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Volunteer Stream Monitoring: A Methods Manual

Acknowledgments

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The West Virginia Save Our Streams Program has adopted the US Environmental Protection Agency's (US EPA) **Volunteer Stream Monitoring**: **A Methods Manual** to provide introductory information regarding various concepts in stream ecology and to help volunteer's better understand the many facets of monitoring wadeable streams, rivers and wetlands. The procedures described throughout this manual are examples of a variety of techniques used by volunteer and state water quality programs throughout the Mid-Atlantic Region.

Note: Certain portions of this manual have been have been modified, for example; additional data sheets to help perform a basic visual assessment from the Izaak Walton league and others have been added to chapter three, and several WV Save Our Streams stream survey data sheets have been added to chapter four.

Refer to the program's standard operating procedure manuals for descriptions of the tasks necessary to complete a survey. These are available for downloading from the WV Save Our Streams website.

Go to US EPA's website at (http://www.epa.gov/owow/monitoring/volunteer/stream/) to review this manual on-line. Limited printed quantities are available upon request; contact the WV Save Our Streams program coordinator by E-mail for details.

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Table of contents

As part of its commitment to volunteer monitoring, the U.S. Environmental Protection Agency (EPA) has worked since 1990 to develop a series of guidance manuals for volunteer programs. *Volunteer Stream Monitoring: A Methods Manual*, the third in the series, is designed as a companion document to *Volunteer Water Monitoring: A Guide for State Managers*. The guide describes the role of volunteer monitoring in state programs and discusses how managers can best organize, implement, and maintain volunteer programs. This document builds on the concepts discussed in the *Guide for State Managers* and applies them directly to streams and rivers.

Streams and rivers are monitored by more volunteer programs than any other waterbody type. According to the fourth edition of the *National Directory of Volunteer Environmental Monitoring Programs* (January 1994), three-quarters of the more than 500 programs listed conduct some sort of stream assessment as part, or all, of their monitoring project.

As the interest in monitoring streams grows, so too does the desire of groups to apply an integrated approach to the design and implementation of programs. More and more, volunteer monitors are interested in taking a combination of physical, chemical, and biological measurements and are beginning to understand how land uses in a watershed influence the health of its waterways. This document includes sections on conducting in-stream physical, chemical, and biological assessments as well as land-use or watershed assessments.

The chemical and physical measurements described in this document can be applied to rivers or streams of any size. However, the biological components (macroinvertebrates and habitat) should be applied only to "wadable" streams (i.e., where streams are small in width and relatively shallow in depth, and where both banks are clearly visible).

The purpose of this manual is not to mandate new methods or override methods currently being used by volunteer monitoring groups. Instead, it is intended to serve as a tool for program managers who want to launch a new stream monitoring program *or* enhance an existing program. *Volunteer Stream Monitoring* presents methods that have been adapted from those used successfully by existing volunteer programs.

Further, it would be impossible to provide monitoring methods that are uniformly applicable to all stream watersheds or all volunteer programs throughout the Nation. Factors such as geographic region, program goals and objectives, and program resources will all influence the specific methods used by each group. This manual therefore urges volunteer program coordinators to work hand-in-hand with state and local water quality professionals or other potential data users in developing and implementing a volunteer monitoring program. Through this partnership, volunteer programs gain improved credibility and access to professional expertise and data; agencies gain credible data that can be used in water quality planning. Bridges between citizens and water resource managers are also the foundation for an active, educated, articulate, and effective constituency of environmental stewards. This foundation is an essential component in the management and preservation of our water resources.

EPA has developed two other methods manuals in this series. *Volunteer Lake Monitoring: A Methods Manual* was published in December 1991. *Volunteer Estuary Monitoring: A Methods Manual* was published in December 1993. To obtain any or all of these documents, contact:

U.S. Environmental Protection Agency
Office of Wetlands, Oceans, and Watersheds
Volunteer Monitoring (4503F)
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1.1 Manual Organization

Volunteer Stream Monitoring: A Methods Manual is organized into six chapters. All chapters include references for further reading.

Chapter One: Introduction

The first chapter introduces the manual and outlines its organization.

Chapter Two: Elements of a Stream Study

Chapter 2 introduces the concept of the stream environment and presents information on the leading sources of pollution affecting streams in the United States. It then discusses in some detail 10 questions volunteer program coordinators must answer in designing a stream study, from knowing why monitoring is taking place to determining how the program will ensure the data collected are credible. The chapter includes a highlight on training volunteer monitors. The chapter concludes with safety and equipment considerations.

Chapter Three: Watershed Survey Methods

This chapter describes how to conduct a watershed survey (also known as a watershed inventory or visual survey), which can serve as a useful first step in developing a stream monitoring program. It provides hints on conducting a background investigation of a watershed and outlines steps for visually assessing the stream and its surrounding land uses.

Chapter Four: Macroinvertebrates and Habitat

In this chapter, three increasingly complex methods of monitoring the biology of streams are presented. The first is a simple stream survey that requires little training or preparation; the second is a widely used macroinvertebrate sampling and stream survey approach that yields a basic stream rating while monitors are still at the stream; and the third is a macroinvertebrate sampling and advanced habitat assessment approach that requires professional and laboratory support but can yield data on comparatively subtle stream impacts.

Chapter Five: Water Quality and Physical Conditions

Chapter 5 summarizes techniques for monitoring 10 different constituents of water: dissolved oxygen/biochemical oxygen demand, temperature, pH, turbidity, phosphorus, nitrates, total solids, conductivity, total alkalinity, and fecal bacteria. The chapter begins with a discussion on preparing sampling containers, highlights basic steps for collecting samples, and discusses taking stream flow measurements. This chapter discusses why each parameter is important, outlines sampling and equipment considerations, and provides instructions on sampling techniques.

Chapter Six: Managing and Presenting Monitoring Data

Chapter 6 outlines basic principles of data management, with an emphasis on proper quality assurance/quality control procedures. Spreadsheets, databases, and mapping software are discussed, as are basic approaches to presenting volunteer data to different audiences. These approaches include simple graphs, summary statistics, and maps. Lastly, the chapter briefly discusses ideas for distributing monitoring results to the public.

Appendices

- Appendix A provides a glossary of terms used in this manual.
- Appendix B lists a number of scientific supply houses where monitoring and analytical equipment can be purchased.
- Appendix C discusses how to determine the latitude and longitude of monitoring locations.

References and Further Reading

Ely, E. 1994. A Profile of Volunteer Monitoring. *Volunteer Monitor.* 6(1):4.

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USEPA. 1996. *The Volunteer Monitor's Guide To Quality Assurance Project Plans.* EPA 841-B-96-003. September. Office of Wetlands, Oceans, and Watersheds, 4503F, Washington, DC 20460.

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USEPA. 1991. *Volunteer Lake Monitoring: A Methods Manual*, EPA 440/4-91-002, December. Office of Wetlands, Oceans, and Watersheds, 4503F, Washington, DC 20460.

USEPA. 1990. *Volunteer Water Monitoring: A Guide for State Managers*, EPA 440/4-90-010, August. Office of Wetlands, Oceans, and Watersheds, 4503F, Washington, DC 20460.

This chapter is divided into three sections. The first section provides a review of basic concepts concerning watersheds, the water cycle, stream habitat, and water quality. This background information is essential for designing a stream monitoring program that provides useful data.

Section 2.2 presents the 10 critical questions that should be answered by program planners. These include: Why is monitoring taking place? Who will use the monitoring data? and What parameters or conditions will be monitored? The last section discusses the importance of safety in the field and laboratory.

2.1 Basic Concepts

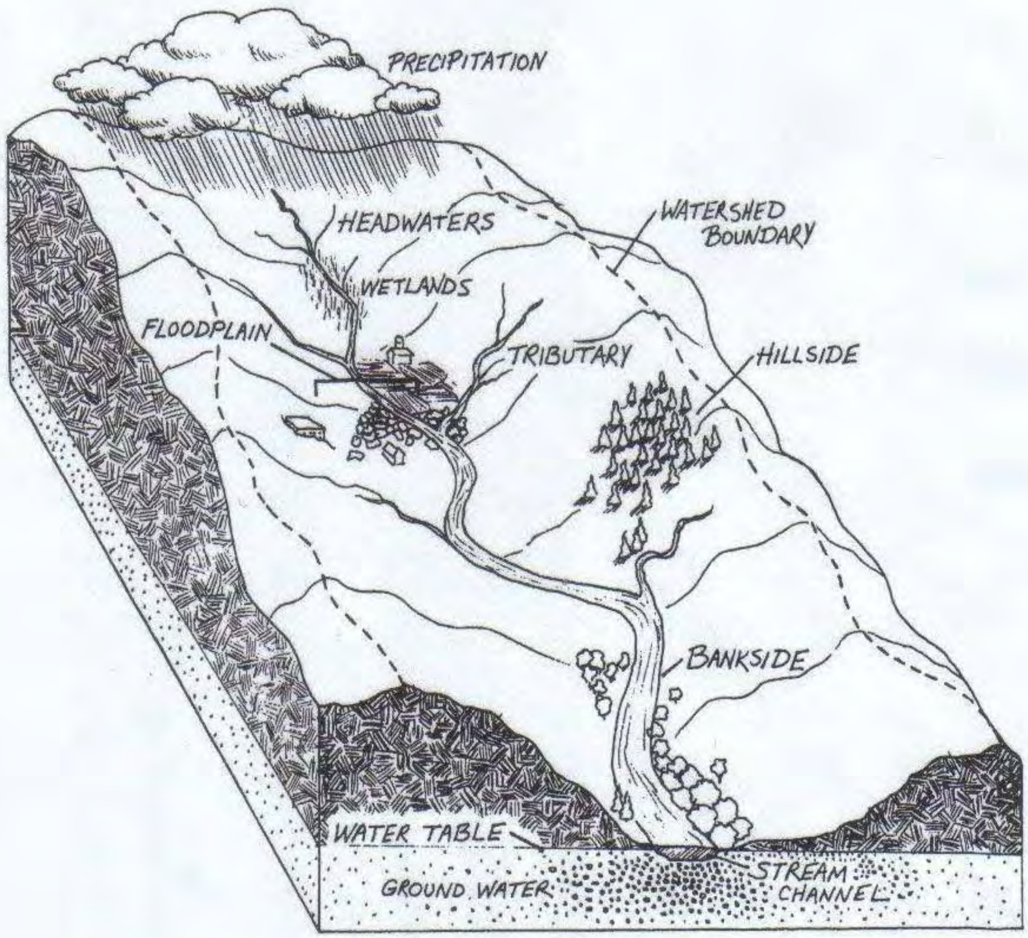
Watersheds

A watershed is the area of land from which runoff (from rain, snow, and springs) drains to a stream, river, lake, or other body of water (Fig. 2.1). Its boundaries can be identified by locating the highest points of lands around the waterbody. Streams and rivers function as the "arteries" of the watershed. They drain water from the land

Figure 2.1

Cross section of a watershed

Volunteers should get to know the watersheds of their study streams.



as they flow from higher to lower elevations.

A watershed can be as small or as large as you care to define it. This is because several watersheds of small streams usually exist within the watershed of a larger river. The watershed of the Mississippi River, for example, is about 1.2 million square miles and contains thousands of smaller watersheds, each defined by a tributary stream that eventually drains into a larger river like the Ohio River or Missouri River and to the Mississippi itself.

The River System

As streams flow downhill and meet other streams in the watershed, a branching network is formed (Fig. 2.2). When observed from the air this network resembles a tree. The trunk of the tree is represented by the largest river that flows into the ocean or large lake. The "tip-most" branches are the headwater streams. This network of flowing water from the headwater streams to the mouth of the largest river is called the river system.

Water resource professionals have developed a simple method of categorizing the streams in the river system. Streams that have no tributaries flowing into them are called first-order streams. Streams that receive only first-order streams are called second-order streams. When two second-order streams meet, the combined flow becomes a third-order stream, and so on.

The Water Cycle

The water cycle is the movement of water through the environment (Fig. 2.3). It is through this movement that water in the river system is replenished. When precipitation falls to earth in a natural (undeveloped) watershed in the mid-Atlantic states, for example, about 40 percent will be returned to the atmosphere by evaporation or transpiration (loss of water vapor by plants). About 50 percent will percolate

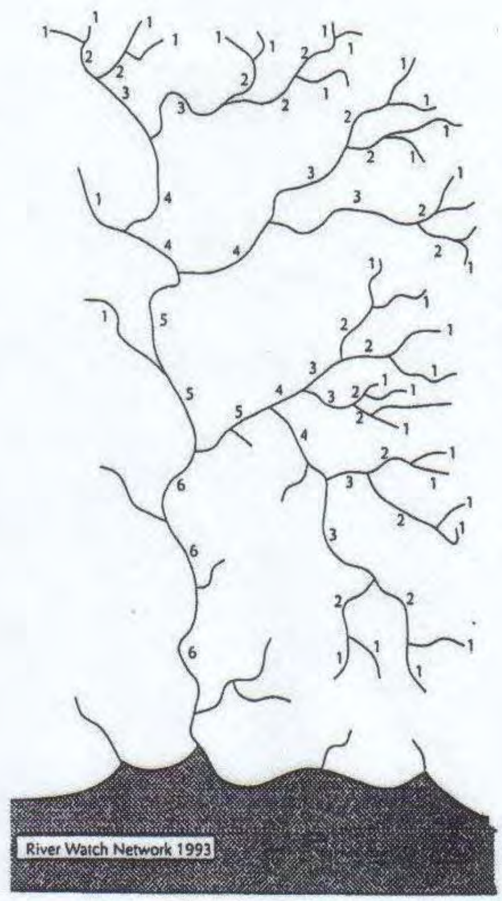


Figure 2.2

A representation of a river network with stream order marked

into the soil. The remaining 10 percent of the precipitation moves across the land as runoff and drains into streams, wetlands, and other bodies of water (Fig. 2.4, left panel).

The water that soaks into the ground is important for maintaining streamflow in the river network during dry weather. Percolating water slowly moves downward through the soil until it drains into an area where all the pores and cracks in the rock are saturated with water. The top of this zone is known as the water table.

Water in this saturated zone moves laterally, following the laws of gravity and/ or water pressure from above. If the path of this moving ground water intercepts a

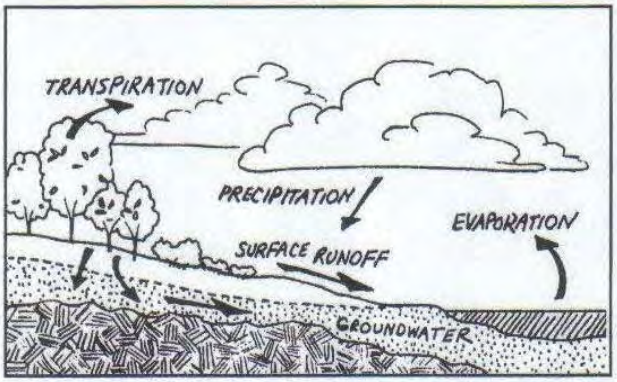


Figure 2.3

The water cycle

Water moving through the water cycle replenishes streams in the watershed.

stream channel, the ground water is discharged into the stream as a spring. The combination of ground water discharges to a stream is defined as its baseflow. At times when there is no surface runoff, the entire flow of a stream might actually be baseflow from ground water (Fig. 2.5).

Some streams, on the other hand, constantly lose water to the ground water. This occurs when the water table is below the bottom of the stream channel. Stream water percolates down through the soil until it reaches the zone of saturation. Other streams alternate between losing and gaining water as the water table moves up and down according to the seasonal conditions or pumpage by area wells.

The interactions between the watershed, soils, and water cycle define the natural water flow (hydrology) of any particular stream. Most significant is the fact that developed land is more impervious than natural land. Instead of percolating into the ground, rain hits the hard surfaces of buildings, pavement, and compacted ground and runs off into a storm drain or other artificial structure designed to move water quickly away from developed areas and into a natural watercourse. These conditions typically change the fate of precipitation in the water cycle (See Fig. 2.4, right panel). For example:

- Less precipitation is evaporated back to the atmosphere. (Water is transported rapidly away via storm drains and is not allowed to stand in pools.)
- Less precipitation is transpired back to the atmosphere from plants. (Natural vegetation is replaced by buildings, pavement, etc.)
- Less precipitation percolates through the soil to become ground water. (This can result in a lower water table and can affect baseflow.)
- More surface runoff is generated and transported to streams. (Streamflow becomes more intense during and immediately after storms.)

Chapter 3, Watershed Survey Methods, is designed to help volunteers learn about their watershed. Using the watershed survey approach, they will become familiar with their watershed's boundaries, its hydrologic features, and the human uses of land and water that might be affecting the quality of the streams within it.

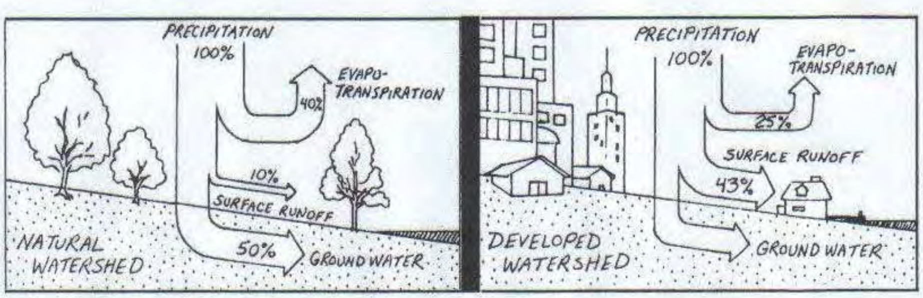
The Living Stream Environment

A healthy stream is a busy place. Wildlife and birds find shelter and food near and in its waters. Vegetation grows

Figure 2.4

The fate of precipitation in undeveloped vs. developed watersheds

Surface runoff increases and ground water recharge decreases as watersheds become developed.



along its banks, shading the stream, slowing its flow in rainstorms, filtering pollutants before they enter the stream, and sheltering animals. Within the stream itself are fish and a myriad of insects and other tiny creatures with very particular needs. For example, stream dwellers need dissolved oxygen to breathe; rocks, overhanging tree limbs, logs, and roots for shelter; vegetation and other tiny animals to eat; and special places to breed and hatch their young. For many of these activities, they might also need water of specific velocity, depth, and temperature.

Human activities shape and alter many of these stream characteristics. We dam up, straighten, divert, dredge, dewater, and discharge to streams. We build roads, parking lots, homes, offices, golf courses, and factories in the watershed. We farm, mine, cut down trees, and graze our livestock in and along stream edges. We also swim, fish, and canoe in the streams themselves.

These activities can dramatically affect the many components of the living stream environment (Fig. 2.6). These components include:

1. The *adjacent watershed* includes the higher ground that captures runoff and drains to the stream. For purposes of this manual, the adjacent watershed is defined as land extending from the riparian zone to 1/4 mile from the stream.
2. The *floodplain* is the low area of land that surrounds a stream and holds the overflow of water during a flood.
3. The *riparian zone* is the area of natural vegetation extending outward from the edge of the stream bank. The riparian zone is a buffer to pollutants entering a stream from runoff, controls erosion, and provides stream habitat and nutrient input into the stream. A healthy stream system generally has a healthy riparian zone. Reductions and impairment of riparian zones occur when roads, parking lots, fields, lawns, and other artificially cultivated areas, bare soil, rocks, or buildings are near the stream bank.
4. The *stream bank* includes both an upper bank and a lower bank. The lower bank normally begins at the normal water line and runs to the bottom of the stream. The upper bank extends from the break in the normal slope of the surrounding land to the normal high water line.
5. The *streamside cover* includes any overhanging vegetation that offers protection and shading for the stream and its aquatic inhabitants.
6. *Stream vegetation* includes emergent, submergent, and floating plants. Emergent plants include plants with true stems, roots, and leaves with most of their vegetative parts above the water. Submergent plants also include some of the same types of plants, but they are completely immersed in water. Floating

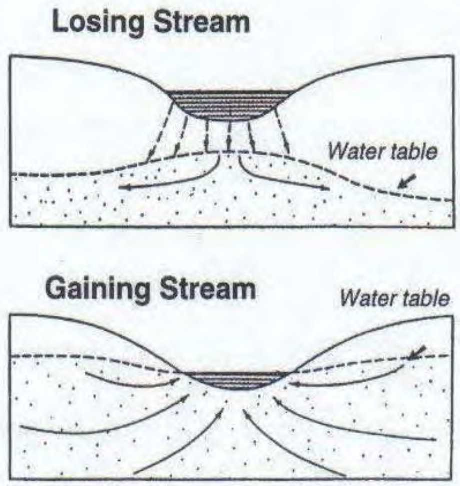


Figure 2.5

Streams losing and gaining water

The position of the water table sometimes plays a role in determining the amount of streamflow.

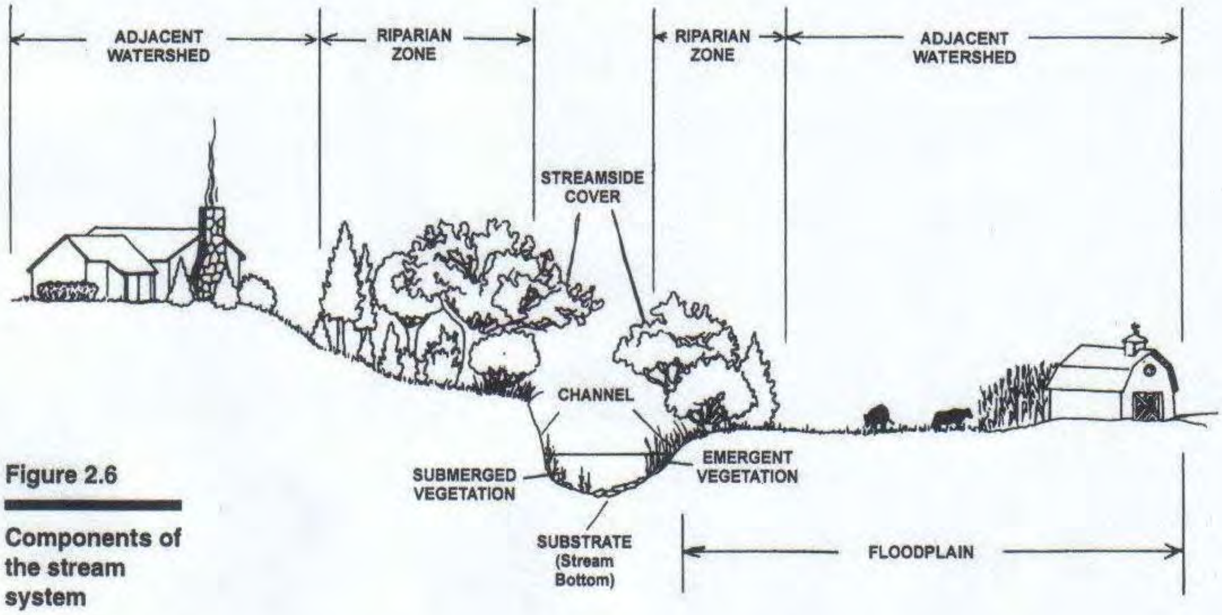


Figure 2.6

Components of the stream system

Volunteers should be aware that the surrounding land affects stream habitat.

plants (e.g., duckweed, algae mats) are detached from any substrate and are therefore drifting in the water.

7. The *channel* of the streambed is the zone of the stream cross section that is usually submerged and totally aquatic.
8. *Pools* are distinct habitats within the stream where the velocity of the water is reduced and the depth of the water is greater than that of most other stream areas. A pool usually an has soft bottom sediments.
9. *Riffles* are shallow, turbulent, but swiftly flowing stretches of water that flow over partially or totally submerged rocks.
10. *Runs* or *glides* are sections of the stream with a relatively low velocity that flow gently and smoothly with little or no turbulence at the surface of the water.
11. The *substrate* is the material that makes up the streambed, such as clay, cobbles, or boulders.

Whether streams are active, fast-moving, shady, cold, and clear or deep, slow-moving, muddy, and warm —or something in between—they are shaped by the land they flow through and by what we do to that land. For example, vegetation in the stream's riparian zone protects and serves as a buffer for the stream's streamside cover, which in turn shades and enriches (by dropping leaves and other organic material) the water in the stream channel.

Furthermore, the riparian zone helps maintain the stability of the stream bank by binding soils through root systems and helps control erosion and prevent excessive siltation of the stream's substrate. If human activities begin to degrade the stream's riparian zone, each of these stream components—and the aquatic insects, fish, and plants that inhabit them—also begins to degrade.

Chapter 4 includes methods that volunteers can use to assess the stream's living environment—specifically, the insects that live in the stream and the physical components of the stream (the habitats) that support them.

Water Quality

The water in a stream is always moving and mixing, both from top to bottom and from one side of the stream to the other. Pollutants that enter the stream travel some distance before they are thoroughly mixed throughout the flow. For example, water upstream of a pipe discharging wastewater might be clean. At the discharge site and immediately downstream, the water might be extremely degraded. Further downstream, in the recovery zone, overall quality might improve as pollutants are diluted with more water. Far downstream the stream as a whole might be relatively clean again. Unfortunately, most streams with one source of pollution often are affected by many others as well.

Pollution is broadly divided into two classes according to its source. Point source pollution comes from a clearly identifiable point such as a pipe which discharges directly into a waterbody. Examples of point sources include factories, wastewater treatment plants, and illegal straight pipes from homes and boats.

Nonpoint source pollution comes from surface water runoff. It originates from a broad area and thus can be difficult to identify. Examples of nonpoint sources include agricultural runoff, mine drainage, construction site runoff, and runoff from city streets and parking lots.

Nationally, the pollutants most often found in the stream environment are not toxic substances like lead, mercury, or oil and grease. More impacts are caused by sediments and silt from eroded land and nutrients such as the nitrogen and phosphorus found in fertilizers, detergents, and sewage treatment plant discharges. Other leading pollutants include pathogens such as bacteria, pesticides, and organic enrichment that leads to low levels of dissolved oxygen. Common sources of pollution to streams include:

- *Agricultural activities* such as crop production, cattle grazing, and maintaining livestock in holding areas or feedlots. These contribute pollutants such as sediments, nutrients, pesticides, herbicides, pathogens, and organic enrichment.
- *Municipal dischargers* such as sewage treatment plants which contribute nutrients, pathogens, organic enrichment, and toxicants.
- *Urban runoff* from city streets, parking lots, sidewalks, storm sewers, lawns, golf courses, and building sites. Common pollutants include sediments, nutrients, oxygen-demanding substances, road salts, heavy metals, petroleum products, and pathogens.

Other commonly reported sources of pollutants are mining, industrial dischargers (factories), forestry activities, and modifications to stream habitat and hydrology.

Chapter 5 describes methods volunteers can use to monitor water quality and detect pollutants from these sources.

2.2 Designing the Stream Study

Before beginning a stream monitoring study, volunteer program officials should develop a design or plan that answers the 10 basic questions listed below. Without answers to these questions, the monitoring program might well end up collecting data that do not meet anyone's needs.

Answering these 10 questions is not easy. A planning committee composed of the program coordinator, key volunteers, scientific advisors, program supporters, and data users should resolve these questions well before the project gets under way. Naturally, the committee should also address other planning questions less directly related to monitoring design, such as how to recruit volunteers and how to secure funding for the project. Answers will likely change as the program matures. For example, program coordinators might find that a method is not producing data of high enough quality, data collection is too labor-intensive or expensive, or additional parameters need to be monitored.

1. Why is the monitoring taking place?

Typical reasons for initiating a volunteer monitoring project include:

- Developing baseline characterization data
- Documenting water quality changes over time
- Screening for potential water quality problems
- Determining whether waters are safe for swimming
- Providing a scientific basis for making decisions on the management of a stream or watershed
- Determining the impact of a municipal sewage treatment facility, industrial facility, or land use activity such as forestry or farming
- Educating the local community or stream users to encourage pollution prevention and environmental stewardship
- Showing public officials that local citizens care about the condition and management of their water resources

Of course, an individual program might be monitoring for a number of reasons. However, it is important to identify one or two top reasons and develop the program based on those objectives.

2. Who will use the monitoring data?

Knowing your data users is essential to the program development process. Potential data users might include:

- State, county, or local water quality analysts
- The volunteers themselves
- Fisheries biologists
- Universities
- Schoolteachers
- Environmental organizations
- Parks and recreation staff
- Local planning and zoning agencies
- State environmental agencies
- State and local health departments
- Soil and water conservation districts
- Federal agencies such as the U.S. Geological Survey or U.S. Environmental Protection Agency

Each of these users will have different data requirements. Some users, such as

Type	Approach	Applications*
Physical condition	Watershed survey	Determine land use patterns; determine presence of current and historical pollution sources; identify gross pollution problems; identify water uses, users, diversions, and stream obstructions
	Habitat assessment	Determine and isolate impacts of pollution sources, particularly land use activities; interpret biological data; screen for impairments
Biological condition	Macroinvertebrate sampling	Screen for impairment; identify impacts of pollution and pollution control activities; determine the severity of the pollution problem and rank stream sites; identify water quality trends; determine support of designated aquatic life uses
Chemical condition	Water quality sampling	Screen for impairment; identify specific pollutants of concern; identify water quality trends; determine support of designated contact recreation uses; identify potential pollution sources

* Beyond education and promoting stewardship

Table 2.1

Some types of monitoring approaches and their application

government analysts and planning/zoning agencies, will have more stringent requirements than others and will require higher levels of quality assurance. As the volunteer monitoring project is being designed, program coordinators should contact as many potential information users as possible to determine their data needs. It is important to have at least one user committed to receiving and using the data. In some cases that user might be the monitoring group itself.

3. How will the data be used?

The range of uses of volunteer data is limited only by the imagination. Volunteer data could be used, for example, to influence local planning decisions about where to site a sewage treatment facility or to publicize a water quality problem and seek community solutions. Collected data could also be used to educate primary school children about the importance of water resources. Other data uses include the support of:

- Local zoning requirements
- A stream protection study
- State preparation of water quality assessments
- Screening waters for potential problems
- The setting of statewide priorities for pollution control

Each data use potentially has different data requirements. Knowing the ultimate uses of the collected volunteer data will help determine the right kind of data to collect and the level of effort required to collect, analyze, store, and report them.

4. What parameters or conditions will be monitored?

Determining what to monitor will depend on the needs of the data users, the intended use of the data, and the resources of the volunteer program. If the program's goal is to determine whether a creek is suitable for swimming, for example, a human-health-related parameter such as fecal coliform bacteria should be monitored. If the objective is to characterize the ability of a stream to support sport fish, volunteers should examine stream habitat characteristics, the aquatic insect community, and water quality parameters such as dissolved oxygen and temperature. Alternatively, if a program seeks to provide baseline data useful to state water quality or natural resource agencies, program designers should consult those agencies to determine which parameters they consider of greatest value.

Money for test kits or meters, available laboratory facilities, help from state or university advisors, and the abilities and desires of volunteers will also clearly have an impact on the choice of parameters to be monitored. For characterization studies, EPA usually recommends an approach that integrates physical, chemical, and biological parameters.

5. How good does the monitoring data need to be?

Some uses require high-quality data. For example, high-quality data are usually needed to prove compliance with environmental regulations, assess pollution impacts, or make land use planning decisions. In other cases the quality of the data is secondary to the actual process of collecting it. This is often the case for monitoring programs that focus on the overall educational aspects of stream monitoring.

Data quality is measured in five ways—accuracy, precision, completeness, representativeness, and comparability (see box—Data Quality Terms).

6. What methods should be used?

The methods adopted by a volunteer program depend primarily on how the data will be used and what kind of data quality is needed. There are, of course, many sampling considerations including:

- How samples will be collected (e.g., using grab samples or measuring directly with a meter)
- What sampling equipment will be used (e.g., disposable Whirl-pak bags, glass bottles, 500-micron mesh size kick net, etc.)
- What equipment preparation methods are necessary (such as container sterilization or meter calibration)
- What protocols will be followed (such as the Winkler method for dissolved oxygen, intensive stream bioassessment approach for habitat and benthic macroinvertebrates, etc.)

Analytical questions must also be addressed such as:

- Will volunteers return to a lab for macroinvertebrate identification or dissolved oxygen titration procedures or conduct them in the field?
- Will a color wheel provide nitrate data of needed quality, or is a more sophisticated approach needed?
- Should visual observation and habitat assessment approaches be combined with turbidity measures to best determine the impact of construction sites?

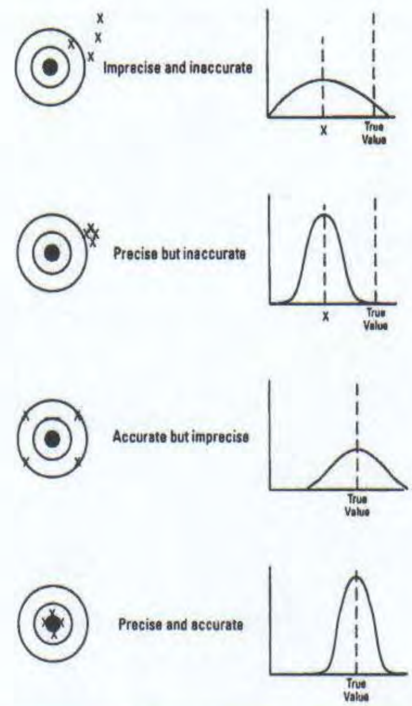
While sophisticated methods usually yield more accurate and precise data (if properly carried out), they are also more costly and time-consuming. This extra effort and expense might be worthwhile if the goal of the program is to produce high-quality data. Programs with an educational focus, however, can often use less sensitive equipment and less sophisticated methods to meet their goals.

7. Where are the monitoring sites?

Sites might be chosen for any number of reasons such as accessibility, proximity to volunteers' homes, value to potential

Data Quality Terms

- ***Accuracy*** is the degree of agreement between the sampling result and the true value of the parameter or condition being measured. Accuracy is most affected by the equipment and the procedure used to measure the parameter.
- ***Precision***, on the other hand, refers to how well you are able to reproduce the result on the same sample, regardless of accuracy. Human error in sampling techniques plays an important role in estimating precision.
- ***Representativeness*** is the degree to which collected data actually represent the stream condition being monitored. It is most affected by site location.
- ***Completeness*** is a measure of the amount of valid data actually obtained vs. the amount expected to be obtained as specified in the original sampling design. It is usually expressed as a percentage. For example, if 100 samples were scheduled but volunteers sampled only 90 times due to bad weather or broken equipment, the completeness record would be 90 percent.
- ***Comparability*** represents how well data from one stream or stream site can be compared to data from another. Most managers will compare sites as part of a statewide or regional report on the volunteer monitoring program; therefore, sampling methods should be the same from site to site.



users such as state agencies, or location in problem areas. If the volunteer program is providing baseline data to characterize a stream or screen for problems, it might wish to monitor a number of sites representing a range of conditions in the stream watershed (e.g., an upstream "pristine" area, above and below towns and cities, in agricultural areas and parks, etc.). For more specific purposes, such as determining whether a stream is safe to swim in, it might only be necessary to sample selected swimming areas. To determine whether a particular land use activity or potential source of pollution is, in fact, having an impact, it might be best to monitor upstream and downstream of the area where the source is suspected. To determine the effectiveness of runoff control measures, a paired watershed approach might be best (e.g., sampling two similar small watersheds, one with controls in place and one without controls).

A program manager might also select one or more sites near professionally monitored sites in order to compare the quality of volunteer-generated data against professional data. It might also be helpful to locate some sites near U.S. Geological Survey gauging stations, which can provide useful data on streamflow. Certainly, for any volunteer program, safety and accessibility (both legal and physical) will be important in determining site location. No matter how sampling sites are chosen, most monitoring programs will need to maintain the same sites over time and identify them clearly in their monitoring program design.

When selecting monitoring sites, ask the following questions. Based on the answers, you may need to eliminate some sites or select alternative locations that meet your criteria:

- Are other groups (local, state, federal agencies; other volunteer groups;

schools or colleges) already monitoring this site?

- Can you identify the site on a map and on the ground?
- Is the site representative of the watershed?
- Does the site have water in it during the times of year that monitoring will take place?
- Is there safe, convenient access to the site (including adequate parking) and a way to safely sample a flowing section of the stream? Is there access all year long?
- Can you acquire landowner permission?
- Can you perform all the monitoring activities and tests that are planned at this site?
- Is the site far enough downstream of drains or tributaries? Is the site near tributary inflows, dams, bridges, or other structures that may affect the results?
- Have you selected enough sites for the study you want to do?

Once you have selected the monitoring sites, you should be able to identify them by latitude and longitude. This location information is critical if your data will potentially be used in Geographical Information Systems (GIS) or in sophisticated data management systems (See Appendix C).

8. When will monitoring occur?

A program should specify:

- What time of day is best for sampling. (Temperature and dissolved oxygen, for example, can fluctuate naturally as the sun rises and aquatic plants release oxygen.)
- What time of year is best for sampling. (For example, there is no point in sampling fecal coliform bacteria at swimming beaches in the winter, when no one is swimming, or sampling intermittent streams at the height of summer, when because of dry conditions the streams hold little water.)
- How frequently should monitoring take place? (It is possible, for example, to conduct too many biological assessments of a stream and thereby deplete the stream's aquatic community. A program designed to determine whether polluted runoff is a problem would do well to monitor after storms and heavy rainfalls.)

In general, monthly chemical sampling and twice-yearly biological sampling are considered adequate to identify water quality changes over time. Biological sampling should be conducted at the same time each year because natural variations in aquatic insect population and streamside vegetation occur as seasons change. Monitoring at the same time of day and at regular intervals (e.g., at 2:00 p.m. every 30 days) helps ensure comparability of data over time.

9. How will monitoring data be managed and presented?

The volunteer program coordinator should have a clear plan for dealing with the data collected each year. Field and lab data sheets should be checked for completeness, data should be screened for outliers, and a database should be developed or adapted to store and manipulate the data. The elements of such a database should be clearly explained in order to allow users to interpret the data accurately and with confidence.

Training Volunteer Monitors

Training should be an essential component of any volunteer stream monitoring project. When volunteers are properly trained in the goals of the volunteer project and its sampling and analytical methods, they:

- Produce higher quality, more credible data.
- Better understand their role in protecting water quality.
- Are more motivated to continue monitoring.
- Save program manager time and effort by becoming better monitors who require less supervision.
- Feel more like part of a dedicated team.

Some of the key elements to consider in developing a training program for volunteers include the following:

1. *Plan ahead.* When you are in the early stages of developing your training program, decide who will do the training, when training will occur, where it will be held, what equipment and handouts volunteers will receive, and what, in they end, they will learn. Plan on at least one initial training session at the start of the sampling season and a quality control session somewhat into the season (to see if volunteers are using the right methods, and to answer questions). If volunteers will be sampling many different chemical parameters or will be conducting intensive biological monitoring, you should probably schedule two initial training sessions—one to introduce volunteers to the program, and the other to cover sampling and analytical methods in detail. You might also want to plan a post-season session that encourages volunteers to air problems, exchange information, and make suggestions for the coming year. Make sure the program planning committee agrees to the training plan.

2. *Put it in writing.* Once you've made these decisions, write them all down. Note the training specifics in the program's quality assurance project plan. It might also help to develop a "job description" for the volunteers that lists the tasks they will perform in the field and lab, and that identifies the obligations to which they will be held and the schedule they will follow. Hand this out at the first training session. Volunteers should leave the session knowing what is expected of them. If they decide not to join after all because the tasks are too onerous, it is better for you to find out after the first session than later in the sampling year.

3. *Be prepared.* Nothing will discourage volunteers more than an ill-planned, chaotic initial training session. The elements of a successful initial training session include:

   - Enthusiastic, knowledgeable trainers
   - Short presentations that encourage audience participation and don't strain attention spans
   - A low ratio of trainers to trainees
   - Presentations that include why the monitoring is needed, what the program hopes to accomplish, and what will be done with the data
   - An agenda that is followed (especially start and finish times)
   - Good acoustics, clear voices, and interesting audiovisual aids
   - Opportunities for all trainees to handle equipment, view demonstrations of sampling protocols, and practice sampling
   - Instruction on safety considerations
   - Refreshments and opportunities for trainees to meet one another, socialize, and have fun
   - Time for questions and answers.

4. *Conduct quality control checks.* After your initial training session(s), schedule opportunities to "check up" on how your volunteers are performing. The purpose of these quality control checks is to ensure that all volunteers are monitoring using proper and consistent protocols, and to emphasize the importance of quality control measures. Some time into the sampling season, observe how volunteers are sampling, analyzing their samples, identifying macroinvertebrates, and recording their results. Either observe volunteers in small groups at their monitoring sites or bring them to a central location for an organized quality control session. If your program is involved in chemical monitoring, you might want all volunteers to analyze the same water sample using their own equipment, or hold a lab exercise in which volunteers read and record results from equipment and kits that have already been set up. For a biological monitoring program, have trainers or seasoned volunteers observe sampling methods in the field and provide preserved samples of macroinvertebrates for volunteers to identify. Reserve time to answer questions, talk about initial findings, and have some fun.

5. *Review the effectiveness of your training program.* At the end of each training session, encourage volunteers to fill out a training evaluation form. This form should help you assess the effectiveness of individual trainers and their styles, the handouts and audiovisual aids, the general atmosphere of the training session, and what the volunteers liked most and least about the session. Use the results of the evaluation to revise training protocols as needed to best meet program and volunteer needs.

Put It in Writing

When you and the volunteer program planning committee have answered the ten project design questions to everyone's satisfaction, your next critical step is to put it all in writing. The written plan, including sampling and analytical methods, sites, parameters, project goals, and data quality considerations, is your bible. With a written plan you:

- Document the particulars of your program for your data users
- Educate newcomers to the program
- Ensure that newcomers will use the same methods as those who came before them
- Keep an historical record for future program leaders, volunteers, and data users

Your written plan may simply consist of a study design and standard operating procedures such as a monitoring and lab methods manual. You may, however, prefer to develop a more comprehensive *quality assurance project plan.* The quality assurance project plan is a document that outlines the procedures you will use to ensure high quality data when conducting sample collection and analysis in your program.

By law, any water quality monitoring program that receives EPA funding is required to have an EPA-approved quality assurance project plan. Even if you don't receive EPA funding, you will find that preparing a written plan helps ensure that your data are used with confidence, now and in the future. (See *The Volunteer Monitor's Guide to Quality Assurance Project Plans* (EPA 841-B-96-003 September 1996) for more information.)

Program coordinators will also have to decide how they want to present data results, not only to the general public and to specific data users, but also to the volunteers themselves. Different levels of analysis might be needed for different audiences. A volunteer group collecting data for state or county use should consult with the appropriate agency before investing in computerized data management software because the agency could have specific needs or recommendations based on its own data management protocols.

10. How will the program ensure that data are credible?

Developing specific answers to questions 1-9 is the first step in ensuring that data are credible. Credible data meet specific needs and can be used with confidence for those needs. Other steps include:

- Properly training, testing, and retraining volunteers
- Evaluating the program's success after an initial pilot stage and making any necessary adjustments
- Assigning specific quality assurance tasks to qualified individuals in the program
- Documenting in a written plan all the steps taken to sample, analyze, store, manage, and present data

A written plan, known as a quality assurance project plan, can be elaborate or simple depending on the volunteer program's goals. Its essential feature, however, is that it documents how the data are to be generated. Without such knowledge, the data cannot be used with confidence. It is also important for educating future volunteers and data users about the program and the data. People might be analyzing the data 5 or 10 or more years later to study trends in stream quality. (Note: EPA requires that any monitoring program sponsored by EPA through grants, contracts, or other formal agreement must carry out a quality assurance/quality control program and develop a quality assurance project plan.)

2.3 Safety Considerations

One of the most critical considerations for a volunteer monitoring program is the safety of its volunteers. All volunteers should be trained in safety procedures and should carry with them a set of safety instructions and the phone number of their program coordinator or team leader. Safety precautions can never be overemphasized.

The following are some basic common sense safety rules. At the site:

- Always monitor with at least one partner. Teams of three or four people are best. Always let someone else know where you are, when you intend to return, and what to do if you don't come back at the appointed time.
- Develop a safety plan. Find out the location and telephone number of the nearest telephone and write it down. Locate the nearest medical center and write down directions on how to get between the center and your site(s) so that you can direct emergency personnel. Have each member of the sampling team complete a medical form that includes emergency contacts, insurance information, and pertinent health information such as allergies, diabetes, epilepsy, etc.
- Have a first aid kit handy (see box below). Know any important medical conditions of team members (e.g., heart conditions or allergic reactions to bee stings). It is best if at least one team member has first aid/CPR training.
- Listen to weather reports. Never go sampling if severe weather is predicted or if a storm occurs while at the site.
- Never wade in swift or high water. Do not monitor if the stream is at flood stage.
- If you drive, park in a safe location. Be sure your car doesn't pose a hazard to other drivers and that you don't block traffic.
- Put your wallet and keys in a safe place, such as a watertight bag you keep in a pouch strapped to your waist. Without proper precautions, wallet and keys might end up downstream.
- Never cross private property without the permission of the landowner. Better yet, sample only at public access points such as bridge or road crossings or public parks. Take along a card identifying you as a volunteer monitor.

First Aid Kit

The minimum first aid kit should contain the following items:

- Telephone numbers of emergency personnel such as the police and an ambulance service.
- Several band-aids for minor cuts.
- Antibacterial or alcohol wipes.
- First aid creme or ointment.
- Several gauze pads 3 or 4 inches square for deep wounds with excessive bleeding.
- Acetaminophen for relieving pain and reducing fever.
- A needle for removing splinters.
- A first aid manual which outlines diagnosis and treatment procedures.
- A single-edged razor blade for minor surgery, cutting tape to size, and shaving hairy spots before taping.
- A 2-inch roll of gauze bandage for large cuts.
- A triangular bandage for large wounds.
- A large compress bandage to hold dressings in place.
- A 3-inch wide elastic bandage for sprains and applying pressure to bleeding wounds.
- If a participant is sensitive to bee stings, include their doctor-prescribed antihistamine.

Be sure you have emergency telephone numbers and medical information with you at the field site for everyone participating in field work (including the leader) in case there is an emergency.

