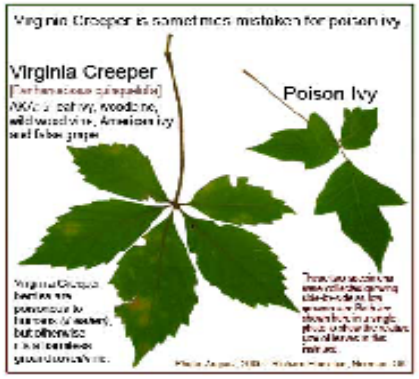
Myth: Dead poison ivy plants are no longer toxic.
Fact: Urushiol remains active for up to five years. Never handle dead plants that look like poison ivy without proper protection.

Myth: Burning is the best way to dispose of poison ivy.
Fact: The toxic oils from poison ivy spread in the smoke and can cause full-body rash and more serious health problems if inhaled.

Zanfel Laboratories provides poison ivy treatment brochures for free to BSA troops. Call 1800 401 4002

If a rash develops From http://poisoncontrol.uchc.edu

•Apply calamine lotion, cool compresses, or over the counter corticosteroid creams to lessen itching. Oatmeal baths can also help. Avoid scratching and cover open blisters to avoid infection. If face or genitals are involved, see a doctor for evaluation. If symptoms are persistent after these treatments see a doctor.



Ticks & Lyme Disease

T I C K S & L Y M E D I S E A S E

What Is Lyme Disease?

Lyme disease is a bacterial infection caused by the bite of an infected deer tick. Untreated, the disease can cause a number of health problems. Patients treated with antibiotics in the early stage of the infection usually recover rapidly and completely.

Where Is Lyme Disease Found?

In the United States, infected ticks can be found in the north-east, including New York State; in the upper Midwest; and along the northwest coast.

What Are the Symptoms of Lyme Disease?

The early symptoms of Lyme disease may be mild and easily missed. If you find a tick attached to your skin, remove the tick with tweezers and watch for the symptoms of Lyme disease. In 60-80% of cases the first symptom is a rash, known as *erythema migrans,* that:

- Occurs at or near the site of the tick bite.
- Is a "bulls-eye" circular patch or solid red patch that grows larger.
- Appears between three days and one month after the tick bite.
- Has a diameter of two to six inches.
- Lasts for about three to five weeks.
- May or may not be warm to the touch.
- Is usually not painful or itchy.
- Sometimes multiple rashes appear.

How Can I Safely Remove a Tick?

If you DO find a tick attached to your skin, do not panic. Not all ticks are infected, and your risk of Lyme disease is greatly reduced if the tick is removed within the first 36 hours.

To remove a tick:

- Use a pair of pointed tweezers to grasp the tick by the head or mouth parts right where they enter the skin. DO NOT grasp the tick by the body.
- Pull firmly and steadily outward. DO NOT jerk or twist the tick.
- Place the tick in a small container of rubbing alcohol to kill it.
- Clean the bite wound with rubbing alcohol or hydrogen peroxide.
- Monitor the site of the bite for the next 30 days, for the appearance of a rash. If you develop a rash or flu-like symptoms, contact your health care provider immediately.

What Else Can Be Done?

- Keep lawns mowed and edges trimmed.
- Clear brush, leaf litter and tall grass around the house, and at the edges of gardens and stone walls.
- Stack woodpiles neatly away from the house and preferably off the ground.
- Clear all leaf litter (including the remains of perennials) out of the garden in the fall.
- Keep the ground under bird feeders clean so as not to attract small animals.
- Locate children's swing sets and other play equipment in sunny, dry areas of the yard, away from the woods.

For more information on Lyme disease, contact your local health department or refer to the NYS Department of Health web site at ***www.health.state.ny.us***

- Do NOT apply repellents directly to children. Apply to your own hands and then put it on the child.
- When applying repellents, avoid the child's face and hands.
- Do not apply repellents on skin damaged by sunburn, cuts, bruises or other conditions, such as psoriasis.
- Avoid prolonged and excessive use of DEET.
- Do NOT apply repellents in enclosed areas.
- Do NOT apply directly on your face.
- Do NOT apply near eyes, nose or mouth.
- Wash treated skin and clothing after returning indoors.
- If you believe you or a child is having an adverse reaction to a repellent containing DEET, wash the treated area immediately and contact your local health care provider or local poison control center.

Also consider these important facts:

- If you tuck pants into socks and shirts into pants, be aware that ticks will climb upward to hidden areas of the head and neck, so spot-check clothes frequently.
- Clothes can be sprayed with DEET or treated with permethrin. Follow label instructions carefully.
- Upon returning home, clothes can be put in a high temperature dryer for 20 minutes to kill any unseen ticks. A shower and shampoo may help to dislodge crawling ticks, but this is not always effective.
- Any contact with vegetation, even playing in the yard, can result in exposure to ticks. Frequent tick checks should be followed by a whole-body examination and tick removal each night. This is the single most effective method for prevention of Lyme disease.

Ticks will attach themselves anywhere including the thighs, groin, trunk, armpits and behind the ears. If you are infected, the rash may be found in one of these areas.

Around the time the rash appears, other symptoms, such as joint pain, chills, fever and fatigue can occur, but they may seem too mild to require medical attention. As Lyme disease progresses, severe fatigue, a stiff aching neck, and tingling or numbness in the arms and legs, or facial paralysis can occur.

The most severe symptoms of Lyme disease may not appear until weeks, months or years after the tick bite. These can include severe headaches, painful arthritis, swelling of the joints, and heart and central nervous system problems.

How Is Lyme Disease Diagnosed?

If you think you have Lyme disease, you should see your health care provider immediately. Early diagnosis of Lyme disease should be made on the basis of symptoms and history of possible exposure to ticks. Blood tests may give false negative results if performed in the first month after the tick bite.

How Is Lyme Disease Treated?

Early treatment of Lyme disease involves antibiotics and almost always results in a full cure. However, the chances of a complete cure decrease if treatment is delayed.

In a small number of cases, Lyme disease can become a chronic condition. However, some patients have reported slow improvement and even an end to symptoms, months or even years after treatment.

How Can I Protect Against Ticks and Prevent Lyme Disease?