- Confirm that you are at the proper site location by checking maps, site descriptions, or directions.
- Watch for irate dogs, farm animals, wildlife (particularly snakes), and insects such as ticks, hornets, and wasps. Know what to do if you get bitten or stung.
- Watch for poison ivy, poison oak, sumac, and other types of vegetation in your area that can cause rashes and irritation.
- Never drink the water in a stream. Assume it is unsafe to drink, and bring your own water from home. After monitoring, wash your hands with antibacterial soap.
- Do not monitor if the stream is posted as unsafe for body contact. If the water appears to be severely polluted, contact your program coordinator.
- Do not walk on unstable stream banks. Disturbing these banks can accelerate erosion and might prove dangerous if a bank collapses. Disturb streamside vegetation as little as possible.
- Be very careful when walking in the stream itself. Rocky-bottom streams can be very slippery and can contain deep pools; muddy-bottom streams might also prove treacherous in areas where mud, silt, or sand have accumulated in sink holes. If you must cross the stream, use a walking stick to steady yourself and to probe for deep water or muck. Your partner(s) should wait on dry land ready to assist you if you fall. Do not attempt to cross streams that are swift and above the knee in depth. Wear waders and rubber gloves in streams suspected of having significant pollution problems.
- If you are sampling from a bridge, be wary of passing traffic. Never lean over bridge rails unless you are firmly anchored to the ground or the bridge with good hand/foot holds.
- **If at any time you feel uncomfortable about the condition of the stream or your surroundings, stop monitoring and leave the site at once. Your safety is more important than the data!**

When using chemicals:

- Know your equipment, sampling instructions, and procedures before going out into the field. Prepare labels and clean equipment before you get started.
- Keep all equipment and chemicals away from small children. Many of the chemicals used in monitoring are poisonous. Tape the phone number of the local poison control center to your sampling kit.
- Avoid contact between chemical reagents and skin, eye, nose, and mouth. Never use your fingers to stopper a sample bottle (e.g., when you are shaking a solution). Wear safety goggles when performing any chemical test or handling preservatives.
- Know chemical cleanup and disposal procedures. Wipe up all spills when they occur. Return all unused chemicals to your program coordinator for safe disposal. Close all containers tightly after use. Do not switch caps.
- Know how to use and store chemicals. Do not expose chemicals or equipment to temperature extremes or long-term direct sunshine.

2.4 Basic Equipment

Much of the equipment a volunteer will need is easily obtained from either hardware stores or scientific supply houses. Other equipment can be found around the house. In either case, the volunteer program should clearly specify the equipment its volunteers will need and where it should be obtained.

Listed below is some basic equipment appropriate for any volunteer field activity. Much of this equipment is optional but will enhance the volunteers' safety and effectiveness.

- Boots or waders; life jackets if you are sampling by boat
- Walking stick of known length for balance, probing, and measuring
- Bright-colored snag- and thorn-resistant clothes; long sleeves and pants are best
- Rubber gloves to guard against contamination
- Insect repellent/sunscreen
- Small first aid kit, flashlight, and extra batteries
- Whistle to summon help in emergencies
- Refreshments and drinking water
- Clipboard, preferably with plastic cover
- Several pencils
- Tape measure
- Thermometer
- Field data sheet
- Information sheet with safety instructions, site location information, and numbers to call in emergencies
- Camera and film, to document particular conditions

Specific equipment lists for the chemical and biological monitoring procedures included in the manual are provided in the relevant chapters.

References and Further Reading

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One of the most rewarding and least costly stream monitoring activities a volunteer program can conduct is the watershed survey. Some programs call it a windshield survey, a visual survey, or a watershed inventory. It is, in essence, a comprehensive survey of the geography, land and water uses, potential and actual pollution sources, and history of the stream and its watershed.

The watershed survey may be divided into two distinct parts:

- *A one-time background investigation of the stream and its watershed.* (To do this, volunteers research town and county records, maps, photos, news stories, industrial discharge records, and oral histories.)
- *A periodic visual assessment of the stream and its watershed.* (To do this, volunteers walk along the stream and drive through the watershed, noting key features.)

The watershed survey requires little in the way of training or equipment. Its chief uses include:

- Screening for pollution problems
- Identifying potential sources of pollution
- Identifying sites for monitoring
- Helping interpret biological and chemical information
- Giving volunteers and local residents a sense of the value of the stream or watershed
- Educating volunteers and the local community about potential pollution sources and the stressors affecting the stream and its watershed
- Providing a blueprint for possible community restoration efforts such as cleanups and tree plantings

To actually determine whether those stressors are, in fact, affecting the stream requires additional monitoring of chemical, physical, or biological conditions.

The watershed survey described in this chapter was developed from survey approaches used by programs such as Rhode Island Watershed Watch, Maryland Save Our Streams, the Delaware Department of Natural Resources and Environmental Control, and Washington's Adopt-A-Stream Foundation. References are provided at the end of this chapter for further information on watershed surveys.

3.1 How to Conduct a Watershed Survey

The Background Investigation

Researching the stream is generally a one-time activity that should yield valuable information about the cultural and natural history of the stream and the uses of the land surrounding it. This information will prove helpful in orienting new volunteers to the purpose of the monitoring program, in building a sense of the importance of the stream and its role in the watershed, and in identifying land use activities in the watershed with a potential to affect the quality of the stream. The program might choose to monitor these areas and activities more intensively in the future.

The background investigation is essentially a "detective investigation" for information on the stream and includes the following steps:

Task 1 **Determine what you want to know about your stream**

Before beginning the background investigation, establish what it is you want to know about the stream you are surveying. Types of information include:

- Location of the stream's headwaters, its length, where it flows, and where it empties
- Name and boundaries of the watershed it occupies, the population in the watershed, and the communities through which it flows
- Roles of various jurisdictions in managing the stream and watershed
- Percentage of watershed land area in each town or jurisdiction
- Land uses in the stream's watershed
- Industries and others that discharge to the stream
- Current uses of the stream (such as fishing, swimming, drinking water supply, irrigation)
- Historical land uses
- History of the stream

Any or all of these types of information should prove valuable to the monitoring program. You might also uncover other important information in the process. At a minimum, the investigation should yield information on the size of the stream, watershed boundaries, and general land use in the area. By establishing categories of information to investigate, program coordinators can assign volunteers to specific activities and end up with a complete picture of the stream that answers many questions of value to the program.

Task 2 **Determine the tools you will need**

Offered below are some of the tools you will need to find answers in your background investigation of the stream.

Stream headwaters, length, tributaries, final stream destination, and watershed boundaries are best determined through maps. Of greatest value are U.S. Geological Survey 7 1/2- minute topographic maps (on a 1:24,000 scale where 1 inch = 2,000 feet). At varying degrees of resolution, they depict landforms, major roads and political boundaries, developments, streams, tributaries, lakes, and other land features. Sporting goods stores and bookstores often carry these maps, especially for recreational areas that are likely to be hiked or camped. The maps can also be ordered through the U.S. Geological Survey (see box–Obtaining USGS Topographic Maps).

Road, state, and county maps might also prove helpful in identifying some of

Obtaining USGS Topographic Maps

The U.S. Geological Survey's Earth Science Information Centers can provide you with a catalog of available USGS topographic maps, a brochure on how to use topographic maps, and general information on ESIC services. Contact the main ESIC office at:

USGS Earth Science Information Center
507 National Center
12201 Sunrise Valley Drive
Reston, VA 22092
1-800-USA-MAPS

You can obtain a free USGS Indexing Catalog to help you identify the map(s) you need by calling 1-800-435-7627. If you know the coordinates of the map you need, you can order it directly from:

USGS
Branch of Information Services
Box 25286
Denver, CO 80225

Place your order in writing and include a check for $4.00 per map plus $3.50 for shipping and handling. The ESIC can also refer you to commercial map distributors that can get you the topographic maps sooner, for a higher fee. USGS topographic maps might also be available from sporting goods stores in your area.

these stream and watershed features. Hydrologic unit maps, also available from the U.S. Geological Survey but at a 1:100,000 scale of resolution (less detail than the 7 1/2-minute maps cited above), might also help you determine hydrologic watershed boundaries. Atlases and other reference materials at libraries can prove helpful in determining facts about population in the watershed.

Land uses in the stream watershed might also be depicted on maps such as those discussed above. You will verify this information in the second half of the watershed survey, when you are actually in the field observing land around the stream. Information from maps is particularly useful in developing a broad statement about general land use in the stream watershed (e.g., land use in the hypothetical Volunteer Creek watershed is 60 percent residential, 20 percent parkland/recreational, and 20 percent light industrial).

Other sources of information include:

- Land use plans from local planning offices, which include information not only for current land uses but for potential uses for which the area is zoned
- Conservation district offices or offices of the agricultural extension service or Natural Resources Conservation Service (Formerly the Soil Conservation Service, these offices might be able to provide information on agricultural land in rural areas)
- Local offices of the U.S. Geological Survey, which might provide a variety of publications, special studies, maps, and photos on land uses and landforms in the area
- Aerial photographs, which might provide current and historical views of land uses

Industries and others that discharge to the stream might be identified at the state, city, or county environmental protection or water quality office. (The name of the agency will vary by locality.) At these offices, you may ask to see records of industries with permits to discharge treated effluent to streams. These records are maintained through the National Pollutant Discharge Elimination System (NPDES). All industrial and municipal dischargers are required to have permits that specify where, when, and what they are allowed to discharge to waters of the United States.

Especially in older metropolitan areas, combined sewers are also potential discharges. Combined sewers are pipes in which sanitary sewer waste overflow and storm water are combined in times of heavy rain. These combined sewers are designed to discharge directly into harbors and rivers during storms when the volume of flow in the sewers exceeds the capacity of the sewer system. The discharge might include raw sanitary sewage waste. Combined sewers do not flow in dry weather. Maps of sewer systems can be obtained from your local water utility.

The state or local environmental agency should also be able to provide location information on other potential pollution sources such as landfills, wastewater treatment plants, and stormwater detention ponds.

Current uses of the stream are established in state water quality standards, which specify what the uses of all state waters should be. These uses can include, for example, cold water fisheries, primary contact recreation (swimming) and irrigation. The state also establishes criteria or limits on pollutants in the waters necessary to maintain sufficient water quality to support those uses, as well as a narrative statement that prohibits degradation of waters below their designated uses.

Section 305(b) of the Clean Water Act requires states to report to the U.S. Environmental Protection Agency on the designated uses of their waters, the extent of the impairment of those uses, and the causes and sources of impairment. This information is kept on file at the state water quality agency. While state reports cannot specify water uses and degree of impairment in all individual streams in the state, they are a good starting point. Write to the state water quality agency for its biennial water quality (section 305(b)) assessment.

You might also be able to obtain a copy of your state's water quality standards or establish contact with a water quality specialist who can give you information on standards for your stream. Again, information on actual water uses will be verified and detailed once you walk the stream during the visual assessment portion of your watershed survey.

Historical land uses and the history of the stream might take some legwork to uncover. Local historical societies, libraries, and newspaper archives are good places to start. Look for historical photos of the area and stories about fishing contests, fish kills, spills, floods, and other major events affecting the stream and its watershed. County or town planning offices might be able to provide information on when residential developments were built and when streams were channelized or diverted. State and local transportation agencies might have records on when highways and bridges were built. State environmental regulatory agencies have records of past or current applications to modify stream hydrology through dredging, channelization, and stream bank stabilization.

Long-time residents are another invaluable source of information on the history of your stream. People who fished or swam in your stream in their youth might have witnessed how the stream has changed. They might remember industries or land use activities of the past—such as

Getting to Know the Boundaries of Your Watershed

Once you've obtained topographic maps of your area, follow these steps to draw your watershed boundaries:

1. Locate and mark the downstream outlet of the watershed. For rivers and streams, this is the farthest downstream point in which you are interested.
2. Locate all water features such as streams, wetlands, lakes, and reservoirs that eventually flow to the outlet. Start with major tributaries, then include smaller creeks and drainage channels. To determine whether a stream is flowing to or from a lake or river, compare the elevation of land features to that of the waterbody.
3. Use arrows to mark the direction of stream or wetland flow.
4. Find and mark the high points (hills, ridges, saddles) on the map. Then connect these points, following ridges and crossing slopes at right angles to contour lines. This line forms the watershed boundary.

If you don't need to know exact watershed boundaries, simply look at the pattern of streamflow and draw lines dividing different stream systems. This will give you an idea of the shape of your watershed and those that border it. Also, once you've identified watershed boundaries, water features, and flow direction, you might want to transfer this information to a road map for easier use.

From: Eleanor Ely, Delineating a Watershed, *The Volunteer Monitor* 6(2), Fall 1994.

Figure 3.1

A topographic map with a delineated watershed.

Volunteers should learn to read a topo map to learn about the natural and cultural features of their study stream's watershed

mines or farms—that could have affected the stream. They might have tales to tell about fish they once caught or floods that led to channelization and dams. Assembling such oral histories is a particularly good activity for school-age volunteers.

Task 3 Conduct the background investigation

It is best to conduct your background investigation of the stream in the early stages of the volunteer program and use the information it uncovers to help design the program's monitoring plan, future activities, and projects.

The investigation might emphasize those aspects which are most important to the volunteers or the watershed, or it might include all the resources and tools listed above. In any case, rely on the interests of the volunteers in designing and conducting the background investigation, and divide duties among different volunteers.

Once the investigation has been conducted, either the program coordinator or an interested volunteer should compile the information collected and present it to other volunteers in written form or at a program-wide meeting. At a minimum, key information on land uses, water uses, watershed boundaries, and dischargers should be maintained in written form for program use and for volunteers who might join the program at a later date. Maps, photographs, and other information on previous water quality studies in the watershed will be of particular value to the program over time.

Obtaining Aerial Photographs

Historic and current aerial photographs can be obtained from local, state, and federal governments, as well as private firms. Try planning offices, highway departments, soil and water conservation districts, state departments of transportation, and universities.

Federal sources of aerial photographs include:

- USGS Earth Science Information Center
  507 National Center
  12201 Sunrise Valley Drive
  Reston, VA 22092
  1-800-USA-MAPS

- USDA Consolidated Farm Service Agencies
  Aerial Photography Field Office
  222 West 2300 South
  P.O. Box 30010
  Salt Lake City, UT 84103-0010
  801-524-5856

- Cartographic and Architectural Branch
  National Archives and Records Administration
  8601 Adelphi Road
  College Park, MD 20740-6001
  301-713-7040

3.2 The Visual Assessment

To conduct the visual stream assessment portion of the watershed survey, volunteers regularly walk, drive, and/or canoe along a defined stretch of stream observing water and land conditions, land and water uses, and changes over time. These observations are recorded on maps and on visual assessment data sheets and passed to the volunteer coordinator, who can decide whether additional action is needed. Volunteers might themselves follow up by reporting on problems such as fish kills, sloppy construction practices, or spills they have identified during the visual assessment.

The basic steps to follow are:

Task 1 **Determine the area to be assessed**

The visual assessment will have most value if the same stream or segment of stream is assessed each time. In this way, you will grow familiar with baseline stream conditions and land and water uses, and will be better able to identify changes over time. You should choose the largest area you feel comfortable assessing and ensure that it has easy, safe, and legal access. The area should have recognizable boundaries that can be marked or identified on road maps or U.S. Geological Survey topographic maps. This will help future volunteers continue the visual assessment in later years and help the program coordinator easily locate any problems that have been identified.

Once you have identified the area to be assessed, define it clearly in words (for example, "Volunteer Creek from Bridge over Highway One to confluence of Happy

Creek at entrance to State Park"). Then, either draw the outline and significant features of the stream and its surroundings on a blank sheet of paper or obtain a more detailed map of the area, such as a plat, road, or neighborhood map. This will serve as the base map you will use to mark stream obstructions, pollution sources, land uses, litter, spills, or other problems identified during your visual assessment.

Task 2 **Determine when to survey**

Because land and water uses can change rapidly and because the natural condition of the stream might change with the seasons, it is best to visually assess the stream or stream segment at least three times a year. In areas with seasonal changes, the best times to survey are:

- Early spring, before trees and shrubs are in full leaf and when water levels are generally high
- Late summer, when trees and shrubs are in full leaf and when water levels are generally low
- Late fall, when trees and shrubs have dropped their leaves but before the onset of freezing weather

In addition, you may wish to spot-check potential problem areas more frequently. These include construction sites, combined sewer overflow discharges, animal feedlots, or bridge/highway crossings. If polluted runoff or failing septic systems are suspected, schedule a survey during or after heavy rainfall. If a stream is diverted for irrigation purposes, surveys during the summer season will identify whether water withdrawals are affecting the stream.

Again, it is important to survey the stream at approximately the same time each season to account for seasonal variations. You might find it productive to drive through the watershed once a year and to walk the stream (or the stream's problem sites) at other times (see Tasks 4 and 5).

Task 3 **Gather necessary equipment**

In addition to the general and safety equipment listed in Chapter 2, the following equipment should be gathered before beginning the visual assessment:

- Reference map such as road map or USGS topographic map, to locate the stream and the area to be assessed
- Base map to record land uses, land characteristics, stream obstructions, sources of pollution, and landmarks
- Field data sheet
- Additional blank paper, to draw maps or take notes if needed
- Relevant information from background investigation (e.g., location of NPDES outfalls, farms, abandoned mines, etc.)

Task 4 **Drive (or walk) the watershed**

The purpose of driving (or walking) the watershed is to get an overall picture of the land that is drained by your stream or stream segment. It will help you understand what problems to expect in your stream, and it will help you know where to look for those problems.

As with all other monitoring activities, you should undertake your watershed drive or walk with at least one partner. If you are driving, one of you should navigate with a road map and mark up the base map and field sheet with relevant discoveries while the other partner drives. You might want to pull over to make detailed observations, particularly near stream crossings. ***Remember never to enter private property without permission*** (see Safety Considerations, Chapter 2).

As you drive or walk the watershed, look for the following:

- *The "lay" of the land*—become aware of hills, valleys, and flat terrain. Does any of this area periodically flood?
- *Bridges, dams, and channels*—look for evidence of how the community has dealt with the stream and its flood potential over the years. Are portions of it running through concrete channels? Is it dammed, diverted, culverted, or straightened? Where the road crosses the stream, is there evidence of erosion and pollution beneath bridges? Is streamflow obstructed by debris hung up beneath bridges?
- *Activities in the watershed*—look for land use activities that might affect your stream. In particular, look for construction sites, parking lots, manicured lawns, farming, cattle crossings, mining, industrial and sewage treatment plant discharges, open dumps, and landfills. Look for the outfalls you identified in your background investigation. Also look for forested land, healthy riparian zones, undisturbed wetlands, wildlife, and the presence of recreational users of the stream such as swimmers or people fishing. (Note that heavy recreational use or large flocks of birds might adversely affect the quality of streams, ponds, lakes, and wetlands.)

Task 5 Walk the stream

Where you have safe public access or permission to enter the stream, stop driving or walking the watershed and go down to the stream. Use all of your senses to observe the general water quality condition. Does the stream smell? Is it strewn with debris or covered with an oily sheen or foam? Does it flow quickly or sluggishly? Is it clear or turbid? Are the banks eroded? Is there any vegetation along the banks? If you see evidence of water quality problems at a particular site, you might want to investigate them in more detail. Drive or walk upstream as far as you can, and try to identify where the water quality problem begins.

Use your field data sheet to record your findings. Always be as specific as possible when noting your location and the water conditions you are observing. Draw new maps or take pictures if that will help you remember what you are observing. Don't be afraid to take too many notes or draw too many pictures. You can always sort through them later.

Take note of the positive conditions and activities you see as well as the negative ones. This, too, will help you characterize the stream and its watershed. Look for such things as people swimming or fishing in the stream; stable, naturally vegetated banks; fish and waterfowl; or other signs that the stream is healthy.

For more information on what to look for in and around the stream, consult Chapter 4 and, in particular, the *Stream Habitat Walk.*

Task 6 Review your maps/field data sheets

The last step of the watershed survey's visual assessment is to review the maps, drawings, photos, and field data sheets you have assembled for your stream or stream segment. What is this information telling you about problem sites, general stream condition, potential for future degradation, and the need for additional action? In most cases you will find that you have put together an interesting picture of your stream. This picture might prompt additional monitoring or community activity, or could urge your program coordinator to bring potential problems to the attention of water quality or public health agencies in your area.

When reviewing your data, be sure maps are legible and properly identified, photos have identifiable references, and field data sheets are filled out completely and accurately. Your program coordinator might ask for your field data sheets, maps, and other material and can probably help interpret the findings of your watershed survey.

For More Information on Your Watershed

EPA's *Surf Your Watershed* internet web site is a service designed to help citizens locate, share, and use information on their watershed or community. While you are conducting your watershed survey, you might find its features of value. *Surf* provides:

- Access to a large listing of protection efforts and volunteer opportunities by watershed.
- Information on water resources, drinking water sources, land use. population, wastewater dischargers, and water quality conditions.
- Capabilities to generate maps of your watershed and determine the latitude and longitude of specific sites within it.
- Opportunity to share your watershed information with other on-line groups through links with other pages and data-bases.

You can reach *Surf Your Watershed* on the web at *www.epa.gov/surf.*

References and Further Reading

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WATERSHED SURVEY VISUAL ASSESSMENT

GENERAL INFORMATION

Stream name: ____________________

Watershed name: ____________________

County: ____________ **State:** ____________

Approximate size of study area (acres): ____________

Investigators: ____________________

Site (description): ____________________

____

____

Date: ____________ **Time:** ____________

Weather in past 24 hours:

- ❑ Storm (heavy rain)
- ❑ Rain (steady rain)
- ❑ Showers (intermittent rain)
- ❑ Overcast
- ❑ Clear/Sunny

Weather now:

- ❑ Storm (heavy rain)
- ❑ Rain (steady rain)
- ❑ Showers (intermittent rain)
- ❑ Overcast
- ❑ Clear/Sunny

LAND USES IN THE WATERSHED

1. **Specific uses identified** *(check as many as apply)*

	Streamside	Within 1/4 mile of Stream	Within Watershed
Residential:			
Single-family housing	❑	❑	❑
Apartment building	❑	❑	❑
Lawns	❑	❑	❑
Playground	❑	❑	❑
Parking lot	❑	❑	❑
Other ____________	❑	❑	❑
Commercial / Industrial / Institutional:			
Commercial development (stores, restaurants)	❑	❑	❑
Auto repair/gas station	❑	❑	❑
Factory/Power plant	❑	❑	❑
Sewage treatment facility	❑	❑	❑
Water treatment facility	❑	❑	❑
Institution (e.g., school, offices)	❑	❑	❑
Landfill	❑	❑	❑
Automobile graveyard	❑	❑	❑
Bus or taxi depot	❑	❑	❑
Other ____________	❑	❑	❑
Forest / Parkland:			
Recreational park	❑	❑	❑
National/State Forest	❑	❑	❑
Woods/Greenway	❑	❑	❑
Other ____________	❑	❑	❑
Agricultural / Rural:			
Grazing land	❑	❑	❑
Cropland	❑	❑	❑
Animal feedlot	❑	❑	❑
Isolated farm	❑	❑	❑
Old (abandoned) field	❑	❑	❑
Fish hatchery	❑	❑	❑
Tree farm	❑	❑	❑
Other ____________	❑	❑	❑

2. **Summary of major land uses in the watershed** *(use approx. percentages)*

Residential ____% Parkland/Forest ____%

Commercial/Industrial/Institutional ____% Other ____%

Agricultural/Rural ____%

3. **Additional activities in the watershed** *(check as many as apply)*

	Streamside	Within 1/4 mile of Stream	Within Watershed
Construction			
Building construction	☐	☐	☐
Roadway	☐	☐	☐
Bridge construction	☐	☐	☐
Other ____________	☐	☐	☐
Logging			
Selective logging	☐	☐	☐
Intensive logging	☐	☐	☐
Lumber treatment facility	☐	☐	☐
Other ____________	☐	☐	☐
Mining			
Strip mining	☐	☐	☐
Pit mining	☐	☐	☐
Abandoned mine	☐	☐	☐
Quarry	☐	☐	☐
Other ____________	☐	☐	☐
Recreation			
Biking/Off-road vehicle trails	☐	☐	☐
Horseback riding trail	☐	☐	☐
Boat ramp	☐	☐	☐
Jogging paths/hiking trail	☐	☐	☐
Swimming area	☐	☐	☐
Fishing area	☐	☐	☐
Picnic area	☐	☐	☐
Golf course	☐	☐	☐
Campground/trailer park	☐	☐	☐
Power boating	☐	☐	☐
Other ____________	☐	☐	☐

4. **Comments on land uses**

Use this space to explain or expand on land use descriptions you have identified above. For example, you might want to identify particular buildings, specify the location of construction sites, note the condition of streamside picnic areas, note the presence of cows in a stream, or note corrective measures such as swales or settling basins.

GENERAL STREAM AND WATERSHED CHARACTERISTICS

5. Note the number of hydrologic modifications *(structures that alter natural stream flow):*

None	________	Waterfalls	________
Dams	________	Stream fords	________
Bridges	________	Beaver dams	________

6. Note the approximate length of stream that is affected by the following:

Stream diversion	________ feet or	________ miles
Stream straightening	________ feet or	________ miles
Concrete streambank/bottom	________ feet or	________ miles

7. Check the categories that best describe the general appearance of the stream:

Litter:

- ❑ No litter visible
- ❑ Small litter occasionally (e.g., cans, paper)
- ❑ Small litter common
- ❑ Large litter occasionally (e.g., tires, carts)
- ❑ Large litter common

Erosion:

- ❑ No streambank erosion or areas of erosion very rare; no artificial stabilization
- ❑ Occasional areas of streambank erosion
- ❑ Areas of streambank erosion common
- ❑ Artificial streambank stabilization (e.g., rip rap) present

Special Problems *(note in detail in comment section below):*

- ❑ Spills of chemicals, oil, etc.
- ❑ Fish kills
- ❑ Wildlife, waterfowl kills
- ❑ Flooding
- ❑ Periods of no flow

8. Comments on general stream characteristics (e.g., date and size of fish kill, increased rate of erosion evident, litter most evident after storms)

PIPE AND DRAINAGE DITCH INVENTORY

In this section, provide information on pipes and drainage ditches found on the banks or in the stream. These pipes/ditches can be abandoned or active. Note this basic information for each pipe or drainage ditch you observe. Attach additional pages to this form.

9. This information applies to a:

❑ Pipe ❑ Drainage ditch ❑ Other ________

10. Location of pipe/ditch:

❑ In stream ❑ In streambank ❑ Near stream

Describe location:

11. Pipe/Ditch # (for mapping/locational purposes) ____________

12. Identify type of pipe (check one)

- ❑ Industrial outfall
- ❑ Sewage treatment plant outfall
- ❑ Storm drain
- ❑ Combined sewer overflow
- ❑ Agricultural field drainage
- ❑ Paddock or feedlot drainage
- ❑ Settlement basin/pond drainage
- ❑ Parking lot drainage
- ❑ Unknown
- ❑ Other ____________

13. Approximate Diameter of Pipe: ________ inches or ________ feet

14. Describe the discharge flow:

Rate of Flow: ❑ None ❑ Intermittent ❑ Trickle ❑ Steady ❑ Heavy

Appearance: ❑ Clear ❑ Foamy ❑ Turbid ❑ Oily sheen ❑ Colored ____________

Odor: ❑ None ❑ Rotten eggs/sewage ❑ Chemical ❑ Chlorine ❑ Other ____________

15. Describe the streambank/stream below pipe or drainage ditch:

- ❑ No problem evident
- ❑ Sewage litter (e.g., toilet paper)
- ❑ Litter (e.g., styrofoam, cans)
- ❑ Eroded
- ❑ Lots of algae
- ❑ Other ____________

16. Comments on pipes and drainage ditches

Use this space to explain or expand on information provided on pipes and discharges you have identified above. For example, you may want to identify particular facilities, or discuss in more detail the condition of the stream below the discharge.

Izaak Walton League of America
Save Our Streams – Stream Walk Survey

Name of Stream:________________________ **County:**______________ **State:**_______ **Date:**_______________

Why take a stream walk?

- to learn more about the health of your local stream,
- to map potential sources of pollution, and;
- to determine needs for more water quality monitoring, clean-up, and enhancement.

Have Fun, Be Prepared, and Be Safe.

- Before you leave, make sure someone knows where you are going and about what time you should return. Do not go alone.
- Take with you: this survey sheet, a notebook, map, and pencils. And if you have these, take them too: a GPS unit, camera, binoculars, thermometer, tape measure, and waterproof boots.
- Most importantly take a First Aid Kit and cell phone for emergencies! Your First Aid Kit should include: adhesive and cloth bandages, surgical tape, tweezers, pain reliever, anti-histamine, antiseptic spray or ointment, hydrogen peroxide, cotton balls, and an instant ice pack.

General Stream Information. *(You can answer a lot of these questions by looking at maps and talking to local and state conservation staff prior to taking your stream walk.)*

- How long is the stream? _________miles
- Where does is begin?________________________ Where does it end?_____________________________
- Do other streams flow into this stream? **YES/NO**_ Which ones? ______________________________________
- Does this stream flow into other streams? **YES/NO** Which ones? _____________________________________
- What type of land uses does the stream flow through in its watershed? *(check all that apply)*
  ☐ Rural, such as farmland, forested land or open grasslands ☐ Other ____________________________
  ☐ Urban, such as cities and towns
  ☐ Suburban, such as housing developments and some open land

Stream Walk Survey Sheet

(Fill this out as your walk along the stream. You may want to use several copies for long walks to represent various areas along the stream, or where an unusual condition appears.)

GPS Coordinates/or Description of Start Location (cross street names, landmarks, etc.):_______________________
GPS Coordinates/or Description of End Location: ____
Avg stream width: _______ft. Avg. stream depth: ________ft.
Water lever/flow rate is: ______High ______Normal ______Low _____Negligible
Weather conditions (last 72 hours):____

What is the stream bed made of? (*Check all that apply. Give an estimated % for each description marked.*)

☐ ___Bedrock (large area of rock covering streambed, cannot be removed)
☐ ___Boulders (watermelon-size and larger)
☐ ___Cobbles (orange-size)
☐ ___Gravel (grape-size)
☐ ___Sand (smaller than grape-size)
☐ ___Silt (smaller than sand and feels silky)

What color is the water? ☐ Clear ☐ Tea-colored ☐ Milky ☐ Muddy ☐ Black ☐ Grey ☐ Other _______________
Does the water appear oily on the surface? **YES/NO** Describe:____
Is there foam on the surface of the water? **YES/NO** Describe:____

Do you see trash in or around the stream? **YES/NO**
Describe the types of trash and how much you see.______________________________________ If collecting trash along your trip, record how many large garbage bags you collect. ________ (*Can some, or all of it, be recycled? If so, you can recycle at your municipal waste facility.)*

Do you smell any unusual smells such as oil, sewage, or rotten eggs? **YES/NO**
Describe the smells. ___________________________________ *(Do not go into any stream with unusual smells. Instead, record a description of the smells and the location, and contact your local environmental government agency).*

Are there any discharge pipes in the stream? **YES/NO** If yes, how many? _________
What types of pipes are they? ☐ Unknown ☐ Runoff (field or stormwater) describe: __________
☐ Sewage Treatment _________ ☐ Industrial: type of industry _____________________

What do you see on the banks of the stream? ☐ Concrete ☐ Soil ☐ Rock ☐ Vegetation/roots

Is there erosion along the banks? **YES/NO** Describe: ☐ Severe ☐ Moderate ☐ Slight; ☐ One side ☐ Both sides

If there is vegetation growing on the streambanks, what types do you see?
☐ Trees (woody plants 6' or taller) ☐ Shrubs (woody plants shorter than 6') ☐ Grasses and Vines

Is the land along the stream:
☐ Paved ☐ Lawn ☐ Trees ☐ Other ____

Circle the land uses you see while walking along the stream:

Roads	Houses	Apartments	Schools
Shopping Malls	Crop Fields	Golf Courses	Pastures
Parks	Mining	Sewer Manholes	Landfill
Forest	Discharge Pipes	Construction Sites	Cut Trees

Are there any land uses not listed above? **YES/NO**
Indicate location (draw a stream map) and describe each land use. ________________________

Do you see any animal tracks? **YES/NO**
Draw pictures of the animal tracks.

Do you see any animal houses, such as beaver dams or bird nests? YES/NO
Describe: ____

Do you see fish? **YES/NO**
What size? _____inches How many? ☐ Scattered Individuals ☐ Scattered Schools (groups)
What kind? *(check box if you can identify the fish you see)*
☐ Unsure ☐ Trout *(pollution sensitive)* ☐ Bass *(somewhat pollution sensitive)*
☐ Catfish *(pollution tolerant)* ☐ Carp *(pollution tolerant)* ☐ Other _______________

What other observations can you make about your stream? Describe them: ________________
____

For more information about stream monitoring and stream enhancement projects go to www.iwla.org/sos.

Founded in 1922, the Izaak Walton League of America protects America's outdoors through community-based conservation, education, and the promotion of outdoor recreation. The League has more than 36,000 members and supporters nationwide

Biological monitoring, the study of biological organisms and their responses, is used to determine environmental conditions. One type of biological monitoring, the biological survey or *biosurvey*, is described in this chapter. The biosurvey involves collecting, processing, and analyzing aquatic organisms to determine the health of the biological community in a stream.

In wadable streams (streams that can be easily walked across, with water no deeper than about thigh-high), the three most common biological organisms studied are fish, algae, and macroinvertebrates. This manual discusses macroinvertebrate monitoring only.

Macroinvertebrates are organisms that are large (macro) enough to be seen with the naked eye and lack a backbone (invertebrate). They inhabit all types of running waters, from fast-flowing mountain streams to slow-moving muddy rivers. Examples of aquatic macroinvertebrates include insects in their larval or nymph form, crayfish, clams, snails, and worms (Fig. 4.1). Most live part or most of their life cycle attached to submerged rocks, logs, and vegetation.

Aquatic macroinvertebrates are good indicators of stream quality because:

- They are affected by the physical, chemical, and biological conditions of the stream.
- They can't escape pollution and show the effects of short- and long-term pollution events.
- They may show the cumulative impacts of pollution.
- They may show the impacts from habitat loss not detected by traditional water quality assessments.
- They are a critical part of the stream's food web.
- Some are very intolerant of pollution.
- They are relatively easy to sample and identify.

The basic principle behind the study of macroinvertebrates is that some are more sensitive to pollution than others. Therefore, if a stream site is inhabited by organisms that can tolerate pollution—and the more pollution-sensitive organisms are missing—a pollution problem is likely.

For example, stonefly nymphs—aquatic insects that are very sensitive to most pollutants—cannot survive if a stream's dissolved oxygen falls below a certain level. If a biosurvey shows that no stoneflies are present in a stream that used to support them, a hypothesis might be that dissolved oxygen has fallen to a point that keeps stoneflies from reproducing—or has killed them outright.

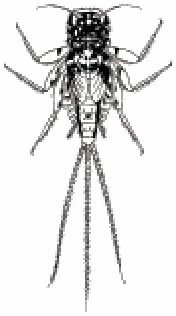
This brings up both the advantage and disadvantage of the biosurvey. The advantage of the biosurvey is that it tells us very clearly when the stream ecosystem is impaired, or "sick," due to pollution or habitat loss. It is not difficult to realize that a stream full of many kinds of crawling and swimming "critters" is healthier than one without much life. The disadvantage of the biosurvey, on the other hand, is that it cannot definitively tell us *why* certain types of creatures are present or absent.

In this case, the absence of stoneflies might indeed be due to low dissolved oxygen. But is the stream under-oxygenated because it flows too sluggishly or because pollutants in the stream are damaging water quality by using up the oxygen? The absence of stoneflies might also be due to other pollutants discharged by factories or running off farmland, water temperatures that are too high, habitat degradation such as excess sand or silt on the stream bottom that has ruined stonefly sheltering areas, or other conditions. Thus a biosurvey should be accompanied by an assessment of *habitat* and *water quality conditions* in order to help explain biosurvey results.

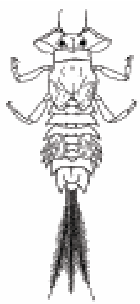
Habitat, as it relates to the biosurvey, is defined as the space occupied by living organisms. In a stream, habitat for macroin-

Visual guide to common macroinvertebrates

Flathead mayfly (L)

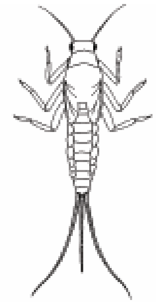


Spiny-crawler mayfly (L)

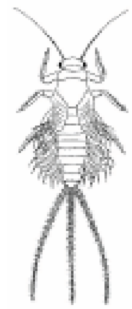


Brush-legged mayfly (L)

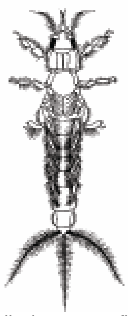
Minnow mayfly (M)



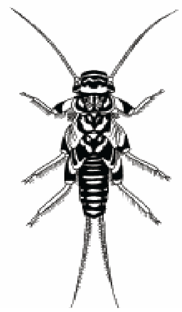
Prong-gilled mayfly (L)



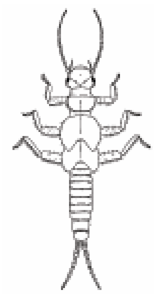
Burrowing mayfly (L)



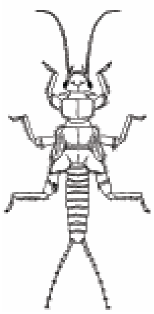
Common stonefly (L)



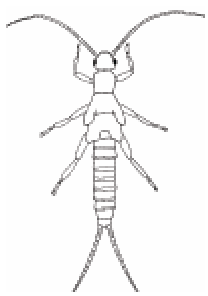
Green stonefly (L)



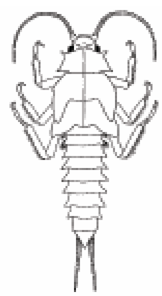
Brown stonefly (L)



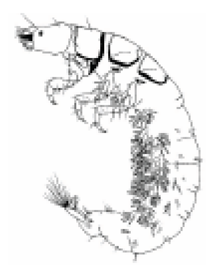
Small winter stonefly (L)



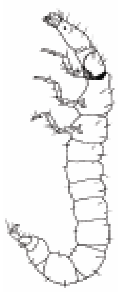
Giant stonefly (L)



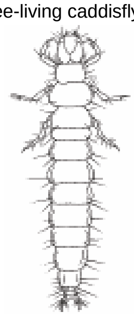
Common netspinner (M)



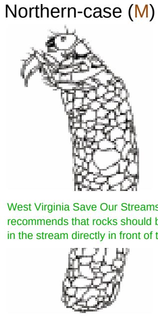
Finger-net caddisfly (L)



Free-living caddisfly (L)



Northern-case (M)

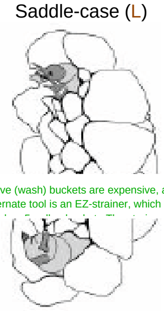


West Virginia Save Our Streams
recommends that rocks should be rubbed
in the stream directly in front of the slightly

Humpless-case (L)

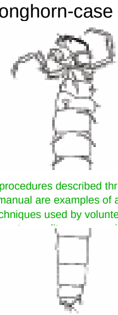
WV Save Our Streams rates the
invertebrate community differently
than the example provided above.

Saddle-case (L)



Sieve (wash) buckets are expensive, an
alternate tool is an EZ-strainer, which fits

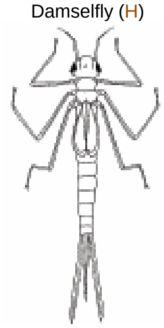
Longhorn-case (L)



The procedures described throughout
this manual are examples of a variety
of techniques used by volunteer and

Dragonfly (M)

Damselfly (H)



Tolerance rating scale

Low (L)			Moderate (M)				High (H)		
1	2	3	4	5	6	7	8	9	10

The tolerance ratings are based upon the organisms' ability to withstand changes to their environment (i.e. pollution), mostly from human influences.

Size ranges (mm)

< 5	5 - 15	15 - 30	30 - 50	> 50
(VS)	(S)	(M)	(L)	(VL)

The purpose of this guide is to provide images and common names of **benthic macro-invertebrates** that volunteers may collect from riffles of streams and rivers. The guide is not complete, it is only meant to distinguish between the most common orders and classes, and a few common families (kinds).

Insect Groups

Mayflies (order **Ephemeroptera**)

Three pairs (6 total) of legs; one hooked claw at the end of each leg; gills on the abdomen (may be covered by plates); 2 or 3 tail filaments and 2 short antennae. (M – L)

Stoneflies (Order **Plecoptera**)

Three pairs of legs (6 total); 2 hooked claws at the end of each leg; no gills on most of the abdomen but may have gills on the legs, thorax and upper abdomen; 2 tail filaments and 2 long antennae. (M – VL)

Caddisflies (Order **Trichoptera**)

Three pairs of legs (6 total); segmented grub-like body; some kinds may have gills along lower and upper portions of the abdomen; small hair-like tails or hooks. Case builders may be enclosed in a case (retreat) that they construct using stream bottom materials such as pebbles, sand grains, woody debris, pieces of plant material or some combination; others construct a net, which consists of materials held together by a silk-like thread. Note: The **free-living caddisfly** does not build a retreat. The case builders often construct a specific case that can sometimes help with their identification. The **common netspinner caddisfly** is more tolerant than most of the group. The abundant gills on the underside of their body, their filamentous tails and their particular motion can distinguish them. (S – L)

Fishflies and **Alderflies** (Order **Megaloptera**)

Three pairs of legs (6 total); filaments along the body starting just below the legs; variable tails at the end of the abdomen. **Alderflies** have a long tapered tail; **hellgrammites** have hooked-tails. They also have gill-tufts under each of their filaments, fishflies and alderflies do not. All members of the group have large pinching jaws on the head, (M – VL)

Beetles (Order **Coleoptera**)

Three pairs of legs (6 total); mainly rounded or oval shape as adults; a few kinds have tails hooks or filaments, hard bodies and visible wing-pads. The most commonly encountered beetles are the **riffle beetle**, which is a small dark beetle and **water penny**, which looks like a penny. The whirligig beetle larva may have many filaments along their bodies similar to fishflies. (VS – L)

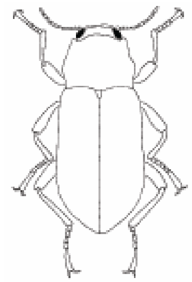
Damselflies and **Dragonflies** (Order **Odonata**)

Damselflies: Three pairs of legs (6 total); long, thin abdomen; large eyes; extended lower lip; 3 fan like structures, which are actually their gills, at the end of the abdomen. (M – L)

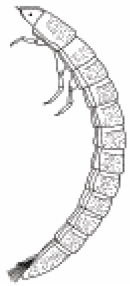
Dragonflies: Three pairs of legs (6 total); extended lower lip; large eyes; rounded or extended abdomen; no gills on the abdomen; no tails but may have knobs or points on the abdomen that resemble tails. (M – VL)

Visual guide to common macroinvertebrates

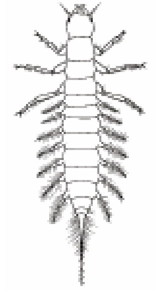
Riffle beetle adult (M)



Riffle beetle larva (M)

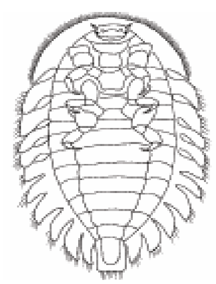


Alderfly (M)

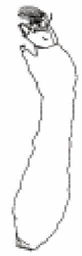


Hellgrammite (L)

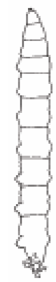
Water penny (L)



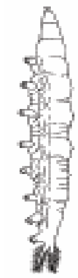
Black fly (M)



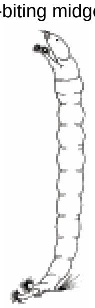
Crane fly (M)



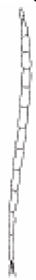
Watersnipe fly (L)



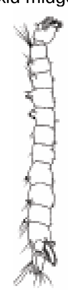
Non-biting midge (H)



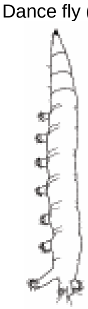
Biting midge (H)



Dixid midge (H)

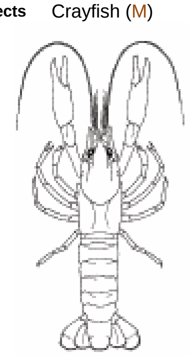


Dance fly (H)



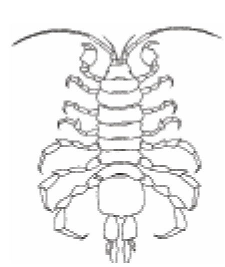
Non-Insects

Crayfish (M)

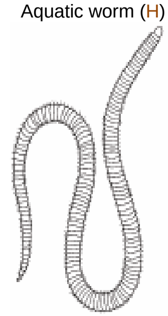


Scud/Sideswimmer (M)

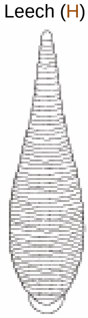
Aquatic sowbug (H)



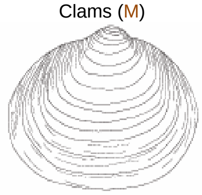
Aquatic worm (H)



Leech (H)



Clams (M)



Mussels (L)

Snails (M)

Note: The image sizes are not proportional nor do they represent actual sizes; in some cases colors can be variable. Color and size should not be used as distinguishing characteristics when attempting to identify benthic macroinvertebrates. Not all families that may be collected are shown here. Most of the images are drawn in lateral of top views.

Learn more at: http://dep.wv.gov/sos

< 5	5 - 15	15 - 30	30 - 50	> 50
(VS)	(S)	(M)	(L)	(VL)

Size ranges (mm)

True Flies (Order **Diptera**)

No legs or may have structures that resemble legs (false-legs); mainly segmented grub-like or worm-like bodies; tiny hair-like tails, lobes, tentacles or other structures at the end of their abdomen (or no tails); often a distinct head can be seen, but on other kinds no head is visible. Many different kinds of flies are encountered, the more common kinds include the **crane flies**, most have no legs, a plump segmented body and numerous tentacles or bulbous structures; **watersnipe flies** has false legs and a forked hooked tail, looks similar to a caterpillar; **black flies** have a bowling pin or vase shape and fan-like structure on their head; **non-biting midges** are usually very small with a thread-like or worm-like body (some are red in color) with a very erratic wriggling motion. There are many more Diptera that are sometimes collected, but the only the images of the biting midge, dixid midge and dance fly are shown here. (VS – L)

Non-Insects

Crayfish, **Scuds** and **Sowbugs** (Sub-phylum **Crustacea**)

More than three pairs (more than 6 total) of legs; claws on the first several pairs of legs, which may be enlarged; long antenna. This group includes the **crayfish**, which looks like a small lobster, **scuds** also called sideswimmers resemble a shrimp and are flattened from side to side, and the **aquatic sowbugs**, which resemble a pill bug and are flattened from top to bottom. (M – VL)

Leeches and **Worms** (Phylum **Annelida**)

Worm-like appearance; no legs and many segments along the entire length of the body. This group includes the **aquatic worms** and **leeches**. The suckers on both ends of their body distinguish the leeches from other annelids. **Flatworms** are also sometimes collected, but they are not truly Annelids, they belong to the phylum Platyhelminthes. An image of the flatworm is not provided. (M – VL)

Clams and **Mussels** (Class **Bivalvia**)

Two cup-shaped shells connected by a hinged structure; the shell is made of calcium carbonate and is usually very strong and hard to open. **Mussels** have an oblong rough, often dark color shell. Most **clams** are smaller and have a rounded shell. The Asian clam can be distinguished from the native pea clam by the raised ridges; pea clams are often smaller and its shell feels smooth to the touch. (S – L)

Snails (Class **Gastropoda**)

Single coiled shell that mostly opens to the right when the point is held facing towards you. **Operculate snails** have an operculum "a door that shuts the shell" and are commonly known as gilled snails. Some have shells that open to the left when the point is held facing towards you; shells also may be rounded flat or coiled. Many of these are **non-operculate snails** that do not have an operculum and are commonly known as pouch or pond snails. (S – L)

Images courtesy of the University of Minnesota's **Water Research Center**; used with permission

vertebrates includes the rocks and sediments of the stream bottom, the plants in and around the stream, leaf litter and other decomposing organic material that falls into the stream, and submerged logs, sticks, and woody debris. Macroinvertebrates need the shelter and food these habitats provide and tend to congregate in areas that provide the best shelter, the most food, and the most dissolved oxygen. A habitat survey examines these aspects and rates the stream according to their quality. This chapter includes both simple and intensive habitat surveys volunteers can conduct.