Deer ticks live in shady, moist areas at ground level. They will cling to tall grass, brush and shrubs, usually no more than 18-24 inches off the ground. They also live in lawns and gardens, especially at the edges of woods and around old stone walls.

Deer ticks cannot jump or fly, and do not drop onto passing people or animals. They get on humans and animals only by direct contact. Once a tick gets on the skin, it generally climbs upward until it reaches a protected area.

In tick-infested areas, your best protection is to avoid contact with soil, leaf litter and vegetation. However, if you garden, hike, camp, hunt, work, or otherwise spend time in the outdoors, you can still protect yourself:

- **Wear light-colored clothing** with a tight weave to spot ticks easily.
- **Wear endosed shoes, long pants and a long-sleeved shirt.** Tuck pant legs into socks or boots and shirt into pants.
- **Check clothes and any exposed skin frequently** for ticks while outdoors.
- **Consider using insect repellent.**
- **Stay on cleared, well-traveled trails. Avoid contacting vegetation.**
- **Avoid sitting directly on the ground or on stone walls.**
- **Keep long hair tied back,** especially when gardening.
- **Do a final, full-body tick check at the end of the day** (also check children and pets), and remove ticks promptly.

What Do Ticks Look Like?

Two common types of ticks are dog ticks and deer ticks. Deer ticks can carry Lyme disease. Dog ticks can carry Rocky Mountain spotted fever but have not been known to carry Lyme disease.

Enlarged View Female Deer Tick

Deer Ticks Actual Size

Female deer ticks have four pairs of legs and are red and black in color, while the male is all black. Young deer ticks - nymphs, are brown, the size of poppy seeds and very difficult to spot. An adult deer tick is only about the size of a sesame seed – still very small.

Enlarged View, Male and Female Dog Ticks

Adult Dog Tick Actual Size

Dog ticks are the most common type of tick, and, while feeding, can be as large as a small pea. They have four pairs of legs, are reddish-brown and are easier to spot. Dog ticks turn gray while feeding. Ticks can be found throughout the year, but they are most active during the spring, early summer and fall, when it is warm and moist.

What About Insect Repellent?

Two active ingredients found in repellents are DEET (the label may say N, N-diethyl-m-toluamide) and permethrin. Permethrin is only used on clothes. DEET repellents or products come in many different concentrations, with percentages as low as five percent or as high as 100 percent. In general, the higher the concentration the higher the protection, but the risk of negative health effects goes up too. Use the lowest concentration that you think will provide the protection you need. The New York State Health Department recommends taking these precautions when using repellents that contain these active ingredients:

- Store out of the reach of children and read all instructions on the label before applying.
- Do NOT allow children to apply repellents themselves.

Cyanobacteria

What is cyanobacteria?
Cyanobacteria, also known as blue-green algae, are naturally occurring bacteria that are present in Lake Champlain and other water bodies around the world. Like plants, they use photosynthesis to convert sunlight into energy. Usually cyanobacteria cannot be seen by the naked eye. However, under certain conditions, the algae grow prolifically and are visible as blooms. The blooms appear as a cloudy pea green accumulation in the water. Generally, these blooms of cyanobacteria occur when there is a balance of certain factors including: an abundance of available nutrients, warm surface water temperatures, and calm winds.

Why should be concerned?
Unfortunately, certain types of blue-green algae produce toxins or poisons. When the algae die and break down, these toxins are released into the water. Exposure to these toxins have health impacts on humans and animals. Human health effects from cyanobacteria blooms vary depending on the type and duration of exposure (including inhalation of water droplets). In the summers of 1999 and 2000, the deaths of several dogs were linked to the cyanobacteria in Lake Champlain.



Photo source: Lake Champlain Basin Program

Identification and Avoidance: ***When in Doubt, Stay Out***
In general, blooms have the appearance of:

- Cloudy water as thick as pea soup or green paint on the water
- While generally green or blue-green in color, they can be brown or even purple
- A thick mat or foam may form as it accumulates onto shore

Blooms usually occur in August or September and can appear and disappear rapidly. There is no accurate way to identify the algae without a microscope. If you are suspicious, simply stay out of and away from the water.

References and Resources:
Check Current Conditions Online:
http://healthvermont.gov/enviro/bg_algae/weekly_status.aspx

Vermont Department of Health's Blue-Green Algae Guidance Document:
http://healthvermont.gov/enviro/bg_algae/documents/BGA_guide.pdf

Websites:
http://healthvermont.gov/enviro/bg_algae/bgalgae.aspx
http://www.lcbp.org/water-environment/human-health/cyanobacteria/

http://www.lakechamplaincommittee.org/lcc-at-work/algae-in-lake/

Photo Galleries:

http://www.lcbp.org/2012/12/photo-gallery-2008-cyanobacteria-blooms/
http://healthvermont.gov/enviro/bg_algae/photos.aspx#bg

Report a Blue-green Algae Bloom:
If you have questions or want to report a suspected bloom:
Call 1-800-439-8550 or 802-863-7220, or
email AHS.VDHBlueGreenAlgae@state.vt.us

If you believe that someone has become ill because of exposure to blue-green algae, seek medical attention and contact the Health Department at 1-800-439-8550.

Measuring Infiltration Rates

This exercise is included in the manual for RACC teachers to use with their classes, if interested. It is not a required data collection task for your participation in the RACC Streams Project and these data will not be uploaded to the Streams Project database.

Introduction:
Infiltration is the movement of water into a soil profile. The rate at which infiltration occurs is controlled both by the inherent properties of the soil and by the ways in which humans have modified the landscape. Infiltration rates, in turn, control runoff rates and soil erosion, which are important because these processes influence the behavior of hillslopes. This exercise is designed to introduce you to a simple method for measuring infiltration rates. You will use a ring infiltrometer to measure infiltration at plots that represent differences in disturbance of the soil surface. You may also measure the soil bulk density and gravimetric moisture content at the measurement sites and compare these to measured infiltration rates.

Methods:
Select two sites for measurement of soil properties and infiltration rates representing (1) a forested site showing no signs of noticeable compaction or human traffic, and (2) a site located on a designated hiking trail or one showing noticeable signs of compaction. You will extract soil cores from a location immediately adjacent to your infiltration test.

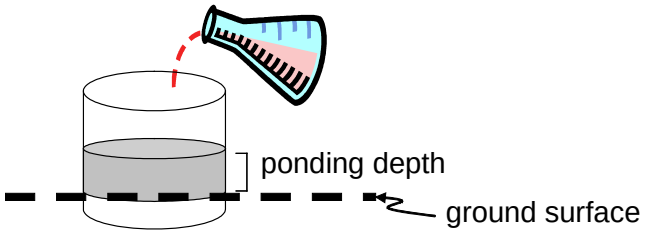
A. Infiltration test

1. Select a level site for your test. Remove loose debris (leaves, sticks) from an area the size of your infiltrometer (but do not pull up rooted plants; this will affect the pores in the soil).

2. Insert the ring infiltrometer several centimeters into the soil. Record this penetration depth. The ring should be inserted deeply enough and sealed adequately to the soil to preclude any leakage from the ring.

3. Fill out the top of the data sheet to record your group members and experimental set up.

4. To conduct the infiltration test, establish a standing pond of water within the ring that you maintain to within about 10% of this depth throughout the test. Once you have established this ponding depth, add water to maintain a constant ponding depth throughout your experiment. This should require frequent additions of water at the start of your experiment and less frequent additions as your test proceeds. Continue to make measurements of water additions for at least one hour, recording additions at least every 10 minutes, but more frequently if needed to maintain a constant ponding depth.



B. Soil extraction for bulk physical properties

1. Immediately adjacent to each of your infiltration tests, extract a bulk sample of the mineral soil using the soil auger. Retain only the center ring of your extracted sample. Be sure to record the dimensions (diameter, length) of the device used to extract your sample.

2. Place the sample into a plastic bag, labeled with your name(s) and indicate whether it is from the "forest" or "trail" site.

3. In the lab, weigh an empty aluminum pan to determine the tare weight, then place your sample in the pan and weigh again. Place the soil sample in the oven for overnight drying at 103^{o}C. When drying is complete, weigh the sample again to determine dry weight.

C. Data reduction, analysis and interpretation

1. Use the data reduction instructions following each data sheet to make calculations from your raw field data.

2. Enter your infiltration data for both sites into a spreadsheet with columns to record time, elapsed time, volume of water added, and depth of water infiltrated at each time step. Your entries should include at least one hour of observations.

3. Plot the data in your spreadsheet as an x,y scatterplot with elapsed time on the x axis and infiltration rate on the y axis (see for example figure 5.4 in your textbook).

4. Estimate a steady state infiltration capacity from your data plot for both sites by taking an average of measurements over a time interval during which infiltration rate shows little or no change.

5. Consider/discuss:
   - How do the steady state infiltration rates differ between the two sites you measured?
   - What factors influence the rate at which infiltration occurs; how do your measurements of bulk density relate to any of these factors?
   - What are the limitations associated with inferring infiltration rates across the landscape based on the measurements you have made?

Infiltration Test Data Sheet

Group member names: _____

Experiment date: _____ Location: _____

Experimental Set-up

Diameter of infiltrometer (d): _____

Site type: forest | trail

Infiltrometer length (cm) _____ Depth inserted into soil (cm) _____

Ponding depth (cm) _____

DATA:

time (hr:min:sec)	volume start (ml)	volume end (ml)	volume added ( )		time (hr:min:sec)	volume start (ml)	volume end (ml)	volume added ( )
0:00:00	--	--	--					
	1000							
			.					.

Adapted from Measuring Infiltration Lab by Beverley Wemple, UVM Geography Department.
Please do not distribute without permission.

Infiltration Test Data Sheet

Group member names: _____

Experiment date: _____ Location: _____

Experimental Set-up

Diameter of infiltrometer (d): _____

Site type: forest | trail

Infiltrometer length (cm) _____ Depth inserted into soil (cm) _____

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DATA:

time (hr:min:sec)	volume start (ml)	volume end (ml)	volume added ( )		time (hr:min:sec)	volume start (ml)	volume end (ml)	volume added ( )
0:00:00	--	--	--					
	1000							
			.					.

Data reduction:

To compute infiltration rates from your experiment, you will need to convert the volume of water to a water depth, then divide by the elapsed time. Follow the steps below to reduce your data and compute infiltration rates for each experiment. In each step, write the formula you use, then clearly show your calculations with units:

1. Calculate the surface area (A) of the infiltrometer from the diameter of the ring. (4 pts)

2. For one time step on one your data sheet, compute depth of water infiltrated (D) as the volume[1] of water (V) divided by the surface area (a) of the infiltrometer. Use an arrow on your data sheet to indicate the time step for which you are making this calculation. (4 pts)

3. For the time step used in #2 above, convert the elapsed time (t) in minutes and seconds to time in hours (this should be a fraction of an hour). (3 pts)

4. Compute infiltration rate (I) by dividing water depth (D) by elapsed time (t). Express your answer in cm/hr (4 pts)

[1] Note: Water volume for the experiment is measured in milliliters. 1 ml = 1 cm^3.

Bulk Density Data Sheet

Plot 1 (circle one): forest | trail

Auger ring diameter (cm) _____ Auger ring length (cm) _____

Sample tare weight (g): _____

Sample field weight (g): _____

Sample dry weight (g): _____

Notes on site conditions:

Plot 2 (circle one): forest | trail

Auger ring diameter (cm) _____ Auger ring length (cm) _____

Sample tare weight (g): _____

Sample field weight (g): _____

Sample dry weight (g): _____

Notes on site conditions:

Data reduction:

To compute bulk density and gravimetric moisture content, you will need to calculate the volume of soil extracted, then weigh it to get mass of the soil and mass of water lost with drying. Follow the steps below to reduce your data. For each step, write the formula you use and clearly show your calculations with units:

Forest site:

1. Calculate ring volume. (3 pts)

2. Calculate the bulk density of the soil sample. (3 pts)

3. Calculate the gravimetric moisture content of the soil sample. (3 pts)

Trail site:

1. Calculate ring volume. (2 pts)

2. Calculate the bulk density of the soil sample. (2 pts)

3. Calculate the gravimetric moisture content of the soil sample. (2 pts)

High School Team Calendar – Independent Projects 2015-16

June 22-26	**Training Week**
July – winter	Identify a research question Collect data / conduct investigation ➢ ➢ ➢ ➢ ➢
December–March	**Project Presentation** ➢ Export data from websites, if needed: www.uvm.edu/epscor/redir/streamsprojectdata and/or other data sites, if applicable ➢ Analyze data ➢ Create a poster or PowerPoint presentation describing your research
February	Submit application for 2016-17 program, if applicable
April, date tbd	Present your research at the 2016 VT EPSCoR Student Research Symposium!