Monitoring for water quality conditions such as low dissolved oxygen, temperature, nutrients, and pH helps identify which pollutants are responsible for impacts to a stream. Water quality monitoring is discussed in Chapter 5.

Uses of the Biosurvey and Habitat Assessment

The information provided by biosurveys and habitat assessments can be used for many purposes.

- *To screen for impairment.* Biosurveys can be used to identify problem sites along a stream. A habitat assessment can help determine whether the problem is due, at least in part, to a habitat limitation such as poor bank conditions.
- *To identify the impact of pollution and of pollution control activities.* Because macroinvertebrates are stationary and are sensitive to different degrees of pollution, changes in their abundance and variety vividly illustrate the impact pollution is having on the stream. Loss of macroinvertebrates in the stream, or of trees along the stream bank, are environmental impacts that a wide segment of society can relate to. Similarly, when a pollution control activity takes place—say, a fence is built to keep cows out of the stream—a biosurvey may show that the sensitive macroinvertebrates have returned and a habitat assessment might find that the formerly eroded stream banks have recovered.
- *To determine the severity of the pollution problem and to rank stream sites.* To use biological data properly, water resource analysts generally compare the results from the stream sites under study to those of sites in ideal or nearly ideal condition (called a *reference condition*). Individual stream sites can then be ranked from best to worst, and priorities can be set for their improvement.
- *To determine support of aquatic life uses.* All states designate their waters for certain specific uses, such as swimming or as cold water fishery. States establish specific standards (limits on pollutants) identifying what concentrations of chemical pollutants are allowable if designated stream uses are to be maintained. Increasingly, states are also developing biological criteria—essentially, statements of what biological conditions should be in various types of streams throughout the state. States are required by the Clean Water Act to report on those waters which do not support their designated uses.

  Biological surveys directly examine the aquatic organisms in streams and the stressors that affect them. Therefore, these surveys are ideal tools to use in determining whether a stream's designated aquatic life uses are supported.
- *To identify water quality trends.* In any given site, biological data can be used to identify water quality trends (increasing or decreasing) over several years.

Designing a Biosurvey Program

In most cases, this manual recommends that local aquatic biologists assist in the development of volunteer biological monitoring programs. This is because the types of habitats and organisms in streams vary widely with geography and climate. Tools as basic as macroinvertebrate identification keys might need to be adapted to local conditions.

Many volunteer monitoring programs rely for assistance on aquatic biologists working for state water-quality or natural resource agencies. Others are assisted by university personnel, hire their own expert staff, or contract out for consulting services. Whatever the source of expertise, professional guidance is essential for creating a successful biosurvey program. **This manual strongly recommends a close level of coordination with state or local agencies that might use the data volunteers collect.**

Monitoring approaches—and the level of professional guidance and assistance needed—clearly vary with the goals and resources of individual volunteer groups. Therefore, this manual presents three different approaches or tiers to biological monitoring.

- *Stream Habitat Walk* (detailed in section 4.1) is for groups focused primarily on educating volunteers about their streams and for identifying severe pollution problems. Volunteers conduct simple visual assessments of habitat to gain a greater appreciation of local stream ecology.

  It is based on a protocol known as Streamwalk developed by the EPA Region 10 Office in Seattle, Washington, and is widely used by volunteers throughout the Pacific Northwest.

- *Streamside Biosurvey* (detailed in section 4.2) trains volunteers to collect macroinvertebrates and identify them to order level (stonefly, mayfly, caddisfly, etc.) in the field. Monitors evaluate the macroinvertebrate community structure by sorting specimens into three general sensitivity categories. In addition, volunteers characterize habitat by conducting a modified Stream Habitat Walk.

  This tier is based on a protocol developed by the Ohio Department of Natural Resources and adapted by the Izaak Walton League of America. It has been used by volunteer monitors nationwide, including programs in Ohio, Tennessee, Georgia, Virginia, Kentucky, Illinois, and West Virginia.

- *Intensive Biosurvey* (detailed in section 4.3) requires that volunteers work under the supervision of professional aquatic biologists. Volunteers undergo formal training and conduct quality-controlled sampling and analysis. Using microscopes in a laboratory setting, macroinvertebrates are identified to

Figure 4.2

Taxonomic classification system

Depending on the program, volunteers might be asked to identify macroinvertebrates to the order level in the field or to the family level if using microscopes in the laboratory.

Taxonomic Classification

Scientists have developed a system for classifying all living creatures based on shared characteristics (taxonomic classification). It is a tiered system that begins on a large scale (i.e., Animal Kingdom/Plant Kingdom) and works its way down to the level of individual species. To illustrate, the burrowing mayfly is classified as follows.

Kingdom:	Animal	**Family:**	Ephemerida
Phylum:	Arthropoda	**Genus:**	Hexagenia
Class:	Insecta	**Species:**	limbata
Order:	Ephemeroptera		

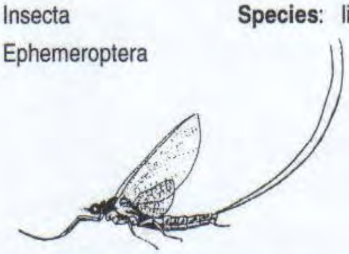


Table 4.1

Tiered framework for volunteer biological monitoring programs

Program designers might choose simple or complex approaches according to program goals and resources.

the family level (what types of stoneflies, mayflies, caddisflies, etc.). Analytical techniques are subsequently applied to the data to draw conclusions about the biological health of the sampled site. This rigorous biosurvey approach results in data that can yield information on subtle stream impacts and trends.

Based primarily on EPA's Rapid Bioassessment Protocols, this approach has been adapted by Maryland Save Our Streams, the River Watch Network and other groups.

We have modified the approaches used by other groups to add to their capabilities or to make them more generally applicable to all U.S. streams. Individual programs might choose to start with the simplest, least resource-intensive approach and work their way toward increasing complexity as resources, expertise, and volunteer interest allow. However, groups might decide to begin with a more complex approach that better suits their program goals. Table 4.1 illustrates some of the key differences in the three biological monitoring approaches discussed in this manual.

Protocol Elements	Stream Habitat Walk	Streamside Biosurvey	Intensive Biosurvey
Program Objectives	■ Education/public awareness ■ Gross problem identification/screening	■ Education/public awareness ■ Problem identification/screening ■ Preliminary ranking of sites for further study	■ Education/public awareness ■ Problem identification/screening ■ Assessing severity of problems ■ Ranking of sites for management action
Complexity of Approach	■ Simple visual assessment of habitat and physical characteristics ■ Basic observational biological data recording general abundance/variety of macroinvertebrates and presence or absence of macrophytes, algae, and fish	■ Visual assessment of habitat and physical characteristics ■ In-stream biota collected and evaluated at streamside for relative sensitivity/tolerance and identified to order/family level	■ Comprehensive habitat and physical assessment ■ Instream biota collected, preserved, and identified in lab to family level (multimetric approach) ■ Reference sites or conditions identified
Resource Investment	■ Scientific personnel assist in project design, preparation of documentation, and orientation of volunteers ■ Minimal equipment (maps, manuals, forms)	■ Scientific personnel involved in project design, preparation of documentation, training, and supervision of biosurveys ■ Sampling gear, maps, manuals, forms, references	■ Scientific personnel active in all levels and mandatory for assessment and data interpretation ■ Laboratory and storage facilities in addition to other equipment ■ Voucher and reference collections required
Training	■ Primarily self-instructional using manuals/documentation (some training is desirable)	■ Periodic workshops and streamside training sessions	■ Formal lab and field training with experienced team leaders before all assessments

4.1 Stream Habitat Walk

The Stream Habitat Walk is an easy-to-use approach for identifying and assessing the elements of a stream's habitat. It is based on a simple protocol known as *Streamwalk*, developed by EPA's Regional Office in Seattle, Washington and consists primarily of visual observation of stream habitat characteristics, wildlife present, and gross physical attributes. A simple in-stream macroinvertebrate evaluation can also be performed. This approach requires little in the way of equipment and training.

The Stream Habitat Walk is most useful as:

- A screening tool to identify severe water quality problems
- A vehicle for learning about stream ecosystems and environmental stewardship

Because the Stream Habitat Walk is not scientifically rigorous, data from this approach are less likely to be used by state and local water quality management agencies than are data from other biological monitoring approaches. However, the Stream Habitat Walk's ease of use, adaptability, and low cost make it a highly attractive approach for many programs whose primary focus is public awareness and citizen involvement.

Step 1—Prepare for the Walk

TASK 1 **Schedule your Habitat Walk**

To provide data that accurately characterize your stream and can be used to document general trends in your area, you should walk the same site at least three times a year, during different seasons. It is usually best to visit your site in early spring, late summer, and fall if you live in a part of the country that experiences seasonal variations in leaf cover, vegetation growth, and water flow. It is a good idea to check with a local aquatic biologist for assistance in determining the best times to schedule monitoring. For purposes of accuracy and consistency, it is best to monitor the same site from year to year and at the same time of the year (e.g., in the spring and, more specifically, in the same month).

TASK 2 **Obtain a U.S. Geological Survey (USGS) topographic map of your area**

One of the most valuable tools for conducting stream monitoring work is a U.S. Geological Survey (USGS) topographic map. These "topo" maps display many important features of the landscape including elevations, waterways, roads, and buildings. They are critical tools for defining the watershed of your study stream. (See Chapter 3 for a discussion of topographic maps.)

TASK 3 **Select and mark the Habitat Walk location(s)**

Choosing the location for stream monitoring is a task defined by the goals of your individual program. Program managers may select sites themselves or in collaboration with local or state water quality personnel. Other programs allow their volunteers to choose the site based on their personal interests. (See Chapter 2 for a discussion on choosing monitoring locations.) If a Watershed Survey is conducted (see Chapter 3), this information should play a role in deciding which areas are the best candidates for the Stream Habitat Walk.

Once a monitoring site is chosen, it should be marked on the topo map. This will document the location and serve as a record in case future volunteers or data users need to find the site.

TASK 4 Become familiar with safety procedures

Volunteers must always keep safety in mind while conducting any stream monitoring activity. Provide all Stream Habitat Walk participants with a list of safety do's and don'ts and have them review this list thoroughly. **Chapter 3 covers several important safety concerns that should be incorporated into a stream monitoring program.** Remember, volunteer safety is more important than the data. Some reminders include:

- Let someone know where you're going and when you expect to return. Make sure you have an "in case of emergency" phone number with you before leaving for the field.
- Do not cross streams in high flows.
- Never go into the field alone; always work in teams of at least two people.
- If for any reason you feel unsafe, do not attempt to monitor on that day.

TASK 5 Gather equipment and tools for the Habitat Walk

There is nothing more frustrating than arriving at a field monitoring site and not having all your equipment and supplies. Providing volunteers with a checklist of necessary items will help keep them organized. In addition to the basic equipment listed in Chapter 2, you will need the following for the Stream Habitat Walk.

For locating the site

- U.S. Geological Survey (USGS) topographic map of the stream area (supplemented by regular street map if needed)

For recording observations

- Stream Habitat Walk field data sheet

For marking-off the stream stretch of study

- Tape measure, string, or twine (25 yards)

For working in and around the stream

- Thermometer for measuring water temperature (Scientific supply houses sell armored thermometers that are best suited for this purpose, although you can obtain a good thermometer from an aquarium store. Some thermometers need to be calibrated before use. See Chapter 5 for instruction on calibrating and using thermometers.)
- Watch with a second hand or a stopwatch

For observing macroinvertebrates (optional)

- A bucket
- A shallow white pan. (Alternatives: white plastic plate or the bottom of a white plastic detergent jug)
- Tweezers or soft brush
- Ice cube trays (for sorting macroinvertebrates)
- Magnifying glass

TASK 6 Become familiar with the Stream Habitat Walk field data sheet and the definitions of its elements

It is important to become familiar with the Stream Habitat Walk field data sheet and its instructions before you begin your Stream Habitat Walk. If you are unclear about any instructions when you are conducting your Walk, just leave that space blank and keep going. You might wish to contact your volunteer program coordinator for further explanation after you have completed your Walk.

At the end of this section is a sample field data sheet. You might find it necessary to modify this sheet slightly to better meet the needs of your volunteers, your ecological region, and your program. When you fill out your field data sheet, base your responses on your best judgment of conditions in a stretch of stream that includes about 50 yards both upstream and downstream of the place where you are standing. If you identify features and problems beyond your chosen 100-yard length, feel free to note them on your map and form. You might want to conduct additional Walks in the area where those features are found.

Instructions on how to fill out the field data sheet are included right on the form. They are also covered in an expanded format, with illustrations, in this text. Although many of the required measures are relatively self-explanatory, it might be a good idea to make copies of these instructions for all volunteer teams to take into the field as an additional training tool.

Step 2—Delineate and sketch your site

TASK 1 **Delineate the site**

Using your tape measure or 25 yards of string or twine, measure off four 25-yard lengths alongside the stream for a total of 100 yards. Start from a point of reference such as a tree, large rock, or bend in the stream.

TASK 2 **Sketch your site on the field data sheet**

On the field data sheet, sketch the 100-yard section of stream. (Fig. 4.3). Drawing the map will familiarize you with the terrain and stream features and provide you and other volunteers with a visual record of your habitat walk. You should walk the 100-yard length from at least one bank.

On your sketch, note features such as riffles, runs, pools, ditches, wetlands, dams, riprap, outfalls, tributaries, landscape features, jogging paths, vegetation, and roads. Use your topo map or a compass to determine which direction is north and mark it on your sketch. If you see important features outside your 100-yard length of stream, mark them on your sketch but note that they are outside the stream reach. Remember to use pencil or waterproof ink when drawing your map or filling out the field data sheets because regular ink will run if wet.

Select a 25-yard section of the site. You will be filling out your field data sheet for this section only. Mark the section on the sketch. If you want to conduct multiple walks, choose another 25-yard section or move to an entirely different location. Even though you will only be completing the data forms for the 25 yard reach, it is important to sketch the full 100-yard section so that you can document the stream features surrounding the evaluated reach.

TASK 3 **Complete the top portion of your field data sheet**

Include stream name, date, and county (or appropriate local designation) of your site, and describe its location as precisely as possible. It is best to stand at or near a permanent marker such as a bridge, abutment, or road. Remember, you or another volunteer will be coming back to the same spot again and again, so be as specific as you can. Some programs might ask you for the latitude and longitude of your location; others might ask for a map reference number or other site identifier.

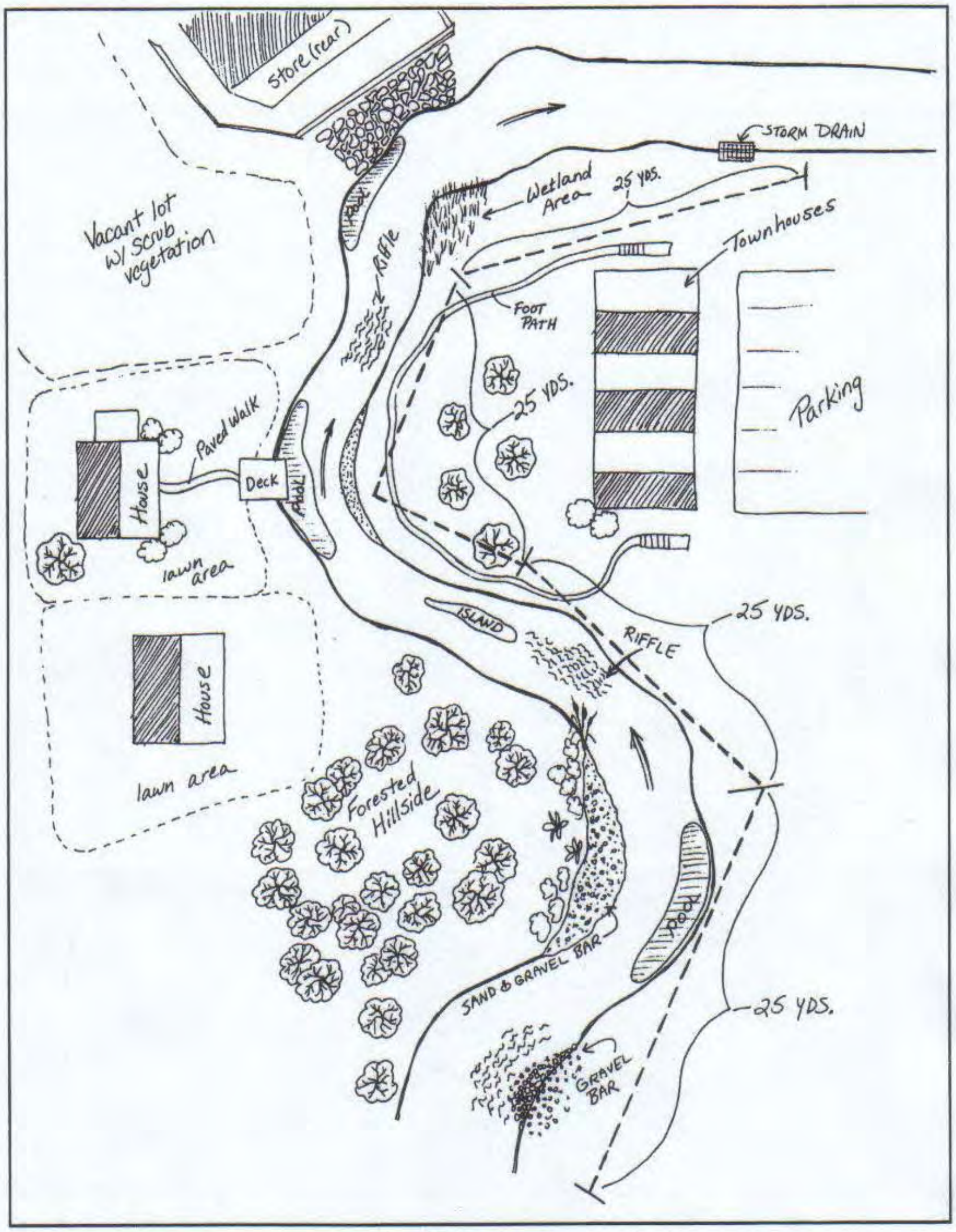
Latitude and longitude information is critical for mapping and for many data management programs. It is also required if the data is to be entered in USEPA's STOrage and RETrieval System (STORET) or used in a Geographical Information System (GIS).

An easy way to determine latitude and longitude is to use a global positioning system (GPS), a hand-held tool that looks like a calculator. GPS units receive signals form orbiting satellites and then use the information from the satellites to calculate the lat/long coordinates of the user. In

Figure 4.3

Example of a stream sketch

Volunteers should note important stream features on their sketch including riffles and pools.



general, these tools are accurate up to 15 meters. GPS units are relatively inexpensive and can be purchased from scientific supply houses and many camping or outdoor stores. Many government agencies are using GPS and might be able to loan a system to your program. Latitude and longitude can also be calculated manually using a USGS topographical map and a ruler (See Appendix C).

Step 3—Conduct the Stream Habitat Walk

Detailed instructions for performing the Stream Habitat Walk begin on page 48 of this section.

TASK 1 **Complete the habitat characterization components of the walk for the 25-yard section of stream: the "In-Stream Characteristics," "Stream Bank and Channel Characteristics," and "Local Watershed Characteristics" sections of the field data sheet**

These elements involve making observations about the stream itself as well as the riparian zone and immediate watershed.

TASK 2 **Complete the "Visual Biological Survey" section of the field data sheet**

This involves simple visual observations of the presence or absence of wildlife and obvious aquatic life in the stream, including fish, aquatic plants, and algae.

TASK 3 **Complete the "Macroinvertebrate Survey" section of the field data sheet**

This is optional and serves as an introduction to the types of life that inhabit some of the microhabitats of the stream—the spaces under and on rocks and in and on twigs and leaves. To conduct this survey, you will need to select the method(s) that best suits your stream. Use the rock-rubbing method in streams with riffles, or use the stick-picking method if your stream does not have riffles. Clumps of submerged leaves may be present in either type of stream and are often an important microhabitat for macroinvertebrates. You may choose to sort through these leaf packs in addition to rock–rubbing or stick–picking.

You will also need some specific equipment (a bucket, tweezers, picnic plate, etc.). Be sure to dress appropriately because you'll probably get wet.

Remember to return the organisms to the stream when you finish the macroinvertebrate survey. Then, check to make sure your field data sheet has been completed as fully as possible.

Step 4—Check data forms for completeness and return forms to program coordinator

After completing the habitat characterization and biological survey, make sure you have completed the field data sheet to the extent possible and that the recorded data are legible. If you are not able to determine how to answer a question on the field data sheet, just leave the space blank. If you leave a space blank, indicate that it is because you are not able to answer the question (e.g., write "not able to answer" or "does not apply" in the space).

Upon completion of the Stream Habitat Walk, present a copy of the field data sheet to your volunteer program coordinator. You may want to keep a copy of the field data sheet, and other appropriate data, for your own records and to evaluate any future discrepancies in the data. **If you have identified an urgent problem, such as leaking drums of chemicals, foul odors, or fish kills, contact your program coordinator or the agency with whom you are working as soon as possible.**

Instructions for completing the Stream Habitat Walk data sheet

For ease of use, the following numbered instructions correspond to the numbers on the field data sheet.

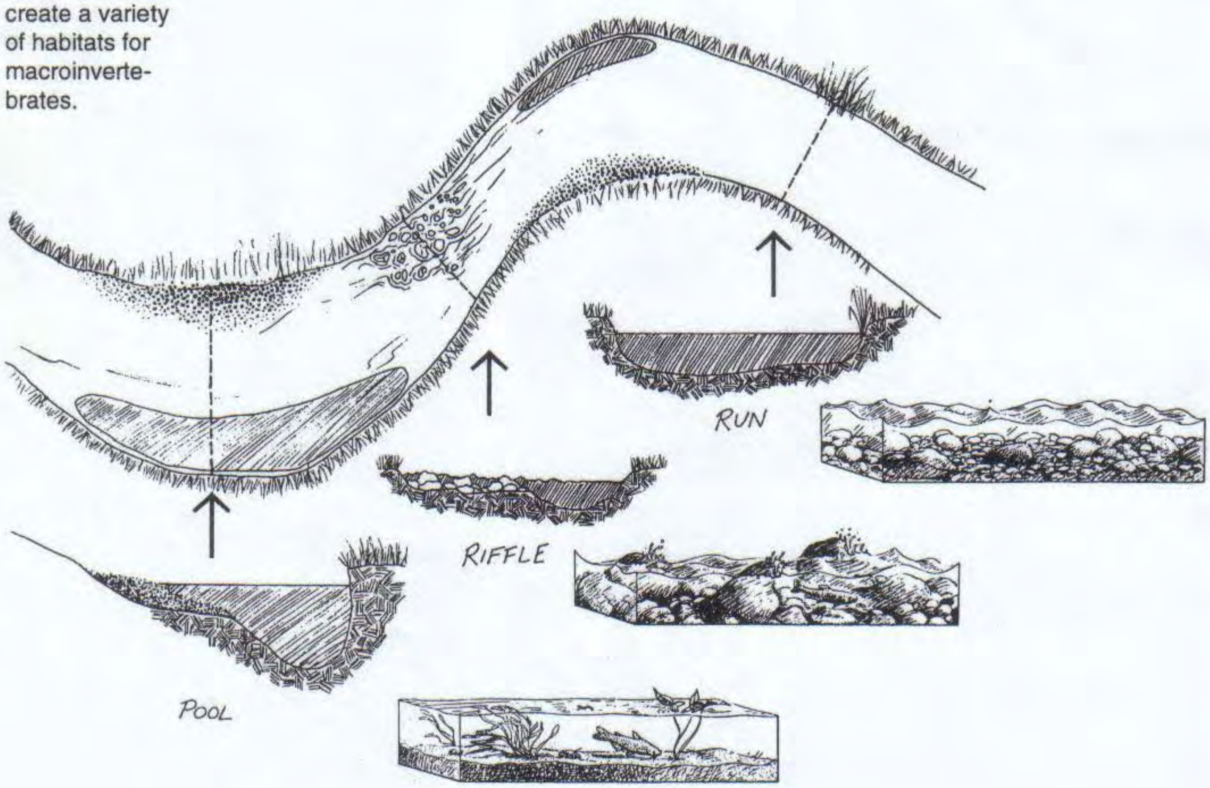
In-stream Characteristics

1. *Pools, riffles, and runs.* A mixture of flows and depths creates a variety of habitats to support fish and invertebrate life. Pools are deep with slow water. Riffles are shallow with fast, turbulent water running over rocks. Runs are deep with fast water and little or no turbulence.
2. *Stream bottom (substrate)* is the material on the stream bottom. Identify what substrate types are present. Substrate types include:
   - *Silt/clay/mud.* This substrate has a sticky, cohesive feeling. The particles are fine. The spaces between the particles hold a lot of water, making the sediments behave like ooze.
   - *Sand (up to 0.1 inch).* A sandy bottom is made up of tiny, gritty particles of rock that are smaller than gravel but coarser than silt (gritty, up to pea size).
   - *Gravel (0.1-2 inches).* A gravel bottom is made up of stones

Figure 4.4

Overview and cross sections of a pool, riffle, and run

Varying flows and depths create a variety of habitats for macroinvertebrates.



ranging from tiny quarter-inch pebbles to rocks of about 2 inches (fine gravel - pea size to marble size; coarse gravel - marble to tennis ball size).

- *Cobbles (2-10 inches).* Most rocks on this type of stream bottom are between 2 and 10 inches (between a tennis ball and a basketball).
- *Boulders (greater than 10 inches).* Most of the rocks on the bottom are greater than 10 inches (between a basketball and a car in size).
- *Bedrock.* This kind of stream bottom is solid rock (or rocks bigger than a car).

3. *Embeddedness* is the extent to which rocks (gravel, cobbles, and boulders) are sunken into the silt, sand, or mud of the stream bottom (Fig. 4.5). Generally, the more rocks are embedded, the less rock surface or space between rocks is available as habitat for aquatic macroinvertebrates and for fish spawning. Excessive silty runoff from erosion can increase a stream's embeddedness. To estimate embeddedness, observe the amount of silt or finer sediments overlying, in between, and surrounding the rocks.
4. *Presence of logs or woody debris (not twigs and leaves) in stream* can slow or divert water to provide important fish habitat such as pools and hiding places. Mark the box that describes the general amount of woody debris in the stream.
5. *Naturally occurring organic material in stream.* This material includes leaves and twigs. Mark the box that describes the general amount of organic matter in the stream.

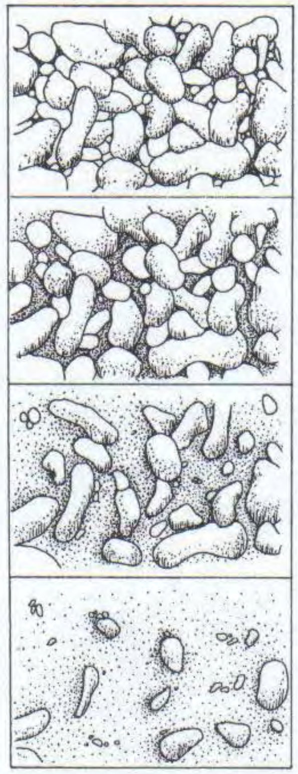


Figure 4.5

A representation of a rocky-bottom stream becoming embedded with sand and silt

As silt settles on the streambed, spaces between the rocks are filled in and the stream becomes more embedded.

6. *Water appearance* can be a physical indicator of water pollution.
   - *Clear* - colorless, transparent
   - *Milky* - cloudy-white or grey, not transparent; might be natural or due to pollution
   - *Foamy* - might be natural or due to pollution, generally detergents or nutrients (foam that is several inches high and does not brush apart easily is generally due to some sort of pollution)
   - *Turbid* - cloudy brown due to suspended silt or organic material
   - *Dark brown* - might indicate that acids are being released into the stream due to decaying plants
   - *Oily sheen* - multicolored reflection might indicate oil floating in the stream, although some sheens are natural

- *Orange* - might indicate acid drainage
- *Green* - might indicate excess nutrients being released into the stream

7. *Water odor* can be a physical indicator of water pollution
   - *No smell or a natural odor*
   - *Sewage* - might indicate the release of human waste material
   - *Chlorine* - might indicate over-chlorinated sewage treatment/ water treatment plant or swimming pool discharges
   - *Fishy* - might indicate the presence of excessive algal growth or dead fish
   - *Rotten eggs* - might indicate sewage pollution (the presence of methane from anaerobic conditions)

8. *Water temperature* can be particularly important for determining the suitability of the stream as aquatic habitat for some species of fish and macroinvertebrates that have distinct temperature requirements. Temperature also has a direct effect on the amount of dissolved oxygen available to the aquatic organisms. Measure temperature by submerging a thermometer for at least 2 minutes in a typical stream run. Repeat once and average the results.

Stream Bank and Channel Characteristics

9. *Depth of runs and pools* should be determined by estimating the vertical distance from the surface to the stream bottom at a representative depth at each of the two habitats.

10. *The width of the stream channel* can be determined by estimating the width of the streambed that is covered by water from bank to bank. If it varies widely, estimate an average width.

11. *Stream velocity* can have a direct influence on the health, variety, and abundance of aquatic communities. If water flows too quickly, organisms might be unable to maintain their hold on rocks and vegetation and be washed downstream; if water flows too slowly, it might provide insufficient aeration for species needing high levels of dissolved oxygen. Stream velocity can be affected by dams, channelization, terrain, runoff, and other factors. To measure stream velocity, mark off a 20-foot section of stream run and measure the time it takes a stick, leaf, or other floating biodegradable object to float the 10 feet. Repeat at least three times and pick the average time. Divide the distance (20 feet) by the average time (seconds) to determine the velocity in *feet per second.* (See Chapter 5, Section 5.1 on flow for a more in-depth discussion of using a float to estimate velocity.)

12. *The shape of the stream bank, the extent of artificial modifications, and the shape of the stream channel* are determined by standing at the downstream end of the 25-yard section and looking upstream.

   (a) The shape of the stream bank (Fig. 4.6) may include.
   - *Vertical or undercut bank* - a bank that rises vertically or overhangs the stream. This type of bank generally provides good cover for macroinvertebrates and fish and is resistant to erosion. If seriously undercut, it might be vulnerable to collapse.
   - *Steeply sloping* - a bank that slopes at more than a 30

degree angle. This type of bank is very vulnerable to erosion.

- *Gradual sloping* - a bank that has a slope of 30 degrees or less. Although this type of stream bank is highly resistant to erosion, it does not provide much streamside cover.

(b) *Artificial bank modifications* include all structural changes to the stream bank such as riprap (broken rock, cobbles, or boulders placed on earth surfaces such as the face of a dam or the bank of a stream, for protection against the action of the water) and bulk-heads. Determine the approximate percentage of each bank (both the left and right) that is artificially covered by the placement of rocks, wood, or concrete.

(c) *The shape of the stream channel* can be described as narrow (less than 6 feet wide from bank to bank), wide (more than 6 feet from bank to bank), shallow (less than 3 feet deep from the stream substrate to the top of the banks) or deep (more than 3 feet from the stream substrate to the top of the banks). Choose the category that best describes the channel.

- Narrow, deep
- Narrow, shallow
- Wide, deep
- Wide, shallow

13. *Streamside cover* information helps determine the quality and extent of the stream's riparian zone. This information is important at the stream bank itself and for a distance away from the stream bank. For example, trees, bushes, and tall grass can contribute shade and cover for fish and wildlife and can provide the stream with needed organic material such as leaves and twigs. Lawns indicate that the stream's riparian zone has been altered, that pesticides and grass clippings are a possible problem, and that little habitat and shading are available. Bare soil and pavement might indicate problems with erosion and runoff. Looking upstream, provide this information for the left and right banks of the stream.

- *Evergreen trees (conifers)* - cone-bearing trees that do not lose their leaves in winter.
- *Hardwood trees (deciduous)* - in general, trees that shed their leaves at the end of the growing season.

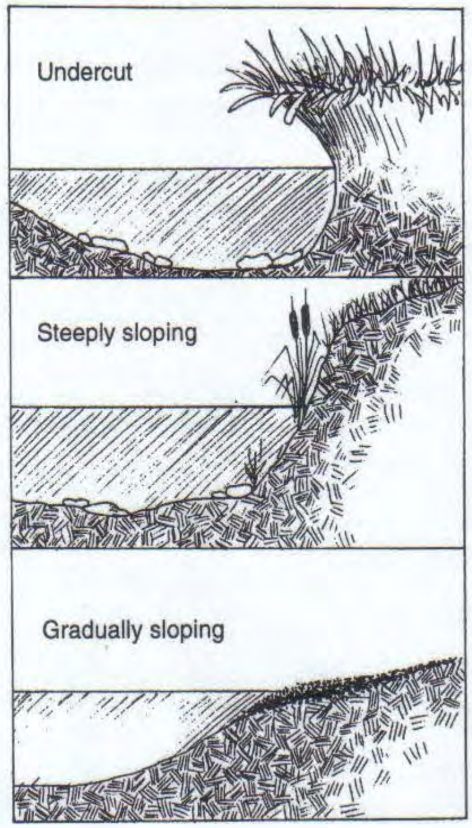


Figure 4.6

Types of streambank shapes

Undercut banks provide good cover for fish and macroinvertebrates.

- *Bushes, shrubs* - conifers or deciduous bushes less than 15 feet high.
- *Tall grass, ferns, etc.* - includes tall natural grasses, ferns, vines, and m 1osses.
- *Lawn* - cultivated and maintained short grass.
- *Boulders* - rocks larger than 10 inches.
- *Gravel/cobbles/sand* - rocks smaller than 10 inches; sand.
- *Bare soil*
- *Pavement, structure* - any structures or paved areas, including paths, roads, bridges, houses, etc.

14. *Stream shading* is a measurement of the extent to which the stream itself is overhung and shaded by the cover identified in 13 above. This shade (or *overhead canopy*) provides several important functions in the stream habitat. The canopy cools the water; offers habitat, protection, and refuge for aquatic organisms; and provides a direct source of beneficial organic matter and insects to the stream. Determine the extent to which vegetation shades the stream at your site.

15. *General conditions of the stream bank and stream channel, and other conditions* that might be affecting the stream are determined by standing at the downstream end of the 25-yard site and looking upstream. Provide observations for the right and left banks of the stream.

    (a) *Stream bank conditions* that might be affecting the stream.

    - *Natural plant cover degraded.* Note whether streamside vegetation is trampled or missing or has been replaced by landscaping, cultivation, or pavement. (These conditions could lead to erosion.)
    - *Banks collapsed/eroded.* Note whether banks or parts of banks have been washed away or worn down. (These conditions could limit habitats in the area.)
    - *Garbage/junk adjacent to the stream.* Note the presence of litter, tires, appliances, car bodies, shopping carts, and garbage dumps.
    - *Foam or sheen on bank.* Note whether there is foam or an oily sheen on the stream bank. Sheen may indicate an oil spill or leak, and foam may indicate the presence of detergent.

    (b) *Stream channel conditions* that might be affecting the stream.

    - *Mud/silt/sand on bottom/ entering stream.* Excessive mud or silt can interfere with the ability of fish to sight potential prey. It can clog fish gills and smother fish eggs in spawning areas in the stream bottom. It can be an indication of poor construction practices, urban area runoff, silviculture (forestry-related activities), or agriculture in the watershed. It can also be a normal condition in slow- moving, muddy-bottom streams.
    - *Garbage or junk in stream.* Note the presence of litter, tires, appliances, car bodies, shopping carts, and garbage.

    (c) *Other general conditions* that might be affecting the stream.

    - *Yard waste (e.g., grass clippings).* Is there evidence that grass clippings, cut branches, and other types of yard waste have been dumped into the stream?

- *Livestock in or with unrestricted access to stream.* Are livestock present, or is there an obvious path that livestock use to get to the water from adjacent fields? Is there streamside degradation caused by livestock?
- *Actively discharging pipes.* Are there pipes with visible openings discharging fluids or water into the stream? Note such pipes even though you may not be able to tell where they come from or what they are discharging.
- *Other pipes.* Are there pipes near or entering the stream? Note such pipes even if you cannot find an opening or see matter being discharged.
- *Ditches.* Are there ditches draining the surrounding land and leading into the stream?

Local Watershed Characteristics

16. *Adjacent land uses* can potentially have a great impact on the quality and state of the stream and riparian areas. Determine the land uses, based on your own judgment of the activities in the watershed surrounding your site within a quarter of a mile. Enter a "1" if a land use is present and a "2" if it is clearly having a negative impact on the stream.

Visual Biological Survey

17. *Wildlife* in or around the stream might indicate that the stream and its adjacent area are of sufficient quality to support animals with food, water, and habitat. Look for signs of frogs, turtles, snakes, ducks, deer, beaver, etc.
18. Are *fish* present in the stream? Fish can indicate that the stream is of sufficient quality for other organisms. Indicate the average size and note any visible barriers to the movement of fish—obstructions that would keep fish from moving freely upstream or downstream.
19. *Aquatic plants* provide food and cover for aquatic organisms. They also might provide very general indications of stream quality. For example, streams that are overgrown with plants could be over-enriched by nutrients. Streams devoid of plants could be affected by extreme acidity or toxic pollutants. Aquatic plants may also be an indicator of stream velocity because plants cannot take root in fast-flowing streams.
20. *Algae* are simple plants that do not grow true roots, stems, or leaves and that mainly live in water, providing food for the food chain. Algae may grow on rocks, twigs, or other submerged materials, or float on the surface of the water. It naturally occurs in green and brown colors. Excessive algal growth may indicate excessive nutrients (organic matter or a pollutant such as fertilizer) in the stream.

STREAM HABITAT WALK

Stream Name: ____________________

County: ____________________ **State:** ____________________

Investigators: ____________________

____

____

Site (description): ____________________

____

____

____

Latitude: ____________________ **Longitude:** ____________________

Site or Map Number: ____________________

Date: ____________________ **Time:** ____________________

Weather in past 24 hours:

- ❑ Storm (heavy rain)
- ❑ Rain (steady rain)
- ❑ Showers (intermittent rain)
- ❑ Overcast
- ❑ Clear/Sunny

Weather now:

- ❑ Storm (heavy rain)
- ❑ Rain (steady rain)
- ❑ Showers (intermittent rain)
- ❑ Overcast
- ❑ Clear/Sunny

Sketch of site

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.

PHYSICAL CHARACTERIZATION

In-Stream Characteristics

1. Check which stream habitats are present: Page 48

(You can check more than 1 habitat)

❑ Pool(s) θ Riffle(s) θ Run(s)

2. Nature of particles in the stream bottom at site Page 48

	None/Little	Some	Most
Silt/Clay/Mud	❑	❑	❑
Sand (up to 0.1" in diam.)	❑	❑	❑
Gravel (0.1 - 2" in diam.)	❑	❑	❑
Cobbles (2 - 10" in diam.)	❑	❑	❑
Boulders (over 10" in diam.)	❑	❑	❑
Bedrock (solid)	❑	❑	❑

3. Pick the category that best describes the extent to which gravel, cobbles, and boulders on the stream bottom are embedded (sunk) in silt, sand, or mud. Page 49

θ Somewhat/not embedded (0-25%) θ Mostly embedded (75%)

θ Halfway embedded (50%) θ Completely embedded (100%)

4. Presence of logs or large woody debris in stream: Page 49

θ None θ Occasional θ Plentiful

5. Presence of naturally-occurring organic material (i.e., leaves and twigs, etc.) in stream: Page 49

θ None θ Occasional θ Plentiful

6. Water appearance: Page 49

θ Clear θ Light brown θ Orange
θ Milky θ Dark brown θ Greenish
θ Foamy θ Oily sheen θ Other ______________
θ Turbid

7. Water odor: Page 50

θ Sewage θ Fishy θ None
θ Chlorine θ Rotten eggs θ Other ______________

8. Water temperature: Page 50

_________ °C *or* _________ °F

Streambank and Channel Characteristics

9. (a) Approximate depth of run(s): Page 50

θ < 1 ft θ 1-2 ft θ > 2 ft

(b) Approximate depth of pool(s):

θ < 1 ft θ 1-2 ft θ > 2 ft

10. Approximate width of stream channel: Page 50

_________ feet θ measured θ estimated

11. Stream velocity: _________ ft/sec. Page 50

12. Looking upstream (100 yds.), pick the description that best fits the shape of the stream bank and the channel. Page 50

(a) Stream bank:

Left		Right
θ	Vertical/undercut	θ
θ	Steeply sloping (> 30°)	θ
θ	Gradual/no slope (< 30°)	θ

(b) Extent of artificial bank modifications:

Left		Right
θ	Bank 0-25% covered	θ
θ	Bank 25-50% covered	θ
θ	Bank 50-75% covered	θ
θ	Bank 75-100% covered	θ

(c) Shape of the channel:

θ Narrow, deep θ Wide, deep
θ Narrow, shallow θ Wide, shallow

13. Looking upstream (100 yds.), describe the streamside cover. Check "1" if present, "2" if common Page 51

(a) Along water's edge and stream bank only:

Left 1	Left 2		Right 1	Right 2
θ	θ	Trees	θ	θ
θ	θ	Bushes, shrubs	θ	θ
θ	θ	Tall grasses, ferns, etc.	θ	θ
θ	θ	Lawn	θ	θ
θ	θ	Boulders/rocks	θ	θ
θ	θ	Gravel/sand	θ	θ
θ	θ	Bare soil	θ	θ
θ	θ	Pavement, structures	θ	θ

(b) From the top of the streambank out to 25 yards.

Left 1	Left 2		Right 1	Right 2
θ	θ	Trees	θ	θ
θ	θ	Bushes, shrubs	θ	θ
θ	θ	Tall grasses, ferns, etc.	θ	θ
θ	θ	Lawn	θ	θ
θ	θ	Boulders/rocks	θ	θ
θ	θ	Gravel/sand	θ	θ
θ	θ	Bare soil	θ	θ
θ	θ	Pavement, structures	θ	θ

14. Pick the category that best describes the extent to which vegetation shades the stream at your site. Page 52

θ 0% θ 25% θ 50% θ 75% θ 100%

15. Looking upstream, note general conditions. Check "1" if present, "2" if severe problem is clearly Page 52

Left 1	Left 2		Right 1	Right 2
1	**2**	**Stream Banks**	**1**	**2**
θ	θ	Natural streamside plant cover degraded	θ	θ
θ	θ	Banks collapsed/eroded	θ	θ
θ	θ	Garbage/junk adjacent to the stream	θ	θ
θ	θ	Foam or sheen on bank	θ	θ
1	**2**	**Stream Channel**	**1**	**2**
θ	θ	Mud, silt, or sand in or entering the stream	θ	θ
θ	θ	Garbage/junk in the stream	θ	θ
1	**2**	**Other**	**1**	**2**
θ	θ	Yard waste on bank (grass, clippings, etc.)	θ	θ
θ	θ	Livestock in or with unrestricted access to stream	θ	θ
θ	θ	Actively discharging pipe(s)	θ	θ
θ	θ	Other pipe(s) entering the stream	θ	θ
θ	θ	Ditches entering the stream	θ	θ

Local Watershed Characteristics

(within about 1/4 mile of the site; adjacent and upstream)

16. Land uses in the local watershed can potentially have an impact on a stream. Check "1" if present, "2" if clearly having an impact on the stream. Page 53

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

BIOLOGICAL CHARACTERIZATION

VISUAL BIOLOGICAL SURVEY

Page 53

17. Wildlife in or around the stream? *(Mark all that apply)*

θ Amphibians θ Waterfowl θ Reptiles θ Mammals

18. Fish in the stream? *(Mark all that apply)* Page 53

θ No θ Yes, but rare θ Yes, abundant

θ Small (1-2 in.) θ Medium (3-6 in.) θ Large (7 in. and above)

Are there any barriers to fish movement?

θ Beaver dams θ Waterfalls > 1' θ None

θ Dams θ Road barriers θ Other ________

19. Aquatic plants in the stream. *(Mark all that apply)* Page 53

θ None θ Occasional θ Plentiful

θ Attached θ Free-floating

θ Stream margin θ Pools θ Near riffle

20. Extent of algae in the stream. *(Mark all that apply)* Page 53

(a) Are the submerged stones, twigs, or other material in the stream coated with a layer of algal "slime"?

θ None θ Occasional θ Plentiful

θ Light coating θ Heavy coating

θ Brownish θ Greenish θ Other ________

(b) Are there any filamentous (string-like) algae?

θ None θ Occasional θ Plentiful

θ Brownish θ Greenish θ Other ________

(c) Are any detached "clumps" or "mats" of algae floating on the water's surface?

θ None θ Occasional θ Plentiful

θ Brownish θ Greenish θ Other ________

MACROINVERTEBRATE SURVEY *(Optional)*

21. If macroinvertebrates were collected from the stream bottom, which type of method/habitat was selected? Page 53

θ Rock-rubbing method: From cobbles and large stones selected from riffles.

θ Stick-picking method: From woody objects in streams with sandy, silty bottoms.

θ Leaf-pack sorting method: From submerged leaves in streams with either a rocky or sandy, silty bottom.

22. Are macroinvertebrates present? Page 54

θ No θ Yes, but rare θ Yes, abundant

23. If present, describe the types of macroinvertebrates found. Page 54

(Mark all that apply)

Wormlike	θ Occasional	θ Plentiful
Snails/clamlike	θ Occasional	θ Plentiful
Insects	θ Occasional	θ Plentiful
Crayfish	θ Occasional	θ Plentiful

COMMENTS: *(Note changes or potential problems such as spills, new construction, type of discharging pipes)*

4.2 Streamside Biosurvey

The Streamside Biosurvey is based on the simple macroinvertebrate sampling approach developed and used by the Ohio Department of Natural Resources and the Izaak Walton League of America's Save Our Streams program and adapted by many volunteer monitoring programs throughout the United States.

This assessment approach has two basic components. The first is a biosurvey of aquatic organisms that involves collecting and identifying macroinvertebrates in the field and calculating an index of stream quality. The second is the habitat characterization method known as the *Streamside Biosurvey Habitat Walk.*

Two methods of macroinvertebrate sampling are detailed in this section—one for rocky-bottom streams (using a kick net) and one for muddy-bottom streams (using a dip net). Figure 4.7 illustrates and describes the nets used for these assessments. Both of these aquatic organism collection procedures have been widely tested and used successfully by many groups. You should consult with a local aquatic scientist to determine which method is appropriate for streams in your area.