High School Team Calendar – Streams Project 2015-16

June 22-26	**Training Week**
July	**Site Preparation** • Teams deploy iButton temperature sensors as early as possible at both sites • If applicable, Janel will install a stage sensor at one site
July – winter	**Stream Monitoring** 1. At initial site visit: a. Conduct Site and Habitat Assessment b. **Upload** Site and Habitat Assessment data to database 2. At site visit every two to three weeks: a. Collect grab samples for nutrients and TSS b. Conduct Water Quality Assessment c. **Upload** Water Quality Assessment data to the website database d. Freeze nutrient samples and refrigerate TSS samples e. Send frozen nutrient samples and cold TSS samples to Janel at SMC after each collection date 3. One-time occurrence: a. Collect macroinvertebrate sample (late summer to mid-fall): • Record macroinvertebrate field data • Collect 4 macroinvertebrate community replicates at each site • (If you choose to you may also want to collect a post-storm macroinvertebrate community sample) b. **Upload** macroinvertebrate field data to the website database 4. Identify macroinvertebrate replicate samples and **upload** identification data to the database 5. When the stream temperature and/or weather gets cold: a. Remove iButton temperature sensors from stream sites, b. Remove stage sensor and housing from stream site, c. Download temperature data using data shuttle, and d. Email temperature csv file to Janel at jroberge@smcvt.edu
December–March	**Research Project** ➢ Identify a research question ➢ Export data from the website: www.uvm.edu/epscor/redir/streamsprojectdata and/or other data sites, if applicable ➢ Analyze data ➢ Create a PowerPoint poster or oral presentation describing your research
February	Submit application for 2016-17 program, if applicable
April, date tbd	Present your research at the 2016 VT EPSCoR Student Research Symposium!

Global Decomposition Project

Background:
Soil decomposers, such as some bacteria and fungi, obtain energy needed for life from dead and decomposing plant and animal remains, known as *soil organic matter*. Soil organic matter is important to local ecosystems because it affects soil structure, regulates soil moisture and temperature, and provides energy and nutrients to soil organisms. It is also important globally, because it stores a large amount carbon, and when microbes "eat", or *decompose*, organic matter they release greenhouse gasses (primarily carbon dioxide (CO_2), but also methane (CH_4) when conditions are right) into the atmosphere, which affects the Earth's climate.

Aerobic decomposition (i.e., decomposition that requires oxygen) is the same chemical process as respiration, in which organisms, including humans, break down sugars to obtain energy:

$$C_6H_{12}O_6 + O_2 \rightarrow CO_2 + H_2O + \text{energy}.$$

In addition to oxygen and sugar, aerobic decomposers require nutrients, water, and suitable temperatures. Other soil conditions, such as pH, as well as the chemical composition of organic matter also affect decomposition. It is important to understand the conditions that affect decomposition because of the role organic matter plays in local ecosystem processes as well as the role of decomposition in global climate.

Student Learning Objectives

1. Students will learn about decomposition and how it relates to respiration.
2. Students will learn about the role of decomposers in the formation of soil.
3. Students will look at conditions that control decomposition in the soil.
4. Students will make connections between greenhouse gasses from decomposition, such as carbon dioxide and methane, and how they affect the atmosphere and the earth's climate.
5. Students will collect meaningful data using lab equipment, such as electronic balances.

Project Objective:
The objective of the Global Decomposition Project (GDP) is to explore local and global patterns of soil organic matter decomposition, to educate students and the general public about soil organic matter and decomposition, and address these questions:

1. How do environmental conditions control decomposition of organic matter in soil?
2. Why do some areas accumulate organic matter and others do not?

We will answer these questions by comparing decomposition rates of a common substrate, cellulose paper, within and across ecosystems and biomes. The protocol below is a standard method for measuring decomposition using cellulose, which is a component of plant cell walls and common component of soil organic matter.

Next Generation Science Standards Learning Outcomes:

HS.Earth Systems

HS-ESS2-2 *Analyze geoscience data to make the claim that one change to Earth's surface can create feedbacks that cause changes to other Earth systems.*

HS-ESS2-6 *Develop a quantitative model to describe the cycling of carbon among the hydrosphere, atmosphere, geosphere, and biosphere.*

HS.Weather and Climate

HS-ESS2-4 *Use a model to describe how variations in the flow of energy into and out of Earth's systems result in changes in climate.*

HS-ESS3-5 *Analyze geoscience data and the results from global climate models to make an evidence-based forecast of the current rate of global or regional climate change and associated future impacts to Earth systems.*

HS.Human Sustainability

HS-ESS3-4 *Evaluate or refine a technological solution that reduces impacts of human activities on natural systems.*

HS-ESS3-6 *Use a computational representation to illustrate the relationships among Earth systems and how those relationships are being modified due to human activity.*

HS.Engineering Design

HS-ETS1-1 *Analyze a major global challenge to specify qualitative and quantitative criteria and constraints for solutions that account for societal needs and wants.*

HS-ETS1-2 *Design a solution to a complex real-world problem by breaking it down into smaller, more manageable problems that can be solved through engineering.*

HS-ETS1-3 *Evaluate a solution to a complex real-world problem based on prioritized criteria and trade-offs that account for a range of constraints, including cost, safety, reliability, and aesthetics as well as possible social, cultural, and environmental impacts.*

HS-ETS1-4 *Use a computer simulation to model the impact of proposed solutions to a complex real-world problem with numerous criteria and constraints on interactions within and between systems relevant to the problem.*

Global Decomposition Project

Cellulose Decomposition Bag Protocol

Protocol overview: Cellulose decomposition bags are made of cellulose paper enclosed in a screen mesh. The bags are placed in the ground for a set period of time, then removed and weighed to determine mass loss. The screen allows decomposing organisms, such as bacteria and fungi, to access and decompose the cellulose.