Like the Stream Habitat Walk described in Section 4.1, the Streamside Biosurvey is useful as a screening tool to identify water quality problems and as an educational tool to teach volunteers about pollution and stream ecology. But instead of randomly picking up rocks or sticks and brushing off macroinvertebrates for simple observation purposes, Streamside Biosurvey volunteers are trained to use special nets and standardized sampling protocols to collect organisms from a measured area of stream habitat. Volunteers identify collected organisms, usually to the order level, and sort them into taxonomic groups based on their ability to tolerate pollution. Using this information, volunteers can then calculate a simple stream quality rating of good, fair, or poor.

Because the Streamside Biosurvey involves a standardized sampling protocol, a basic level of training, professional assistance, and a simple stream rating based on macroinvertebrate diversity and abundance, this approach is more effective than the Stream Habitat Walk in characterizing stream health and determining general water quality trends over several years. However, this method is not generally suited to determining subtle pollution impacts due, in part, to its uncomplicated level of macroinvertebrate identification

Note

The Streamside Biosurvey is based on protocols developed and widely used by programs such as the Ohio Department of Natural Resources, the Izaak Walton League of America, and others. This manual recommends some modifications to their established protocols. These include:

- A finer mesh size for the kick and dip nets used to sample for macroinvertebrates
- In rocky-bottom streams, compositing three samples into one before identifying macroinvertebrates rather than identifying macroinvertebrates in three separate samples and choosing the best result. Compositing generally provides a more representative sample of the macroinvertebrate community than a discrete sample taken from one part of the riffle. Riffle areas have what is known as a patchy distribution of organisms, meaning that different types of organisms are naturally found in different parts of the riffle. In order to more accurately assess the macroinvertebrate community in a rocky-bottom stream site, it is important to take a representative sample that includes organisms found in different microhabitats—such as in different parts of the riffle or in riffles of various flows and depths.
- A new method for calculating the stream quality rating. This modification incorporates a weighting factor to take into account the abundance of organisms in each pollution tolerance category (pollution-sensitive, somewhat tolerant, and tolerant).
- In muddy-bottom streams, varying how much each habitat type is sampled depending on its abundance at the sampling site.

and analysis. This, of course, is also one of the Streamside Biosurvey's greatest strengths, since volunteers can be easily trained in its methods.

Key features of the Streamside Biosurvey are as follows:

- It includes the Streamside Biosurvey Habitat Walk as its physical habitat characterization and visual biological characterization components. This protocol is a somewhat more detailed version of the Stream Habitat Walk described in Section 4.1.
- It centers around a macroinvertebrate survey in which organisms are collected according to specific protocols, identified in the field (generally to taxonomic order), and are then released back into the stream.
- For the identification process, volunteers group macroinvertebrates into three categories based on their pollution tolerance or sensitivity. Volunteers then calculate a water quality index by counting the specimens in each sensitivity category and determining whether they are rare, common, or dominant; multiplying the number of taxa in each category by a weighting factor; adding all the scores; and comparing results to a water quality rating scale that has been determined by a locally knowledgeable biologist/ecologist.
- The Streamside Biosurvey requires some equipment and training. Training can be conducted at the stream site, although some advance preparation is required. For example, a biologist with regional experience should assist in developing the macroinvertebrate key and the tolerance category groupings on the field data sheets. A reference collection is recommended to help volunteers identify macroinvertebrates.

Step 1—Prepare for the Streamside Biosurvey field work

Much of the preparation work for this approach is similar to that of the Stream Habitat Walk (section 4.1). Refer back to that section for relevant information on the following tasks:

- Scheduling the biosurvey
- Obtaining a USGS topographical map
- Selecting and marking monitoring locations
- Becoming familiar with safety procedures

TASK 1 Gather tools and equipment for the Streamside Biosurvey

In addition to the basic equipment listed in Section 2.4, you should collect the following equipment needed for the macroinvertebrate collection of the Streamside Biosurvey:

- Vial with tight cap filled about one-half full with 70 percent ethyl alcohol
- Buckets (2)
- Hand lens, magnifying glass, or field microscope
- Tweezers, eyedropper, or spoon
- Plastic bag
- Large, shallow, white pans, such as dishpans (2)
- Spray water bottle
- Plastic ice cube tray
- Taxonomic key to aquatic organisms
- Calculator
- *For rocky-bottom streams*—Kick net, a fine mesh (500 µm) nylon net approximately 3x3 feet with a 3-foot long supporting pole on each side is recommended—Fig.4.7).

Nets recommended in this manual

Kick net

For rocky-bottom stream sampling, a kick net of 590 µm (a #30 mesh size) or 500 µm (#35 mesh size) is recommended. (Mesh size is usually measured in microns, µm. The higher the number, the coarser the mesh.)

D-frame net

For muddy-bottom stream sampling, a long-handled D-frame or dip net is recommended for reaching into vegetation that grows along stream banks or is attached to the stream bottom, and for sweeping up macroinvertebrates dislodged from woody debris. D-frame nets also come in different mesh sizes.

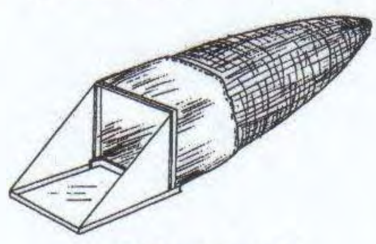
This manual recommends that volunteer programs purchase their macroinvertebrate sampling nets from scientific supply houses to ensure a standard degree of net quality and known mesh size. Some supply houses might sell the components of the net separately. Volunteer programs then buy the net material commercially, supply their own handles, and build the nets using volunteer labor.

Many programs use coarser mesh than is recommended in this manual. Coarser mesh is generally less expensive. However, smaller organisms can be lost through the mesh during sampling. If you are in doubt as to what mesh size to use, consult your technical advisor. If possible—and especially if you want your volunteer data to be used by state and local water managers—it is best to use nets of the same type and size as those which water quality professionals use in your state.

Other types of commonly used nets

Rectangular-style kick net

Used by the West Virginia Save Our Streams Program in both rocky-bottom and muddy-bottom streams.



Surber sampler

Used by professional monitoring programs, this sampler delineates an exact stream bottom area to be disturbed.

Figure 4.7

Examples of macroinvertebrate sampling nets

Nets used by professionals and volunteers vary in overall size, design, and mesh size.

- *For muddy-bottom streams*—D-frame net (a dip net with a frame 12 inches wide with a fine nylon mesh, usually about 500 µm, attached to the frame).

Step 2—Collect and Sort Macroinvertebrates

The method you use to collect macroinvertebrates using this approach depends on the type of stream you are sampling. Rocky-bottom streams are defined as those with bottoms made up of gravel, cobbles, and boulders in any combination and usually have definite riffle areas. Riffle areas are fairly well oxygenated and, therefore, are prime habitats for benthic macroinvertebrates. In these streams, use the *rocky-bottom sampling method.*

Muddy-bottom streams have muddy, silty, or sandy bottoms and lack riffles. Generally, these are slow moving, low-gradient streams (i.e., streams that flow along relatively flat terrain). In such streams, macroinvertebrates generally attach themselves to overhanging plants, roots, logs, submerged vegetation, and stream substrate where organic particles are trapped. In these streams, use the *muddy-bottom sampling method.*

Both methods are detailed below. Regardless of which collection method is used, the process for counting, identifying, and analyzing the macroinvertebrate sample for the Streamside Biosurvey is the same.

Rocky-Bottom Sampling Method

Use the following method of macroinvertebrate sampling in streams that have riffles and gravel/cobble substrates. You will collect three samples at each site and composite (combine) them to obtain one large total sample.

TASK 1 **Identify the sampling location**

You should have already located your site on a map along with its latitude and longitude (see Task 3, page 45).

1. You are going to sample in three different spots within a 100-yard stream reach. These spots may be three separate riffles; one large riffle with different current velocities; or, if no riffles are present, three run areas with gravel or cobble substrate. Combinations are also possible (if, for example, your site has only one small riffle and several run areas).

   Mark off your 100-yard stream reach. If possible, it should begin at least 50 yards upstream of any human-made modification of the channel, such as a bridge, dam, or pipeline crossing, Avoid walking in the stream, since this might dislodge macroinvertebrates and alter your sampling results.
2. Sketch the 100-yard sampling area. Indicate the location of your three sampling spots on the sketch. Mark the most downstream site as Site 1, the middle site as Site 2, and the upstream site as Site 3. (See Fig. 4.8.)

TASK 2 **Get into place**

1. **Always approach your sampling locations from the downstream end and sample the site farthest downstream first (Site 1)** (see Fig. 4.9, Panel #1). This minimizes the possibility of biasing your second and third collections with dislodged sediment or macroinvertebrates.

   Always use a clean kick net, relatively free of mud and debris from previous uses. Fill a bucket about one third full with stream water and fill your spray bottle.

2. Select a 3-foot by 3-foot riffle area for sampling at Site 1. One member of the team, the net holder, should position the net at the downstream end of this sampling area. Hold the net handles at a 45 degree angle to the water's surface (see Fig. 4.9, Panel #2). Be sure that the bottom of the net fits tightly against the streambed so no macroinvertebrates escape under the net. You may use rocks from the sampling area to anchor the net against the stream bottom. Don't allow any water to flow over the net.

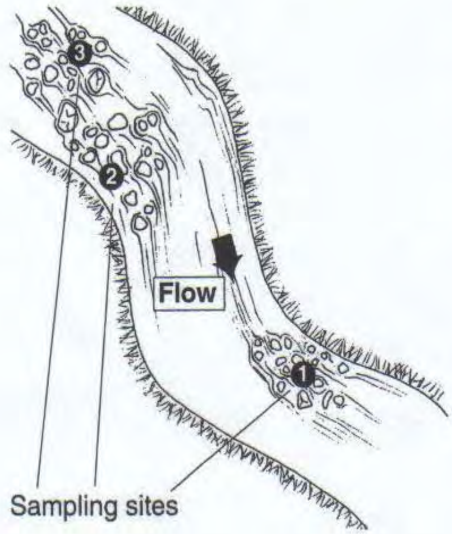


Figure 4.8

Location of sample sites in a rocky-bottom stream with riffles

Within a 100 yard reach volunteers begin their sampling at the most downstream site and then work their way upstream.

TASK 3 **Dislodge the macroinvertebrates**

1. Pick up any large rocks in the 3-foot by 3-foot sampling area and rub them thoroughly over the partially-filled bucket so that any macroinvertebrates clinging to the rocks will be dislodged into the bucket (see Fig. 4.9, Panel #3). Then place each cleaned rock outside of the sampling area. After sampling is completed, rocks can be returned to the stretch of stream they came from.
2. The member of the team designated as the "kicker" should thoroughly stir up the sampling area with their feet, starting at the upstream edge of the 3-foot by 3-foot sampling area and working downstream, moving toward the net. All dislodged organisms will be carried by the stream flow into the net (see Fig. 4.9, Panel #4). Be sure to disturb the first few inches of stream sediment to dislodge burrowing organisms. As a guide, disturb the sampling area for about 3 minutes, or until the area is thoroughly worked over.
3. Any large rocks used to anchor the net should be thoroughly rubbed into the bucket as above.

TASK 4 **Remove the net**

1. Next, remove the net without allowing any of the organisms it contains to wash away. While the net holder grabs the top of the net handles, the kicker grabs the bottom of the net handles and the net's bottom edge. Remove the net from the stream with a forward scooping motion (see Fig. 4.9, Panel #5).
2. Roll the kick net into a cylinder shape and place it vertically in the partially filled bucket. Pour or spray water down the net to flush its contents into the bucket (see Fig. 4.9, Panel #6). If necessary, pick debris and organisms from the net by hand. Release back into the stream any fish, amphibians, or reptiles caught in the net.

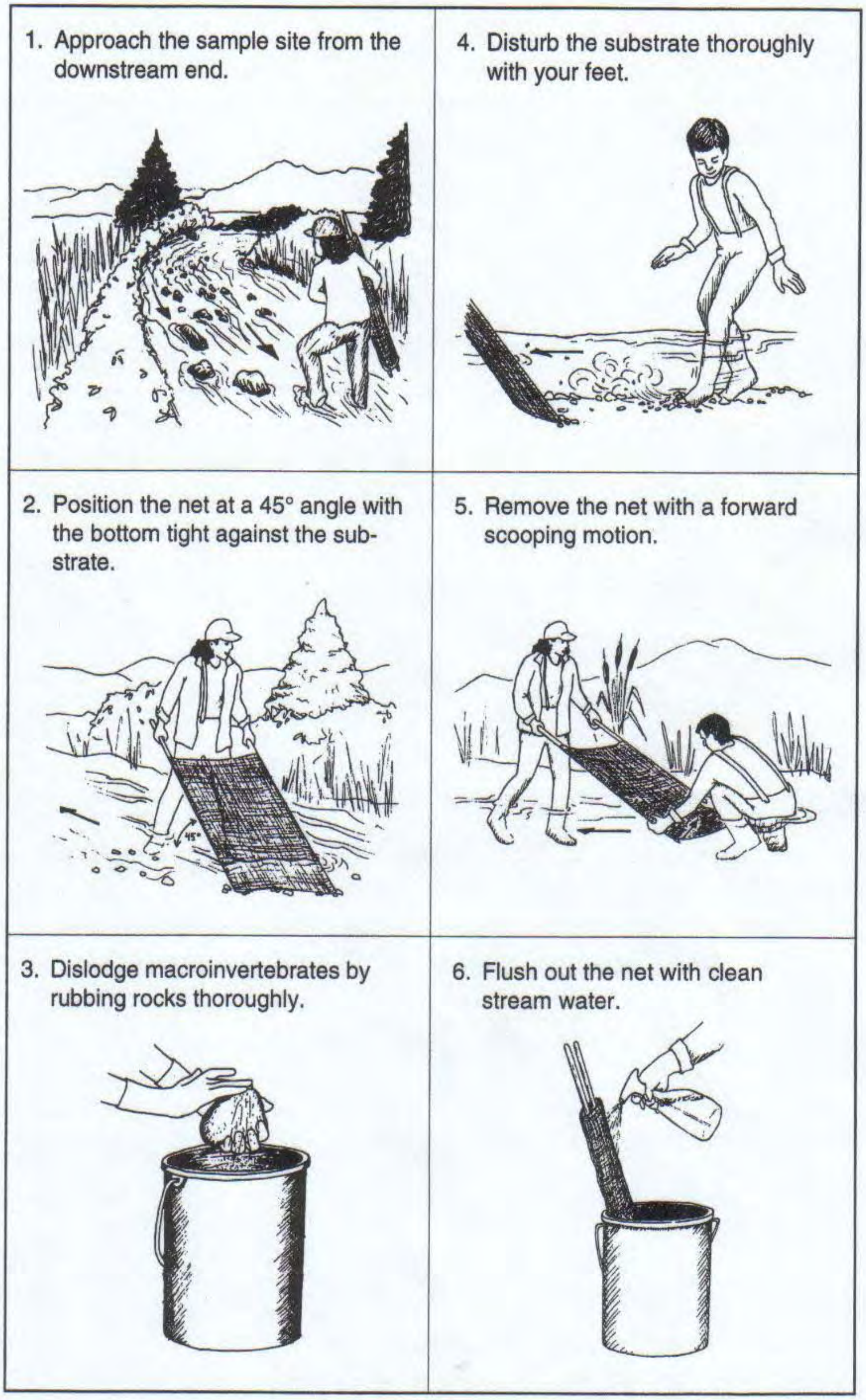
TASK 5 **Collect the second and third samples**

Once you have removed all the organisms from the net repeat these tasks at Sites 2 and 3. Put the samples from all three sites into the same bucket. Combining the debris and organisms from all three sites into the same bucket is called *compositing*.

Figure 4.9

Procedures for collecting a macroinvertebrate sample in a rocky-bottom stream

Volunteers must follow set protocol to collect an unbiased sample.



West Virginia Save Our Streams recommends that rocks should be rubbed in the stream directly in front of the slightly into the net. Bucket-rubbing is only used during very low water levels.

Hint: If your bucket is nearly full of water after you have washed the net clean, let the debris and organisms settle to the bottom of the bucket. Then cup the net over the bucket and pour the water through the net into a second bucket. Inspect the water in the second bucket to be sure no organisms came through.

TASK 6 **Sort macroinvertebrates**

Pour the contents of the bucket into a large, shallow, white pan. Add some stream water to the pan, and fill the ice cube tray with stream water. Using tweezers, eye dropper, or spoon, pick through the leaf litter and organic material looking for anything that swims, crawls, or seems to be hiding in a shell, like a snail. Look carefully; many of these creatures are quite small and fast-swimming. Sort similar organisms into the ice cube tray.

Note: Instructions for counting, identifying, and analyzing the macroinvertebrate sample follow the muddy-bottom sampling method. (See page 70, Step 3)

Muddy-Bottom Sampling Method

In muddy-bottom streams, as in rocky-bottom streams, the goal is to sample the most productive habitats available and look for the widest variety of organisms. The most productive habitats are the ones that harbor a diverse population of pollution sensitive-macroinvertebrates. Volunteers should sample by using a D-frame net to jab at the habitat and scoop up the organisms that are dislodged. The objective is to collect a combined sample from 20 jabs taken from a variety of habitats.

TASK 1 **Determine which habitats are present**

Picking Bugs

Some monitoring programs find it easier to collect organisms from the net by hand-picking them rather than washing the sample into a pan and then trying to pick through the floating debris. The advantage to placing the organisms in a pan is that they are more likely to survive while in the pan and their characteristic movements will help in organism identification.

If you prefer to pick bugs directly off the net, a white background, such as a white plastic trash bag under the net, will help you see the bugs more clearly. In addition, periodically wetting the net with a water bottle will help keep the bugs alive and moving.

Identification can be made easier if you sort the organisms into groups based on physical similarities and place them together in sections of an ice cube tray as you pick them from the pan or net.

Muddy-bottom streams usually have four habitats (Fig. 4.10). It is generally best to concentrate sampling efforts on the most productive habitat available, yet to sample other principal habitats if they are present. This ensures that you will secure as wide a variety of organisms as possible. Not all habitats are present in all streams or present in significant amounts. If your sampling areas have not been preselected, try to determine which of the following habitats are present. (Avoid standing in the stream while making your habitat determinations.)

- *Vegetated bank margins.* This habitat consists of overhanging bank vegetation and submerged root mats attached to banks. The bank margins may also contain submerged, decomposing leaf packs trapped in root wads or lining the streambanks. This is generally a highly productive habitat in a muddy–bottom stream, and it is often the most abundant type of habitat.
- *Snags and logs.* This habitat consists of submerged wood, primarily dead trees, logs, branches, roots, cypress knees and leaf packs lodged between rocks or logs. This is also a very productive muddy-bottom stream habitat.

- *Aquatic vegetation beds and decaying organic matter.* This habitat consists of beds of submerged, green/leafy plants that are attached to the stream bottom. This habitat can be as productive as vegetated bank margins, and snags and logs.
- *Silt/sand/gravel substrate.* This habitat includes sandy, silty, or muddy stream bottoms; rocks along the stream bottom; and/or wetted gravel bars. This habitat may also contains algae-covered rocks (sometimes called Aufwuchs). This is the least productive of the four muddy-bottom stream habitats, and it is always present in one form or another (e.g., silt, sand, mud, or gravel might predominate).

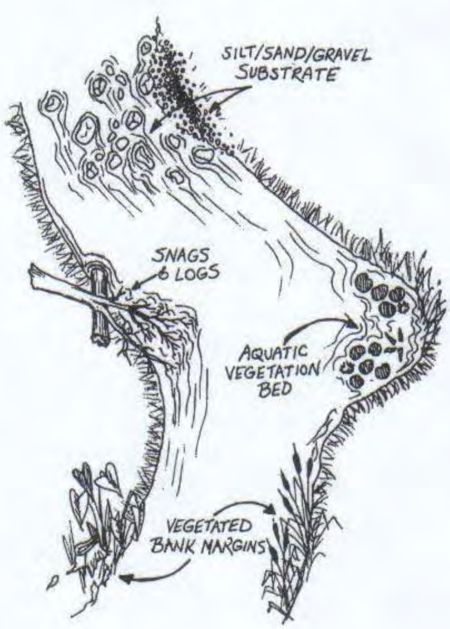
TASK 2 **Determine how many times to jab in each habitat type**

Your goal is to jab a total of 20 times. The D-frame net is 1 foot wide, and a jab should be approximately 1 foot in length. Thus, 20 jabs equals 20 square feet of combined habitat.

Figure 4.10

Four habitats found in muddy-bottom streams

Volunteers will likely find the most macroinvertebrates in vegetated habitats and snags and logs.



- If all four habitats are present in plentiful amounts, jab the vegetated banks 10 times and divide the remaining 10 jabs among the remaining 3 habitats.
- If three habitats are present in plentiful amounts and one is absent, jab the silt/sand/gravel substrate—the least productive habitat—5 times and divide the remaining 15 jabs among the other two more productive habitats.
- If only two habitats are present in plentiful amounts, the silt/sand/gravel substrate will most likely be one of those habitats. Jab the silt/sand/gravel substrate 5 times and the more productive habitat 15 times.
- If some habitats are plentiful and others are sparse, sample the sparse habitats to the extent possible, even if you can take only one or two jabs. Take the remaining jabs from the plentiful habitat(s). This rule also applies if you cannot reach a habitat because of unsafe stream conditions. Jab a total of 20 times.

Because you might need to make an educated guess to decide how many jabs to take in each habitat type, it is critical that you note, on the field data sheet, how many jabs you took in each habitat. This information can be used to help characterize your findings.

TASK 3 **Get into place**

Outside and downstream of your first sampling location (1st habitat), rinse the dip net and check to make sure it does not contain any macroinvertebrates or debris from the last time it was used. Fill a bucket approximately one-third full with clean stream water. Also, fill the spray bottle with clean stream water. This bottle will be used to wash down the net between jabs and after sampling is completed.

This method of sampling requires only one person to disturb the stream habitats. While one person is sampling, a second person should stand outside the sampling area, holding the bucket and spray bottle. After every few jabs, the sampler should hand the net to the second person, who then can rinse the contents of the net into the bucket.

TASK 4 **Dislodge the macroinvertebrates**

Approach the first sample site from downstream, and sample as you walk upstream. Here is how to sample in the four habitat types:

- Sample vegetated bank margins by jabbing vigorously, with an upward motion, brushing the net against vegetation and roots along the bank. The entire jab motion should occur underwater.
- To sample snags and logs, hold the net with one hand under the section of submerged wood you are sampling. With the other hand (which should be gloved), rub about 1 square foot of area on the snag or log. Scoop organisms, bark, twigs, or other organic matter you dislodge into your net. Each combination of log rubbing and net scooping is one jab (Fig. 4.11).
- To sample aquatic vegetation beds, jab vigorously, with an upward motion, against or through the plant bed. The entire jab motion should occur underwater.
- To sample a silt/sand/gravel substrate, place the net with one edge against the stream bottom and push it forward about a foot (in an upstream direction) to dislodge the first few inches of silt, sand, gravel, or rocks. To avoid gathering a netful of mud, periodically sweep the mesh bottom of the net back and forth in the water, making sure that water does not run over the top of the net. This will allow fine silt to rinse out of the net.

When you have completed all 20 jabs, rinse the net thoroughly into the bucket. If necessary, pick any clinging organisms from the net by hand and put them in the bucket.

Figure 4.11

Collecting a sample from a log

Volunteer rubs the log with one hand and catches dislodged organisms and other material in the net.

TASK 5 **Sort the macroinvertebrates**

Pour the contents of the bucket (water, organisms, and organic material) into a large, shallow, white pan and fill the ice cube tray with clean stream water. Using tweezers, eye dropper, or spoon, pick through the leaf litter and organic material looking for anything that swims, crawls, or seems to be hiding in a shell (like a snail). Look carefully; many of these creatures are quite small and fast-swimming. Sort similar organisms into the plastic ice cube tray.

Step 3—Identify Macroinvertebrates and Calculate Stream Rating

The following methods are used for both the rocky- and muddy-bottom assessments.

Task 1 **Identify Macroinvertebrates**

1. Identify the collected macroinvertebrates. Using the hand lens or magnifying glass and the aquatic organism identification key, carefully observe the collected macroinvertebrates. Refine your initial sort so that like individuals are placed in the same section(s) of the ice cube tray. If you cannot identify an organism, place one or two specimens in the alcohol-filled vial and forward it to your program coordinator for identification.
2. On your field data sheet, note the number of individuals of each type of organism you have identified (Section 3 of the field data sheet—See Fig. 4.12.).

Note: When you feel that you have identified all the organisms to the best of your ability, return the macroinvertebrates to the stream.

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3. Assign one of the following abundance codes to each type of organism. Record the code next to the actual count on the field data sheet.

R (rare)	=	if 1-9 organisms are found in the sample
C (common)	=	if 10-99 organisms are found in the sample
D (dominant)	=	if 100 or more organisms are found in the sample

Your field data sheet should be organized to help you sort macroinvertebrates into three groups based on their ability to tolerate pollution. **A local authority (such as a state biologist or entomologist) should determine which organisms belong in each pollution tolerance category for your region.**

Generally, the three tolerance groups are as follows:

- *Group I* (sensitive organisms) includes pollution- sensitive organisms such as mayflies, stoneflies, and non net-spinning caddisflies, which are typically found in good-quality water.
- *Group II* (somewhat sensitive organisms) includes somewhat pollution-tolerant organisms such as net-spinning caddisflies, crayfish, sowbugs, and clams, found in fair-quality water.
- *Group III* (tolerant organisms) includes pollution-tolerant organisms such as worms, leeches, and midges, found in poor-quality water.

TASK 2 **Calculate the stream quality rating**

The stream water quality rating takes into account the pollution sensitivity of the organisms and their relative abundance. This is accomplished through use of a weighting system.

Figure 4.12

Sample macro-invertebrate count for (hypothetical) Volunteer Creek

MACROINVERTEBRATE COUNT

Identify the macroinvertebrates in your sample and assign them letter codes based on their abundance: R (rare) = 1-9 organisms; C (common) = 10-99 organisms; and D (dominant) = 100 plus organisms.

Group I Sensitive	Group II Somewhat-Sensitive	Group III Tolerant
C (50) Water penny larvae	*R (4)* Beetle larvae	*R (5)* Aquatic worms
R (2) Hellgrammites	____ Clams	____ Blackfly larvae
____ Mayfly nymphs	____ Crane fly larvae	____ Leeches
____ Gilled snails	*R (6)* Crayfish	____ Midge larvae
____ Riffle beetle adult	____ Damselfly nymphs	*C (50)* Snails
C (25) Stonefly nymphs	*D (100)* Scuds	
____ Non net-spinning caddisfly larvae	*D (150)* Sowbugs	
	R (8) Fishfly larvae	
	____ Alderfly larvae	
	C (27) Net-spinning caddisfly larvae	

The weighting system acknowledges the most desirable combinations of pollution sensitivity and abundance by assigning these extra weights within a 5, 3, and 1 point scale. Pollution-sensitive organisms receive a weighting factor based on a 5-point scale. Somewhat sensitive organisms are weighted on a 3-point scale, and tolerant organisms are weighted on a 1-point scale. As can be seen in Table 4.2, a sample's ideal combination of organisms would be "sensitive" and "somewhat sensitive" organisms in common abundance (10-99 organisms), and pollution "tolerant" organisms in rare abundance (less than 10 organisms). This is because it is never ideal for any given type of organism to dominate a sample, and because it is best to have a wide variety of organisms including a few pollution-tolerant individuals.

1. Add the number of R's, C's and D's in each of the 3 pollution tolerance groupings. Then, for each grouping, multiply the total number of R's, C's and D's by the relevant weighting factor. Table 4.3 illustrates sample calculations for determining the water quality rating for (hypothetical) Volunteer Creek.

Note: The tolerance category groupings shown on the Biosurvey Data Sheet were developed for streams in the mid-Atlantic (Maryland, Virginia, West Virginia, District of Columbia, Pennsylvania). These groupings may not totally apply in other regions of the United States. **It is important that a local aquatic biologist take a look at these categories and make any changes necessary for your region.**

In addition, depending on the level of taxonomic training volunteers receive, you might consider separating out some other families of organisms. For instance, the tolerance groupings given here separate caddisflies into net-spinning and non net-spinning families. Mayflies might also be separated into different tolerance groupings. It is not recommended here, however, because of the difficulty in distinguishing mayfly families in the field without a microscope.

Some volunteer programs, like the one coordinated by the Audubon Naturalist Society in Maryland, conduct intensive field identification training workshops and teach volunteers to distinguish several families in the field. Creating more specific tolerance groupings may be an option for your program if you have the resources and expertise to conduct more intensive taxonomic field training.

Table 4.2

Weighting factors used in calculating stream water quality ratings

Abundance	Weighting Factor		
	Group I Sensitive	Group II Somewhat Sensitive	Group III Tolerant
Rare (R)	5.0	3.2	1.2
Common (C)	5.6	3.4	1.1
Dominant (D)	5.3	3.0	1.0

Table 4.3

Sample calculations of index values for Volunteer Creek

Group I Sensitive	Group II Somewhat Sensitive	Group III Tolerant
1 (No. of R's) x 5.0 = 5.0	3 (No. of R's) x 3.2 = 9.6	1 (No. of R's) x 1.2 = 1.2
2 (No. of C's) x 5.6 = 11.2	1 (No. of C's) x 3.4 = 3.4	1 (No. of C's) x 1.1 = 1.1
	2 (No. of D's) x 3.0 = 6.0	
Index Value for Group I = 16.2	Index Value for Group II = 19.0	Index Value for Group III = 2.3

Table 4.4

Tentative rating scale for streams in Maryland

Score	Rating
>40	Good
20-40	Fair
< 20	Poor

2. To obtain a water quality rating for the site, total the values for each group and add them together. The total score for the sample stream site is: 16.2 (Group I) + 19.0 (Group II) + 2.3 (Group III) = 37.5.
3. The final step is to compare the score to water quality ratings (good to poor) established by a trained biologist familiar with local stream fauna. Table 4.4 presents a tentative rating scale for streams in Maryland. Assuming Volunteer Creek is located in Maryland, the stream would receive a rating of "Fair."

Note: In addition to adjusting the rating scale according to regional location, it might also need to be adjusted for muddy-bottom vs. rocky-bottom streams. An experienced stream biologist can calculate the best rating system for your area's streams by examining data from several streams.

In a healthy stream, the sensitive (Group I) organisms will be well represented in a sample. It is important to remember that macroinvertebrate populations can fluctuate seasonally and that these natural fluctuations can affect your results. Therefore, it is best to compare the results by season from year to year. (Compare your spring sampling results to each other, not to fall results.)

WV Save Our Streams rates the invertebrate community differently than the example provided above. The organisms are rated according to an abundance rating scale and the number of different kinds are also determined. Using this method, a modified biotic index and two diversity indices can be calculated.

Step 4—Conduct the Streamside Biosurvey: Habitat Walk

You will conduct a habitat assessment (which will include measuring general characteristics and local land use) in a 100-yard section of stream that includes the riffles from which organisms were collected.

TASK 1 **Delineate the habitat assessment boundaries**

1. Begin by identifying the most downstream riffle that was sampled for macroinvertebrates. Using your tape measure or twine, mark off a 100-yard section extending 25 yards below the downstream riffle and about 75 yards upstream.
2. Complete the identifying information on your field data sheet for your habitat assessment site. On your stream sketch, be as detailed as possible, and be sure to note which riffles were sampled.

TASK 2 **Complete the Physical Characteristics, Local Watershed Characteristics, and Visual Biological Survey sections of the field sheet**

For safety reasons as well as to protect the stream habitat, it is best to estimate these characteristics rather than actually wading into the stream to measure them.

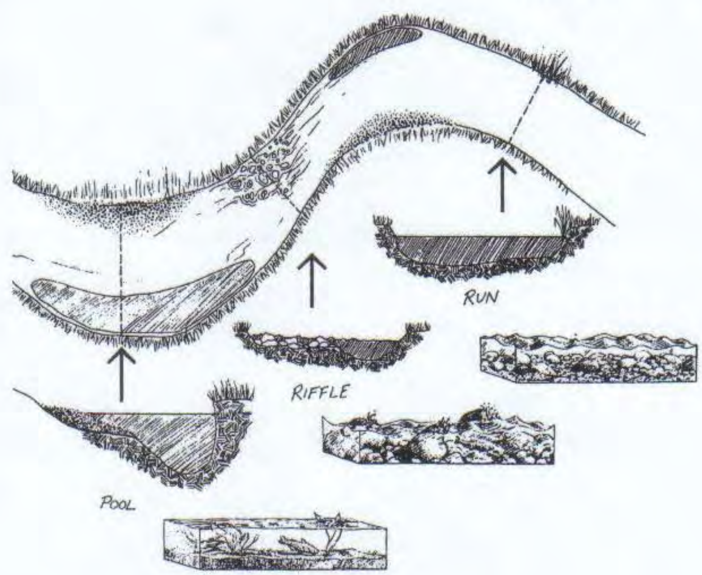
In-stream Characteristics

1. *Pools, riffles, and runs* create a mixture of flows and depths and provide a variety of habitats to support fish and invertebrate life. Pools are deep with slow water. Riffles are shallow with fast, turbulent water running over rocks. Runs are deep with fast water and little or no turbulence.
2. *Stream bottom (substrate)* is the material on the stream bottom. Identify what substrate types are present. Substrate types include:
   - *Silt/clay/mud*—This substrate has a sticky, cohesive feeling. The particles are fine. The spaces between the particles hold a lot of water, making the sediments behave like ooze.
   - *Sand (up to 0.1 inch)*—A sandy bottom is made up of tiny, gritty particles of rock that are smaller than gravel but coarser than silt (gritty, up to pea size).
   - *Gravel (0.1-2 inches)*—A gravel bottom is made up of stones ranging from tiny quarter-inch pebbles to rocks of about 2 inches (fine gravel - pea size to marble size; coarse gravel - marble to tennis ball size).
   - *Cobbles (2-10 inches)*—Most rocks on this type of stream bottom are between 2 and 10 inches (between a tennis ball and a basketball).

Figure 4.13

Overview and cross sections of a pool, riffle, and run

Varying flows and depths create a variety of habitats for macroinvertebrates.



- *Boulders (greater than 10 inches)*—Most of the rocks on the bottom are greater than 10 inches (between a basketball and a car in size).
- *Bedrock*—is solid rock (or rocks bigger than a car).

Estimate the percentage of substrate types at your site.

3. *Embeddedness* is the extent to which rocks (gravel, cobbles, and boulders) are sunken into the silt, sand, or mud of the stream bottom (Fig. 4.14). Generally, the more rocks are embedded, the less rock surface or space between rocks is available as habitat for aquatic macroinvertebrates and for fish spawning. Excessive silty runoff from erosion can increase the embeddedness in a stream. To estimate the embeddedness, observe the amount of silt or finer sediments overlying, in between, and surrounding the rocks.
4. *Streambed stability* can provide additional clues to the amount of siltation in a stream. When you walk in the stream, note whether your feet sink significantly into sand or mud.
5. *Presence of logs or woody debris (not twigs and leaves) in stream* can slow or divert water to provide important fish habitat such as pools and hiding places. Mark the box that describes the general amount of woody debris in the stream.
6. *Naturally occurring organic material in stream.* This material includes leaves and twigs. Mark the box that describes the general amount of organic matter in the stream.
7. *Water appearance* can be a physical indicator of water pollution.
   - *Clear* - colorless, transparent
   - *Milky* - cloudy-white or grey, not transparent; might be natural or due to pollution
   - *Foamy* - might be natural or due to pollution, generally detergents or nutrients (foam that is several inches high and does not brush apart easily is generally due to some sort of pollution)
   - *Turbid* - cloudy brown due to suspended silt or organic material
   - *Dark brown* - might indicate that acids are being released into the stream due to decaying plants
   - *Oily sheen* - multicolored reflection might indicate oil floating in the stream, although some sheens are natural
   - *Orange* - might indicate acid drainage
   - *Green* - might indicate excess nutrients being released into the stream
8. *Water odor* can be a physical indicator of water pollution
   - *No smell or a natural odor*
   - *Sewage* - might indicate the release of human waste material
   - *Chlorine* - might indicate over-chlorinated sewage treatment/ water treatment plant or swimming pool discharges
   - *Fishy* - might indicate the presence of excessive algal growth or dead fish
   - *Rotten eggs* - might indicate sewage pollution (the presence of methane from anaerobic conditions)
9. *Water temperature* can be particularly important for determining the suitability of the stream as aquatic habitat for some species of fish and macroinvertebrates that have distinct

temperature requirements. Temperature also has a direct effect on the amount of dissolved oxygen available to the aquatic organisms. Measure temperature by submerging a thermometer for at least 2 minutes in a typical stream run. Repeat once and average the results.

Stream Bank and Channel Characteristics

10. *Depth of runs and pools* should be determined by estimating the vertical distance from the surface to the stream bottom at a representative depth at each of the two habitats.
11. *The width of the stream channel* can be determined by estimating the width of the streambed that is covered by water from bank to bank. If it varies widely, estimate an average width.
12. *Stream velocity* can have a direct influence on the health, variety, and abundance of aquatic communities. If water flows too quickly, insects might be unable to maintain their hold on rocks and vegetation and be washed downstream; if water flows too slowly, it might provide insufficient aeration for species needing high levels of dissolved oxygen. Stream velocity can be affected by dams, channelization, terrain, runoff, and other factors. To measure stream velocity, mark off a 20-foot section of stream run and measure the time it takes a stick, leaf, or other floating biodegradable object to float the 20 feet. Repeat 5 times and pick the average time. Divide the distance (20 feet) by the average time (seconds) to determine the velocity in feet per second. (See Chapter 5, Section 1 on flow for a more in-depth discussion on using floats to estimate velocity.)

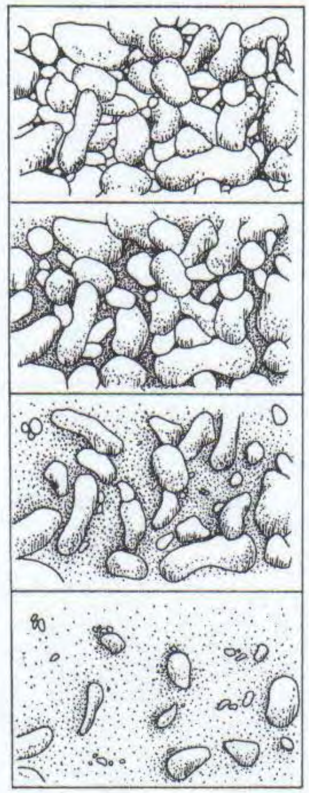


Figure 4.14

A representation of a rocky-bottom stream becoming embedded with sand and silt

As silt settles on the streambed, spaces between the rocks are filled in and the stream becomes more embedded.

13. *The shape of the stream bank, the extent of artificial modifications, and the shape of the stream channel* are determined by standing at the downstream end of the 25-yard section and looking upstream.

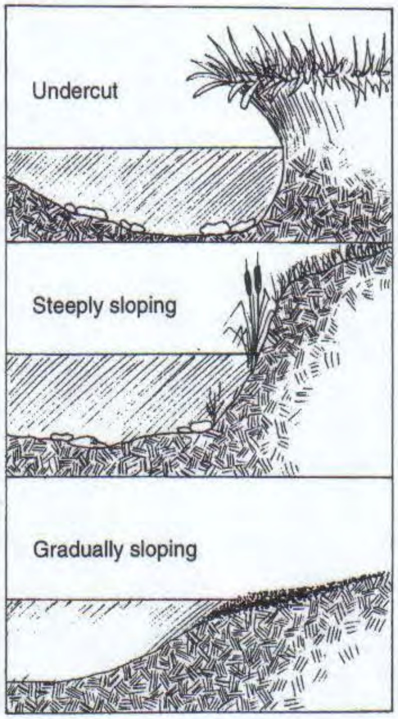
(a) The *shape of the stream bank* (Fig. 4.15) may include.

- *Vertical or undercut bank* - a bank that rises vertically or overhangs the stream. This type of bank generally provides good cover for macroinvertebrates and fish and is resistant to erosion. However, if seriously undercut, it might be vulnerable to collapse.
- *Steeply sloping* - a bank that slopes at more than a 30 degree angle. This type of bank is very vulnerable to erosion.

Figure 4.15

Types of streambank shapes

Undercut banks provide good cover for fish and macroinvertebrates.



- *Gradual sloping* - a bank that has a slope of 30 degrees or less. Although this type of stream bank is highly resistant to erosion, it does not provide much streamside cover.

(b) *Artificial bank modifications* include all structural changes to the stream bank such as riprap (broken rock, cobbles, or boulders placed on earth surfaces such as the face of a dam or the bank of a stream, for protection against the action of the water) and bulkheads. Determine the approximate percentage of each bank (both the left and right) that is artificially covered by the placement of rocks, wood, or concrete.

(c) *The shape of the stream channel* can be described as narrow (less than 6 feet wide from bank to bank), wide (more than 6 feet from bank to bank), shallow (less than 3 feet deep from the stream substrate to the top of the banks) or deep (more than 3 feet from the stream substrate to the top of the banks). Choose the category that best describes the channel.

- Narrow, deep
- Narrow, shallow
- Wide, deep
- Wide, shallow

14. *Streamside cover* information helps determine the quality and extent of the stream's riparian zone. This information is important at the stream bank itself and for a distance away from the stream bank. For example, trees, bushes, and tall grass can contribute shade and cover for fish and wildlife and can provide the stream with needed organic material such as leaves and twigs. Lawns indicate that the stream's riparian zone has been altered, that pesticides and grass clippings are a possible problem, and that little habitat and shading are available. Bare soil and pavement might indicate problems with erosion and runoff. Looking upstream, provide an estimate of the percentage of the stream bank (left and right stream banks) covered by the following:

- *Trees*
- *Bushes, shrubs* - conifers or deciduous bushes less than 15 feet high
- *Tall grass, ferns, etc.* - includes tall natural grasses, ferns, vines, and mosses
- *Lawn* - cultivated and maintained short grass
- *Boulders* - rocks larger than 10 inches
- *Gravel/cobbles/sand* - rocks smaller than 10 inches; sand

- *Bare soil*
- *Pavement, structure* - any man-made structures or paved areas, including paths, roads, bridges, houses, etc.

15. *Stream shading* is a measurement of the extent to which the stream itself is overhung and shaded by the cover identified in 14 above. This shade (or *overhead canopy*) provides several important functions in the stream habitat. It cools the water; offers habitat, protection, and refuge for aquatic organisms; and provides a direct source of beneficial organic matter and insects to the stream. Determine the extent that vegetation shades the stream at the site.

16. *General conditions of the stream bank and stream channel, and other conditions* that might be affecting the stream are determined by standing at the downstream end of the 25-yard site and looking upstream. Provide observations for the right and left banks of the stream.

(a) *Stream bank conditions* that might be affecting the stream.

- *Natural plant cover degraded*—note whether streamside vegetation is trampled or missing or has been replaced by landscaping, cultivation, or pavement. (These conditions could lead to erosion.)
- *Banks collapsed/eroded*—note whether banks or parts of banks have been washed away or worn down. (These conditions could limit habitats in the area.)
- *Garbage/junk* adjacent to the stream—note the presence of litter, tires, appliances, car bodies, shopping carts, and garbage dumps.
- *Foam or sheen on bank*—note whether there is foam or an oily sheen on the stream bank. Sheen may indicate an oil spill or leak, and foam may indicate the presence of detergent.

(b) *Stream channel conditions* that might be affecting the stream.

- *Mud/silt/sand on bottom/entering stream*—can interfere with the ability of fish to sight potential prey. It can clog fish gills and smother fish eggs in spawning areas in the stream bottom. It can be an indication of poor construction practices, urban area runoff, silviculture (forestry-related activities), or agriculture in the watershed. It can also be a normal condition, especially in a slow-moving, muddy-bottom stream.
- *Garbage or junk in stream*—note the presence of litter, tires, appliances, car bodies, shopping carts, and garbage.

(c) *Other general conditions* that might be affecting the stream.

- *Yard waste* (e.g., grass clippings)—is there evidence that grass clippings, cut branches, and other types of yard waste have been dumped into the stream?
- *Livestock in or with unrestricted access to stream*—are livestock present, or is there an obvious path that livestock use to get to the water from adjacent fields? Is there streamside degradation caused by livestock?
- *Actively discharging pipes*—are there pipes with visible openings discharging fluids or water into the stream? Note such pipes even though you may not be able to tell where they come from or what they are discharging.

- *Other pipes*—are there pipes near or entering the stream? Note such pipes even if you cannot find an opening or see matter being discharged.
- *Ditches*—are there ditches, draining the surrounding land and leading into the stream?

Local watershed characteristics

17. *Adjacent land uses* can potentially have a great impact on the quality and state of the stream and riparian areas. Determine the land uses, based on your own judgment of the activities in the watershed surrounding your site within a quarter of a mile. Enter a "1" if a land use is present and a "2" if it is clearly having a negative impact on the stream.

Visual biological survey

18. Are *fish* present in the stream? Fish can indicate that the stream is of sufficient quality for other organisms.
19. *Barriers to the movement of fish* in the stream are obstructions that would keep fish from moving freely upstream or downstream.
20. *Aquatic plants* provide food and cover for aquatic organisms. Plants also might provide very general indications of stream quality. For example, streams that are overgrown with plants could be over enriched by nutrients. Streams devoid of plants could be affected by extreme acidity or toxic pollutants. Aquatic plants may also be an indicator of stream velocity because plants cannot take root in fast-flowing streams.
21. *Algae* are simple plants that do not grow true roots, stems, or leaves and that mainly live in water, providing food for the food chain. Algae may grow on rocks, twigs, or other submerged materials, or float on the surface of the water. It naturally occurs in green and brown colors. Excessive algal growth may indicate excessive nutrients (organic matter or a pollutant such as fertilizer) in the stream.

Step 4—Complete all the field data sheets

After you have completed macroinvertebrate sampling, analysis of findings, and the habitat characterization, make sure you have completed the field data sheet to the extent possible and that the recorded data are legible. If you are not able to determine how to answer a question on the field data sheet, just leave the space blank. Return all completed forms to your program coordinator.

Level-One Survey Data Sheet

(**1**) Determine your stream-reach boundary; this is a stream length up to 100-meters, which may be more or less under certain circumstances. (**2**) Near the lower end of the reach (in the deepest portion of the run), collect water samples and analyze using the chemical tests you have available. You may use your collection container to observe watercolor and clarity and to determine water odors. (**3**) Measure the width-depth and velocity, and estimate the water level. (**4**) Using a **kick-net**, collect a minimum of three benthic macro-invertebrate samples from the best riffles or runs within your stream reach. Use the tally sheet on page three to record information about your collections. (**5**) Evaluate the physical and habitat conditions, and record information about known land use activities. (**6**) Sketch your reach or submit photographs with the survey, and add any other comments that you feel are important for evaluating the conditions of your study site.

Stream name ______________________ Survey date ____________

Watershed ______________________ County ____________

Latitude ____________ Longitude ____________ Directions ____________

Start/end times ____________

Survey completed by ______________________ Station code ____________

Affiliation ______________________ E-mail ____________

Mailing address ______________________ Phone number ____________

Water chemistry: Use the boxes below to record the results of your water chemistry analysis; attach additional sheets if necessary.