We use cellulose because it is a main component of plants, which are the primary source of organic matter in soil. By using a common substrate, we can ask questions about how different environments affect decomposition. Another interesting question is how different substrates (e.g., leaves from different plant species) differ in their decomposition rates. While substrate differences are not addressed in this protocol, you can design an experiment to explore this question using substrates of local interest or importance.

You are encouraged to add experiments to the basic protocol to answer questions your students develop about decomposition in your area. To understand this process at larger scales (i.e., continent or globe), we would like to compare and share results with all participants, which will only be possible if data collection is standardized. One outcome of this project will be a global database and interactive map of decomposition rates. That database will be available to all GDP participants and to the broader public**. In order for this component of the project to succeed, it is important that you follow the methods outlined below and submit your final data back to the GDP.**

The protocol is broken down into four steps:

I. **Field deployment**
II. **Removing bags**
III. **Processing the bags**
IV. **Optional: Making more bags**

<u>Supplies needed</u>

Deployment

1. Serrated knife
2. Trowel or small flat shovel
3. Fishing line and flags, or another method of marking your bag locations
4. Decomposition bags

Processing

1. Drying oven or you can air dry
2. Paint brushes
3. Tweezers
4. Scale that weighs in increments of 0.001g (0.01 would also work).

The filters weigh ~ 0.45 g prior to deployment, so a 10% mass loss requires a scale that can detect ~ 0.04g. If you do not have access to a scale, mail your clean filters to the GDP address provided, or contact a scientist in your area. There are many reasonably priced 0.01 g increment scales available online. Please let me know if you find one you would recommend.

Optional: Making more bags

1. Cellulose filter paper (Fisher; Whatman P8; product # 09-802-1B), or lignin-free paper purchased from an office supply store
2. Fiberglass window screening (14 x 18 mesh), purchased at any hardware store
3. Aluminum tags (e.g., from Forestry Suppliers) for marking bags. You can also fold up a piece of aluminum foil to make a tag, or mark the bags in some other way that will stand up to several weeks or months of burial in the soil.
4. Heat sealer or an iron
5. Stapler
6. Scale that weighs in increments of 0.001g or 0.01g. See above for more information.
7. Optional for cutting: Foam mat, rotary cutter, ruler. Scissors and/or a paper cutter work.

I. Field deployment

You can place all of your bags in one location, or you can explore different ecosystems that may vary in soil nutrients, soil moisture, plant cover, or other factors. Place 3-5 replicate bags in each location with bags at least 1m apart.

1. Place decomposition bags in desired location. Insert bag by cutting 10 cm deep and 10 cm wide slit into the ground with a serrated knife (Fig. 1a). Open the slit in the ground with your hands or a trowel, and insert the bag (Fig. 1b). Insert bag vertically with the tag at the top, making sure that the bag does not fold when you insert (Fig. 1c). Close the opening in the soil with your hands. The bags should be placed so that the top of the screen is barely visible from the surface and the bottom of the bag is at 10 cm (Fig. 1d).

Figure 1. To install decomposition bags: A. Cut slit in ground; B. open slit with hands or trowel; C. insert bag vertically. D. Top of bag should just be visible.

2. **Important:** Mark bag locations so you can find the bags again! One option is to tie fishing line to the bags before deploying and attach the other end of the line to a flag. Record the flag location.

3. Record relevant information about your field site: location (city, state, country as well as latitude and longitude with a GPS if you have one), date bags deployed, description of site type (e.g., deciduous forest, lawn, wetland), air temperature on the day the bag was buried, other information that is relevant to describe your location. Collecting good field notes is a key skill for young scientists to develop, so this is an important exercise for student participation.

II. Removing the Bags

1. Determine the length of time you will leave your bags in the field. This will vary greatly by the temperature, precipitation, and nutrient status of your location. Below are some suggested deployment lengths. These are coarse estimates and will vary based on local site conditions. *Please help us to refine these times with information from your field site*!

Ecosystem Type	Mass loss/day	Suggested deployment Length*
Tropical rain forest	1-4% mass loss/day	2 weeks
Sub-tropical wetland	0.5-6% mass loss/day	2-3 weeks
Temperate forest	0.2-0.5% mass loss/day	2-3 months
Tropical, arid	0.5-2% mass loss/day	3-6 weeks
Arctic/subarctic	0.1% mass loss/day	3 months +

* Estimates are for spring/summer seasons

2. Remove the bags from the ground. Carefully remove soil from the outside of the bags, and place decomposition bags inside a sealed plastic bag. **If you cannot process bags immediately, place them in the freezer to stop/slow decomposition.** Record the date and weather conditions when the bags were removed from the ground.

3. *Optional:* When you remove the bags, collect ~ 50 g of soil (0-10 cm depth) near bag locations to determine soil moisture content.

Gravimetric water content = weight of water/weight of dry soil
=[(wet soil)-(dry soil)]/dry soil

4. *Optional:* Ship 5-10 g dry weight of each labeled soil sample to the WHRC (address at the end of this protocol) for carbon and nitrogen analysis.

III. Processing the Bags

1. Carefully wash the bags (the filter should still be encased in the screen mesh) in water to remove any soil attached to the outside surface of the screen. Either run water over the bags or wash them all in a bin of water. Wash the bags enough to clean them but not too much that the paper inside disintegrates.

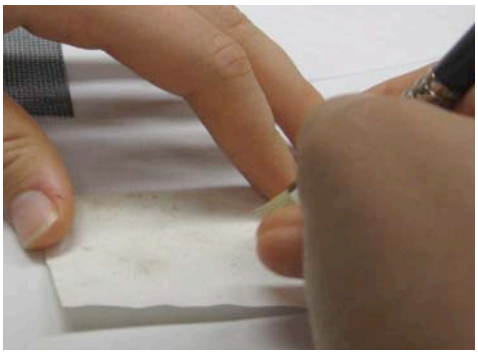

Figure 2: Clean off soil/debris using a dry and clean paintbrush.

2. Dry the bags (with filter paper still encased in the mesh) in a lab drying oven at 60° C for 48 hours, or at room temperature in a dry and sunny location until the bags are no longer losing water weight. If you have a balance, weigh the bags daily until the weight is constant. If you do not have a scale, then dry bags until filter papers are dry to the touch.