	Result	units		Result	units		Result	units
Temperature (C/F)			Conductivity			Alkalinity		
Dissolved oxygen			Nitrate/Nitrite			Metals (describe)		
pH			Phosphate			Fecal/E-coli		

Additional tests (describe and record results) ______________________

Physical conditions: Use the check boxes below to describe the conditions that closely resemble those of your stream. The extra lines are provided to write in any additional comments. You may see more than one type of condition; if so, be sure to indicate these on your survey (check all that apply). If multiple conditions are observed, always indicate the most dominant condition. Note: If the condition you observe is not listed, describe it in the comment section.

Water clarity		Water color		Water odor		Surface foam	
Clear		None		None		None	
Murky		Brown		Fishy		Slight	
Milky		Black		Musky		Moderate	
Muddy		Orange/red		Rotten egg		High	
Other (describe)		Gray/White		Sewage			
		Green		Chemical			

Algae color		Algae abundance		Algae growth habit		Streambed color	
Light green		None		Even coating		Brown	
Dark green		Scattered		Hairy		Black	
Brown		Moderate		Matted		Green	
Other (describe)		Heavy		Floating		White/gray	
						Orange/red	

Physical condition comments: ______________________

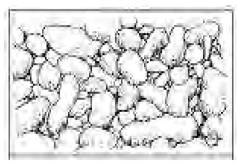
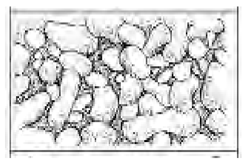
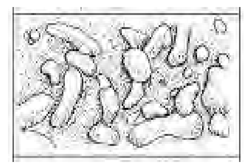
Estimate the % of your reach that is shaded	> 80	80 - 60	60 - 40	< 40
	Excellent	Good	Fair	Poor

Circle your estimate

Width and depth measurements: Record the wetted width and average depth from at least one of the channel's habitats (riffle, run or pool). Record the average depth from a minimum of five measurements (one of these should be from the deepest part of the habitat). The width should be measured from the widest section of the feature. Be sure to indicate the type(s) of habitat that you have chosen. **It is best to complete this task during your discharge measurements**.

				Riffle	Run	Pool
1. Width (feet)	______	Depth (feet)	______			
2. Width (feet)	______	Depth (feet)	______			

Habitat conditions: Rate the habitat conditions by choosing the best description for the reach. Bank stability and riparian buffer width are assessed on both the **left** and **right** side of the stream. Indicate your choice by writing **O**, **S**, **M** or **P** in the spaces provided.

Point values	8	6	4	2
Embeddedness	 Fine sediments surrounds <10% of the spaces between the gravel, cobble and boulders.	 Fine sediment surrounds 10-30% of the spaces between the gravel, cobble and boulders.	 Fine sediment surrounds 30-60% of the spaces between the gravel, cobble and boulders.	 Fine sediment surrounds > 60% of the spaces between the gravel, cobble and boulders.
	Optimal	**S**uboptimal	**M**arginal	**P**oor

Embeddedness should be evaluated in riffles/runs prior to or during your macroinvertebrate collections.

Sediment deposition	Little or no formation of depositional features; < 20% of the reach affected. **See below for examples**	Some increase in depositional features; 20-40% of the reach affected.	Moderate amounts of depositional features; 40-60% of the reach affected.	Heavy amounts of deposition; > 60% of the reach affected.
	Optimal	**S**uboptimal	**M**arginal	**P**oor

The next two conditions are evaluated on both the left and the right sides of the stream.

Point values	4	3	2	1
Bank stability	Banks are stable; no evidence of erosion or bank failure; little or no potential for future problems; < 10% of the reach affected.	Banks are moderately stable; infrequent areas of erosion occur, mostly shown by banks healed over or a few bare spots; 10-30 % of the reach affected.	Banks are moderately unstable; 30-50% of the reach has some areas of erosion; high potential for erosion during flooding events.	Banks are unstable; many have eroded areas (bare soils) along straight sections or bends; obvious bank collapse or failure; > 50% affected.
	Optimal	**S**uboptimal	**M**arginal	**P**oor
Riparian buffer width	Mainly undisturbed vegetation > 60 ft; no evidence of human impacts such as parking lots, road beds, clear-cuts, mowed areas, crops, lawns etc.	Zone of undisturbed vegetation 40-60 ft; some areas of disturbance evident.	Zone of undisturbed vegetation 20-40 ft; disturbed areas common throughout the reach.	Zone of undisturbed vegetation < 20 ft; disturbed areas common throughout the entire reach.
	Optimal	**S**uboptimal	**M**arginal	**P**oor
Totals	> 26	26 – 20	19 – 13	< 13
	Optimal	Suboptimal	Marginal	Poor

Habitat condition comments: ____________________

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.

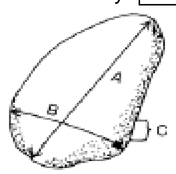
Station code ____________ Date(s) ____________

Streambed composition: You should always collect information about the composition of your reach. You can either estimate the proportions or you use a **pebble count** for a more accurate measure of composition. At a minimum you should estimate composition of the riffles within your reach. The size categories are determined by the (B) axis measured in millimeters. Did you estimate ______ or count ______ ? Use the table below to record your data.

Woody debris ______
Includes sticks, leaves etc.

Silt/clay < 0.06	Sand 0.06 – 2	Fine gravel 2 - 24	Coarse gravel 25 - 64	Cobble 65 - 255	Boulder 256 - 1096	Bedrock > 1096
Very small; having a smooth slick feel	Very small; having a grainy feel	Pea to tennis ball		Tennis ball to basketball	Basketball to car size	Usually larger than a car; solid surface

Riffle only ______ Entire reach ______



(A) Long axis (**Length**)
(B) Intermediate axis (**Width**)
(C) Short axis (**Height**)

Pebble counts require two people, one in the stream and one on shore. The person in the stream walks upstream from bank to bank using a zigzag pattern. After each step the person reaches down without looking, picks up the first particle touched, and measures the intermediate axis with a ruler. The on-shore partner records the measurement. The process continues until **100** pebbles have been measured or the reach has been walked. For a quick estimate, the coordinator recommends that **50** be collected from the entire reach and **20** if collecting from riffles only.

Land use: Indicate the land uses that you believe may be having an impact on your stream station. Use the letters (**S**) streamside, (**M**) within ¼ mile and (**W**) somewhere in the watershed, to indicate the approximate location of the disturbance and the numbers (**1**) slight, (**2**) moderate or (**3**) high, to represent the level of disturbance.

Active construction			Pastureland			Single-family residences		
Mountaintop mining			Cropland			Sub-urban developments		
Deep mining			Intensive feedlots			Parking lots, strip-malls etc.		
Abandoned mining			Unpaved Roads			Paved Roads		
Logging			Trash dumps			Bridges		
Oil and gas wells			Landfills			Other (describe)		
Recreation (parks, trails etc.)			Industrial areas					

Pipes? Yes No

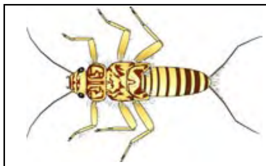
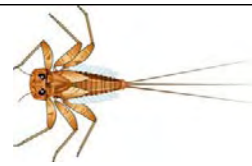
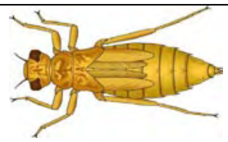
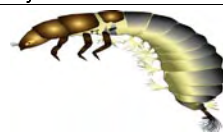
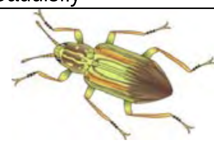
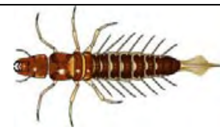
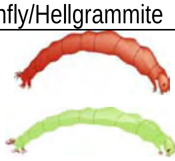
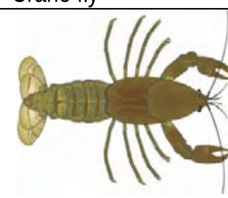
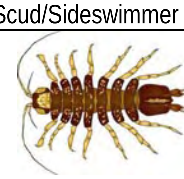
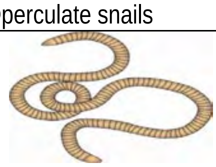
Describe the types of pipes observed and indicate if there is any discharge from the pipes. Also describe the colors and odors of the discharge, and provide any other land-use comments. ________________________________

____

Photograph and **sketch your study reach**: Use the space below to draw your study reach. Indicate the direction of flow, sample locations and important features of the reach. Photographs are an excellent method for tracking changes, especially changes related to the condition of the habitat. Choose at least two locations from which to take your photos and submit your photos with your survey data sheet.

Level-one survey data sheet

Benthic macroinvertebrates: Use the table below to record information about your collections. Record their abundance using these codes: (**A**) > **50**, (**C**) **5** – **50** and (**R**) < **5** and also record the number of different kinds. The **# of kind's** box indicates groups in which multiple kinds (families) are possible. If collected, include the free-living caddisfly with the other net-spinners. Illustrations courtesy of the Cacapon Institute; Jennifer Gillies, artist

 Stoneflies	# of kinds	 Mayflies	# of kinds	 Caddisflies	**Case-builders** # of kinds
 Dragonflies	# of kinds	 Caddisfly	**Common netspinner**	 Caddisflies	**Other net-spinners** # of kinds
 Damselflies	# of kinds	 Riffle beetle		 Water penny	
 Fishfly/Hellgrammite		 Alderfly		 Other beetles	# of kinds
 Midges		 Black fly		 Crane fly	
 Watersnipe fly		 Other flies	# of kinds	 Crayfish	
 Clams	# of kinds	 Mussel		 Scud/Sideswimmer	
 Operculate snails	# of kinds	 Non-operculate snails	# of kinds	 Aquatic sowbug	
 Aquatic worm		 Leech		 Flatworm	

Other aquatic life observed or collected: ______________________________

Station code ____________________

Date(s) ____________________

Stream Score

After the sorting and identifications is complete, the macroinvertebrates are assessed using four metrics. First, transform your abundance rating into numbers using this code (**A** = **6; C** = **3**; **R** = **1**) and follow the instructions below to complete all calculations. The light gray shading indicates that multiple kinds are possible.

1. **Biotic Index**: Multiply the abundance number by the tolerance value to calculate the tolerance score. Add the entire tolerance score column and the abundance column. Divide the tolerance total by the abundance total.
2. **Total Taxa**: Calculate the total number of kinds.
3. **EPT Taxa**: Calculate the total number of kinds from the stoneflies, mayflies, and all caddisflies.

The final step is to determine a **point value** for each metric. These points are added together to determine your overall **stream score** and integrity rating.

Benthic macroinvertebrates	Abundance	Tolerance Value	Tolerance Score	Number of Kinds
Stoneflies (Order Plecoptera)		2		
Mayflies (Order Ephemeroptera)		3		
Case-building caddisflies (Order Trichoptera)		3		
Net-spinning caddisflies (Order Trichoptera)		4		
Common netspinner (Family Hydropsychidae)		5		
Dragonflies (Sub-order Anisoptera)		4		
Damselflies (Sub-order Zygoptera)		7		
Riffle beetle (Family Elmidae)		4		
Water penny (Family Psephenidae)		3		
Other beetles (Order Coleoptera)		6		
Hellgrammite (Family Corydalidae)		3		
Alderfly (Family Sialidae)		6		
Non-biting midge (Family Chironomidae)		8		
Black fly (Family Simuliidae)		6		
Crane fly (Family Tipulidae)		4		
Watersnipe fly (Family Athericidae)		3		
Other true flies (Order Diptera)		7		
Water mite (Order Hydrachnida)		6		
Crayfish (Order Decapoda)		5		
Sideswimmer (Order Amphipoda)		5		
Aquatic sowbug (Order Isopoda)		7		
Operculate snails (Sub-class Prosobranchia)		4		
Non-operculate snails (Sub-class Pulmonata)		7		
Clams (Order Veneroida)		6		
Mussel (Family Unionidae)		4		
Aquatic worm (Class Oligochaeta)		10		
Leech (Class Hirudinea)		10		
Flatworm (Class Turbellaria)		7		
Other invertebrates (describe)	Total Abundance		Total Tolerance	Total Taxa (Kinds)

Metrics	Results	**Points**	**10**	**7**	**5**	**3**
1. Total Taxa			> 18	18 - 13	12 - 8	< 8
2. EPT Taxa			> 10	10 - 7	6 - 4	< 4
3. Biotic Index			< 4.0	4.0 – 5.0	5.1 – 6.0	> 6.0

Stream Score ____

Integrity Rating

> 24	24 - 19	18 - 12	< 12
Optimal	Suboptimal	Marginal	Poor

Station code ____________________

Date(s) ____________________

Discharge: Determine the discharge by using a flow meter (if available) or other methods such as the float method or the velocity head rod method (VHR). Discharge should always be measured from a **run**. The more measurements collected the more accurate your discharge results will be. To convert inches into feet divide by 12. For example, if your depth measurement was 6-inches the result in feet would be 0.5. Indicate the method and use the tables to record your results.

Discharge method used

☐ Float ☐ Velocity Head Rod ☐ Flow meter

Channel width ______________ feet

Water Level

☐ Low ☐ Normal ☐ High ☐ Dry

Tape distance (ft)	Depth (ft)	Velocity (ft/sec)	VHR (Rise-inches)	Float (sec)	Discharge (cfs)
1					
2					
3					
4					
5					
6					
7					
8					
9					
10					
11					
12					
13					
14					
15					
16					
Totals/Averages					

Cross Sectional Area (CSA) ______________ ft^2

(CSA = Average Depth x Width)

Discharge = CSA x Velocity

= ______________ x ______________

= ______________ cfs (ft^3/sec)

If you use a float record your distance below and the number of seconds it took to travel the distance in the column indicated.

Float distance (feet) ____________

VHR rises and velocities.

Rise (R)	Velocity	Rise (R)	Velocity
¼	1.2	3 ¼	4.2
½	1.6	3 ½	4.3
¾	2.0	3 ¾	4.5
1	2.3	4	4.6
1 ¼	2.6	4 ¼	4.8
1 ½	2.8	4 ½	4.9
1 ¾	3.1	4 ¾	5.0
2	3.3	5	5.2
2 ¼	3.5	5 ¼	5.3
2 ½	3.7	5 ½	5.4
2 ¾	3.8	5 ¾	5.5
3	4.0	6	5.7

Submit a clear copy or the original data sheet to the coordinator at address below. If you submit the original, always keep a copy for your records.

West Virginia Dept. of Environmental Protection
Save Our Streams Program
601 57th Street, SE
Charleston, WV 25304

If you have questions or comments contact the program coordinator by E-mail at: timothy.d.craddock@wv.gov, call (304) 926-0499 Ext. 1040 or visit the program's web page at: http://www.dep.wv.gov/sos.

Station code ______________

Date(s) ______________

4.3 Intensive Stream Biosurvey

The Intensive Stream Biosurvey is based on the habitat assessment and macroinvertebrate sampling approach developed by EPA in its *Rapid Bioassessment Protocols for Streams and Rivers* (Protocol II) and adapted by volunteer monitoring programs such as Maryland Save Our Streams and River Watch Network.

Like the Stream Habitat Walk and Streamside Biosurvey, this approach includes a study of macroinvertebrates and habitat. However, the Intensive Stream Biosurvey approach is more rigorous; it requires substantial volunteer training in habitat and macroinvertebrate sampling methods and in macroinvertebrate identification. This approach also requires the involvement of a stream biologist to advise the program participants regarding everything from the selection of reference conditions to taxonomy and data analysis.

Because of the need for training and professional assistance, the Intensive Stream Biosurvey approach can be expensive and labor-intensive for the volunteer program. Its benefits, however, are equally clear: with proper quality control and volunteer training, the Intensive Stream Biosurvey can yield credible information on subtle stream impacts and water quality trends. Key features of the Intensive Stream Biosurvey are as follows:

- *It relies on comparing the results for the sampling site to regional or local reference conditions.* This type of study is used to determine how streams in a given area compare to the best possible conditions. The reference condition is a composite of the best attainable (minimally impaired) stream conditions within the region and should be determined by an experienced aquatic biologist familiar with the characteristics of the ecological region.
- *It includes a detailed habitat assessment that requires the volunteer to rate 10 parameters on a scale of 0 to 20.* The results of the habitat assessment are compared to the score received by the stream's reference condition, and a percent similarity score is calculated.
- *The methods for collecting macroinvertebrates are similar to those of the Streamside Biosurvey.* However, rather than being processed streamside, the entire sample of macroinvertebrates is preserved and returned to a laboratory. A portion, or subsample, of the total organisms collected at each location is randomly selected and identified to taxonomic family level in the lab. After identification, a series of indices (or metrics) are calculated to provide a broad range of information about the stream site. The subsample and the rest of the collected organisms are maintained as a voucher collection, which serves as a quality assurance component.
- *The Intensive Stream Biosurvey requires that volunteers be extensively trained before habitat assessment and macroinvertebrate sampling and before attempting macroinvertebrate identification in the laboratory.* An experienced aquatic biologist is needed to determine and evaluate the regional reference conditions; train volunteers in habitat characteristics; and supervise and train volunteers in the collection, processing, and identification of sample macroinvertebrates. A laboratory (with microscopes) and a macroinvertebrate sample storage facility are required.

Step 1—Prepare for the Intensive Stream Biosurvey field work

Preparing for the Intensive Stream Biosurvey might take several months from the initial planning stages to the time when actual sampling occurs. An aquatic biologist should be centrally involved in all aspects of technical program development.

Issues that should be considered in planning the program include the following:

- Availability of reference conditions for your area
- Appropriate dates to sample in each season
- Appropriate sampling gear
- Sampling station location
- Availability of laboratory facilities and trainers
- Sample storage
- Data management
- Appropriate taxonomic keys, metrics, or measurements for macroinvertebrate analysis
- Habitat assessment consistency

Some of the preparation work for this approach is similar to that of the Stream Habitat Walk (section 4.1) and Streamside Biosurvey (section 4.2). Refer back to those sections for relevant information on the following tasks:

- Obtaining a USGS topographical map
- Becoming familiar with safety procedures

TASK 1 **Select monitoring locations**

If possible, the program coordinator, in conjunction with technical advisor(s), should preselect sampling locations for each stream. This adds an element of quality control to the sampling process. You might want to consider sampling at a few locations that are also sampled by state or local professionals, as a way to compare your results to theirs. *Be sure to secure approval to do so, however, and coordinate your sampling so as not to affect professional results.*

Provide detailed hand-drawn maps of the locations selected to the monitors. Know the latitude and longitude of your monitoring locations. This is critical for mapping and for many data management programs. Latitude and longitude can be calculated manually (see Appendix C) or by using a hand-held Global Positioning System (GPS).

TASK 2 **Schedule the field portion of the biosurvey**

Schedule your Intensive Stream Biosurvey for a time of year for which reference conditions have been established. Reference conditions might vary by season. It is also essential that seasonal data be collected within the same index period, or window of time, each year. In other words, if you sample during the last two weeks of March this year, do the same next year.

Another factor to keep in mind is weather. It is best to wait at least a week after a heavy rain or snow event before sampling. Heavy rains can have a scouring effect on macroinvertebrates, washing them downstream. If this happens, samples collected will not accurately reflect biological conditions. However, if you are studying the possible impact of runoff from a particular source (such as a construction site), you might decide to sample within a short time after heavy precipitation.

TASK 3 **Gather tools and equipment for the Intensive Stream Biosurvey**

In addition to the basic sampling equipment listed for the Stream Habitat Walk, collect the following equipment needed for the macroinvertebrate collection and habitat assessment of the Intensive Stream Biosurvey:

- Jars (2, at least quart size), plastic, wide-mouth with tight cap; one should be empty and the other filled about two thirds full with 70 percent ethyl alcohol. (Jars can be purchased from a scientific supply company or you might try using large pickle, mayonnaise, or quart mason jars.)
- Hand lens, magnifying glass, or field microscope
- Fine-point forceps
- Heavy-duty rubber gloves (kitchen gloves will work fine)
- Plastic sugar scooper or ice-cream scooper
- Kick net (rocky bottom stream) or dip net (muddy bottom stream) (see Fig. 4.7, page 63)
- Buckets (2)
- String or twine (50 yards); tape measure
- Stakes (4)
- Orange (a stick, an apple, or a fish float may also be used in place of an orange) to measure velocity
- Reference maps indicating general information pertinent to the sampling area, including the surrounding roadways, as well as hand-drawn station map

Sieve Buckets

Most professional biological monitoring programs employ sieve buckets as a holding container for composited samples. These buckets have a mesh bottom that allows water to drain out while the organisms and debris remain. This material can then be easily transferred to the alcohol-filled jars. However, sieve buckets can be expensive. Many volunteer programs employ alternative equipment, such as the two regular buckets described in this section. Regardless of the equipment, the process for compositing and transferring the sample is basically the same. The decision is one of cost and convenience.

- Station ID tags
- Spray water bottle
- Pencils (at least 2)

TASK 4 **Become familiar with field data sheets and instructions/definitions for conducting the macroinvertebrate collection and Habitat Assessment portions of the Intensive Biosurvey**

Step 2—Conduct the Intensive Biosurvey field work

The method you use to collect macroinvertebrates using this approach depends on the type of stream you are sampling.

Rocky-bottom streams are defined as those with bottoms made up of gravel, cobbles, and boulders in any combination. They usually have definite riffle areas. Riffle areas are fairly well oxygenated and, therefore, are prime habitats for benthic macroinvertebrates. In these streams, use the Rocky-Bottom sampling method.

Muddy-bottom streams have muddy, silty, or sandy bottoms that lack riffles. Usually, these are slow-moving, low-gradient streams (i.e., streams that flow along flat terrain). In such streams, macroinvertebrates generally attach to overhanging plants, roots, logs, submerged vegetation, and stream substrate where organic particles are trapped. In these streams, use the Muddy Bottom sampling method.

Each method is detailed below. Regardless of which collection method is used, the process for counting, identifying, and analyzing the macroinvertebrate sample for the Intensive Stream Biosurvey is the same. Following the discussion of both approaches to macroinvertebrate collection and habitat assessment procedures is a section on analyzing the sample.

Sieve (wash) buckets are expensive, an alternate tool is an EZ-strainer, which fits inside a 5-gallon bucket. The strainer can be ordered in the appropriate size mesh for macroinvertebrate assessments.

Rocky-Bottom Streams
Part 1: Macroinvertebrate Sampling Method

Use the following method of macroinvertebrate sampling in streams that have riffles and gravel/cobble substrates. You will collect three samples at each site and composite them to obtain one large total sample.

TASK 1 Identify the sampling location

You should already have located your site on a map along with its latitude and longitude (see Task 3, page 45)

1. You are going to sample in three different spots within a 100-yard stream site. These spots may be three separate riffles; one large riffle with different current velocities; or, if no riffles are present, three run areas with gravel or cobble substrate. Combinations are also possible (if, for example, your site has only one small riffle and several run areas).

   Mark off your 100-yard stream site. If possible, it should begin at least 50 yards upstream of any human-made modification of the channel, such as a bridge, dam, or pipeline crossing, Avoid walking in the stream, since this might dislodge macroinvertebrates and alter your sampling results.
2. Sketch the 100-yard sampling area. Indicate the location of your three sampling spots on the sketch. Mark the most downstream site as Site 1, the middle site as Site 2, and the upstream site as Site 3. (See Fig. 4.8.)

TASK 2 Get into place

1. **Always approach your sampling locations from the downstream end and sample the site farthest downstream first (Site 1).** This keeps you from biasing your second and third collections with dislodged sediment or macroinvertebrates. Always use a clean kick-seine, relatively free of mud and debris from previous uses. Fill a bucket about one third full with stream water and fill your spray bottle.
2. Select a 3-foot by 3-foot riffle area for sampling at Site 1. One member of the team, the net holder, should position the net at the downstream end of this sampling area. Hold the net handles at a 45 degree angle to the water's surface. Be sure that the bottom of the net fits tightly against the streambed so no macroinvertebrates escape under the net. You may use rocks from the sampling area to anchor the net against the stream bottom. Don't allow any water to flow over the net.

TASK 3 Dislodge the macroinvertebrates

1. Approach the sample site from the downstream end.

2. Position the net at a 45° angle with the bottom tight against the substrate.

3. Dislodge macroinvertebrates by rubbing rocks thoroughly.

4. Disturb the substrate thoroughly with your feet.

5. Remove the net with a forward scooping motion.

6. Flush out the net with clean stream water.

1. Pick up any large rocks in the 3-foot by 3-foot sampling area and rub them thoroughly over the partially-filled bucket so that any macroinvertebrates clinging to the rocks will be dislodged into the bucket. Then place each cleaned rock outside of the sampling area. After sampling is completed, rocks can be returned to the stretch of stream they came from.
2. The member of the team designated as the "kicker" should thoroughly stir up the sampling area with their feet, starting at the upstream edge of the 3-foot by 3-foot sampling area and working downstream, moving toward the net. All dislodged organisms will be carried by the stream flow into the net. Be sure to disturb the first few inches of stream sediment to dislodge burrowing organisms. As a guide, disturb the sampling area for about 3 minutes, or until the area is thoroughly worked over.
3. Any large rocks used to anchor the net should be thoroughly rubbed into the bucket as above.

TASK 4 **Remove the net**

1. Next, remove the net without allowing any of the organisms it contains to wash away. While the net holder grabs the top of the net handles, the kicker grabs the bottom of the net handles and the net's bottom edge. Remove the net from the stream with a forward scooping motion.
2. Roll the kick net into a cylinder shape and place it vertically in the partially filled bucket. Pour or spray water down the net to flush its contents into the bucket. If necessary, pick debris and organisms from the net by hand. Release back into the stream any fish, amphibians, or reptiles caught in the net.

TASK 5 **Collect the second and third samples**

Once you have removed all the organisms from the net repeat these steps at Sites 2 and 3. Put the samples from all three sites into the same bucket. Combining the debris and organisms from all three sites into the same bucket is called *compositing*.

Hint: If your bucket is nearly full of water after you have washed the net clean, let the debris and organisms settle to the bottom of the bucket. Then cup the net over the bucket and pour the water through the net into a second bucket. Inspect the water in the second bucket to be sure no organisms came through.

TASK 6 **Preserve the sample**

1. After collecting and compositing all three samples, it is time to preserve the sample. All team members

should leave the stream and return to a relatively flat section of stream bank with all their equipment. The next step will be to remove large pieces of debris (leaves, twigs, and rocks) from the sample. Carefully remove the debris one piece at a time. While holding the material over the bucket, use the forceps, spray bottle, and your hands to pick, rub, and rinse the leaves, twigs, and rocks to remove any attached organisms. Use your magnifying lens and forceps to find and remove small organisms clinging to the debris. When you are satisfied that the material is clean, discard it back into the stream.

2. You will need to drain off the water before transferring material to the jar. This process will require two team members. Place the kick net over the second bucket, which has not yet been used and should be completely empty. One team member should push the center of the net into bucket #2, creating a small indentation or depression. Then, hold the sides of the net closely over the mouth of the bucket. The second person can now carefully pour the remaining contents of bucket #1 onto a small area of the net to drain the water and concentrate the organisms. Use care when pouring so that organisms are not lost over the side of the net (Fig. 4.16).

   Use your spray bottle, forceps, sugar scoop, and gloved hands to remove all the material from bucket #1 onto the net. When you are satisfied that bucket #1 is empty, use your hands and the sugar scoop to transfer all the material from the net into the empty jar.

   Bucket #2 captured the water and any organisms that might have fallen through the netting during pouring. As a final check, repeat the process above, but this time, pour bucket #2 over the net, into bucket #1. Transfer any organisms on the net into the jar.

3. Now, fill the jar (so that all material is submerged) with the alcohol from the second jar. Put the lid tightly back onto the jar and gently turn the jar upside down two or three times to distribute the alcohol and remove air bubbles.

4. Complete the Sampling Station ID tag. Be sure to use a pencil, not a pen, because the ink will run in the alcohol! The tag includes your station number, the stream, location (e.g., upstream from a road crossing), date, time, and the names of the members of the collecting crew. Place the ID tag into the sample container—writing side facing out, so that identification can be seen clearly.

Fig. 4.16

Pouring sample water through the net

Rocky-Bottom Streams
Part 2: Habitat Assessment Method

You will conduct a habitat assessment (which will include measuring general characteristics and local land use) in a 100-yard section of stream that includes the riffles from which organisms were collected.

TASK 1 **Delineate the habitat assessment boundaries**

1. Begin by identifying the most downstream riffle that was sampled for macroinvertebrates. Using your tape measure or twine, mark off a 100-yard section extending 25 yards below the downstream riffle and about 75 yards upstream.
2. Complete the identifying information on your field data sheet for your habitat assessment site. On your stream sketch, be as detailed as possible, and be sure to note which riffles were sampled.

TASK 2 **Complete the General Characteristics and Local Land Use sections of the field sheet**

For safety reasons as well as to protect the stream habitat, it is best to estimate these characteristics rather than actually wading into the stream to measure them.

General Characteristics

1. *Water appearance* can be a physical indicator of water pollution.
   - *Clear* - colorless, transparent
   - *Milky* - cloudy-white or grey, not transparent; might be natural or due to pollution
   - *Foamy* - might be natural or due to pollution, generally detergents or nutrients (foam that is several inches high and does not brush apart easily is generally due to pollution)
   - *Turbid* - cloudy brown due to suspended silt or organic material
   - *Dark brown* - might indicate that acids are being released into the stream due to decaying plants
   - *Oily sheen* -multicolored reflection might indicate oil floating in the stream, although some sheens are natural
   - *Orange* - might indicate acid drainage
   - *Green* - might indicate excess nutrients being released into the stream
2. *Water odor* can be a physical indicator of water pollution.
   - *None or natural smell*
   - *Sewage* - might indicate the release of human waste material
   - *Chlorine* - might indicate that a sewage treatment plant is over-chlorinating its effluent
   - *Fishy* - might indicate the presence of excessive algal growth or dead fish
   - *Rotten eggs* - might indicate sewage pollution (the presence of a natural gas)
3. *Water temperature* can be particularly important for determining whether the stream is suitable as habitat for some species of fish and macroinvertebrates that have distinct temperature requirements. Temperature also has a direct effect on the amount of dissolved oxygen available to aquatic organisms. Measure temperature by submerging a thermometer for at least 2 minutes in a typical stream run. Repeat once and average the results.

4. The *width of the stream channel* can be determined by estimating the width of the streambed that is covered by water from bank to bank. If it varies widely along the stream, estimate an average width.

Local Land Use

5. *Local land use* refers to the part of the watershed within 1/4 mile upstream of and adjacent to the site. Note which land uses are present, as well as which ones seem to be having a negative impact on the stream. Base your observations on what you can see, what you passed on the way to the stream, and, if possible, what you notice as you leave the stream.

TASK 3 **Conduct the habitat assessment**

The following information describes the parameters you will evaluate for rocky-bottom habitats. Use these definitions when completing the habitat assessment field data sheet.

The first two parameters should be assessed directly at the riffle(s) or run(s) that were used for the macroinvertebrate sampling.

1. *Attachment sites for macroinvertebrates* are essentially the amount of living space or hard substrates (rocks, snags) available for aquatic insects and snails. Many insects begin their life underwater in streams and need to attach themselves to rocks, logs, branches, or other submerged substrates. The greater the variety and number of available living spaces or attachment sites, the greater the variety of insects in the stream. Optimally, cobble should predominate and boulders and gravel should be common. The availability of suitable living spaces for macroinvertebrates decreases as cobble becomes less abundant and boulders, gravel, or bedrock become more prevalent.

2. *Embeddedness* refers to the extent to which rocks (gravel, cobble, and boulders) are surrounded by, covered, or sunken into the silt, sand, or mud of the stream bottom. Generally, as rocks become embedded, fewer living spaces are available to macroinvertebrates and fish for shelter, spawning and egg incubation.

   To estimate the percent of embeddedness, observe the amount of silt or finer sediments overlying and surrounding the rocks. If kicking does not dislodge the rocks or cobbles, they might be greatly embedded.

The following eight parameters should be assessed in the entire 100-yard section of the stream.

3. *Shelter for fish* includes the relative quantity and variety of natural structures in the stream, such as fallen trees, logs, and branches; cobble and large rocks; and undercut banks that are available to fish for hiding, sleeping, or feeding. A wide variety of submerged structures in the stream provide fish with many living spaces; the more living spaces in a stream, the more types of fish the stream can support.

4. *Channel alteration* is basically a measure of large-scale changes in the shape of the stream channel. Many streams in urban and agricultural areas have been straightened, deepened (e.g., dredged), or diverted into concrete channels, often for flood control purposes. Such streams have far fewer natural habitats for fish, macroinvertebrates, and plants than do naturally meandering

streams. Channel alteration is present when the stream runs through a concrete channel; 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, bridges, and flow-altering structures such as combined sewer overflow (CSO) pipes are present; when the stream is of uniform depth due to dredging; and when other such changes have occurred. Signs that indicate the occurrence of dredging include straightened, deepened, and otherwise uniform stream channels, as well as the removal of streamside vegetation to provide dredging equipment access to the stream.

5. *Sediment deposition* is a measure of the amount of sediment that has been deposited in the stream channel and the changes to the stream bottom that have occurred as a result of the deposition. High levels of sediment deposition create an unstable and continually changing environment that is unsuitable for many aquatic organisms.

   Sediments are naturally deposited in areas where the stream flow is reduced, such as pools and bends, or where flow is obstructed. These deposits can lead to the formation of islands, shoals, or point bars (sediments that build up in the stream, usually at the beginning of a meander) or can result in the complete filling of pools. To determine whether these sediment deposits are new, look for vegetation growing on them: new sediments will not yet have been colonized by vegetation.

6. *Stream velocity and depth combinations* are important to the maintenance of healthy aquatic communities. Fast water increases the amount of dissolved oxygen in the water; keeps pools from being filled with sediment; and helps food items like leaves, twigs, and algae move more quickly through the aquatic system. Slow water provides spawning areas for fish and shelters macroinvertebrates that might be washed downstream in higher stream velocities. Similarly, shallow water tends to be more easily aerated (i.e., it holds more oxygen), but deeper water stays cooler longer. Thus the best stream habitat includes all of the following velocity/depth combinations and can maintain a wide variety of organisms.

   slow (<1 ft/sec), shallow (<1.5 ft)
   slow, deep
   fast, deep
   fast, shallow

   Measure stream velocity by marking off a 10-foot section of stream run and measuring the time it takes a stick, orange, or other floating biodegradable object to float the 10 feet. Repeat 5 times, in the same 10-foot section, and determine the average time. Divide the distance (10 feet) by the average time (seconds) to determine the velocity in feet per second.

   Measure the stream depth by using a stick of known length and taking readings at various points within your stream site, including riffles, runs, and pools. Compare velocity and depth at various points within the 100-yard site to see how many of the combinations are present.

7. *Channel flow status* is the percent of the existing channel that is filled with water. The flow status changes as the channel enlarges 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 living area for aquatic organisms is limited.

For the last three parameters, evaluate the condition of the right and left stream banks separately. Define the " left" and "right" banks by standing at the downstream end of your study stretch and looking upstream. Each bank is evaluated on a scale of 0-10.

8. *Bank vegetative protection* measures the amount of the stream bank that is covered by natural (i.e., growing wild and not obviously planted) vegetation. The root systems of plants growing on stream banks help hold soil in place, reducing erosion. Vegetation on banks provides shade for fish and macroinvertebrates and serves as a food source by dropping leaves and other organic matter into the stream. Ideally, a variety of vegetation should be present, including trees, shrubs, and grasses. Vegetative disruption can occur when the grasses and plants on the stream banks are mowed or grazed, or when the trees and shrubs are cut back or cleared.
9. *Condition of banks* measures erosion potential and whether the stream banks are eroded. Steep banks are more likely to collapse and suffer from erosion than are gently sloping banks and are therefore considered to have a high erosion potential. Signs of erosion include crumbling, unvegetated banks, exposed tree roots, and exposed soil.
10. The *riparian vegetative zone width* is defined here as the width of natural vegetation from the edge of the stream bank. The riparian vegetative zone is a buffer zone to pollutants entering a stream from runoff. It also controls erosion and provides stream habitat and nutrient input into the stream.

    A wide, relatively undisturbed riparian vegetative zone reflects a healthy stream system; narrow, far less useful riparian zones occur when roads, parking lots, fields, lawns, and other artificially cultivated areas, bare soil, rocks, or buildings are near the stream bank. The presence of "old fields" (i.e., previously developed agricultural fields allowed to revert to natural conditions) should rate higher than fields in continuous or periodic use. In arid areas, the riparian vegetative zone can be measured by observing the width of the area dominated by riparian or water-loving plants, such as willows, marsh grasses, and cottonwood trees.

Note: Instructions on sample processing, macroinvertebrate identification, and data analysis follow the sections on muddy-bottom macroinvertebrate sampling and habitat assessment. (See Step 3, page 101)

Muddy-Bottom Sampling
Part 1: Macroinvertebrate Sampling

In muddy-bottom streams, as in rocky-bottom streams, the goal is to sample the most productive habitat available and look for the widest variety of organisms. The most productive habitat is the one that harbors a diverse population of pollution-sensitive macroinvertebrates. Volunteers should sample by using a D-frame net to jab at the habitat and scoop up the organisms that are dislodged. The idea is to collect a total sample that consists of 20 jabs taken from a variety of habitats.

TASK 1 **Determine which habitats are present**

Muddy-bottom streams usually have four habitats (Fig. 4.17). It is generally best to concentrate sampling efforts on the most productive habitat available, yet to sample other principal habitats if they are present. This ensures that you will secure as wide a variety of organisms as possible. Not all habitats are present in all streams or present in significant amounts. If your sampling areas have not been preselected, try to determine which of the following habitats are present. (Avoid standing in the stream while making your habitat determinations.)

- *Vegetated bank margins* consist of overhanging bank vegetation and submerged root mats attached to banks. The bank margins may also contain submerged, decomposing leaf packs trapped in root wads or lining the streambanks. This is generally a highly productive habitat in a muddy–bottom stream, and it is often the most abundant type of habitat.
- *Snags and logs* consist of submerged wood, primarily dead trees, logs, branches, roots, cypress knees and leaf packs lodged between rocks or logs. This is also a very productive muddy-bottom stream habitat.
- *Aquatic vegetation beds and decaying organic matter* consist of beds of submerged, green/leafy plants that are attached to the stream bottom. This habitat can be as productive as vegetated bank margins, and snags and logs.
- *Silt/sand/gravel substrate* includes sandy, silty, or muddy stream bottoms; rocks along the stream bottom; and/or wetted gravel bars. This habitat may also contains algae-covered rocks (sometimes called Aufwuchs). This is the least productive of the four muddy-bottom stream habitats, and it is always present in one form or another (e.g., silt, sand, mud, or gravel might predominate).

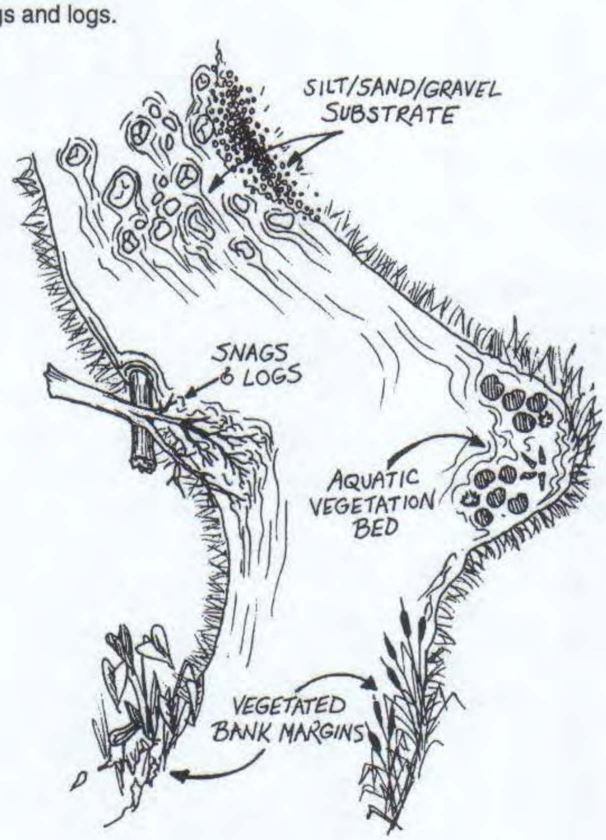


Figure 4.17

Four habitats found in muddy-bottom streams

Volunteers will likely find the most macroinvertebrates in vegetated habitats and snags and logs.

TASK 2 **Determine how many times to jab in each habitat type**

Your goal is to jab a total of 20 times. The D-frame net is 1 foot wide, and a jab should be approximately 1 foot in length. Thus, 20 jabs equals 20 square feet of combined habitat.

- If all four habitats are present in plentiful amounts, jab the vegetated banks 10 times and divide the remaining 10 jabs among the remaining 3 habitats.

- If three habitats are present in plentiful amounts and one is absent, jab the silt/sand/gravel substrate—the least productive habitat—5 times and divide the remaining 15 jabs among the other two more productive habitats.
- If only two habitats are present in plentiful amounts, the silt/sand/gravel substrate will most likely be one of those habitats. Jab the silt/sand/gravel substrate 5 times and the more productive habitat 15 times.
- If some habitats are plentiful and others are sparse, sample the sparse habitats to the extent possible, even if you can take only one or two jabs. Take the remaining jabs from the plentiful habitat(s). This rule also applies if you cannot reach a habitat because of unsafe stream conditions. Jab a total of 20 times.

Because you might need to make an educated guess to decide how many jabs to take in each habitat type, it is critical that you note, on the field data sheet, how many jabs you took in each habitat. This information can be used to help characterize your findings.

TASK 3 Get into place

Outside and downstream of your first sampling location (1st habitat), rinse the dip net and check to make sure it does not contain any macroinvertebrates or debris from the last time it was used. Fill a bucket approximately one-third full with clean stream water. Also, fill the spray bottle with clean stream water. This bottle will be used to wash down the net between jabs and after sampling is completed.

This method of sampling requires only one person to disturb the stream habitats. While one person is sampling, a second person should stand outside the sampling area, holding the bucket and spray bottle. After every few jabs, the sampler should hand the net to the second person, who then can rinse the contents of the net into the bucket.

TASK 4 Dislodge the macroinvertebrates

Approach the first sample site from downstream, and sample as you walk upstream. Here is how to sample in the four habitat types:

- Sample vegetated bank margins by jabbing vigorously, with an upward motion, brushing the net against vegetation and roots along the bank. The entire jab motion should occur underwater.
- To sample snags and logs, hold the net with one hand under the section of submerged wood you are sampling (Fig. 4.18). With the other hand (which should be gloved), rub about 1 square foot of area on the snag or log. Scoop organisms, bark, twigs, or other organic matter you dislodge into your net. Each combination of log rubbing and net scooping is one jab.
- To sample aquatic vegetation beds, jab vigorously, with an upward motion, against or through the plant bed. The entire jab motion should occur underwater.
- To sample a silt/sand/gravel substrate, place the net with one edge against the stream bottom and push it forward about a foot (in an upstream direction) to dislodge the first few inches of silt, sand, gravel, or rocks. To avoid gathering a netful of mud, periodically sweep the mesh bottom of the net back and forth in the water, making sure that water does not run over the top of the net. This will allow fine silt to rinse out of the net. When you have com-

Figure 4.18

Collecting a sample from a log

Volunteer rubs the log with one hand and catches dislodged organisms and other material in the net.

pleted all 20 jabs, rinse the net thoroughly into the bucket. If necessary, pick any clinging organisms from the net by hand and put them in the bucket.

TASK 5 **Preserve the sample**

1. Look through the material in the bucket and immediately return any fish, amphibians, or reptiles to the stream. Carefully remove large pieces of debris (leaves, twigs, and rocks) from the sample. While holding the material over the bucket, use the forceps, spray bottle, and your hands to pick, rub, and rinse the leaves, twigs, and rocks to remove any attached organisms. Use your magnifying lens and forceps to find and remove small organisms clinging to the debris. When you are satisfied that the material is clean, discard it back into the stream.
2. You will need to drain off the water before transferring material to the jar. This process will require two team members. One person should place the net into the second bucket, like a sieve (this bucket, which has not yet been used, should be completely empty) and hold it securely. The second person can now carefully pour the remaining contents of bucket #1 onto the center of the net to drain the water and concentrate the organisms.

   Use care when pouring so that organisms are not lost over the side of the net. Use your spray bottle, forceps, sugar scoop, and gloved hands to remove all the material from bucket #1 onto the net. When you are satisfied that bucket #1 is empty, use your hands and the sugar scoop to transfer all the material from the net into the empty jar. You can also try to carefully empty the contents of the net directly into the jar by turning the net inside out into the jar.

   Bucket #2 captured the water and any organisms that might have fallen through the netting. As a final check, repeat the process above, but this time, pour bucket #2 over the net, into bucket #1. Transfer any organisms on the net into the jar.
3. Fill the jar (so that all material is submerged) with alcohol. Put the lid tightly back onto the jar and gently turn the jar upside down two or three times to distribute the alcohol and remove air bubbles.
4. Complete the sampling station ID tag. Be sure to use a pencil, not a pen, because the ink will run in the alcohol. The tag should include your station number, the stream, location (e.g., upstream from a road crossing), date, time, and the names of the members of the collecting crew. Place the ID tag into the sample container, writing side facing out, so that identification can be seen clearly (Fig. 4.19).

You will conduct a habitat assessment (which will include measuring general characteristics and local land use) in a 100-yard section of the stream that includes the habitat areas from which organisms were collected.

TASK 1 **Delineate the habitat assessment boundaries**

1. Begin by identifying the most downstream point that was sampled for macroinvertebrates. Using your tape measure or twine, mark off a 100-yard section extending 25 yards below the downstream sampling point and about 75 yards upstream.
2. Complete the identifying information on your field data sheet for your habitat assessment site. On your stream sketch, be as detailed as possible, and be sure to note which habitats were sampled.

TASK 2 **Complete the General Characteristics and Local Land Use sections of the field sheet**

For safety reasons as well as to protect the stream habitat, it is best to estimate these characteristics rather than actually wading into the stream to measure them. For instructions on completing these sections of the field data sheet, see the rocky-bottom habitat assessment instructions.

TASK 3 **Conduct the habitat assessment**

The following information describes the parameters you will evaluate for muddy-bottom habitats. Use these definitions when completing the habitat assessment field data sheet.

STATION ID TAG

Station #: ____________

Stream: ____________

Location: ____________

Date/Time: ____________

Team members:

____

____

____

Figure 4.19

Example of a Station ID tag

To prevent samples from being mixed up, volunteers should place the ID tag *inside* the sample jar.

1. *Shelter for fish and attachment sites for macroinvertebrates* are essentially the amount of living space and shelter (rocks, snags, and undercut banks) available for fish, insects, and snails. Many insects attach themselves to rocks, logs, branches, or other submerged substrates. Fish can hide or feed in these areas. The greater the variety and number of available shelter sites or attachment sites, the greater the variety of fish and insects in the stream.

   Many of the attachment sites result from debris falling into the stream from the surrounding vegetation. When debris first falls into the water, it is termed new fall and it has not yet been "broken down" by microbes (conditioned) for macroinvertebrate colonization. Leaf material or debris that is conditioned is called old fall. Leaves that have been in the stream for some time lose their color, turn brown or dull yellow, become soft and supple with age, and might be slimy to the touch. Woody debris becomes blackened or dark in color; smooth bark becomes coarse and partially disintegrated, creating holes and crevices. It might also be slimy to the touch.

2. *Pool substrate characterization* evaluates the type and condition of bottom substrates found in pools. Pools with firmer sediment types (e.g., gravel, sand) and rooted aquatic plants support a wider variety of organisms than do pools with substrates dominated by mud or bedrock and no plants. In addition, a pool with one uniform substrate type will support far fewer types of organisms than will a pool with a wide variety of substrate types.
3. *Pool variability* rates the overall mixture of pool types found in the stream according to size and depth. The four 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 habitats to support a diverse aquatic community.
4. *Channel alteration* (See description in habitat assessment for rocky-bottom streams.)
5. *Sediment deposition* (See description for rocky-bottom streams.)
6. *Channel sinuosity* evaluates the sinuosity or meandering of the stream. Streams that meander provide a variety of habitats (such as pools and runs) and stream velocities and reduce the energy from current surges during storm events. Straight stream segments are characterized by even stream depth and unvarying velocity, and they are prone to flooding. To evaluate this parameter, imagine how much longer the stream would be if it were straightened out.
7. *Channel flow status* (See description in habitat assessment for rocky-bottom streams.)
8. *Bank vegetative protection* (See description for rocky-bottom streams.)
9. *Condition of banks* (See description for rocky-bottom streams.)
10. The *riparian vegetative zone width* (See description for rocky-bottom streams.)

Reference Collection

A reference collection is a sample of locally-found macroinvertebrates that have been identified, labelled, and preserved in alcohol. The program advisor, along with a professional biologist/entomologist, should assemble the reference collection, properly identify all samples, preserve them in vials, and label them. This collection may then be used as a training tool and, in the field, as an aid in macroinvertebrate identification.

Step 3—Leave the field, complete data forms, clean the site, and return material

After completing the stream characterization and habitat assessment, make sure that all of the field data sheets have been completed properly and that the information is legible. Be sure to include the site's identifying name and the sampling date on each sheet. These will function as a quality control element. If you can't determine how to answer a question on the field data sheet, just leave the space blank.

Before you leave the stream location, make sure that all your equipment has been collected and rinsed properly. Double-check to see that sample jars are tightly closed and properly identified. All samples, field sheets, and equipment should be returned to the coordinator at this point. You might want to keep a copy of the field data sheet for comparison with future monitoring trips and for personal records.

Step 4—Prepare for macroinvertebrate laboratory work

This step includes all the work needed to set up a laboratory for processing samples into subsamples and identifying macroinvertebrates to the family level. A professional biologist/entomologist or the program advisor should supervise the identification procedure. All interested volunteers should be encouraged to participate. In general it is a good idea to train volunteers in identification procedures before each lab session and to start new volunteers with less diverse samples. Refresher workshops for experienced volunteers are strongly encouraged.

TASK 1 **Gather tools and equipment for the laboratory**

The following lab equipment is recommended for the macroinvertebrate identification process. Enough of each will need to be provided for each volunteer work station:

- Reference collection and taxonomic keys
- Fine-point forceps
- Petri dishes or small, shallow, clear container
- Alcohol preservative (used in field and lab): 70 percent ethyl alcohol, denatured; no other preservatives used
- Microscope, dissecting microscope, and magnifying glass, or hands lens
- Sample containers, preferably shatterproof with poly-seal caps that prevent evaporation of the preservative (jars or vials are used in field and lab). Shatterproof vials with poly-seal caps are available from scientific supply houses.
- Wash bottles or spray bottles
- Shallow, rectangular white pans (large enough to hold entire macroinvertebrate sample)
- Additional shallow white containers (heavy duty plastic plates with a rim, white pans, or cafeteria trays are all possible choices).
- Plastic spoons or unslotted spatulas
- Sieve, purchased from scientific supply company (#30) or homemade (with same mesh size as sampling net)
- Permanent ink markers
- Ruler
- Macroinvertebrate assessment worksheet
- Pencils
- Note paper for counting

TASK 2 **Create gridded subsampling pans**

Using the ruler, measure the inside width and length of the large rectangular white pan. Draw a grid of evenly sized squares on the inside of the pan, using permanent ink. The grid should fill the entire inside of the pan. Number each square. One pan will be needed for each work station. Volunteers will use these pans for randomizing the sample and selecting a subsample of organisms.

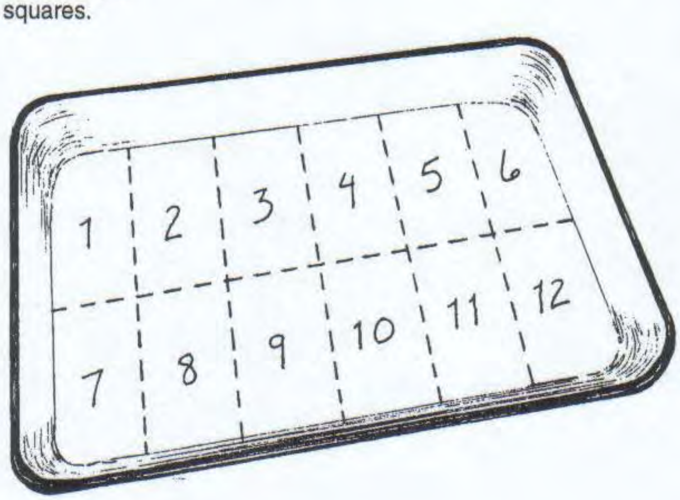
TASK 3 **Prepare the lab and the individual work stations**

Before volunteers enter the lab, the program manager will need to prepare work stations. Make sure that all microscopes are functioning properly and that each station has access to all other equipment. The reference collection should be centrally located as should any other visual training displays. The lab itself should be well lit and well ventilated. A copy of lab safety instructions should be visible to all volunteers.

Figure 4.20

A gridded subsampling pan

Volunteers collect a subsample of organisms by picking them from randomly selected grid squares.



Step 5—Conduct macroinvertebrate processing and identification

If possible, before beginning the subsampling and identification processes, all volunteers should become familiar with the lab equipment, microscope(s), the reference collection, and the taxonomic key chosen by the advisor.

Processing a subsample and identifying the organisms are two separate activities. Some programs might prefer to split these tasks into separate lab sessions.

Session 1: Picking a subsample of aquatic organisms

TASK 1 **Prepare the sample**

1. Carefully remove the station ID tag from the sample container and put it aside. You will need it later.
2. Cover the bottom of the gridded pan with about 1/4 inch of clean water.
3. Pour the preserved sample (alcohol and debris) into the sieve and wash off preservative over a sink, using a spray or wash bottle filled with water.
4. Transfer the sample to the white gridded pan by turning the sieve upside down over the pan. Tap it several times to empty the contents onto the pan. Squirt a small amount of water over the bottom of the sieve to flush the organisms into the pan.
5. With your hands and by gently shaking the pan, evenly disperse the sample over the entire bottom of the pan, making sure that even the corners are covered. The water will help in distributing the sample throughout the pan. This is called randomizing the sample.

TASK 2 **Randomly select a square for the subsample**

1. Randomly choose a square to start sorting organisms. You may use a random numbers table, draw numbers from a hat, or roll a pair of dice. The most important thing to remember is that the grid selection should be random. Indicate the square number selected on the lab sheet.
2. Using a plastic spoon or unslotted spatula, remove *all* the material from the square and transfer it to another container (another pan, tray, or plate) for sorting. The organisms in this container will become your subsample.

TASK 3 **Pick the subsample**

1. Prepare a container to house the subsample by filling a vial or jar one-half full of alcohol. Place the new label into the vial, writing side out. Keep the vial on a flat, stable area.
2. Using forceps, carefully and systematically remove all organisms from the pan or tray and place them one by one into the prepared subsample vial. Examine all debris such as leaves or sticks for clinging organisms. Count each organism as it is transferred. Keep a written count of the number of organisms you have transferred. The objective is have at least 100 individual organisms in your subsample. If you reach 100 and there are still organisms remaining in your subsample plate or tray, continue picking until *all* the organisms are removed even though you might end up with more than 100.

   When you think all the organisms have been transferred from the plate or tray to the subsample vial, have a second volunteer check to confirm that all organisms have been removed. On your lab sheet, record how many organisms are in the subsample.
3. If you finish picking the contents of the first square selected and have fewer than 100 organisms, randomly select another square and repeat the process of removing the contents of the square to the subsample plate or tray; picking organisms with the forceps and transferring them to the vial (all organisms that will be part of the subsample should be transferred to the same vial). Record the number of organisms you obtain from the second square. Repeat this process until at least 100 organisms have been placed into the vial or until the entire sample in the gridded pan has been picked clean. Remember, any square started must be picked clean.

   If, after picking the entire gridded pan clean, you have fewer than 100 organisms, and your reference site

SUBSAMPLE ID TAG

Station #: ______________

Stream: ______________

Location: ______________

Date/Time: ______________

Subsample team members:

____

____

____

Figure 4.21

Example of a Subsample ID tag

To prevent subsamples from being mixed up, volunteers should place the ID tag *inside* the subsample jar.

produced 100 or more organisms, either your site is impaired or your sampling technique is flawed. It is also possible that recent heavy rains might have washed many organisms downstream. If you do not find 100 organisms in the entire sample, be sure to note the potential cause for such a problem on the Habitat Assessment Data Sheet.

TASK 4 Label and store the subsample

Fill out a new Subsample ID Tag (Fig. 4.21) for the subsample. Remember to use pencil because ink will run in the alcohol. The vial housing the subsample must be labeled with the same station number, stream name, location, and date found on the original sample ID tag. The vial tag should also include information on when the subsample was picked (i.e., 100 or more organisms counted) and by whom. Place the tag in the vial with the writing side out. Make sure the vial is tightly closed before giving the subsample in the vial to the program coordinator.

TASK 5 Replace remainder of original sample back into the sample jar

Place the remaining sample back into the original container. Be sure that the original station ID tag is included, writing side out. Fill the jar with 70 percent alcohol. This sample will be retained as part of a voucher collection. Make sure the jar is tightly closed before returning it to the program coordinator.

Session 2: Identifying the subsample to family level

TASK 1 Prepare for the ID

1. Make sure that you have several petri dishes, fresh alcohol, and fresh water close at hand. Also have your taxonomic keys handy for all stages of the ID process. Check to make sure that your microscope is working properly.
2. Carefully remove the station ID tag from the subsample vial and put it aside. You will need it later. Be sure no organisms are clinging to it. If they are, remove them with forceps.
3. Using the information on the station ID tag, complete the first section of the Macroinvertebrate Assessment Sheet with your name, date, the stream name, station number, and any other information requested.

TASK 2 Identify the sample to order level

1. Place a few of the macroinvertebrates in a petri dish (or other small, shallow container) and examine them under the microscope. Include some ethyl alcohol in the dish to ensure that the organisms do not dry out. Compare the organisms in the dish to those in the taxonomic key and/or reference collection.
2. Roughly sort organisms by taxonomic order into petri dishes. Many volunteers find it helpful to use one dish for every major taxonomic order found in the subsample. Place any organism that you cannot identify into another dish for the biological advisor to examine.

TASK 3 **Identify the organisms within each order to family level**

1. Starting with one order, and using the taxonomic keys, reference collection, and assistance of the biological advisor, identify each individual to family level.
2. Keep a running count of how many individuals there are in each family on a piece of scratch paper.
3. Place any organisms that you cannot identify into a separate container. Make sure that the biological advisor sees them and assists you with the ID.
4. After all organisms have been identified, note the total number of organisms in each family on the Macroinvertebrate Assessment Sheet. Write in pencil and make sure your writing is legible. These lab sheets will be the basis for the data analysis. It is important that they are accurate and easy to read.

TASK 4 **Return the organisms to the vial**

1. After you have identified and counted all organisms in the subsample, return them to the subsample vial and replace the subsample ID Tag, writing side out.
2. Refill the subsample vial with 70 percent ethyl alcohol (new or re-cycled). Be sure to secure the caps on the vial tightly to prevent the organisms from drying out.
3. Return the subsample vial and the assessment worksheet to the program manager.

Voucher Collection

Maintaining a voucher collection adds another layer of credibility to the program by documenting the accuracy of the volunteer identifications. It substantiates and provides evidence to support the analysis of the data—a powerful quality control element. However, an important issue to consider is how long to keep the samples. Program managers, in collaboration with technical advisors, will have to consider the following in keeping a voucher collection.

- *Sample maintenance.* Even jars and vials with tight fitting lids require maintenance on a regular basis (every 2-3 months) to ensure that alcohol levels are adequate.
- *Fire safety.* When you are dealing with alcohol, you will need to consider fire safety and ventilation issues to make sure that you are in line with local codes.
- *Availability of storage space.* In addition to needing well-ventilated and fire-proof storage cabinet, you will need a well-ventilated room to store samples. Samples should not be stored in someone's office for any length of time.
- *Length of storage.* How long samples should be maintained is an issue determined by program goals. Data collected for regulatory purposes will probably require longer storage than other samples. Generally, 1-5 years is recommended for storage.

Step 6—Performing habitat assessment data analysis

To evaluate the condition of your stream site properly, you should compare it to an optimal or best condition found in the region. This is called a reference condition. In an ideal world, the reference condition would reflect the water quality, habitat, and aquatic life characteristics of pristine sites in the same ecological region as your stream. In real life, however, few pristine sites remain. The reference condition is, therefore, a composite of sites that reflect the best physical, chemical, and biological conditions existing in your ecological region. State water quality or natural resource agencies might have already established reference conditions for the ecological regions in your state.

Table 4.5

Reference scores for sampling site comparison

If a score falls at or near the break between categories, use your best judgement to determine the appropriate score.

% Similarity to Reference Score	Habitat Quality Category	Attributes
> 90 %	Excellent	Comparable to the best situation to be expected within an ecoregion. Excellent overall habitat structure conducive to supporting healthy biological community.
75 - 88%	Good	Habitat structure slightly impaired. Generally, diverse instream habitat well-developed; some degradation of riparian zone and banks; a small amount of channel alteration may be present.
60 - 73%	Fair	Loss of habitat compared to reference. Habitat is a major limiting factor to supporting a healthy biological community.
< 58%	Poor	Severe habitat alteration at all levels.

Your program's consulting biologist should work in cooperation with the state agency to identify the reference condition(s) you will need to conduct an Intensive Stream Biosurvey. The biologist will use the reference condition to establish a water quality rating system against which to rank your monitored stream sites.

To perform the habitat assessment data analysis for the Intensive Stream Biosurvey, perform the following tasks.

TASK 1 **Determine the habitat index score**

Add together the scores of all 10 habitat parameters. This sum is the habitat index score for the study stretch.

TASK 2 **Determine the percent similarity to the reference score**

Divide the habitat index score by the reference index score and then multiply the result by 100. This number is the percent similarity to the reference score.

TASK 3 **Determine the stream habitat quality rating**

Compare the percent similarity of your results with the range of percent similarity numbers in the stream habitat rating table to obtain the habitat quality category for your site(s) (Table 4.5). Enter the appropriate descriptive rating (excellent, good, fair, or poor) on the field data sheet. If your score falls at or near the break between habitat quality categories, use your best judgment to determine an appropriate rating.

Step 7—Conduct macroinvertebrate data analysis

In general, the program's biological advisor, rather than the volunteers, should analyze the results of the Intensive Stream Biosurvey's macroinvertebrate identification. The advisor's knowledge of local ecological conditions will help in the interpretation of the data findings and will lend additional credibility to the sampling effort. Volunteers can contribute significantly to the advisor's data analysis by interpreting field notes, assisting with macroinvertebrate identification, and counting organisms on the aquatic macroinvertebrate assessment worksheet. Relay the results of the data analysis to the volunteers as soon after the sampling date as possible.

TASK 1 **Determine which metrics or measurements are appropriate**

A number of metrics (or measures) can be used to calculate stream health using benthic macroinvertebrates. These metrics should be calculated for both the sample site and the reference condition. By comparing the two, the program advisor can reach a clear understanding of the biological health of the sampling site.

The Intensive Stream Biosurvey recommends the use of four basic metrics (taxa richness, number of EPT taxa, percent abundance of EPT, and sensitive taxa index) plus two optional metrics (percent abundance of scrapers and percent abundance of shredders). These metrics are discussed briefly below. Refer to the reference list for more information.

The term *taxa* (plural for taxon), used below, refers to the specific taxonomic groupings to which organisms have been identified. For the Intensive Stream Biosurvey, organisms are identified to the taxon of family. Your volunteer monitoring program should identify organisms to a specific taxonomic grouping if it is to compare results over time and between sites. The following metrics are generally applicable throughout the country (but confirm this with a local biologist).

1. *Number of taxa (taxa richness)*—this measure is a count of the number of taxa (e.g., families) found in the sample. A high diversity or variety is good.
2. *Number of EPT taxa (EPT richness)*—this measure is a count of the number of taxa in each of three generally pollution-sensitive orders: Ephemeroptera (mayflies), Plecoptera (stoneflies), and Trichoptera (caddisflies). A high diversity or variety is good.
3. *Percent dominance*—this measure is the percent composition of the most abundant family from your station. It indicates how dominant a single taxon is at a particular site. A high percent dominance is not good.
4. *Sensitive taxa index (modified Hilsenhoff index)*—this measure is calculated by multiplying the number of organisms in each taxon by the pollution tolerance value assigned to each taxon, adding these for all taxa represented in the sample, and dividing by the total number of taxa in the sample. A high index number is not good.

$$\text{Sensitive taxa index} = \frac{\sum (X_i t)}{n}$$

where:

$\sum$ = the summation of $X_i t$

X_i = the number of individuals in each taxon

t = tolerance value for each taxon in the sample

n = number of individuals in the sample

The following optional metrics can be used in rocky-bottom streams if at least 10 scraper and shredder organisms are collected.

5. *Percent abundance of scrapers*—in the majority of rocky-bottom streams, the basic food source for many aquatic organisms is algae covering the rocks in the stream.

   Macroinvertebrates that "scrape" or graze on these algae are known as scrapers. To compute the percent

Selecting Metrics to Determine Stream Health

Metrics are used to analyze and interpret biological data by condensing lists of organisms into relevant biological information. In order to be useful, metrics must be proven to respond in predictable ways to various types and intensities of stream impacts. This manual recommends using a multimetric approach that combines several metrics into a total Biosurvey Score. The four primary and two optional metrics discussed in this chapter have been tested extensively in the mid-Atlantic region and have been shown to respond in predictable ways to stream impacts. In other parts of the country, other metrics and scoring systems may be more appropriate. For example, the Benthic Index of Biotic Integrity (B-IBI), developed by Dr. James Karr, is another multimetric approach, using different metrics, that has been tested in the Tennessee Valley, the Midwest, and the northwest. The River Watch Network suggests that, while you should always use multiple metrics to summarize your data, you shouldn't rely solely on an overall score to interpret your data; individual metrics can also provide a wealth of information. In any case you will need to select metrics that have been proven to respond predictably to various impacts. As always, consult with your program's biological advisor for help in selecting appropriate metrics for your region and for determining whether an overall biosurvey score is recommended.

Below are metrics that are commonly used in rocky bottom streams. This is only a partial list of the dozens of metrics used by monitoring programs throughout the country. These metrics fall under four general categories: 1) taxa richness and composition, 2) pollution tolerance and intolerance, 3) feeding ecology, and 4) population attributes. Metrics marked with a (*) are included in the recommended suite of metrics in this manual. The River Watch Network's *Benthic Macroinvertebrate Monitoring Manual* contains detailed guidance on selecting, calculating, aggregating, and interpreting the metrics discussed below. (See Dates, G. and J. Byrne in References and Further Reading)

Taxa Richness and Composition Metrics

- *Total Number of Taxa *:* the total number of taxa found in the sample.
- *Number of EPT Taxa *:* the combined number of mayfly (E), stonefly (P) and caddisfly (T) taxa found in the sample. The number of taxa in each of these macroinvertebrate orders can also be reported separately since each order may respond differently to various impacts.
- *Number of Long-Lived Taxa:* the number of organism families found in the sample (such as giant stoneflies and dobson flies) that live more than one season.
- *Percent Abundance of the Major Groups *:* the percent of the sample that is comprised of individuals in each of the selected major groups (mostly orders).
- *Percent Model Affinity* (Bode, 1991): used in conjunction with *Percent Composition of the Major Groups*, this metric measures the similarity of the sample to a model "nonimpacted" community of organisms (adjusted for ecoregional conditions) based on the percent composition of the major groups.
- *Quantitative Similarity Index* (from Shackleford, 1988): used in conjunction with *Percent Composition of the Major Groups*, this metric shows the percent similarity between two sites based on the percent of the sample in each of the major groups.
- *Dominants in Common* (from Shackleford, 1988): the number of dominant (5 most abundant families) families common to two sites.

Tolerance and Intolerance Metrics

- *Number of Intolerant Taxa:* the number of taxa in the sample that are in the 10-15% of the least tolerant taxa in a region or that have a pollution tolerance value of 1 (based on the Hilsenhoff scale of 0-10).
- *Percent of Individuals in Tolerant Taxa:* the number of taxa in the sample that are in the 10-15% of the most tolerant taxa in a region or that have a pollution tolerance value of 10 (based on the Hilsenhoff scale of 0-10).
- *Number of Clinger Taxa:* the number of families in the sample that live by clinging to the bottom of the stream.
- *Sensitive Taxa Index *:* the pollution tolerance values (based on the Hilsenhoff scale of 0-10) assigned to each family aggregated into an overall pollution tolerance value for the sample.

Feeding Ecology Metrics

- *Percent Composition of Functional Feeding Groups:* the percentage of the total number of individuals in the sample that belong to each of the five functional feeding groups (scrapers, shredders, filtering collectors, gathering collectors, and predators).
- *Percent Abundance of Scrapers *:* the percent of the total number of individuals in the sample that use bottom-growing algae as their primary food source.
- *Percent Abundance of Shredders *:* the percent of the total number of individuals in the sample that use leaves and other plant debris as their primary food source.
- *Percent Abundance of Predators:* the percent of the total number of individuals in the sample that eat other animals as their primary food source.

Population Attributes Metrics

- *Percent Dominance (of the most abundant family) *:* the percentage of the total number of individuals in the sample that are in the sample's most abundant family.
- *Percent Dominance (of the three most abundant families):* the percentage of the total number of individuals in the sample that are in the sample's three most abundant families.
- *Organism Density Per Sample (total abundance):* the total number of individuals in the sample (calculated if a subsample is used).

abundance of the scrapers in the macroinvertebrate community, divide the number of organisms classified as grazers or scrapers by the total number of organisms in the sample. A high percent abundance of scrapers is good.

6. *Percent abundance of shredders*—leaf litter and other plant debris are broken down and processed by organisms called shredders. To compute the percent abundance of shredders in the macroinvertebrate community, divide the number of organisms classified as shredders by the total number of organisms in the sample. A high percent abundance of shredders is good.

The following optional metrics can be used in muddy-bottom streams as additional metrics to provide more information about the condition of the macroinvertebrate assemblage.

7. *Percent abundance of EPT*—this measure compares the number of organisms in the EPT orders to the total number of organisms in the sample. (The number of organisms in the EPT orders is divided by the total number of organisms in the sample to calculate a percent abundance.) A high percent abundance of EPT orders is good.

8. *Percent abundance of midge larvae*—this measure compares the number of midges to the total number of organisms in the sample. (The number of organisms in the chironomidae family is divided by the total number of organisms in the sample to calculate a percent composition.) A low percent abundance of midge larvae is good.

TASK 2 **Calculate a score for the site**

The metric worksheets Tables 4.6 and 4.7 are designed to help calculate a total score for the monitored site. Table 4.8 provides an example of a sample metric worksheet for the fictional Volunteer Creek (rocky-bottom stream). This score should be compared to reference conditions to determine the biological condition of the stream at that site. You should also note that these worksheets were developed for use in mid-Atlantic states; they might need to be modified to reflect local conditions.

To calculate a score for your stream site using one of these worksheets, enter the metric values at the monitored site in the (M) column. Compare each metric value from your monitored site to the value ranges presented in the biosurvey score columns. Choose the matching range and circle it; this gives you the corresponding score (6, 3, or 0) for your metric value. Add the metric scores to obtain the total biosurvey score (see instructions in Tables 4.6 and 4.7).

TASK 3 **Determine the biological condition**

To determine the biological condition of the site, refer to Table 4.9, Biosurvey Scoring Guide.

TASK 4 **Return the lab sheets and metric worksheets to the program coordinator**

All remaining worksheets should be returned to the program coordinator once the site's final score has been determined. The program coordinator will determine how to proceed with the findings of the biological assessment (e.g., the data may be entered into a database or shared with a state or local agency). It is important that the biological advisor include documentation of any problems encountered in the process of monitoring, identifying macroinvertebrates, or analyzing the data.

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Note: References marked with **(k)** contain macroinvertebrate taxonomic keys.

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Save Our Streams

A citizens volunteer stream monitoring program

(**1**) Determine your stream-reach boundary; this is a stream length up to 100-meters, which may be more or less under certain circumstances. (**2**) Near the lower end of the reach (in the deepest portion of the run), collect water samples and analyze using the chemical tests you have available. You may use your collection container to observe watercolor and clarity and to determine water odors. (**3**) Measure the width-depth and velocity, and estimate the water level. (**4**) If you use a two-pole **kick-net**, collect a minimum of three benthic macro-invertebrate samples from the best riffle or runs within your stream reach. Use the table on page five to record information about your collections. (**5**) Evaluate the physical and habitat conditions; record information about known land use activities. (**6**) Sketch your reach or submit photographs with the survey, and add any other comments that you feel are important for evaluating the conditions of your stream study site.

Stream name ______________________ Survey date __________
Watershed ______________________ Station code __________
Latitude __________ Longitude __________ Directions to site __________

____

Survey completed by ______________________
Current weather conditions ______________________
Past weather conditions (last 3-days) ______________________
Affiliation ______________________ Email ______________________
Mailing address ______________________ Phone number __________
____

Water chemistry: Use the spaces below to record the results of your water chemistry analysis; attach additional sheets if necessary.

	Result	units		Result	units		Result	units
Temperature (C/F)			Conductivity			Alkalinity		
Dissolved oxygen			Nitrate/Nitrite			Metals (describe)		
pH			Phosphate			Fecal/E-coli		

Additional tests (describe and record results) ______________________

____

____

Physical conditions: Use the check boxes below to describe the conditions that closely resemble those of your stream. The extra lines are provided to write in any additional comments. You may see more than one type of condition; if so, be sure to indicate these on your survey (check all that apply). If multiple conditions are observed, always indicate the most dominant condition. If the condition you observe is not listed, describe it in the comment section.

Water clarity		Water color		Water odor		Surface foam	
Clear		None		None		None	
Murky		Brown		Fishy		Slight	
Milky		Black		Musky		Moderate	
Muddy		Orange/red		Rotten egg		High	
Other (describe)		Gray/White		Sewage			
		Green		Chemical			

Algae color		Algae abundance		Algae growth habit		Streambed color	
Light green		None		Even coating		Brown	
Dark green		Scattered		Hairy		Black	
Brown		Moderate		Matted		Green	
Other (describe)		Heavy		Floating		White/gray	
						Orange/red	

Physical condition comments: ______________________

____

____

Estimate the percentage of your reach that is shaded.

> 80	80-60	60-40	< 40
Excellent	Good	Marginal	Poor

Circle your estimate

Width and depth: Record the wetted width and depth of the channel's habitats (riffles, runs or pools). Choose one or more features to measure. Record the average depth from a minimum of four measure-ments (one of these should be from the deepest part of the habitat). The width should be measured from the widest section of the feature. Be sure to indicate the type(s) of habitat that you have chosen. **It is best to measure the width and depth when you determine the discharge**.

1. Width (feet)	________	Depth (feet)	________	Riffle	Run	Pool
2. Width (feet)	________	Depth (feet)	________			

Channel profiles: Width and depth measurements can be used to create a cross section profile within your reach. Choose a location in your reach across one of the channel types above. Stretch a tape from bank to bank and anchor it at both ends. Move from left to right facing in an upstream direction; measure the distance from the stream bottom to the top of the tape at selected intervals (i.e. every foot). Record your measurements in the table below. The table provides enough spaces for 20 measurements; if more are necessary you can create your own table on a separate piece of paper. Your tape measure will probably not start at zero so make sure to record the actual position of the tape as you measure across the channel.

Width intervals

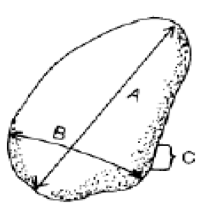
1	2	3	4	5	6	7	8	9	10
11	12	13	14	15	16	17	18	19	20

Depth measurements

1	2	3	4	5	6	7	8	9	10
11	12	13	14	15	16	17	18	19	20

Pebble count: Collect a minimum of 100-particles from your reach using a Zigzag method, percent habitat method or specific transects (e.g. every 10-meter). If you do not complete a pebble count, **always estimate** streambed composition from the riffles/runs chosen for your macroinvertebrate sample collections.

Indicate your method from the choices below.		Size Classes (Intermediate axis in millimeters)						
		Silt/clay < 0.06	Sand 0.06 – 2	Fine Gravel 2 – 24	Coarse Gravel 25 – 64	Cobble 65 – 255	Boulder 256 – 1096	Bedrock > 1096
Zigzag								
% Habitat								
10-m Transects								
Woody Debris Includes sticks, roots, leaves etc.								
Totals								

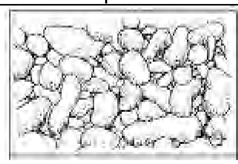
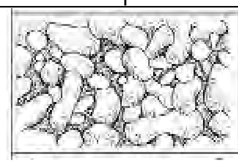
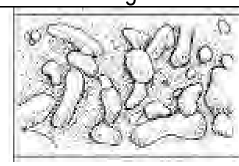


(A) Long axis (**Length**)
(B) Intermediate axis (**Width**)
(C) Short axis (**Height**)

Pebble counts require two people, one in the stream and one on shore. The person in the stream slowly walks upstream from bank to bank using one of the methods above. After each step the person reaches down without looking, picks up the first particle touched, and measures the intermediate axis with a ruler. The on-shore partner records the measurement. The process continues until 100 pebbles have been measured or the reach has been walked.

Station ____________ Date(s) ____________

Habitat conditions: Score each habitat condition using the scales provided. Add all of the scores to determine your overall habitat score and integrity rating. Feel free to describe additional features that you feel are important. See the next page for more information about sediment deposition.

Point values	20 19 18 17 16	15 14 13 12 11	10 9 8 7 6	5 4 3 2 1
Sediment deposition	Little or no formation of depositional features; < 20% of the reach affected.	Some increase in depositional features; 20-40% of the reach affected.	Moderate amounts of depositional features; 40-60% of the reach affected.	Heavy amounts of deposition; > 60% of the reach affected.
Rating	**O**ptimal	**S**uboptimal	**M**arginal	**P**oor
Embeddedness	 Fine sediments surrounds <10% of the spaces between the gravel, cobble and boulders.	 Fine sediment surrounds 10-30% of the spaces between the gravel, cobble and boulders.	 Fine sediment surrounds 30-60% of the spaces between the gravel, cobble and boulders.	 Fine sediment surrounds > 60% of the spaces between the gravel, cobble and boulders.
Rating	**O**ptimal	**S**uboptimal	**M**arginal	**P**oor

Embeddedness should be evaluated in riffles, prior to or during your macroinvertebrate collections.

Point values	10 9 8	7 6 5	4 3 2	1
Bank vegetative protection	> 90% of the banks are covered by natural vegetation; all levels (trees, shrubs and herbs) represented; disruption from grazing, mowing etc. minimal or absent; all plants allowed to grow naturally.	70-90% of the banks covered by natural vegetation; one level of plants may be missing or not well represented; some disruption of vegetation evident; > 50% of the potential plant height remains.	50-70% of the banks covered by natural vegetation; patches of bare soil may be present and closely cropped vegetation is common; < 50% of the potential plant heights remains.	< 50% of the banks covered by natural vegetation; disruption is high; vegetation has been removed or the potential plant heights are greatly reduced.
Left Right	**O**ptimal	**S**uboptimal	**M**arginal	**P**oor
Bank stability	Banks are stable; no evidence of erosion or bank failure; little or no potential for future problems.	Banks are moderately stable; infrequent areas of erosion occur, mostly shown by banks healed over.	Banks are moderately unstable; 60% of the reach has some areas of erosion; high potential for erosion during flooding events.	Banks are unstable; many have eroded areas (bare soils) along straight sections or bends; obvious bank collapse or failure; > 60% of the reach has erosion scars.
Left Right	**O**ptimal	**S**uboptimal	**M**arginal	**P**oor
Riparian buffer width	Mainly undisturbed vegetation > 60 ft; no evidence of human impacts such as parking lots, road beds, clear-cuts, mowed areas, crops, lawns etc.	Zone of undisturbed vegetation 40-60 ft; some areas of disturbance evident.	Zone of undisturbed vegetation 20-40 ft; disturbed areas common throughout the reach.	Zone of undisturbed vegetation < 20 ft; disturbed areas common throughout the entire reach.
Left Right	**O**ptimal	**S**uboptimal	**M**arginal	**P**oor

Totals		> 80	80 - 60	59 - 40	< 40
		Optimal	**S**uboptimal	**M**arginal	**P**oor

Habitat comments: ______________________________

 Station ____________ Date(s) ____________

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.

Land use: Indicate the land uses that you believe may be having an impact on your stream station. Use the letters (**S**) streamside, (**M**) within ¼ mile and (**W**) somewhere in the watershed, to indicate the approximate location of the disturbance and the numbers (**1**) slight, (**2**) moderate or (**3**) high, to represent the level of disturbance.

Active Construction			Pastureland			Single-family residences		
Mountaintop mining			Cropland			Sub-urban developments		
Deep mining			Intensive feedlots			Parking lots, strip-malls etc.		
Abandoned mining			Unpaved Roads			Paved Roads		
Logging			Trash dumps			Bridges		
Oil and gas wells			Landfills			Other (describe)		
Recreation (parks, trails etc.)			Industrial areas					

Land use comments: ______________________________ Pipes? | Yes | No |

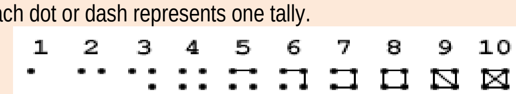
Describe the types of pipes observed and indicate if there is any discharge from the pipes. Also describe the colors and odors of the discharge. ______________________________

Photograph and **sketch your reach**: Use the space below or a separate piece of paper to draw your study reach. Indicate the direction of flow, north, sample locations and important features of the reach. Photographs are an excellent method for tracking changes, especially changes related to the condition of the habitat. Choose a minimum of two permanent locations from which to take your photos. Submit your photos with your survey data sheet.

Benthic macroinvertebrates: Assess your macroinvertebrate collections by counting and identifying to the family-level if possible. Use the table on the **next two pages** to record your collections data.

Note: Although streamside identification is possible, WV Save Our Streams Coordinator recommends preserving your samples using a full count or standard sub-sampling procedure in a well-lit and more comfortable setting.

The dot-dash tally method is a convenient way to record your data. Each dot or dash represents one tally.



Station ______________ Date(s) ______________

Insect Groups

Patterned stoneflies Taxa ___ Total ___	Winter stoneflies Taxa ___ Total ___	Roach-like stonefly Total ___
Giant stonefly Total ___	Brown stonefly Total ___	Spiny crawler mayfly Total ___
Square-gilled mayfly Total ___	Minnow mayflies Taxa ___ Total ___	Flatheaded mayfly Total ___
Brush-legged mayfly Total ___	Burrowing mayflies Taxa ___ Total ___	Net-spinning caddisflies Taxa ___ Total ___
Case-building caddisflies Taxa ___ Total ___	Free-living caddisfly Total ___	Common netspinner Total ___
Dragonflies Taxa ___ Total ___	Damselflies Taxa ___ Total ___	Riffle beetle Total ___
Long-toed beetle Total	Water penny Total ___	Other beetles (true bugs) Taxa ___ Total ___
Hellgrammite/Fishfly Total ___	Alderfly Total ___	Aquatic moth Total ___
Non-biting midge Total ___	Black fly Total ___	Crane fly Total ___
Watersnipe fly Total ___	Dance fly Total ___	Dixid midge Total ___

Station ______________________ Date(s) ______________________

Net-wing midge Total	Horse fly Total	Other fly larva Taxa Total

Non-Insect Groups

Crayfish Total	Scud/Sideswimmer Total	Aquatic sowbug Total
Water mite Total	Operculate snails Taxa Total	Non-operculate snails Taxa Total
Pea clam Total	Asian clam Total	Mussel Total
Flatworms Total	Aquatic worms Total	Leeches Total
Other aquatic invertebrates Taxa Total	Comments:	

Total Taxa	Total Number

Describe other aquatic life (e.g. fish, amphibians) collected or observed, as well as other indications that the reach is being used by other animals (i.e. birds, mammals, reptiles).

Discharge

Determine the discharge by using a flow meter (if available) or other methods such as the **float method** or a **velocity head rod** (VHR). Discharge should be measured from a run (area of the channel with fast moving water with no breaks in the surface such as protruding rocks). The more measurements collected the more accurate your discharge results will be. To convert inches into feet divide by 12. For example, if your depth measurement was 6-inches the result in feet would be 0.5. Indicate the methods chosen to measure the discharge and use the tables to record your results. Use the table on the next page to record your measurements.

Discharge method used

Float	Velocity Head Rod	Flow meter

Water Level

Low	Normal	High	Dry

Channel width ____________ feet

Use the table on the next page to record your velocity data

 Station ____________ Date(s) ____________

Distance (ft)	Depth (ft)	Velocity (ft/sec)	VHR (Rise-inches)	Float (sec)	Discharge (cfs)
1					
2					
3					
4					
5					
6					
7					
8					
9					
10					
11					
12					
13					
14					
15					
16					
17					
18					
19					
20					

Average Depth ____________________ feet

Cross Sectional Area (CSA) ________________ ft^2
(CSA = Average Depth x Width)

Discharge = CSA x Velocity
= ____________________ x ____________________
= ____________________ cfs (ft^3/sec)

If you use a float record your distance below and the number of seconds it took to travel the distance in the column indicated.

Float distance (feet) ______________

Use the table below to determine **VHR velocity** from the rises recorded above. The rises below are in inches.

Rise (R)	Velocity	Rise (R)	Velocity
¼	1.2	3 ¼	4.2
½	1.6	3 ½	4.3
¾	2.0	3 ¾	4.5
1	2.3	4	4.6
1 ¼	2.6	4 ¼	4.8
1 ½	2.8	4 ½	4.9
1 ¾	3.1	4 ¾	5.0
2	3.3	5	5.2
2 ¼	3.5	5 ¼	5.3
2 ½	3.7	5 ½	5.4
2 ¾	3.8	5 ¾	5.5
3	4.0	6	5.7

Additional comments: ____

Submit an original or clear copy of your survey to the coordinator at the address provided below.

WV Department of Environmental Protection
Save Our Streams Program
601 57th Street, SE
Charleston, WV 25304

Office: (304) 926-0499 (1040); Mobile: (304) 289-7630
E-mail: timothy.d.craddock@wv.gov
Web page: http://www.dep.wv.gov/sos

 Station ____________________ Date(s) ____________________

Level-two assessment

The **light blue** shaded boxes indicate that multiple families (**kinds**) are possible; tolerance values are provided.

Macroinvertebrates	Totals	Tolerance score	Number of kinds	Macroinvertebrates	Totals	Tolerance score	Number of kinds
1 Patterned stoneflies				6 Aquatic moth			
2 Winter stoneflies				4 Riffle beetle			
1 Roach-like stonefly				5 Long-toed beetle			
1 Giant stonefly				3 Water penny			
2 Little brown stonefly				5 Whirligig beetle			
3 Spiny crawler mayfly				7 Other beetles/bugs			
5 Square-gilled mayflies				3 Hellgrammite/Fishfly			
4 Minnow mayflies				6 Alderfly			
3 Flatheaded mayfly				8 Non-biting midge			
3 Brush-legged mayfly				6 Black fly			
5 Burrowing mayflies				4 Crane fly			
4 Net-spinning caddisflies				3 Watersnipe fly			
3 Case-building caddisflies				6 Dance fly			
5 Common netspinner				5 Dixid midge			
3 Free-living caddisfly				2 Net-wing midge			
4 Dragonflies				7 Horse fly			
7 Damselflies				7 Other fly larva			
Non-Insect Groups							
5 Crayfish				5 Pea clam			
5 Scud/Sideswimmer				6 Asian clam			
7 Aquatic sowbug				4 Mussel			
6 Water mite				5 Operculate snails			
10 Aquatic worms				7 Non-operculate snails			
10 Leeches				Other invertebrates (**Describe**)			
7 Flatworms							

Complete your calculations using the metrics below. These metrics are combined to determine your overall score and integrity rating.	**Total Number**	**Total Tolerance**	**Total Kinds**

Comments: ______________________

Metrics	Results	Points	**8**	**6**	**4**	**2**
1. Total Taxa			> 18	18 - 13	12 - 8	< 8
2. EPT Taxa			> 10	10 - 7	6 - 4	< 4
3. Biotic Index			< 4.0	4.0 – 5.0	5.1 – 6.0	> 6.0
4. % EPT Abundance			> 80	80 - 60	59.9 - 40	< 40
5. % Tolerant			< 2	2 - 10	10.1 - 30	> 30

Stream Score ______

Integrity Rating

> 32	32 - 24	23 - 16	< 16
Optimal	Suboptimal	Marginal	Poor

1. **Total Taxa** is simply the total number of families collected.
2. **EPT Taxa** is the total number of families within the orders of **Ephemeroptera**, **Plecoptera** and **Trichoptera**.
3. **Biotic Index** is calculated by multiplying the organism by its tolerance value to determine a tolerance score. The total tolerance is then divided by the total number of organisms collected.
4. **% EPT Abundance** is calculated by dividing total number of organisms within the orders Ephemeroptera, Plecoptera and Trichoptera by the total number collected. This result is multiplied by 100 to determine the percentage.
5. **% Tolerant** is calculated by dividing the number of tolerant organisms (≥ 7) by the total number collected. This result is multiplied by 100 to determine the percentage.

- The **Stream Score** is the sum of all five point values.

Station ______________ Date(s) ______________

Water quality monitoring is defined here 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.

Volunteers, as well as state and local water quality professionals, 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.

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 might 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 antidegradation statements that 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 (see Chapter 4) 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.

Designing a water quality monitoring program

The first step in designing a water quality monitoring program is to determine the purpose of the monitoring. This will help you select which parameters to monitor. The program steering committee should make this decision based on factors such as:

- Types of water quality problems and pollution sources that will likely be encountered (Table 5.1)
- Cost of available monitoring equipment
- Precision and accuracy of available monitoring equipment
- Capabilities of the volunteers

Because of the expense and difficulty involved, volunteers generally do not monitor for toxic substances such as heavy metals and organic chemicals (e.g., pesticides, herbicides, solvents, and PCBs). They might, however, collect water samples for analysis at accredited labs.

The parameters most commonly monitored by volunteers in streams are discussed in detail in this chapter. They include stream flow, dissolved oxygen and biochemical oxygen demand, temperature, pH, turbidity, phosphorus, nitrates, total solids, conductivity, total alkalinity, and fecal bacteria. Of these, the first five are the most basic and should form the foundation of almost any volunteer water quality monitoring program.

Relatively inexpensive and simple-to-use kits are available from scientific supply houses to monitor these pollutants. Many volunteer programs use these kits effectively. Meters and sophisticated lab equipment may be more accurate, but they are also more expensive, less flexible (e.g., meters generally have to be read in the field), and require periodic calibration. This chapter discusses specific equipment and sampling considerations for each parameter, and usually describes several ap-

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, enterococcis), 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

Table 5.1

Sources and associated pollutants

A volunteer water quality monitoring program should be geared to the types of watershed land uses most often encountered.

proaches to monitor them. Table 5-2 lists methods available for monitoring key parameters, including the preferred testing site (lab or field).

General preparation and sampling considerations

The sections that follow will detail specific sampling and equipment considerations and analytical procedures for each of the most common water quality parameters. There are, however, two general tasks that are accomplished anytime water samples are taken. These are discussed below.

Task 1 — Preparation of Sampling Containers

Reused sample containers and glassware must be cleaned and rinsed before the first sampling run and after each run by following either Method A or Method B described below. The most suitable method depends on the parameter being measured.

Method A: General Preparation of Sampling Containers

The following method should be used when preparing all sample containers and glassware for monitoring conductivity, total solids, turbidity, pH, and total alkalinity. Wear latex gloves!

1. Wash each sample bottle or piece of glassware with a brush and phosphate-free detergent.
2. Rinse three times with cold tap water.
3. Rinse three times with distilled or deionized water.

Method B: Acid Wash Procedure for Preparing Sampling Containers

This method should be used when preparing all sample containers and glassware for monitoring nitrates and phosphorus. Wear latex gloves!

1. Wash each sample bottle or piece of glassware with a brush and phosphate-free detergent.
2. Rinse three times with cold tap water.
3. Rinse with 10 percent hydrochloric acid.
4. Rinse three times with deionized water.

Task 2 — Collecting Samples

In general, sample away from the streambank in the main current. Never sample stagnant water. 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, carefully wade into the center current to collect the sample.

A boat will be required for deep sites. Try to maneuver the boat into the center of the main current to collect the water sample.

When collecting a water sample for analysis in the field or at the lab, follow the steps below.

For Whirl-pak® Bags

1. Label the bag with the site number, date, and time.
2. Tear off the top of the bag along the perforation above the wire tab just prior to sampling (Fig. 5.1).

Figure 5.1

Sketch of a Whirl-pak® bag

Volunteers can be easily trained to use these factory-sealed, disposable water sample collection bags.

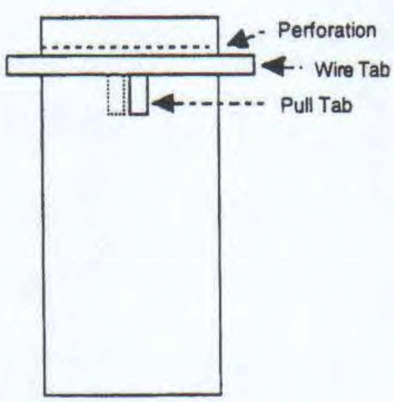


Table 5.2

Summary of chemical monitoring methods

Volunteers can measure some parameters in the field or in the laboratory.