3. Clean the cellulose paper: Once the filters are dry, open each bag (the edges of the sealed screen will easily pull apart), remove the filter, and use a paintbrush to clean soil from the filters before weighing them (Figure 2). You may also need to use tweezers to remove debris that is stuck onto the paper (Figure 3). If your paper is in many pieces, be careful not to lose any of the pieces during this step.

Figure 3. Some debris may be easier to remove with tweezers.

4. Weigh the cellulose paper: After the decomposition paper has been cleaned (Figure 4), weigh the paper (Figure 5). If you plan to clean all the decomposition papers first before weighing them (which may be necessary if you don't have a scale), then place each cleaned decomposition paper in a separate envelope/bag labeled with the decomposition bag number.

Figure 4. This paper has been cleaned. Although it still looks a little dirty, some stains on the paper won't come off.

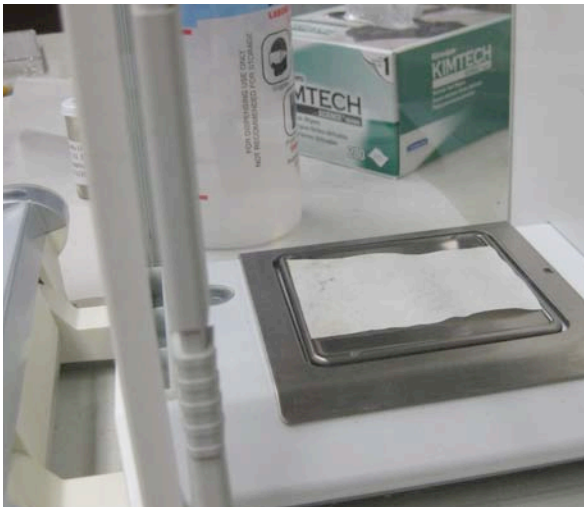

Figure 5. Weigh cleaned filter paper, making sure to record sample number.

5. Enter your data into an excel file (See template) and calculate percent mass loss over time:

% mass loss = [(initial paper weight – final paper weight)/initial paper weight] * 100

% mass loss/day = % mass loss/# days bags were in the ground

6. Create a notes tab on your excel file that includes your "Metadata"-- relevant field and lab notes, and information about your site and samples (See template).

7. Send data to snatali@whrc.org. I will archive data, and once we obtain a critical mass, will post the data online as a database and an interactive map. Until that point, all data will be available to GDP participants or anyone who requests the data.

You can mail cleaned filter papers and soil samples (only U.S. samples) to:

Global Decomposition Project
c/o Sue Natali
Woods Hole Research Center
149 Woods Hole Road
Falmouth, MA 02540

Questions can be addressed to Dr. Sue Natali: snatali@whrc.org

Protocol updated on 11 February 2015 by S. Natali

Extending the Cellulose Decomposition Bag Protocol

You may want to extend this project by making more cellulose bags or using a different substrate. Here are some ideas for extending this project:

1. How does screen size affect decomposition?
2. How do different materials vary in their decomposition rates?
3. How does experimental soil warming (e.g., by placing black plastic over soil to warm it), drying, wetting, fertilization affect decomposition?
4. How does mass loss change over time? Does mass loss/unit day depend on the number of days the bags were placed in the ground?

We would love to hear additional ideas and projects that you and your students develop!

IV. Making More Bags

1. Cut the mesh screen into 10.5 cm x 10.5 cm**.** Screen can be reused after you take bags out of the field. Wash them in tap water, then reuse.

2. Cut the filter paper into 7.5 cm x 7.5 cm squares.

3. Use a heat sealer or an iron to seal the edges of two mesh pieces together on three sides.

4. Cut the aluminum tags, engrave them with the Bag ID's (a ballpoint pen works well for this), and staple them to the upper left corner of the mesh bag.

5. Weigh each filter paper, record weight, and place in the mesh bag.

6. Finish sealing the mesh bags by sealing the remaining edge.

7. Attach bags to ~ 1m of fishing line for field deployment.

Glossary

#

#30 sieve - A strainer that the contents of the kick net is emptied into to remove unwanted debris. The sample material remaining is placed in whirl-paks®.

A

Attached Algae - Algae that has grown attached to a solid object or organism. For example: planktonic algae (slime texture on a rock) or filamentous algae.

B

Bank Full Width - Width of a stream bank at full flood stage. Look for clues such as a high water mark on the bank or on nearby trees, location of trees, or signs of deposition.

Bank Stability - The ability of a stream bank to counteract erosion or gravitational forces.

Baseline Sample - A sample of the quality of water when the body of water is at a normal or resting state. This can be used later on as a comparison to samples that are taken during or after storm events.

Benthic Macroinvertebrates - Organisms that do not have spines, and are generally small, but visible without a microscope. They are abundant near bodies of water and surrounding ecosystems, and usually live in water at some stage of their lives.

Berm - A level space, shelf, or raised barrier separating two areas. These are constructed to control runoff and direct flow.

Bioassessment (or Biological Assessment) - A method of assessing aquatic conditions by surveying biological organisms, such as macroinvertebrates, fish, or plants.

Biological Sampling - Conducting a survey of biological organisms used for beneficial research.

C

Canopy Cover - The amount of sky covered by trees and vegetation over a stream bank.

Channel - (In the context of this research) The physical confinement of a stream that the water flows through, consisting of the stream bed and banks.

Channelization - The straightening and modification of a river corridor as a way to control the water. However, it is difficult to maintain a straight river, as the water tends to erode along the banks to return to a natural winding river.

Channel Sinuosity - A streams natural ability to bend and wind, an important characteristic of rivers to divert high flows and carry/deposit sediment.

Chemical Constituents - The amount of oil, alcohols, aldehydes, esters, ketones, lactones, phenols and terpenes in a water sample.

Cross Sectional Area - The area of a slice of river, perpendicular to flow; used to help determine stream velocity.

D

Deposition - The accumulation of materials, especially sand, out of the water and onto the stream bed.

Didymo (Rock Snot) - A type of freshwater algae that is a nuisance when it blooms, creating thick, brown mats on the streambed. It is found in certain areas of Vermont, therefore waders and nets are decontaminated after use to avoid spreading it.

Discharge (Flow) - The rate that a volume of water (and its associated suspended solids, dissolved chemicals, and biological materials) flows over a specific time. Usually provided in cubic feet per second.