Method	Location (Lab or Field)	Comments
Dissolved Oxygen (DO)		
Winkler with eye dropper	Either	If lab, the sample is fixed in field and titrated in lab; must be measured within 8 hours of collection.
Winkler with digital titrator or buret	Either	
Meter	Field	The meter is fragile and must be handled carefully.
Biochemical Oxygen Demand (BOD)		
Winkler with eye dropper	1st part - Either 2nd part - Lab	If lab, the sample is fixed in field and titrated in lab; must be measured within 6 hours of collection.
Winkler with digital titrator or buret	1st part - Either 2nd part - Lab	If lab, the sample is fixed in field and titrated in lab; must be measured within 6 hours of collection.
Meter	1st part - Either 2nd part - Lab	The meter is fragile and must be handled carefully; must be measured within 6 hours of collection.
Temperature		
Thermometer	Field	Cannot be done in the lab.
pH		
Color comparator	Either	If lab, measured ASAP within 2 hours of collection.
pH "Pocket Pal"	Either	If lab, measured ASAP within 2 hours of collection.
Meter	Either	If lab, measured ASAP within 2 hours of collection.
Turbidity		
Meter	Either	If lab, measured within 24 hours of collection.
Total Orthophosphate		
Ascorbic acid w/ color comparator	Either	If lab, measured within 48 hours of collection.
Ascorbic acid w/ spectrophotometer	Either	If lab, measured within 48 hours of collection.
Nitrate		
Cadmium reduction w/ color comparator	Either	If lab, measured within 48 hours of collection.
Cadmium reduction w/ spectrophotometer	Either	If lab, measured within 48 hours of collection.
Total Solids		
Oven drying/weighing	Lab	Must be measured within 7 days of collection.
Conductivity		
Meter	Either	If lab, measured within 28 days of collection.
Total Alkalinity		
Titration	Either	If lab, measured within 24 hours of collection.
Fecal Bacteria		
Membrane filtration	Lab	Must be measured within 6 hours of collection.

Avoid touching the inside of the bag. If you accidentally touch the inside of the bag, use another one.

3. *Wading*. Try to disturb as little bottom sediment as possible. In any case, be careful not to collect water that contains bottom sediment. Stand facing upstream. Collect the water sample in front of you.

   Boat. Carefully reach over the side and collect the water sample on the upstream side of the boat.

4. Hold the two white pull tabs in each hand and lower the bag into the water on your upstream side with the opening facing upstream. Open the bag midway between the surface and the bottom by pulling the white pull tabs. The bag should begin to fill with water. You may need to "scoop" water into the bag by drawing it through the water upstream and away from you. Fill the bag no more than 3/4 full!

5. Lift the bag out of the water. Pour out excess water. Pull on the wire tabs to close the bag. Continue holding the wire tabs and flip the bag over at least 4-5 times quickly to seal the bag. Don't try to squeeze the air out of the top of the bag. Fold the ends of the wire tabs together at the top of the bag, being careful not to puncture the bag. Twist them together, forming a loop.

6. Fill in the bag number and/or site number on the appropriate field data sheet. This is important! It is the only way the lab coordinator know which bag goes with which site.

7. If samples are to be analyzed in a lab, place the sample in the cooler with ice or cold packs. Take all samples to the lab.

For Screw-cap Bottles

To collect water samples using screw-cap sample bottles, use the following procedures (Fig. 5.2 and 5.3):

Figure 5.2

Getting into position to take a water sample

Volunteers should sample in the main current, facing upstream.

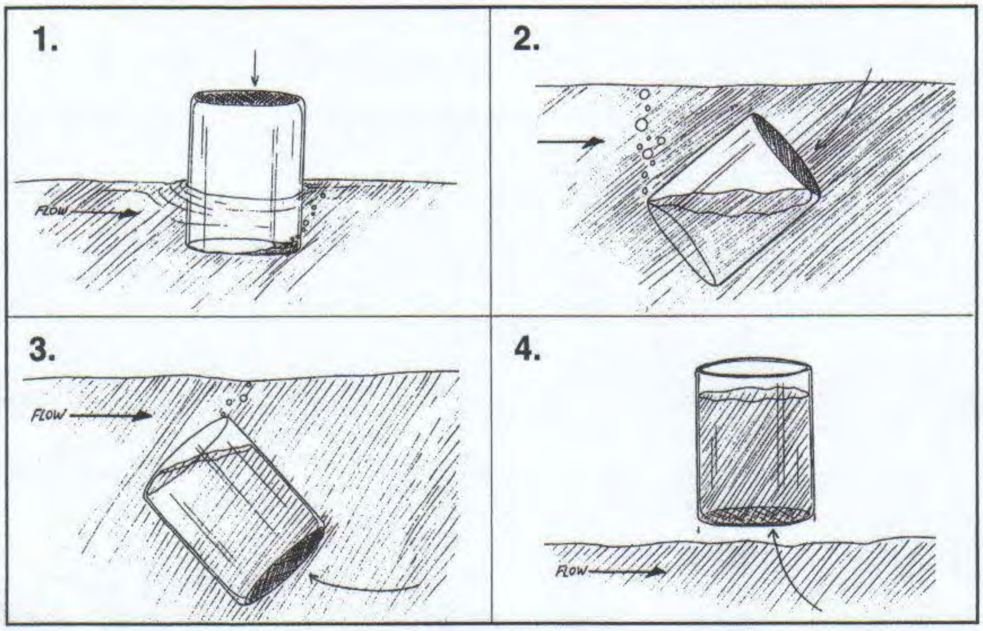


Figure 5.3

Taking a water sample

Turn the bottle into the current and scoop in an upstream direction.

1. Label the bottle with the site number, date, and time.
2. Remove the cap from the bottle just before sampling. Avoid touching the inside of the bottle or the cap. If you accidentally touch the inside of the bottle, use another one.
3. *Wading*. Try to disturb as little bottom sediment as possible. In any case, be careful not to collect water that has sediment from bottom disturbance. Stand facing upstream. Collect the water sample on your upstream side, in front of you. You may also tape your bottle to an extension pole to sample from deeper water.

   Boat. Carefully reach over the side and collect the water sample on the upstream side of the boat.
4. Hold the bottle near its base and plunge it (opening downward) below the water surface. If you are using an extension pole, remove the cap, turn the bottle upside down, and plunge it into the water, facing upstream. 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.
5. Turn the bottle underwater into the current and away from you. In slow-moving stream reaches, push the bottle underneath the surface and away from you in an upstream direction.
6. Leave a 1-inch air space (Except for DO and BOD samples). Do not fill the bottle completely (so that the sample can be shaken just before analysis). Recap the bottle carefully, remembering not to touch the inside.
7. Fill in the bottle number and/or site number on the appropriate field data sheet. This is important because it tells the lab coordinator which bottle goes with which site.
8. If the samples are to be analyzed in the lab, place them in the cooler for transport to the lab.

QUALITY ASSURANCE, QUALITY CONTROL, and QUALITY ASSESSMENT MEASURES

Quality assurance/quality control measures are those activities you undertake to demonstrate the accuracy (how close to the real result you are) and precision (how reproducible your results are) of your monitoring. Quality Assurance (QA) generally refers to a broad plan for maintaining quality in all aspects of a program. This plan should describe how you will undertake your monitoring effort: proper documentation of all your procedures, training of volunteers, study design, data management and analysis, and specific quality control measures. Quality Control (QC) consists of the steps you will take to determine the validity of specific sampling and analytical procedures. Quality assessment is your assessment of the overall precision and accuracy of your data, after you've run the analyses.

Quality Control and Assessment Measures: Internal Checks

Internal checks are performed by the project field volunteers, staff, and lab.

- *Field Blanks.* A trip blank (also known as a field blank) is de-ionized water which is treated as a sample. It is used to identify errors or contamination in sample collection and analysis.
- *Negative and Positive Plates (for bacteria).* A negative plate results when the buffered rinse water (the water used to rinse down the sides of the filter funnel during filtration) has been filtered the same way as a sample. This is different from a field blank in that it contains reagents used in the rinse water. There should be no bacteria growth on the filter after incubation. It is used to detect laboratory bacteria contamination of the sample. Positive plates result when water known to contain bacteria (such as wastewater treatment plant influent) is filtered the same way as a sample. There should be plenty of bacteria growth on the filter after incubation. It is used to detect procedural errors or the presence of contaminants in the laboratory analysis that might inhibit bacteria growth.
- *Field Duplicates.* A field duplicate is a duplicate river sample collected by the same team or by another sampler or team at the same place, at the same time. It is used to estimate sampling and laboratory analysis precision.
- *Lab Replicates.* A lab replicate is a sample that is split into subsamples at the lab. Each subsample is then analyzed and the results compared. They are used to test the precision of the laboratory measurements. For bacteria, they are used to obtain an optimal number of bacteria colonies on filters for counting purposes.
- *Spike Samples.* A known concentration of the indicator being measured is added to the sample. This should increase the concentration in the sample by a predictable amount. It is used to test the accuracy of the method.
- *Calibration Blank.* A calibration blank is de-ionized water processed like any of the samples and used to "zero" the instrument. It is the first "sample" analyzed and used to set the meter to zero. This is different from the field blank in that it is "sampled" in the lab. It is used to check the measuring instrument periodically for "drift" (the instrument should always read "0" when this blank is measured). It can also be compared to the field blank to pinpoint where contamination might have occurred.
- *Calibration Standards.* Calibration standards are used to calibrate a meter. They consist of one or more "standard concentrations" (made up in the lab to specified concentrations) of the indicator being measured, one of which is the calibration blank. Calibration standards can be used to calibrate the meter before running the test, or they can be used to convert the units read on the meter to the reporting units (for example, absorbance to milligrams per liter).

Quality Control And Assessment Measures: External Checks

External checks are performed by non-volunteer field staff and a lab (also known as a "quality control lab"). The results are compared with those obtained by the project lab.

- *External Field Duplicates.* An external field duplicate is a duplicate river sample collected and processed by an independent (e.g., professional) sampler or team at the same place at the same time as regular river samples. It is used to estimate sampling and laboratory analysis precision.
- *Split Samples.* A split sample is a sample that is divided into two subsamples at the lab. One subsample is analyzed at the project lab and the other is analyzed at an independent lab. The results are compared.
- *Outside Lab Analysis of Duplicate Samples.* Either internal or external field duplicates can be analyzed at an independent lab. The results should be comparable with those obtained by the project lab.

- *Knowns.* The quality control lab sends samples for selected indicators, labeled with the concentrations, to the project lab for analysis prior to the first sample run. These samples are analyzed and the results compared with the known concentrations. Problems are reported to the quality control lab.
- *Unknowns.* The quality control lab sends samples to the project lab for analysis for selected indicators, prior to the first sample run. The concentrations of these samples are unknown to the project lab. These samples are analyzed and the results reported to the quality control lab. Discrepancies are reported to the project lab and a problem-identification and solving process follows.

The table below shows the applicability of common quality control measures to the water quality indicators covered in this manual.

Steps To Quality Control

1. Consult with your technical committee and/or program advisor to help you determine quality assurance/quality control measures you will use to answer your questions and meet your data quality requirements
2. Locate a quality control lab — an independent lab that can run external checks for you.
3. Determine which quality checks you have the resources and capabilities to carry out. Your human and financial resources and expertise might limit the water quality indicators you can monitor.

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USEPA. 1984. *Guidance for preparation of combined work/quality assurance project plans for environmental monitoring.* ORWS QA-1, U.S. Environmental Protection Agency, Office of Water Regulations and Standards. Washington DC, May.

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Common Quality Control Measures

	Dissolved Oxygen	Temp-erature	ph	Tur-bidity	Phos-phorus	Nitrates	Total Solids	Con-ductivity	Total Alkalinity	Fecal Bacteria
Internal Checks										
Field blanks				✓	✓	✓	✓	✓		✓
Field duplicates	✓	✓	✓	✓	✓	✓	✓	✓	✓	✓
Lab replicates	✓		✓	✓	✓	✓	✓	✓	✓	✓ [b]
Positive plates										✓
Negative plates										✓
Spike samples					✓	✓			✓	✓
Calibration blank				✓	✓	✓		✓		
Calibration standard	✓ [a]		✓	✓	✓	✓		✓		
External Checks										
External field duplicates			✓	✓	✓	✓	✓	✓	✓	✓
Split lab analysis			✓	✓	✓	✓	✓	✓	✓	✓
Outside lab analysis	✓			✓	✓	✓	✓	✓	✓	✓
Knowns	✓		✓	✓	✓	✓		✓	✓	✓
Unknowns	✓		✓	✓	✓	✓	✓	✓	✓	✓

a - using an oxygen-saturated sample
b - using subsamples of different sizes

5.1 Stream Flow

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 volunteer 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:

$$\text{Flow} = \frac{A\,L\,C}{T}$$

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

How to Measure and Calculate Stream Flow

TASK 1 **Prepare before leaving for the sampling site**

Refer to pages 19-21 for details on confirming sampling date and time, safety considerations, checking supplies, and checking weather and directions. In addition to the standard sampling equipment and apparel, when measuring and calculating flow, include the following equipment:

- Ball of heavy-duty string, four stakes, and a hammer to drive the stakes into the ground. The string will be stretched across the width of the stream perpendicular to shore at two locations. The stakes are to anchor the string on each bank to form a transect line.
- Tape measure (at least 20 feet)
- Waterproof yardstick or other implement to measure water depth
- Twist ties (to mark off intervals on the string of the transect line)
- An orange and a fishing net (to scoop the orange out of the stream)
- Stopwatch (or watch with a second hand)
- Calculator (optional)

TASK 2 **Select a stretch of stream**

The stream stretch chosen for the measurement of discharge should be straight (no bends), at least 6 inches deep, and should not contain an area of slow water such as a pool. Unobstructed riffles or runs are ideal. The length that you select will be equal to *L* in solving the flow equation. Twenty feet is a standard length used by many programs. Measure your length and mark the upper and lower end by running a transect line across the stream perpendicular to the shore using the string and stakes (Fig. 5.4). The string should be taut and near the water surface. The upstream transect is Transect #1 and the downstream one is Transect #2.

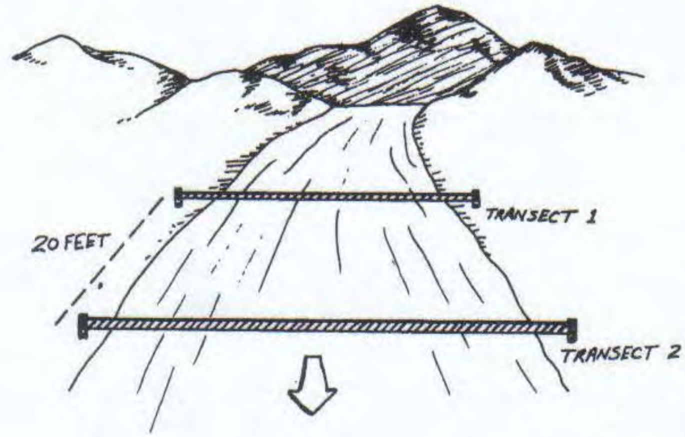


Figure 5.4

A diagram of a 20- foot transect

TASK 3 **Calculate the average cross-sectional area**

Cross-sectional area (*A* in the formula) is the product of stream width multiplied by average water depth. To calculate the average cross-sectional area for the study stream reach, volunteers should determine the cross-sectional area for each transect, add the results together, and then divide by 2 to determine the average cross-sectional area for the stream reach.

To measure cross-sectional area:

1. Determine the average depth along the transect by marking off equal intervals along the string with the twist ties. The intervals can be one-fourth, one-half, and three-fourths of the distance across the stream. Measure the water's depth at each interval point (Fig. 5.5). To calculate average depth for each transect, divide the total of the three depth measurements by 4. (You divide by 4 instead of 3 because you need to account for the 0 depths that occur at the shores.) In the example shown in

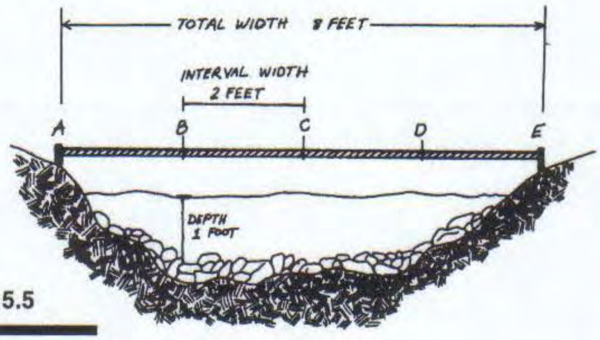


Figure 5.5

A cross section view to measure stream width and depth

Figure 5.6

A sample calculation of average cross-sectional area.

Figure 5.6, the average depth of Transect #1 is 0.575 feet and the average depth of Transect #2 is 0.625 feet.

2. Determine the width of each transect by measuring the distance from shoreline to shoreline. Simply add together all the interval widths for each transect to determine its width. In the Figure 5.6 example, the width of Transect #1 is 8 feet and the width of Transect #2 is 10 feet.
3. Calculate the cross-sectional area of each transect by multiplying width times average depth. The example given in Figure 5.6 shows that the average cross-sectional area of Transect #1 is 4.60 square feet and the average cross-sectional area of Transect #2 is 6.25 square feet.
4. To determine the average cross-sectional area of the entire stream reach (*A* in the formula), add together the average cross-sectional area of each transect and then divide by 2. The average cross-sectional area for the stream reach in Figure 5.6 is 5.42 square feet.

Determining Average Cross-Sectional Area (A)

Transect #1 (upstream)

	Interval width (feet)	Depth (feet)	
A to B =	2.0	1.0	(at B)
B to C =	2.0	0.8	(at C)
C to D =	2.0	0.5	(at D)
D to E =	2.0	0.0	(shoreline)
Totals	8.0	2.3	

Average depth = 2.3 / 4 = 0.575 feet

Cross-sectional area of Transect #1

= Total width X Average depth
= 8 ft X 0.575
= 4.60 ft²

Transect #2 (downstream)

	Interval width (feet)	Depth (feet)	
A to B =	2.5	1.1	(at B)
B to C =	2.5	1.0	(at C)
C to D =	2.5	0.4	(at D)
D to E =	2.5	0.0	(shoreline)
	10.0	2.5	

Average depth = 2.5 / 4 = 0.625 feet

Cross-sectional area of Transect #2

= Total width X Average depth
= 10.0 ft X 0.625
= 6.25 ft²

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

= (4.60 ft² + 6.25 ft²) / 2
= 5.42 ft²

Task 4 **Measure travel time**

Volunteers should time with a stop-watch how long it takes for an orange (or some other object) to float from the up-stream to the downstream transect. An orange is a good object to use because it has enough buoyancy to float just below the water surface. It is at this position that maximum velocity typically occurs.

The volunteer who lets the orange go at the upstream transect should position it so it flows into the fastest current. The clock stops when the orange passes fully under the downstream transect line. Once under the transect line, the orange can be scooped out of the water with the fishing net. This "time of travel" measurement should be conducted at least three times and the results averaged—the more trials you do, the more accurate your results will be. The averaged results are equal to *T* in the formula. It is a good idea to float the orange at different distances from the bank to get various velocity estimates. You should discard any float trials if the object gets hung up in the stream (by cobbles, roots, debris, etc.)

Task 5 **Calculate flow**

Recall that flow can be calculated using the equation:

$$\text{Flow} = \frac{A\,L\,C}{T}$$

Continuing the example in Fig. 5.6. say the average time of travel for the orange between Transect #1 and #2 is 15 seconds and the stream had a rocky bottom. The calculation of flow would be:

A = 5.42 ft^2

L = 20 ft

C = 0.8 (coefficient for a rocky-bottom stream)

T = 15 seconds

$$\text{Flow} = \frac{(5.42\ ft^2)\ (20\ ft)\ (0.8)}{15\ sec.}$$

$$\text{Flow} = \frac{86.72\ ft^3}{15\ sec.}$$

$$\text{Flow} = 5.78\ ft^3/sec.$$

Task 6 **Record flow on the data form**

On the following page is a form volunteers can use to calculate flow of a stream.

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.

West Virginia Save Our Streams recommends using the velocity head rod method to measure velocity in small streams. This is a relatively simple and very inexpensive procedure. Go to the program's web page for more information.

5.2 Dissolved Oxygen and Biochemical Oxygen Demand

What is dissolved oxygen and why is it important?

The stream system both produces and consumes oxygen. It gains oxygen from the atmosphere and from plants as a result of photosynthesis. Running water, because of its churning, dissolves more oxygen than still water, such as that in a reservoir behind a dam. Respiration by aquatic animals, decomposition, and various chemical reactions consume oxygen.

Wastewater from sewage treatment plants often contains organic materials that are decomposed by microorganisms, which use oxygen in the process. (The amount of oxygen consumed by these organisms in breaking down the waste is known as the biochemical oxygen demand or BOD. A discussion of BOD and how to monitor it is included at the end of this section.) Other sources of oxygen-consuming waste include stormwater runoff from farmland or urban streets, feedlots, and failing septic systems.

Oxygen is measured in its dissolved form as dissolved oxygen (DO). If more oxygen is consumed than is produced, dissolved oxygen levels decline and some sensitive animals may move away, weaken, or die.

DO levels fluctuate seasonally and over a 24-hour period. They vary with water temperature and altitude. Cold water holds more oxygen than warm water (Table 5.3) and water holds less oxygen at higher altitudes. Thermal discharges, such as water used to cool machinery in a manufacturing plant or a power plant, raise the temperature of water and lower its oxygen content. Aquatic animals are most vulnerable to lowered DO levels in the early morning on hot summer days when stream flows are low, water temperatures are high, and aquatic plants have not been producing oxygen since sunset.

Sampling and Equipment Considerations

In contrast to lakes, where DO levels are most likely to vary vertically in the water column, the DO in rivers and streams changes more horizontally along the course of the waterway. This is especially true in smaller, shallower streams. In larger, deeper rivers, some vertical stratification of dissolved oxygen might occur. The DO levels in and below riffle areas, waterfalls, or dam spillways are typically higher than those in pools and slower-moving stretches. If you wanted to measure the effect of a dam, it would be important to sample for DO behind the dam, immediately below the spillway, and upstream of the dam. Since DO levels are critical to fish, a good place to sample is in the pools that fish tend to favor or in the spawning areas they use.

An hourly time profile of DO levels at a sampling site is a valuable set of data because it shows the change in DO levels from the low point just before sunrise to the high point sometime in the midday. However, this might not be practical for a volunteer monitoring program. It is important to note the time of your DO sampling to help judge when in the daily cycle the data were collected.

DO is measured either in milligrams per liter (mg/L) or "percent saturation." Milligrams per liter is the amount of oxygen in a liter of water. Percent saturation is the amount of oxygen in a liter of water relative to the total amount of oxygen that the water can hold at that temperature.

Temperature (°C)	DO (mg/L)	Temperature (°C)	DO (mg/L)
0	14.60	23	8.56
1	14.19	24	8.40
2	13.81	25	8.24
3	13.44	26	8.09
4	13.09	27	7.95
5	12.75	28	7.81
6	12.43	29	7.67
7	12.12	30	7.54
8	11.83	31	7.41
9	11.55	32	7.28
10	11.27	33	7.16
11	11.01	34	7.05
12	10.76	35	6.93
13	10.52	36	6.82
14	10.29	37	6.71
15	10.07	38	6.61
16	9.85	39	6.51
17	9.65	40	6.41
18	9.45	41	6.31
19	9.26	42	6.22
20	9.07	43	6.13
21	8.90	44	6.04
22	8.72	45	5.95

Table 5.3

Maximum dissolved oxygen concentrations vary with temperature

DO samples are collected using a special BOD bottle: a glass bottle with a "turtleneck" and a ground glass stopper. You can fill the bottle directly in the stream if the stream is wadable or boatable, or you can use a sampler that is dropped from a bridge or boat into water deep enough to submerse the sampler. Samplers can be made or purchased.

Dissolved oxygen is measured primarily either by using some variation of the Winkler method or by using a meter and probe.

Winkler Method

The Winkler method involves filling a sample bottle completely with water (no air is left to bias the test). The dissolved oxygen is then "fixed" using a series of reagents that form an acid compound that is titrated. Titration involves the drop-by-drop addition of a reagent that neutralizes the acid compound and causes a change in the color of the solution. The point at which the color changes is the "endpoint" and is equivalent to the amount of oxygen dissolved in the sample. The sample is usually fixed and titrated in the field at the sample site. It is possible, however, to prepare the sample in the field and deliver it to a lab for titration.

Dissolved oxygen field kits using the Winkler method are relatively inexpensive, especially compared to a meter and probe. Field kits run between $35 and $200, and each kit comes with enough reagents to run 50 to 100 DO tests. Replacement reagents are inexpensive, and you can buy them already measured out for each test in plastic pillows.

You can also buy the reagents in larger quantities, in bottles, and measure them out with a volumetric scoop. The advantage of the pillows is that they have a longer shelf life and are much less prone to contamination or spillage. The advantage of buying larger quantities in bottles is that the cost per test is considerably less.

The major factor in the expense of the kits is the method of titration they use—eyedropper, syringe-type titrator, or digital titrator. Eyedropper and syringe-type titration is less precise than digital titration because a larger drop of titrant is allowed to pass through the dropper opening and, on a micro-scale, the drop size (and thus the volume of titrant) can vary from drop to drop. A digital titrator or a buret (which is a long glass tube with a tapered tip like a pipet) permits much more precision and uniformity in the amount of titrant that is allowed to pass.

If your program requires a high degree of accuracy and precision in DO results, use a digital titrator. A kit that uses an eye dropper-type or syringe- type titrator is suitable for most other purposes. The lower cost of this type of DO field kit might be attractive if you are relying on several teams of volunteers to sample multiple sites at the same time.

Meter and Probe

A dissolved oxygen meter is an electronic device that converts signals from a probe that is placed in the water into units of DO in milligrams per liter. Most meters and probes also measure temperature. The probe is filled with a salt solution and has a selectively permeable membrane that allows DO to pass from the stream water into the salt solution. The DO that has diffused into the salt solution changes the electric potential of the salt solution and this change is sent by electric cable to the meter, which converts the signal to milligrams per liter on a scale that the volunteer can read.

DO meters are expensive compared to field kits that use the titration method. Meter/probe combinations run between $500 and $1,200, including a long cable to connect the probe to the meter. The advantage of a meter/probe is that you can measure DO and temperature quickly at any point in the stream that you can reach with the probe. You can also measure the DO levels at a certain point on a continuous basis. The results are read directly as milligrams per liter, unlike the titration methods, in which the final titration result might have to be converted by an equation to milligrams per liter.

However, DO meters are more fragile than field kits, and repairs to a damaged meter can be costly. The meter/probe must be carefully maintained, and it must be calibrated before each sample run and, if you are doing many tests, in between samplings. Because of the expense, a volunteer program might have only one meter/probe. This means that only one team of samplers can sample DO and they will have to do all the sites. With field kits, on the other hand, several teams can sample simultaneously.

Laboratory Testing of Dissolved Oxygen

If you use a meter and probe, you must do the testing in the field; dissolved oxygen levels in a sample bottle change quickly due to the decomposition of organic material by microorganisms or the production of oxygen by algae and other plants in the sample. This will lower your DO reading. If you are using a variation of the Winkler method, it is possible to "fix" the sample in the field and then deliver it to a lab for titration. This might be preferable if you are sampling under adverse conditions or if you want to reduce the time spent collecting samples. It is also a little easier to titrate samples in the lab, and more quality control is possible because the same person can do all the titrations.

How to collect and analyze samples

The procedures for collecting and analyzing samples for dissolved oxygen consist of the following tasks:

TASK 1 **Prepare before leaving for the sampling site**

Refer to pages 19-21 for details on confirming sampling date and time, safety considerations, checking supplies, and checking weather and directions. In addition to the standard sampling equipment and apparel, when sampling for dissolved oxygen, include the following equipment:

If Using the Winkler Method

- Labels for sample bottles
- Field kit and instructions for DO testing
- Enough reagents for the number of sites to be tested
- Kemmerer, Van Dorn, or home-made sampler to collect deep-water samples
- A numbered glass BOD bottle with a glass stopper (1 for each site)
- Data sheet for dissolved oxygen to record results

If Using a Meter and Probe

- DO meter and probe (electrode) (NOTE: Confirm that the meter has been calibrated according to the manufacturer's instructions.)
- Operating manual for the meter and probe
- Extra membranes and electrolyte solution for the probe
- Extra batteries for the meter
- Extension pole
- Data sheet for dissolved oxygen to record results

TASK 2 Confirm that you are at the proper location

The directions for sampling should provide specific information about the exact point in the stream from which you are to sample; e.g., "approximately 6 feet out from the large boulder downstream from the west side of the bridge." If you are not sure you are in the exact spot, record a detailed description of where you took the sample so that it can be compared to the actual site later.

TASK 3 Collect samples and fill out the field data sheet

Winkler Method

Use a BOD bottle to collect the water sample. The most common sizes are 300 milliliters (mL) and 60 mL. Be sure that you are using the correct volume for the titration method that will be used to determine the amount of DO. There is usually a white label area on the bottle, and this may already be numbered. If so, be sure to record that number on the field data sheet. If your bottle is not already numbered, place a label on the bottle (not on the cap because a cap can be inadvertently placed on a different bottle) and use a waterproof marker to write in the site number.

If you are collecting duplicate samples, label the duplicate bottle with the correct code, which should be determined prior to sampling by the lab supplying the bottles. Use the following procedure for collecting a sample for titration by the Winkler method:

1. Remember that the water sample must be collected in such a way that you can cap the bottle while it is still submerged. That means that you must be able to reach into the water with both arms and the water must be deeper than the sample bottle.
2. Carefully wade into the stream. Stand so that you are facing one of the banks.
3. Collect the sample so that you are not standing upstream of the bottle. Remove the cap of the BOD bottle. Slowly lower the bottle into the water, pointing it downstream, until the lower lip of the opening is just submerged. Allow the water to fill the bottle very gradually, avoiding any turbulence (which would add oxygen to the sample). When the

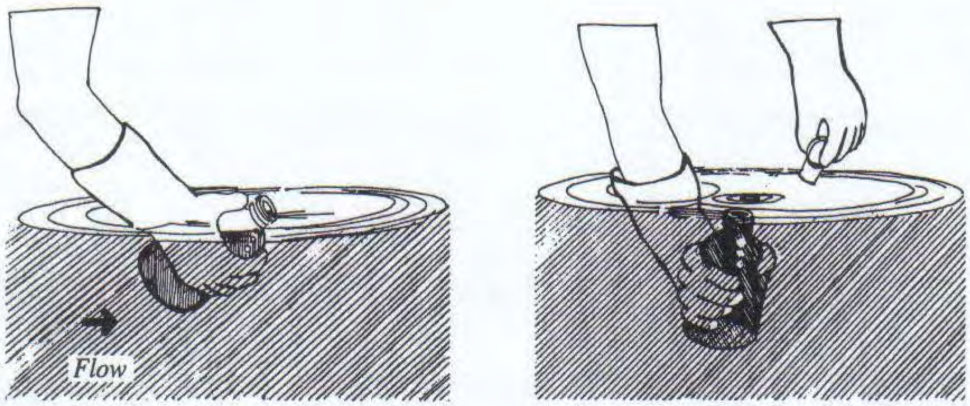


Figure 5.7

Taking a water sample for DO analysis

Point the bottle downstream and fill gradually. Cap underwater when full.

water level in the bottle has stabilized (it won't be full because the bottle is tilted), slowly turn the bottle upright and fill it completely. Keep the bottle under water and allow it to overflow for 2 or 3 minutes to ensure that no air bubbles are trapped.

4. Cap the bottle while it is still submerged. Lift it out of the water and look around the "collar" of the bottle just below the bottom of the stopper. If you see an air bubble, pour out the sample and try again.
5. "Fix" the sample immediately following the directions in your kit:
   - Remove the stopper and add the fixing reagents to the sample.
   - Immediately insert the stopper so air is not trapped in the bottle and invert several times to mix. This solution is caustic. Rinse your hands if you get any solution on them. An orange-brown flocculent precipitate will form if oxygen is present.
   - Wait a few minutes until the floc in the solution has settled. Again invert the bottle several times and wait until the floc has settled. This ensures complete reaction of the sample and reagents. The sample is now fixed, and atmospheric oxygen can no longer affect it.

   If you are taking the sample to the lab for titration, no further action is necessary. You can store the sample in a cooler for up to 8 hours before titrating it in a lab. If you are titrating the sample in the field, see *Task 4: Analyze the Samples.*

Using a DO Meter

If you are using a dissolved oxygen meter, be sure that it is calibrated immediately prior to use. Check the cable connection between the probe and the meter. Make sure that the probe is filled with electrolyte solution, that the membrane has no wrinkles, and that there are no bubbles trapped on the face of the membrane. You can do a field check of the meter's accuracy by calibrating it in saturated air according to the manufacturer's instructions. Or, you can measure a water sample that is saturated with oxygen, as follows. (NOTE: You can also use this procedure for testing the accuracy of the Winkler method.)

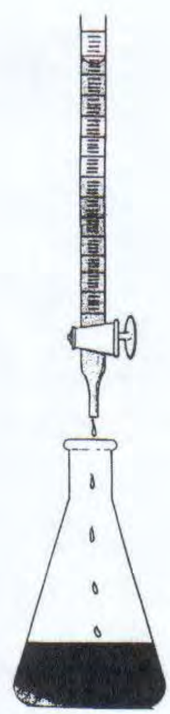
1. Fill a l-liter beaker or bucket half full of tap water. (You may want to bring a gallon jug with water in it for this purpose.) Mark the bottle number as "tap" on the lab sheet.
2. Pour this water back and forth into another beaker 10 times to saturate the water with oxygen.

3. Use the meter to measure the water temperature and record it in the water temperature column on the field data sheet.
4. Find the water temperature of your "tap" sample in Table 5.3. Use the meter to compare the dissolved oxygen concentration of your sample with the maximum concentration at that temperature in the table. Your sample should be within 0.5 mg/L. If it is not, repeat the check and if there is still an error, check the meter's batteries and follow the troubleshooting procedures in the manufacturer's manual.

Once the meter is turned on, allow 15 minute equilibration before calibrating. After calibration, do not turn the meter off until the sample is analyzed. Once you have verified that the meter is working properly, you are ready to measure the DO levels at the sampling site.

Figure 5.8

Titrating a DO sample using a buret



You might need an extension pole (this can be as simple as a piece of wood) to get the probe to the proper sampling point. Simply secure the probe to the end of the extension pole. A golfer's ball retriever works well because it is collapsible and easy to transport. To use the probe, proceed as follows:

1. Place the probe in the stream below the surface.
2. Set the meter to measure temperature, and allow the temperature reading to stabilize. Record the temperature on the field data sheet.
3. Switch the meter to read dissolved oxygen.
4. Record the dissolved oxygen level on the field data sheet.

TASK 4 Analyze the samples

Three types of titration apparatus can be used with the Winkler method: droppers, digital titrators, and burets. The dropper and digital titrator are suited for field use. The buret is more conveniently used in the lab (Fig. 5.8) Volunteer programs are most likely to use the dropper or digital titrator.

For titration with a dropper or syringe, which is relatively simple, follow the manufacturer's instructions. The following procedure is for using a digital titrator to determine the quantity of dissolved oxygen in a fixed sample:

1. Select a sample volume and sodium thiosulfate titration cartridge for the digital titrator corresponding to the expected dissolved oxygen concentration according to Table 5.4. In most cases, you will use the 0.2 N cartridge and the 100-mL sample volume.
2. Insert a clean delivery tube into the titration cartridge.
3. Attach the cartridge to the titrator body.

4. Hold the titrator with the cartridge tip up. Turn the delivery knob to eject air and a few drops of titrant. Reset the counter to 0 and wipe the tip.
5. Use a graduated cylinder to measure the sample volume (from the "fixed" sample in the 300-mL BOD bottle) according to Table 5.4.
6. Transfer the sample into a 250-mL Erlenmeyer flask, and place the flask on a magnetic stirrer with a stir bar. If you are in the field, you can manually swirl the flask to mix.
7. Place the delivery tube tip into the solution and turn the stirrer on to stir the sample while you're turning the delivery knob.
8. Titrate to a pale yellow color.
9. Add two dropperfuls of starch indicator solution and swirl to mix. A strong blue color will develop.
10. Continue to titrate until the sample is clear. Record the number of digits required. (The color might reappear after standing a few minutes, but this is not a cause for concern. The "first" disappearance of the blue color is considered the endpoint.)
11. Calculate mg/L of DO = digits required X digit multiplier (from Table 5.4).
12. Record the results in the appropriate column of the data sheet.

Some water quality standards are expressed in terms of percent saturation. To calculate percent saturation of the sample:

1. Find the temperature of your water sample as measured in the field.
2. Find the maximum concentration of your sample at that temperature as given in Table 5.3.
3. Calculate the percent saturation, by dividing your actual dissolved oxygen by the maximum concentration at the sample temperature.

Expected Range	Sample Volume	Titration Cartridge	Digit Multiplier
1-5 mg/L	200 mL	0.2 N	0.01
2-10 mg/L	100 mL	0.2 N	0.02
10+ mg/L	200 mL	2.0 N	0.10

Table 5.4

Sample volume selection and corresponding values for Winkler titration

Example: You measured a dissolved oxygen concentration of 5 mg/L at 20 °C. Divide 5 mg/L by 9.07, the maximum concentration at 20 °C. The percent saturation would be 55 percent.

4. Record the percent saturation in the appropriate column on the data sheet.

TASK 5 **Return the samples and the field data sheets to the lab/drop-off point**

If you are using the Winkler method and delivering the samples to a lab for titration, double-check to make sure that you have recorded the necessary information for each site on the field data sheet, especially the bottle number and corresponding site number and the times the samples were collected. Deliver your samples and field data sheets to the lab. If you have already obtained the dissolved oxygen results in the field, send the data sheets to your sampling coordinator.

What is biochemical oxygen demand and why is it important?

Biochemical oxygen demand, or BOD, measures the amount of oxygen consumed by microorganisms in decomposing organic matter in stream water. BOD also measures the chemical oxidation of inorganic matter (i.e., the extraction of oxygen from water via chemical reaction). A test is used to measure the amount of oxygen consumed by these organisms during a specified period of time (usually 5 days at 20 °C). The rate of oxygen consumption in a stream is affected by a number of variables: temperature, pH, the presence of certain kinds of microorganisms, and the type of organic and inorganic material in the water.

BOD directly affects the amount of dissolved oxygen in rivers and streams. The greater the BOD, the more rapidly oxygen is depleted in the stream. This means less oxygen is available to higher forms of aquatic life. The consequences of high BOD are the same as those for low dissolved oxygen: aquatic organisms become stressed, suffocate, and die.

Sources of BOD include leaves and woody debris; dead plants and animals; animal manure; effluents from pulp and paper mills, wastewater treatment plants, feedlots, and food-processing plants; failing septic systems; and urban stormwater runoff.

Sampling Considerations

BOD is affected by the same factors that affect dissolved oxygen (see above). Aeration of stream water—by rapids and waterfalls, for example—will accelerate the decomposition of organic and inorganic material. Therefore, BOD levels at a sampling site with slower, deeper waters might be higher for a given volume of organic and inorganic material than the levels for a similar site in highly aerated waters.

Chlorine can also affect BOD measurement by inhibiting or killing the microorganisms that decompose the organic and inorganic matter in a sample. If you are sampling in chlorinated waters, such as those below the effluent from a sewage treatment plant, it is necessary to neutralize the chlorine with sodium thiosulfate. (See APHA, 1992.)

BOD measurement requires taking two samples at each site. One is tested immediately for dissolved oxygen, and the second is incubated in the dark at 20 °C for 5 days and then tested for the amount of dissolved oxygen remaining. The difference in oxygen levels between the first test and the second test, in milligrams per liter (mg/L), is the amount of BOD. This represents the amount of oxygen consumed by microorganisms to break down the organic matter present in the sample bottle during the incubation period. Because of the 5-day incubation, the tests should be conducted in a laboratory.

Sometimes by the end of the 5-day incubation period the dissolved oxygen level is zero. This is especially true for rivers and streams with a lot of organic pollution. Since it is not known when the zero point was reached, it is not possible to tell what the BOD level is. In this case it is necessary to dilute the original sample by a factor that results in a final dissolved oxygen level of at least 2 mg/L. Special dilution water should be used for the dilutions. (See APHA, 1992.)

It takes some experimentation to determine the appropriate dilution factor for a particular sampling site. The final result is the difference in dissolved oxygen between the first measurement and the second after multiplying the second result by the dilution factor. More details are provided in the following section.

How to Collect and Analyze Samples

The procedures for collecting samples for BOD testing consist of the same steps described for sampling for dissolved oxygen (see above), with one important difference. At each site a second sample is collected in a BOD bottle and delivered to the lab for DO testing after the 5-day incubation period. Follow the same steps used for measuring dissolved oxygen with these additional considerations:

- Make sure you have two BOD bottles for each site you will sample. The bottles should be black to prevent photosynthesis. You can wrap a clear bottle with black electrician's tape if you do not have a bottle with black or brown glass.
- Label the second bottle (the one to be incubated) clearly so that it will not be mistaken for the first bottle.
- Be sure to record the information for the second bottle on the field data sheet.

The first bottle should be analyzed just prior to storing the second sample bottle in the dark for 5 days at 20 °C. After this time, the second bottle is tested for dissolved oxygen using the same method that was used for the first bottle. The BOD is expressed in milligrams per liter of DO using the following equation:

DO (mg/L) of first bottle
- DO (mg/L) of second bottle
= BOD (mg/L)

References

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

5.3 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 5.5 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 streambank 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. 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.

How to sample

The procedures for measuring temperature consist of the following tasks.

TASK 1 **Prepare before leaving for the sampling site**

Refer to pages 19-21 for details on confirming sampling date and time, safety considerations, checking supplies, and checking weather and directions. In addition to the standard sampling equipment and apparel, when measuring temperature you will need:

- A thermometer or meter
- A data sheet for temperature to record results

Table 5.5

Maximum weekly average temperatures for growth and short-term maximum temperatures for selected fish (°C and °F)

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) [c]
Largemouth bass	32°C (90°F)	34°C (93°F)	21°C (70°F)	27°C (81°F) [c]
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) [c]
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

(Brungs and Jones 1977)

Be sure to let someone know where you are going and when you expect to return

TASK 2 **Measure the temperature**

In general, sample away from the streambank 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 is not possible, tape your thermometer to an extension pole or use a boat. Reach out from the shore or boat as far as safely possible. If you use an extension pole, read the temperature quickly before it changes to the air temperature.

If you are doing a horizontal or vertical temperature profile, make sure you can safely reach all the points where a measurement is required before trying.

Measure temperature as follows:

1. Place the thermometer or meter probe in the water as least 4 inches below the surface or halfway to the bottom if in a shallow stream.
2. If using a thermometer, allow enough time for it to reach a stable temperature (at least 1 minute). If using a meter, allow the temperature reading to stabilize at a constant temperature reading.
3. If possible, try to read the temperature with the thermometer bulb beneath the water surface. If it is not possible, quickly remove the thermometer and read the temperature.
4. Record the temperature on the field data sheet.

TASK 3 **Return the field data sheets to the lab/dropoff point.**

References

Brungs, W.S. and B.R. Jones. 1977. *Temperature 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.

5.4 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 present the pH of some common liquids.

pH affects many chemical and biological processes in the water. For example, different organisms flourish within different ranges of pH. The largest 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 rainbow 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 ($H^+ + OH^- = H_2O$). 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.

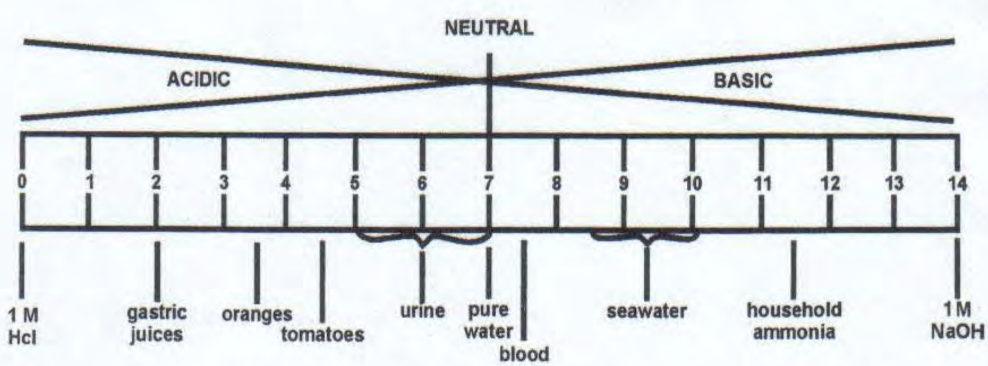
Analytical and equipment considerations

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.

If your program requires a high degree of accuracy and precision in pH results, the pH should be measured with a laboratory quality pH meter and electrode. Meters of this quality range in cost from around $250 to $1,000. Color comparators and pH

Figure 5.9

pH of selected liquids



"pocket pals" are suitable for most other purposes. The cost of either of these is in the $50 range. The lower cost of the alternatives might be attractive if you are relying on several teams of volunteers sampling multiple sites at the same time.

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*.

There is a wide variety of meters, but the most important part of the pH meter is the electrode. Buy a good, reliable electrode and follow the manufacturer's instructions for proper maintenance. Infrequently used or improperly maintained electrodes are subject to corrosion, which makes them highly inaccurate.

pH "Pocket Pals" and Color Comparators

pH "pocket pals" are electronic handheld "pens" that are dipped in the water and provide a digital readout of the pH. They can be calibrated to one pH buffer (lab meters, on the other hand, can be calibrated to two or more buffer solutions and thus are more accurate over a wide range of pH measurements).

Color comparators involve adding a reagent to the sample that colors the sample water. The intensity of the color is proportional to the pH of the sample. This color is then matched against a standard color chart. The color chart equates particular colors to associated pH values. The pH can be determined by matching the colors from the chart to the color of the sample.

How to collect and analyze samples

The field procedures for collecting and analyzing samples for pH consist of the following tasks.

TASK 1 **Prepare the sample containers**

Sample containers (and all glassware used in this procedure) must be cleaned and rinsed before the first run and after each sampling run by following the procedure described under Method A on page 128. Remember to wear latex gloves.

TASK 2 **Prepare before leaving for the sampling site**

Refer to pages 19-21 for details on confirming sampling date and time, picking up and checking supplies, and checking weather and directions. In addition to the standard sampling equipment and apparel, when sampling for pH, include the following equipment:

- pH meter with combination temperature and reference electrode, or pH "pocket pal" or color comparator
- Wash bottle with deionized water to rinse pH meter electrode (if appropriate)
- Data sheet for pH to record results

Before you leave for the sampling site, be sure to calibrate the pH meter or "pocket pal." The pH meter and "pocket pal" should be calibrated prior to sample analysis and after every 25 samples according to the instructions that come with them.

If you are using a "pocket pal," use the buffer recommended by the manufacturer. If you are using a laboratory grade meter, use two pH standard buffer solutions: 4.01 and 7.0. (Buffers can be purchased from test kit supply companies, such as Hach or LaMotte.) Following are notes regarding buffers.

- The buffer solutions should be at room temperature when you calibrate the meter.
- Do not use a buffer after its expiration date.
- Always cap the buffers during storage to prevent contamination.
- 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.
- Do not reuse buffer solutions!