Dissolved Oxygen (DO) - A relative measure of the amount of oxygen that is dissolved or carried in the stream water. Low DO indicates a stressed aquatic ecosystem and can cause fish kills. High DO environments are expected to be found in fast moving streams with riffles and rapids.

Dredging - The scooping and removal of sediment, etc. from the bottom of a stream by humans.

E

Ecological Integrity - The abundance and diversity of organisms at all levels, and the ecological patterns, processes, and structural attributes responsible for that biological diversity and for ecosystem resilience.

Eddies - The swirling of stream water, usually downstream and past a barrier.

Embeddedness - How much of an object is submerged into the substrate under the water. This is provided as the percent of the object, such as a rock, that is buried into the streambed.

Epifaunal - Animals that live on the surface of substrate, such as rocks, pilings, vegetation, or the streambed itself.

EPSCoR – An acronym that stands for "Experimental Program to Stimulate Competitive Research". It is supported by the National Science Foundation (NSF) and seeks to strengthen scientific research and education within the United States.

Ethanol - A form of alcohol that is used to clean lab materials, as well as to preserve insect specimens.

F

Floating Algae - Algae that is not attached to anything, typically refers to mats of algae that have accumulated and are growing together on the water's surface.

Free Floating Algae - Algae that is not attached to anything, such as duckweed.

H

Habitat Assessment Data Sheet - A field sheet used to determine habitat parameters of a stream site.

Headwaters - A tributary stream of a river close to or forming part of its source.

I

iButton - A sensor that measures and records temperature. It works by transferring data in and out of the sensor when it is connected by a USB device.

iButton Capsule - A capsule that protects the iButton from environmental conditions such as temperature, moisture, pressure, and solvents, and allows the iButton to be securely mounted in a stream environment.

Infiltration - The movement of water into and through soil.

In Situ Measurements - Standard parameters that can be taken on the stream site with a water quality instrument.

J

K

Kick Net - A net that is placed, with the opening facing upstream, into the riverbed with the motive of capturing benthic macroinvertebrates. While holding the net stable again the stream bottom, the researcher kicks and stirs up the sediment in front of the net, capturing any organisms living in and around the area.

L

Large Woody Debris (LWD) - Large pieces of wood found in streams, that act as important habitat for aquatic organisms.

M

Macroinvertebrates - see *Benthic Macroinvertebrates*

Macroinvertebrate Data Sheet - A lab sheet that is used to record the taxonomy of the collected macroinvertebrates. This sheet also includes sample identification data as well as squares picked data.

Macroinvertebrate Habitat Data Sheet - A field sheet that is used to record the conditions of the stream including pebble count, canopy cover, temperature, water velocity, pH, and width data. It is also used to record macroinvertebrate collecting locations.

N

Nitrogen - An odorless and colorless element that makes up about 78% of the earth's atmosphere and is necessary for life to exist. Too much dissolved nitrogen in a water source can lead to eutrophication.

NOAA (National Oceanic and Atmospheric Administration) - A Department of Commerce agency that maps out oceans, predicts climate changes, provides weather and natural disaster reports, and helps conserve oceanic resources.

O

One-WireViewer - iButton temperature sensor software for your computer. A Java demonstration application for iButton that features from your PC.

Orthophosphate - A lone phosphate molecule, PO_4, a phosphorus atom connected to four oxygen atoms. This is usually how phosphorous exists in the environment. To calculate the total phosphorus of a water sample, all forms of phosphorus in the sample are converted to orthophosphate.

Outfalls - The place where a river, drain, or sewer empties into the sea, a river, or a lake.

P

Pebble Count - The tallying of 100 or more random sediment samples, measured by walking up and downstream in a zig-zag pattern and selecting random points to measure along the way.

Phosphorus - A solid, nonmetal element (P) that is necessary for life and typically exists in nature as a phosphate molecule (PO_4). Inorganic and organic phosphorus can be dissolved or suspended in water and too much phosphorus in a water source can lead to eutrophication.

Pools - Deep parts of streams that typically occur after riffles.

Poison Ivy - A toxic, flowering plant with three leaves that is common locally. It is known for irritating skin that comes in contact with it.

Poison Parsnip - A common, local, flowering plant with yellow flowers. Can be an irritant if the inner sap is exposed and comes in contact with skin.

Q

Quaternary Ammonium Disinfectant - A combination of water and quaternary ammonium (QUAT) that is used to sanitize waders after using them; ensuring that nothing harmful is transmitted when they are transported.

R

RACC - Stands for "Research on Adaptation to Climate Change" which is the current grant funding Vermont EPSCoR research. The research aims to answer the following overarching question: How will the interaction of climate change and land use alter hydrological processes and nutrient transport from the landscape, internal processing and eutrophic state within the lake and what are the implications for adaptive management strategies?

Replicate - When multiple samples are taken in the field to strengthen the dataset and to account for naturally variability.

Riffles - A rocky or shallow part of a stream or river with rough water that is typically high in dissolved oxygen.

Riparian Zone – The interface between land and a river or stream. An important factor in correlating land use and stream health

Riprap - Loose stone used to form a foundation for a breakwater or other structure.

Rooted Emergent - Refers to a plant that is rooted in sediment below a body of water, such as cattails.

Rooted Floating - Refers to an aquatic plant that is rooted below a body of water that floats to the top, such as lilies.

Rooted Submergent - Refers to an aquatic plant that is rooted below a body of water that does not stick out, such as water milfoil.

S

Sample ID Number - Located at the top of the Macroinvertebrate Data Sheet, this ID Number consists of yy/mm/dd and the Replicate number.

Sensor Field Data Sheet - To be completed at each field site; records temperature and sage sensor data.

Snag - In aquatic systems, this refers to trees and branches that have fallen into the stream.

Stage Sensor (HOBO Water Level Logger) - A battery powered device that is used by RACC which measures stage or water level of fresh water streams.

Stream Gradient - The slope of a stream. How to know if your stream site is high or low gradient:

1. Determine the stream type using this chart below.
    a. Is your stream site confined by valley walls?
    b. What is the general valley slope of your site?
        i. Valley width is important because it is an indicator of how confined the stream is and whether it will have access to a floodplain at different flood levels. To determine valley width differences look for relative changes in the distance between toes of opposing valley walls. The toe of a valley wall can be identified as the bottom of the more steeply sloped portion of the valley.
        ii. If your site is unconfined by valley walls and <2% slope (think fairly flat, not down a steep hill, the water has access to a floodplain when it rains, etc.) you'd classify it as a type C stream.
        iii. If your site has a steeper slope and valley walls that confine the stream (does it have room to meander or change course?), you'd classify it as a type A stream.

Table 2.2 Phase 1 – Reference Stream Typing Chart

Reference Stream Type	Confinement (Valley Type)	Valley Slope
A	Narrowly confined (NC)	Very Steep > 6.5 %
A	Confined (NC)	Very Steep 4.0 - 6.5 %
B	Confined or Semi-confined (NC, SC)	Steep 3.0 - 4.0 %
B	Confined or Semi-confined or Narrow (NC, SC, NW)	Mod.- Steep 2.0 – 3.0 %
C or E	Unconfined (NW, BD, VB)	Mod.- Gentle < 2.0 %
D	Unconfined (NW, BD, VB)	Mod.- Gentle < 4.0 %

2. Once you know what your stream type is, you can use the table below to determine if your site is high or low gradient.
    a. If your site is a type C stream, think about the substrate. Is the stream mostly gravel, cobble, or boulders? If so, you're in a high gradient stream.
    b. If your site is a type C stream but has mostly sand or fine gravel substrate, your site is a low gradient stream.

When to use high gradient RHA field form	**When to use low gradient RHA field form**
- reference stream type is A or B	- reference stream type is E
- reference stream type is C characterized by riffle/pool bed features and a dominant substrate size of gravel or larger	- reference stream type is C with ripple/dune or riffle/pool bed features and dominant substrate size is fine gravel, sand or smaller

For example, our training week field sites are classified below:

Potash Brook:

Stream Type: C
Substrate: Gravel and larger (cobbles)
Classification: High Gradient

Allen Brook:

Stream Type: C
Substrate: Sand and silt
Classification: Low Gradient

Munroe Brook:
Stream Type: B
Classification: High Gradient

Indian Brook (by Essex High School):
Stream Type: C
Substrate: Sand and silt
Classification: Low Gradient

Indian Brook (by Mill Pond):
Stream Type: C
Substrate: Gravel and larger (cobbles)
Classification: High Gradient

Stream Reach - A section of stream having relatively uniform physical attributes, such as confinement, valley slope, sinuosity, dominant bed material, sediment regime, tributary influence, and bed form. Reach determinations do not take into account human disturbances, but rather are based on variables related to valley setting, stream morphology, and their inherent fluvial processes.

Stream Site Code - A code given to any stream being tested so it can be easily identified in a lab. Site Codes are designated by the VT EPSCoR Streams Project staff.

Stream Site General Assessment Data Sheet - A field sheet that is filled out annually for a stream site. It provides general information about the location, surrounding area, and watershed features (such as a nearby dam or bridge).

Stream Stage - The height (typically in feet) of water from an established point, typically from stream bottom to surface. Often maintained by the USGS and can be measured in a variety of ways.

Substrate - Represents the variety of material that is present in the stream, ranging from clay and gravel, to boulder and bedrock, and includes woody debris. Refer to the following table for sizes:

Clay/Silt/Sand	< 0.004-2.0	Fine, granular pieces of sediment measuring under 2.0 cm
Gravel	2.0-16	Small rocks measuring 16 cm or less
Course gravel	16-64	Larger (softball size or bigger) rocks that are smaller than 64 cm
Cobble	64-256	Chunks of rock that are not large enough to be boulders but are still noticeably sizeable.
Boulder	>256	Large Rock measuring above 256 cm, tall (relative to surrounding sediment) and above the bedrock.
Bedrock	---	Solid rock, providing a base layer over which there are other sediments.

T

Thalweg - A line connecting the lowest or deepest points of successive cross-sections along the course of a valley or river. This where the largest volume of water flows within the stream.

Ticks - Small, parasitic (blood sucking) organisms found locally. May transmit diseases including Lyme disease. Following time in the field, researchers should check for ticks on clothing and skin.

Total Suspended Solids (TSS) - The total amount of suspended solids in a sample of water; listed as a pollutant in the US Clean Water Act and is therefore measured as a water quality indication. Includes mostly sediment and algae.

Total Phosphorus (test) - A test that measures all phosphorus forms, such as orthophosphate, condensed phosphate, and organic phosphate, in a given sample of water.

Tributaries - A river or stream flowing into a larger river or lake.

Turbidity - The cloudiness of water caused by small particles.

U

USB Adaptor - An adapter that allows information to be directed between the iButton and a computer via a USB port.

USEPA (United States Environment Protection Agency) - A US federal agency that protects human health and the environment through enforcing regulations and laws passed by Congress.

USGS (United States Geological Survey) - A US federal agency that studies the landscape of the United States and its natural resources and hazards.

V

Valley Slope - While you don't need to calculate the actual valley slope, it is good to know how the calculation is done.

Example – Calculating Valley Slope

1140 ft	upstream elevation
- 1000 ft.	downstream elevation
140 ft	change in elevation

$$\frac{\text{difference in elevation (ft.)}}{\text{length of valley (ft.)}} = \frac{140}{4{,}000} = 0.035 \times 100 = 3.5\ \%\ \text{valley slope}$$

Velocity - In this context, the speed at which the water is flowing downstream.

W

Water Quality Assessment - An evaluation of the conditions of a body of water. Specifically, biologically and chemically assessing and analyzing components such as flow, pH, TSS and nutrients (TP/TN) of the body of water.

Water Quality Monitoring - Sampling and analysis of water constituents and conditions such as pollutants, natural components, dissolved chemicals, bacteria, etc. to know the base condition and target changes that may occur.

Water Quality Parameters - The general measurements of water that help scientists determine the health of a body of water.

Watershed - An area or ridge of land that separates waters flowing to different rivers, basins, or seas.

Wetted Width - The width of the water in a stream bank. This will always be different and will change depending on recent weather events.

Whirl-paks® - Small bags that captured specimen are placed in after being captured in the kick net. Following this step, add ethanol for preservation.

X

Y

Z