TASK 3 **Collect the sample**

Refer to page 128 for details on how to collect water samples using screw-cap bottles or Whirl-pak® bags.

TASK 4 **Measure pH**

The procedure for measuring pH is the same whether it is conducted in the field or lab.

If you are using a "pocket pal" or color comparator, follow the manufacturer's instructions. Use the following steps to determine the pH of your sample if you are using a meter.

1. Rinse the electrode well with deionized water.
2. Place the pH meter or electrode into the sample. Depress the dispenser button once to dispense electrolyte. Read and record the temperature and pH in the appropriate column on the data sheet. Rinse the electrode well with deionized water.
3. Measure the pH of the 4.01 and 7.0 buffers periodically to ensure that the meter is not drifting off calibration. If it has drifted, recalibrate it.

TASK 4 **Return the field data sheets and samples to the lab or drop-off point.**

Samples for pH must be analyzed within 2 hours of collection. If the samples cannot be analyzed in the field, keep the samples on ice and take them to the lab or drop-off point as soon as possible within the 2-hour limit.

References

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.

5.5 Turbidity

What is turbidity and why is it important?

Turbidity is a measure of water clarity—how much the material suspended in water decreases the passage of light through the water. Suspended materials include soil particles (clay, silt, and sand), algae, plankton, microbes, and other substances. These materials are typically in the size range of 0.004 mm (clay) to 1.0 mm (sand). Turbidity can affect the color of the water.

Higher turbidity increases water temperatures because suspended particles absorb more heat. This, in turn, reduces the concentration of dissolved oxygen (DO) because warm water holds less DO than cold. Higher turbidity also reduces the amount of light penetrating the water, which reduces photosynthesis and the production of DO. Suspended materials can clog fish gills, reducing resistance to disease in fish, lowering growth rates, and affecting egg and larval development. As the particles settle, they can blanket the stream bottom, especially in slower waters, and smother fish eggs and benthic macroinvertebrates. Sources of turbidity include:

- Soil erosion
- Waste discharge
- Urban runoff
- Eroding stream banks
- Large numbers of bottom feeders (such as carp), which stir up bottom sediments
- Excessive algal growth.

Sampling and equipment considerations

Turbidity can be useful as an indicator of the effects of runoff from construction, agricultural practices, logging activity, discharges, and other sources. Turbidity often increases sharply during a rainfall, especially in developed watersheds, which typically have relatively high proportions of impervious surfaces. The flow of stormwater runoff from impervious surfaces rapidly increases stream velocity, which increases the erosion rates of streambanks and channels. Turbidity can also rise sharply during dry weather if earth-disturbing activities are occurring in or near a stream without erosion control practices in place.

Regular monitoring of turbidity can help detect trends that might indicate increasing erosion in developing watersheds. However, turbidity is closely related to stream flow and velocity and should be correlated with these factors. Comparisons of the change in turbidity over time, therefore, should be made at the same point at the same flow.

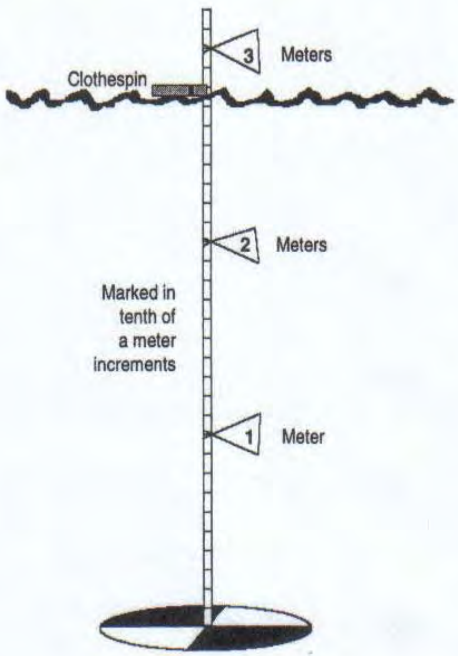
Turbidity is not a measurement of the amount of suspended solids present or the rate of sedimentation of a steam since it measures only the amount of light that is scattered by suspended particles. Measurement of total solids is a more direct measure of the amount of material suspended and dissolved in water (see section 5.9).

Turbidity is generally measured by using a turbidity meter. Volunteer programs may also take samples to a lab for analysis. Another approach is to measure transparency (an integrated measure of light scattering and absorption) instead of turbidity. Water clarity/transparency can be measured using a Secchi disk or transparency tube. The Secchi disk can only be used in deep, slow moving rivers; the transparency tube, a comparatively new development, is gaining acceptance in

Figure 5.10

Using a Secchi disk to measure transparency.

The disk is lowered until it is no longer visible. That point is the Secchi disk depth.



programs around the country but is not yet in wide use (see *Using a Secchi Disk or Tranparency Tube*).

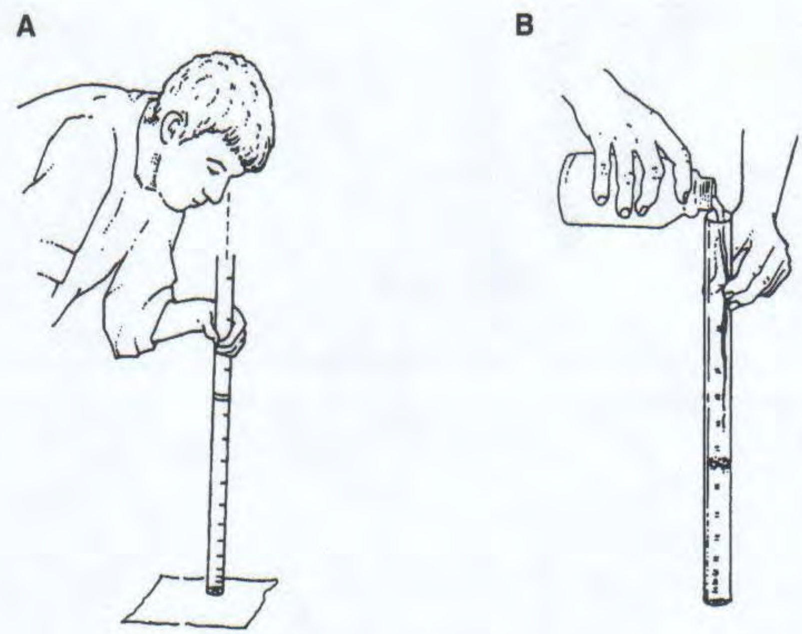
A turbidity meter consists of a light source that illuminates a water sample and a photoelectric cell that measures the intensity of light scattered at a 90° angle by the particles in the sample. It measures turbidity in nephelometric turbidity units or NTUs. Meters can measure turbidity over a wide range—from 0 to 1000 NTUs. A clear mountain stream might have a turbidity of around 1 NTU, whereas a large river like the Mississippi might have a dry-weather turbidity of around 10 NTUs. These values can jump into hundreds of NTU during runoff events. Therefore, the turbidity meter to be used should be reliable over the range in which you will be working. Meters of this quality cost about $800. Many meters in this price range are designed for field or lab use.

Although turbidity meters can be used in the field, volunteers might want to collect samples and take them to a central

Figure 5.11

Using a transparency tube.

(A) Prepare the transparency tube to take a reading. Place the tube on a white surface and look vertically down the tube to see the wave pattern at the bottom.
(B) Slowly pour water sample into the tube stopping intermittently to see if the wave pattern has disappeared.



Using a Secchi Disk or Transparency Tube

Secchi Disk

A Secchi disk is a black and white disk that is lowered by hand into the water to the depth at which it vanishes from sight (Figure 5.10). The distance to vanishing is then recorded. The clearer the water, the greater the distance. Secchi disks are simple to use and inexpensive. For river monitoring they have limited use, however, because in most cases the river bottom will be visible and the disk will not reach a vanishing point. Deeper, slower moving rivers are the most appropriate places for Secchi disk measurement although the current might require that the disk be extra-weighted so it does not sway and make measurement difficult. Secchi disks cost about $50 and can be homemade.

The line attached to the Secchi disk must be marked according to units designated by the volunteer program, in waterproof ink. Many programs require volunteers to measure to the nearest 1/10 meter. Meter intervals can be tagged (e.g., with duct tape) for ease of use.

To measure water clarity with a Secchi disk:

- Check to make sure that the Secchi disk is securely attached to the measured line.
- Lean over the side of the boat and lower the Secchi disk into the water, keeping your back toward the sun to block glare.
- Lower the disk until it disappears from view. Lower it one third of a meter and then slowly raise the disk until it just reappears. Move the disk up and down until the exact vanishing point is found.
- Attach a clothespin to the line at the point where the line enters the water. Record the measurement on your data sheet. Repeating the measurement will provide you with a quality control check.

The key to consistent results is to train volunteers to follow standard sampling procedures and, if possible, have the same individual take the reading at the same site throughout the season.

Transparency Tube

Pioneered by Australia's Department of Conservation, the transparency tube is a clear, narrow plastic tube marked in units with a dark pattern painted on the bottom. Water is poured into the tube until the pattern disappears (Figure 5.11). Some U.S. volunteer monitoring programs (e.g., the Tennessee Valley Authority (TVA) Clean Water Initiative and the Minnesota Pollution Control Agency (MPCA)) are testing the transparency tube in streams and rivers. MPCA uses tubes marked in centimeters, and has found tube readings to relate fairly well to lab measurements of turbidity and total suspended solids (although they do not recommend the transparency tube for applications where precise and accurate measurement is required or in highly colored waters).

The TVA and MPCA recommend the following sampling considerations:

- Collect the sample in a bottle or bucket in mid-stream and mid-depth if possible. Avoid stagnant water and sample as far from the shoreline as is safe. Avoid collecting sediment from the bottom of the stream.
- Face upstream as you fill the bottle or bucket.
- Take readings in open but shaded conditions. Avoid direct sunlight by turning your back to the sun.
- Carefully stir or swish the water in the bucket or bottle until it is homogeneous, taking care not to produce air bubbles (these will scatter light and affect the measurement). Then pour the water slowly in the tube while looking down the tube. Measure the depth of the water column in the tube when the symbol just disappears.

For more information on using a transparency tube, see the references at the end of this section. Many programs have begun making their own tubes. They now may also be purchased in the U.S. (see Appendix B—Scientific Supply Houses).

point for turbidity measurements. This is because of the expense of the meter (most programs can afford only one and would have to pass it along from site to site, complicating logistics and increasing the risk of damage to the meter) and because the meter includes glass cells that must remain optically clear and free of scratches.

Volunteers can also take turbidity samples to a lab for meter analysis at a reasonable cost.

How to sample

The procedures for collecting samples and analyzing turbidity consist of the following tasks:

TASK 1 **Prepare the sample containers**

If factory-sealed, disposable Whirl-pak® bags are used to sample, no preparation is needed. Reused sample containers (and all glassware used in this procedure) must be cleaned before the first run and after each sampling run by following Method A described on page 128.

TASK 2 **Prepare before leaving for the sampling site**

Refer to pages 19-21 for details on confirming sampling date and time, safety consideration, checking supplies, and checking weather and directions. In addition to the standard sampling equipment and apparel, when sampling for turbidity, include the following equipment:

- Turbidity meter
- Turbidity standards
- Lint-free cloth to wipe the cells of the meter
- Data sheet for turbidity to record results

Be sure to let someone know where you are going and when you expect to return.

TASK 3 **Collect the sample**

Refer to page 128 for details on how to collect water samples using screw-cap bottles or Whirl-pak® bags.

TASK 4 **Analyze the sample**

The following procedure applies to field or lab use of the turbidity meter.

1. Prepare the turbidity meter for use according to the manufacturer's directions.
2. Use the turbidity standards provided with the meter to calibrate it. Make sure it is reading accurately in the range in which you will be working.
3. Shake the sample vigorously and wait until the bubbles have disappeared. You might want to tap the sides of the bottle gently to accelerate the process.
4. Use a lint-free cloth to wipe the outside of the tube into which the sample will be poured. Be sure not to handle the tube below the line where the light will pass when the tube is placed in the meter.
5. Pour the sample water into the tube. Wipe off any drops on the outside of the tube.
6. Set the meter for the appropriate turbidity range. Place the tube in the meter and read the turbidity measurement directly from the meter display.
7. Record the result on the field or lab sheet.
8. Repeat steps 3-7 for each sample.

TASK 5 **Return the samples and the field data sheets to the lab/drop-off point.**

If you are sending your samples to a lab for analysis, they must be tested within 24 hours of collection. Keep samples in the dark and on ice or refrigerated.

References and Further Reading

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5.6 Phosphorus

Why is phosphorus important?

Both phosphorus and nitrogen are essential nutrients 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).

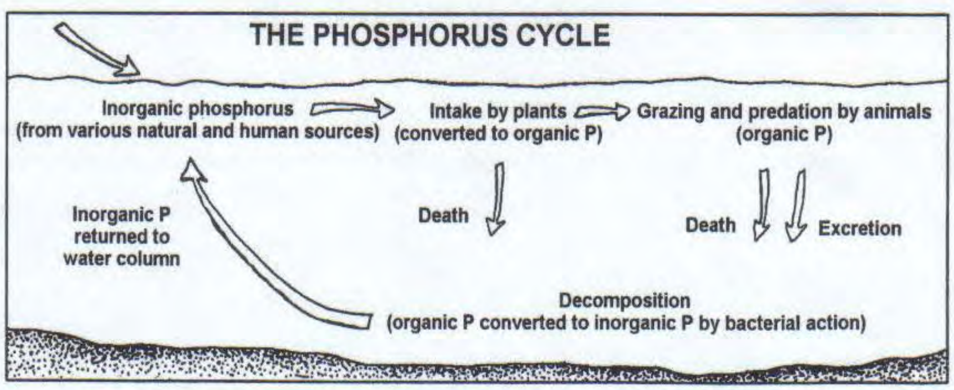
The phosphorus cycle

Phosphorus cycles through the environment, changing form as it does so (Fig. 5.12). 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.

Figure 5.12

The phosphorus cycle

Phosphorus changes form as it cycles through the aquatic environment.



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.
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.

This manual does not address laboratory methods. Refer to the references cited at the end of this section.

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 Labware

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 labware. All labware 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 combined liquid or prepackaged powder reagent, consisting of sulfuric acid, potassium antimonyl tartrate, ammonium molybdate, and ascorbic acid (or comparable compounds), is added to either 50 or 25 mL of the water sample. This colors the sample blue in direct proportion to the amount of orthophosphate in the sample. Absorbance or transmittance is then measured after 10 minutes, but before 30 minutes, using a color comparator with a scale in milligrams per liter that increases with the increase in color hue, or an electronic meter that measures the amount of light absorbed or transmitted at a wavelength of 700 - 880 nanometers (again depending on manufacturer's directions).

A color comparator may be useful for identifying heavily polluted sites with high concentrations (greater than 0.1 mg/L). However, matching the color of a treated sample to a comparator can be very subjective, especially at low concentrations, and can lead to variable results.

A field spectrophotometer or colorimeter with a 2.5-cm light path and an infrared photocell (set for a wavelength of 700-880 nm) is recommended for accurate determination of low concentrations (between 0.2 and 0.02 mg/L). Use of a meter requires that you prepare and analyze known standard concentrations ahead of time in order to convert the absorbance readings of your stream sample to milligrams per liter, or that your meter reads directly as milligrams per liter.

How to prepare standard concentrations

Note that this step is best accomplished in the lab before leaving for sampling. Standards are prepared using a phosphate standard solution of 3 mg/L as phosphate (PO_4). This is equivalent to a concentration of 1 mg/L as Phosphorus (P). All references to concentrations and results from this point on in this procedure will be expressed as mg/L as P, since this is the convention for reporting results.

Six standard concentrations will be prepared for every sampling date in the range of expected results. For most samples, the following six concentrations should be adequate:

0.00 mg/L	0.12 mg/L
0.04 mg/L	0.16 mg/L
0.08 mg/L	0.20 mg/L

Proceed as follows:

1. Set out six 25-mL volumetric flasks—one for each standard. Label the flasks 0.00, 0.04, 0.08, 0.12, 0.16, and 0.20.
2. Pour about 30 mL of the phosphate standard solution into a 50 mL beaker.
3. Use 1-, 2-, 3-, 4-, and 5-mL Class A volumetric pipets to transfer corresponding volumes of phosphate standard solution to each 25-mL volumetric flask as follows:

Standard Concentration	mL of Phosphate Standard Solution
0.00	0
0.04	1
0.08	2
0.12	3
0.16	4
0.20	5

Note: The standard solution is calculated based on the equation: $A = (B \times C) \div D$

Where:

A = mL of standard solution needed

B = desired concentration of standard

C = final volume (mL) of standard

D = concentration of standard solution

For example, to find out how much phosphate standard solution to use to make a 0.04-mg/L standard:

$A = (0.04 \times 25) \div 1$

$A = 1$ mL

Before transferring the solution, clear each pipet by filling it once with the standard solution and blowing it out. Rinse each pipet with deionized water after use.

4. Fill the remainder of each 25 mL volumetric flask with distilled, deionized water to the 25 mL line. Swirl to mix.
5. Set out and label six 50-mL Erlenmeyer flasks: 0.00, 0.04, 0.08, 0.12, 0.16, and 0.20. Pour the standards from the volumetric flasks to the Erlenmeyer flasks.
6. List the standard concentrations (0.00, 0.04, 0.08, 0.12, 0.16, and 0.20) under "Bottle #" on the lab sheet.
7. Analyze each of these standard concentrations as described in the section below.

How to collect and analyze samples

The field procedures for collecting and analyzing samples for phosphorus consist of the following tasks:

TASK 1 **Prepare the sample containers**

If factory-sealed, disposable Whirl-pak® bags are used for sampling, no preparation is needed. Reused sample containers (and all glassware used in this procedure) must be cleaned (including acid rinse) before the first run and after each sampling run by following the procedure described in Method B on page 128. Remember to wear latex gloves.

TASK 2 **Prepare before leaving for the sample site**

Refer to page 19-21 for details on confirming sampling date and time, safety considerations, checking supplies, and checking weather and directions. In addition to sample containers and the standard sampling apparel, you will need the following equipment and supplies for total reactive phosphorus analysis:

- Color comparator or field spectrophotometer with sample tubes for reading the absorbance of the sample
- Prepackaged reagents (combined reagents) to turn the water blue
- Deionized or distilled water to rinse the sample tubes between uses
- Wash bottle to hold rinse water
- Mixing container with a mark at the recommended sample volume (usually 25 mL) to hold and mix the sample
- Clean, lint-free wipes to clean and dry the sample tubes

Note that prepackaged reagents are recommended for ease and safety.

TASK 3 **Collect the sample**

Refer to page 128 for details on how to collect water samples using screw-cap bottles or Whirl-pak® bags.

TASK 4 **Analyze the sample in the field (for total orthophosphate only) using the ascorbic acid method.**

If using an electronic spectrophotometer or colorimeter:

1. "Zero" the meter (if you are using one) using a reagent blank (distilled water plus the reagent powder) and following the manufacturer's directions.
2. Pour the recommended sample volume (usually 25 mL) into a mixing container and add reagent powder pillows. Swirl to mix. Wait the recommended time (usually at least 10 minutes) before proceeding.
3. Pour the first field sample into the sample cell test tube. Wipe the tube with a lint-free cloth to be sure it is clean and free of smudges or water droplets. Insert the tube into the sample cell.
4. Record the bottle number on the field data sheet.
5. Place the cover over the sample cell. Read the absorbance or concentration of this sample and record it on the field data sheet.
6. Pour the sample back into its flask.
7. Rinse the sample cell test tube and mixing container three times with distilled, deionized water. Avoid touching the lower portion of the sample cell test tube. Wipe with a clean, lint-free wipe. Be sure that the lower part of the sample cell test tube is clean and free of smudges or water droplets.

Be sure to use the same sample cell test tube for each sample. If the test tube breaks, use a new one and repeat step 1 to "zero" the meter.

If using a color comparator:

1. Follow the manufacturer's directions. Be sure to pay attention to the direction of your light source when reading the color development. The light source should be in the same position relative to the color comparator for each sample. Otherwise, this is a source of significant error. As a quality check, have someone else read the comparator after you.
2. Record the concentration on the field data sheet.

TASK 5 **Return the samples (for lab analysis for other tests) and the field data sheets to the lab/drop-off point.**

Samples for different types of phosphorus must be analyzed within a certain time period. For some types of phosphorus, this is a matter of hours; for others, samples can be preserved and held for longer periods. Samples being tested for orthophosphate must be analyzed within 48 hours of collection. In any case, keep the samples on ice and take them to the lab or drop-off point as soon as possible.

TASK 6 **Analyze the samples in the lab.**

Lab methods for other tests are described in the references below (APHA. 1992; Hach Company, 1992; River Watch Network, 1992; USEPA, 1983).

TASK 7 **Report the results and convert to milligrams per liter**

First, absorbance values must be converted to milligrams per liter. This is done by constructing a "standard curve" using the absorbance results from your standard concentrations.

1. Make an absorbance versus concentration graph on graph paper:
   - Make the "y" (vertical) axis and label it "absorbance." Mark this axis in 0.05 increments from 0 as high as the graph paper will allow.
   - Make the "x" (horizontal) axis and label it "concentration: mg/L as P." Mark this axis with the concentration of the standards: 0, 0.04, 0.08, 0.12, 0.16, 0.20.
2. Plot the absorbance of the standard concentrations on the graph.
3. Draw a "best fit" straight line through these points. The line should touch (or almost touch) each of the points. If it doesn't, make up new standards and repeat the procedure.

Example: Suppose you measure the absorbance of the six standard concentrations as follows:

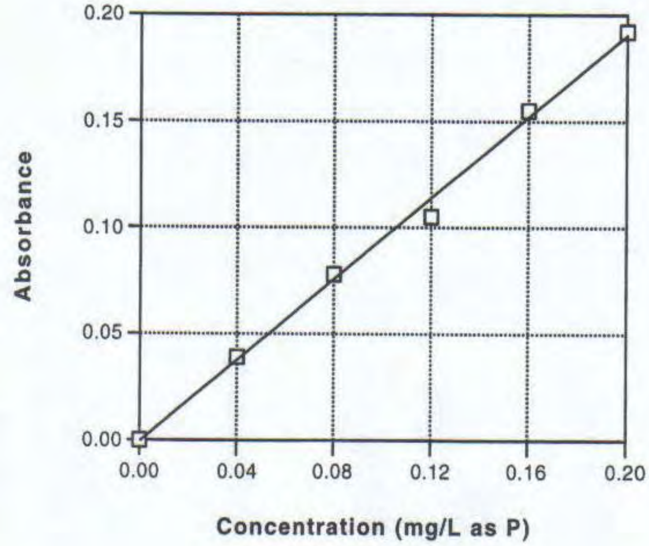
Concentration	Absorbance
0.00	0.000
0.04	0.039
0.08	0.078
0.12	0.105
0.16	0.155
0.20	0.192

The resulting standard curve is displayed in Fig. 5.13.

4. For each sample, locate the absorbance on the "y" axis, read horizontally over to the line, and then more down to read the concentration in mg/L as P.
5. Record the concentration on the lab sheet in the appropriate column. NOTE: The detection limit for this test is 0.01 mg/L. Report any results less than 0.01 as "<0.01." Round off all results to the nearest hundredth of a mg/L.

Figure 5.13

Absorbance of standard concentrations, when plotted, should result in a straight line



Results can either be reported "as P" or "as PO_4." Remember that your results are reported as milligrams per liter—weight per unit of volume. Since the PO_4 molecule is three times as heavy as the P atom, results reported as PO_4 are three times the concentration of those reported as P. For example, if you measure 0.06 mg/L as PO_4, that's equivalent to 0.02 mg/L as P. To convert PO_4 to P, divide by 3. To convert P to PO_4, multiply by 3. To avoid this confusion, and since most state water quality standards are reported as P, this manual recommends that results always be reported as P.

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5.7 Nitrates

What are nitrates and why are they important?

Nitrates are a form of nitrogen, which is found in several different forms in terrestrial and aquatic ecosystems. These forms of nitrogen include ammonia (NH_3), nitrates (NO_3), and nitrites (NO_2). Nitrates are essential plant nutrients, but in excess amounts they can cause significant water quality problems. Together with phosphorus, nitrates in excess amounts can accelerate eutrophication, causing dramatic increases in aquatic plant growth and changes in the types of plants and animals that live in the stream. This, in turn, affects dissolved oxygen, temperature, and other indicators. 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.

Sources of nitrates include wastewater treatment plants, runoff from fertilized lawns and cropland, failing on-site septic systems, runoff from animal manure storage areas, and industrial discharges that contain corrosion inhibitors.

Sampling and equipment 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.

Water that is polluted with nitrogen-rich organic matter might show low nitrates. Decomposition of the organic matter lowers the dissolved oxygen level, which in turn slows the rate at which ammonia is oxidized to nitrite (NO_2) and then to nitrate (NO_3). Under such circumstances, it might be necessary to also monitor for nitrites or ammonia, which are considerably more toxic to aquatic life than nitrate. (See Standard Methods section 4500-NH_3 and 4500-NO_2 for appropriate nitrite methods; APHA, 1992)

Water samples to be tested for nitrate should be collected in glass or polyethylene containers that have been prepared by using Method B in the introduction.

Volunteer monitoring programs usually use two methods for nitrate testing: the cadmium reduction method and the nitrate electrode. The more commonly used cadmium reduction method produces a color reaction that is then measured either by comparison to a color wheel or by use of a spectrophotometer. A few programs also use a nitrate electrode, which can measure in the range of 0 to 100 mg/L nitrate. A newer colorimetric immunoassay technique for nitrate screening is also now available and might be applicable for volunteers.

Cadmium Reduction Method

The cadmium reduction method is a colorimetric method that involves contact of the nitrate in the sample with cadmium particles, which cause nitrates to be converted to nitrites. The nitrites then react with another reagent to form a red color whose intensity is proportional to the original amount of nitrate. The red color is then measured either by comparison to a color wheel with a scale in milligrams per liter that increases with the increase in color hue, or by use of an electronic spectrophotometer that measures the amount of light absorbed by the treated sample at a 543-nanometer wavelength. The absorbance value is then converted to the equivalent concentration of nitrate by using a standard curve. Methods for making standard solutions and standard curves are presented at the end of this section.

This curve should be created by the program advisor before each sampling run. The curve is developed by making a set of standard concentrations of nitrate, reacting them and developing the corresponding color, and then plotting the absorbance value for each concentration against concentration. A standard curve could also be generated for the color wheel.

Use of the color wheel is appropriate only if nitrate concentrations are greater than 1 mg/L. For concentrations below 1 mg/L, a spectrophotometer should be used. Matching the color of a treated sample at low concentrations to a color wheel (or cubes) can be very subjective and can lead to variable results. Color comparators can, however, be effectively used to identify sites with high nitrates.

This method requires that the samples being treated are clear. If a sample is turbid, it should be filtered through a 0.45-micron filter. Be sure to test whether the filter is nitrate-free. If copper, iron, or other metals are present in concentrations above several mg/L, the reaction with the cadmium will be slowed down and the reaction time will have to be increased.

The reagents used for this method are often prepackaged for different ranges, depending on the expected concentration of nitrate in the stream. For example, the Hach Company provides reagents for the following ranges: low (0 to 0.40 mg/L), medium (0 to 4.5 mg/L), and high (0 to 30 mg/L). You should determine the appropriate range for the stream being monitored.

Nitrate Electrode Method

A nitrate electrode (used with a meter) is similar in function to a dissolved oxygen meter. It consists of a probe with a sensor that measures nitrate activity in the water; this activity affects the electric potential of a solution in the probe. This change is then transmitted to the meter, which converts the electric signal to a scale that is read in millivolts. The millivolts are then converted to mg/L of nitrate by plotting them from a standard curve (see above). The accuracy of the electrode can be affected by high concentrations of chloride or bicarbonate ions in the sample water. Fluctuating pH levels can also affect the reading by the meter.

Nitrate electrodes and meters are expensive compared to field kits that employ the cadmium reduction method. (The expense is comparable, however, if a spectrophotometer is used rather than a color wheel.) Meter/probe combinations run between $700 and $1,200 including a long cable to connect the probe to the meter. If the program has a pH meter that displays readings in millivolts, it can be used with a nitrate probe and no separate nitrate meter is needed. Results are read directly as milligrams per liter.

Although nitrate electrodes and spectrophotometers can be used in the field, they have certain disadvantages. These devices are more fragile than the color comparators and are therefore more at risk of breaking in the field. They must be carefully maintained and must be calibrated before each sample run and, if you are doing many tests, between samplings. This means that samples are best tested in the lab. Note that samples to be tested with a nitrate electrode should be at room temperature, whereas color comparators can be used in the field with samples at any temperature.

How to collect and analyze samples

The procedures for collecting and analyzing samples for nitrate consist of the following tasks:

TASK 1 **Prepare the sample containers**

If factory-sealed, disposable Whirl-pak® bags are used for sampling, no preparation is needed. Reused sample containers (and all glassware used in this procedure) must be cleaned before the first run and after each sampling by following the method described on page 128 under Method B. Remember to wear latex gloves.

TASK 2 **Prepare before leaving for the sampling site**

Refer to pages 19-21 for details on confirming sampling date and time, safety considerations, checking supplies, and checking weather and directions. In addition to the standard sampling equipment and apparel, the following equipment is needed when analyzing nitrate nitrogen in the field:

- Color comparator or field spectrophotometer with sample tubes (for reading absorbance of the sample)
- Reagent powder pillows (reagents to turn the water red)
- Deionized or distilled water to rinse the sample tubes between uses
- Wash bottle to hold rinse water
- Waste bottle with secure lid to hold used cadmium particles, which should be clearly labeled and returned to the lab, where the cadmium will be properly disposed of
- Mixing container with a mark at the sample volume (usually 25 mL) to hold and mix the sample
- Clean, lint-free wipes to clean and dry the sample tubes

TASK 3 **Collect the sample**

Refer to page 128 for details on collecting a sample using screw-cap bottles or Whirl-pak® bags.

TASK 4 **Analyze the sample in the field**

Cadmium Reduction Method With a Spectrophotometer

The following is the general procedure to analyze a sample using the cadmium reduction method with a spectrophotometer. However, this should not replace the manufacturer's directions if they differ from the steps provided below:

1. Pour the first field sample into the sample cell test tube and insert it into the sample cell of the spectrophotometer.
2. Record the bottle number on the lab sheet.
3. Place the cover over the sample cell. Read the absorbance or concentration of this sample and record it on the field data sheet.
4. Pour the sample back into the waste bottle for disposal at the lab.

Cadmium Reduction Method With a Color Comparator

To analyze a sample using the cadmium reduction method with a color comparator, follow the manufacturer's directions and record the concentration on the field data sheet.

TASK 5 **Return the samples and the field data sheets to the lab/ drop-off point for analysis**

Samples being sent to a lab for analysis must be tested for nitrates within 48 hours of collection. Keep samples in the dark and on ice or refrigerated.

TASK 6 **Determine results (for spectrophotometer absorbance or nitrate electrode) in lab**

Preparation of Standard Concentrations

Cadmium Reduction Method With a Spectrophotometer

First determine the range you will be testing (low, medium, or high). For each range you will need to determine the lower end, which will be determined by the detection limit of your spectrophotometer. The high end of the range will be the endpoint of the range you are using. Use a nitrate nitrogen standard solution of appropriate strength for the range in which you are working. A 1-mg/L nitrate nitrogen (NO_3-N) solution would be suitable for low-range (0 to 1.0 mg/L) tests. A 100-mg/L standard solution would be appropriate for medium- and high-range tests. In the following example, it is assumed that a set of standards for a 0 to 5.0 mg/L range is being prepared.

Example:

1. Set out six 25-mL volumetric flasks (one for each standard). Label the flasks 0.0, 1.0, 2.0, 3.0, 4.0, and 5.0.
2. Pour 30 mL of a 25-mg/L nitrate nitrogen standard solution into a 50-mL beaker.
3. Use 1-, 2-, 3-, 4-, and 5-mL Class A volumetric pipets to transfer corresponding volumes of nitrate nitrogen standard solution to each 25-mL volumetric flask as follows:

Standard Solution	mL of Nitrate Nitrogen Standard Solution
0.0	0
1.0	1
2.0	2
3.0	3
4.0	4
5.0	5

Analysis of the Cadmium Reduction Method Standard Concentrations

Use the following procedure to analyze the standard concentrations.

1. Add reagent powder pillows to the nitrate nitrogen standard concentrations.
2. Shake each tube vigorously for at least 3 minutes.
3. For each tube, wait at least 10 minutes but not more than 20 minutes to proceed.
4. "Zero" the spectrophotometer using the 0.0 standard concentration and following the manufacturer's directions. Record the absorbance as "0" in the absorbance column on the lab sheet. Rinse the sample cell three times with distilled water.
5. Read and record the absorbance of the 1.0-mg/L standard concentration.
6. Rinse the sample cell test tube three times with distilled or deionized water. Avoid touching the lower part of the sample cell test tube. Wipe with a clean, lint-free wipe. Be sure that the lower part of the sample cell test tube is clean and free of smudges or water droplets.
7. Repeat steps 3 and 4 for each standard.
8. Prepare a calibration curve and convert absorbance to mg/L as follows:
   - Make an absorbance versus concentration graph on graph paper:

     (a) Make the vertical (y) axis and label it "absorbance." Mark this axis in 1.0 increments from 0 as high as the graph paper will allow.

     (b) Make the horizontal (x) axis and label it "concentration: mg/L as nitrate nitrogen." Mark this axis with the concentrations of the standards: 0.0, 1.0, 2.0, 3.0, 4.0, and 5.0.

- Plot the absorbance of the standard concentrations on the graph.
- Draw a "best fit" straight line through these points. The line should touch (or almost touch) each of the points. If it doesn't, the results of this procedure are not valid.
- For each sample, locate the absorbance on the "y" axis, read over horizontally to the line, and then move down to read the concentration in mg/L as nitrate nitrogen.
- Record the concentration on the lab sheet in the appropriate column.

For Nitrate Electrode

Standards are prepared using nitrate standard solutions of 100 and 10 mg/L as nitrate nitrogen (NO_3-N). All references to concentrations and results in this procedure will be expressed as mg/L as NO_3-N. Eight standard concentrations will be prepared:

100.0 mg/L	0.40 mg/L
10.0 mg/L	0.32 mg/L
1.0 mg/L	0.20 mg/L
0.8 mg/L	0.12 mg/L

Use the following procedure:

1. Set out eight 25-mL volumetric flasks (one for each standard). Label the flasks 100.0, 10.0, 1.0, 0.8, 0.4, 0.32, 0.2, and 0.12.
2. To make the 100.0-mg/L standard, pour 25 mL of the 100-mg/L nitrate standard solution into the flask labeled 100.0.
3. To make the 10.0-mg/L standard, pour 25 mL of the 10-mg/L nitrate standard solution into the flask labeled 10.0.
4. To make the 1.0-mg/L standard, use a 10- or 5-mL pipet to measure 2.5 mL of the 10-mg/L nitrate standard solution into the flask labeled 1.0. Fill the flask with 22.5 mL distilled, deionized water to the fill line. Rinse the pipet with deionized water.
5. To make the 0.8-mg/L standard, use a 10- or 5-mL pipet or a 2-mL volumetric pipet to measure 2 mL of the 10-mg/L nitrate standard solution into the flask labeled 0.8. Fill the flask with about 23 mL distilled, deionized water to the fill line. Rinse the pipet with deionized water.
6. To make the 0.4-mg/L standard, use a 10- or 5-mL pipet or a 1-mL volumetric pipet to measure 1 mL of the 10-mg/L nitrate standard solution into the flask labeled 0.4. Fill the flask with about 24 mL distilled, deionized water to the fill line. Rinse the pipet with deionized water.
7. To make the 0.32-, 0.2-, and 0.12-mg/L standards, follow step 4 to make a 25-mL volume of 1.0 mg/L standard solution. Transfer this to a beaker. Pipet the following volumes into the appropriately labeled volumetric flasks:

Standard Solution	mL of Nitrate Nitrogen Standard Solution
0.32	8
0.20	5
0.12	3

Fill each flask up to the fill line. Rinse pipets with deionized water.

Analysis of the Nitrate Electrode Standard Concentrations

Use the following procedure to analyze the standard concentrations.

1. List the standard concentrations (100.0, 10.0, 1.0, 0.8, 0.4, 0.32, 0.2, and 0.12) under "bottle #" on the lab sheet.

2. Prepare a calibration curve and convert to mg/L as follows:

   - Plot absorbance or mV readings for the 100-, 10-, and 1-mg/L standards on semi-logarithmic graph paper, with concentration on the logarithmic (x) axis and the absorbance or millivolts (mV) on the linear (y) axis.

     For the nitrate electrode curve, a straight line with a slope of 58 ± 3 mV/decade at 25°C should result. That is, measurements of 10- and 100-mg/L standard solutions should be no more than 58 ± 3 mV apart.

   - Plot absorbance or mV readings for the 1.0-, 0.8-, 0.4-, 0.32-, 0.2-, and 0.12-mg/L standards on semi-logarithmic graph paper, with concentration on the logarithmic (x) axis and the millivolts (mV) on the linear (y) axis.

     For the nitrate electrode, the result here should be a curved line since the response of the electrode at these low concentrations is not linear.

   - For the nitrate electrode, recalibrate the electrodes several times daily by checking the mV reading of the 10-mg/L and 0.4-mg/L standards and adjusting the calibration control on the meter until the reading plotted on the calibration curve is displayed again.

References

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

5.8 Total Solids

What are total 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.

Higher concentrations of suspended solids can serve as carriers of toxics, which readily cling to suspended particles. This is particularly a concern where pesticides are being used on irrigated crops. Where solids are high, pesticide concentrations may increase well beyond those of the original application as the irrigation water travels down irrigation ditches. Higher levels of solids can also clog irrigation devices and might become so high that irrigated plant roots will lose water rather than gain it.

A high concentration of total solids will make drinking water unpalatable and might have an adverse effect on people who are not used to drinking such water. Levels of total solids that are too high or too low can also reduce the efficiency of wastewater treatment plants, as well as the operation of industrial processes that use raw water.

Total solids also affect water clarity. Higher 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 total 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 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.

Total solids 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 total solids can help detect trends that might indicate increasing erosion in developing watersheds. Total solids are related closely to stream flow and velocity and should be correlated with these factors. Any change in total solids over time should be measured at the same site at the same flow.

Total solids are measured by weighing the amount of solids present in a known volume of sample. This is done by weighing a beaker, filling it with a known volume, evaporating the water in an oven and completely drying the residue, and then weighing the beaker with the residue. The total solids concentration is equal to the difference between the weight of the beaker with the residue and the weight of the beaker without it. 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 or Mettler balances, and they are expensive (around $3,000). The technique requires that the beakers 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 beaker. 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.

How to collect and analyze samples

The procedures for collecting and analyzing samples for total solids consist of the following tasks:

TASK 1 **Prepare the sample containers**

Factory-sealed, disposable Whirl-pak® bags are easy to use because they need no preparation. Reused sample containers (and all glassware used in this procedure) must be cleaned and rinsed before the first sampling run and after each run by following the procedure described in Method A on page 128.

TASK 2 **Prepare before leaving for the sampling site**

Refer to pages 19-21 for details on confirming sampling information. Be sure to let someone know where you are going and when you expect to return.

TASK 3 **Collect the sample**

Refer to page 128 for details on how to collect water samples using screw-cap bottles or Whirl-pak® bags.

TASK 4 **Return samples and field sheets to the lab/drop-off point for analysis.**

Samples that are sent to a lab for total solids analysis must be tested within seven days of collection. Keep the samples on ice or refrigerated.

References

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

5.9 Conductivity

What is conductivity and why is it important?

Conductivity is a measure of the ability of water to pass an electrical current. Conductivity in water is affected by the presence of inorganic dissolved solids such as chloride, nitrate, sulfate, and phosphate anions (ions that carry a negative charge) or sodium, magnesium, calcium, iron, and aluminum cations (ions that carry a positive charge). Organic compounds like oil, phenol, alcohol, and sugar do not conduct electrical current very well and therefore have a low conductivity when in water. Conductivity is also affected by temperature: the warmer the water, the higher the conductivity. For this reason, conductivity is reported as conductivity at 25 degrees Celsius (25 °C).

Conductivity in streams and rivers is affected primarily by the geology of the area through which the water flows. Streams that run through areas with granite bedrock tend to have lower conductivity because granite is composed of more inert materials that do not ionize (dissolve into ionic components) when washed into the water. On the other hand, streams that run through areas with clay soils tend to have higher conductivity because of the presence of materials that ionize when washed into the water. Ground water inflows can have the same effects depending on the bedrock they flow through.

Discharges to streams can change the conductivity depending on their make-up. A failing sewage system would raise the conductivity because of the presence of chloride, phosphate, and nitrate; an oil spill would lower the conductivity.

The basic unit of measurement of conductivity is the mho or siemens. Conductivity is measured in micromhos per centimeter (µmhos/cm) or microsiemens per centimeter (µs/cm). Distilled water has a conductivity in the range of 0.5 to 3 µmhos/cm. The conductivity of rivers in the United States generally ranges from 50 to 1500 µmhos/cm. Studies of inland fresh waters indicate that streams supporting good mixed fisheries have a range between 150 and 500 µhos/cm. Conductivity outside this range could indicate that the water is not suitable for certain species of fish or macroinvertebrates. Industrial waters can range as high as 10,000 µmhos/cm.

Sampling and equipment Considerations

Conductivity is useful as a general measure of stream water quality. Each stream tends to have a relatively constant range of conductivity that, once established, can be used as a baseline for comparison with regular conductivity measurements. Significant changes in conductivity could then be an indicator that a discharge or some other source of pollution has entered a stream.

Conductivity is measured with a probe and a meter. Voltage is applied between two electrodes in a probe immersed in the sample water. The drop in voltage caused by the resistance of the water is used to calculate the conductivity per centimeter. The meter converts the probe measurement to micromhos per centimeter and displays the result for the user. NOTE: Some conductivity meters can also be used to test for total dissolved solids and salinity. The total dissolved solids concentration in milligrams per liter (mg/L) can also be calculated by multiplying the conductivity result by a factor between 0.55 and 0.9, which is empirically determined (see Standard Methods #2510, APHA 1992).

Suitable conductivity meters cost about $350. Meters in this price range should also measure temperature and automatically compensate for temperature in the conductivity reading. Conductivity can be measured in the field or the lab. In most cases, it is probably better if the samples are collected in the field and taken to a lab for testing. In this way several teams of volunteers can collect samples simultaneously. If it is important to test in the field, meters designed for field use can be obtained for around the same cost mentioned above.

If samples will be collected in the field for later measurement, the sample bottle should be a glass or polyethylene bottle that has been washed in phosphate-free detergent and rinsed thoroughly with both tap and distilled water. Factory-prepared Whirl-pak® bags may be used.

How to sample

The procedures for collecting samples and analyzing conductivity consist of the following tasks:

TASK 1 **Prepare the sample containers**

If factory-sealed, disposable Whirl-pak® bags are used for sampling, no preparation is needed. Reused sample containers (and all glassware used in this procedure) must be cleaned before the first run and after each sampling run by following Method A as described on page 128.

TASK 2 **Prepare before leaving for the sampling site**

Refer to pages 19-21 for details on confirming sampling date and time, safety considerations, checking supplies, and checking weather and directions. In addition to the standard sampling equipment and apparel, when sampling for conductivity, include the following equipment:

- Conductivity meter and probe (if testing conductivity in the field)
- Conductivity standard appropriate for the range typical of the stream
- Data sheet for conductivity to record results

Be sure to let someone know where you are going and when you expect to return.

TASK 3 **Collect the sample (if samples will be tested in the lab)**

Refer to page 128 for details on how to collect water samples using screw-cap bottles or Whirl-pak® bags.

TASK 4 **Analyze the sample (field or lab)**

The following procedure applies to field or lab use of the conductivity meter.

1. Prepare the conductivity meter for use according to the manufacturer's directions.
2. Use a conductivity standard solution (usually potassium chloride or sodium chloride) to calibrate the meter for the range that you will be measuring. The manufacturer's directions should describe the preparation procedures for the standard solution.
3. Rinse the probe with distilled or deionized water.
4. Select the appropriate range beginning with the highest range and working down. Read the conductivity of the water sample. If the reading is in the lower 10 percent of the range, switch to the next lower range. If the conductivity of the sample exceeds the range of the instrument, you may dilute the sample. Be sure to perform the dilution according to the manufacturer's directions because the dilution might not have a

simple linear relationship to the conductivity.

5. Rinse the probe with distilled or deionized water and repeat step 4 until finished.

TASK 5 **Return the samples and the field data sheets to the lab/ drop-off point.**

Samples that are sent to a lab for conductivity analysis must be tested within 28 days of collection. Keep the samples on ice or refrigerated.

References

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

Hach Company. 1992. *Hach water analysis handbook.* 2nd ed. Loveland, CO.

Mississippi Headwaters River Watch. 1991. *Water quality procedures.* Mississippi Headwaters Board. March.

5.10 Total Alkalinity

What is total alkalinity and why is it important?

Alkalinity is a measure of the capacity of water to neutralize acids (see pH description). Alkaline compounds in the water such as bicarbonates (baking soda is one type), carbonates, and hydroxides remove H+ ions and lower the acidity of the water (which means increased pH). They usually do this by combining with the H+ ions to make new compounds. Without this acid-neutralizing capacity, any acid added to a stream would cause an immediate change in the pH. Measuring alkalinity is important in determining a stream's ability to neutralize acidic pollution from rainfall or wastewater. It's one of the best measures of the sensitivity of the stream to acid inputs.

Alkalinity in streams is influenced by rocks and soils, salts, certain plant activities, and certain industrial wastewater discharges.

Total alkalinity is measured by measuring the amount of acid (e.g., sulfuric acid) needed to bring the sample to a pH of 4.2. At this pH all the alkaline compounds in the sample are "used up." The result is reported as milligrams per liter of calcium carbonate (mg/L $CaCO_3$).

Analytical ana equipment considerations

For total alkalinity, a double endpoint titration using a pH meter (or pH "pocket pal") and a digital titrator or buret is recommended. This can be done in the field or in the lab. If you will analyze alkalinity in the field, it is recommended that you use a digital titrator instead of a buret because the buret is fragile and more difficult to set up and use in the field. The alkalinity method described below was developed by the Acid Rain Monitoring Project of the University of Massachusetts Water Resources Research Center.

Burets, titrators, and digital titrators for measuring alkalinity

The total alkalinity analysis involves titration. In this test, titration is the addition of small, precise quantities of sulfuric acid (the reagent) to the sample until the sample reaches a certain pH (known as an endpoint). The amount of acid used corresponds to the total alkalinity of the sample. Alkalinity can be measured using a buret, titrator, or digital titrator (described below).

- A *buret* is a long, graduated glass tube with a tapered tip like a pipet and a valve that is opened to allow the reagent to drip out of the tube. The amount of reagent used is calculated by subtracting the original volume in the buret from the volume left after the endpoint has been reached. Alkalinity is calculated based on the amount used.
- *Titrators* forcefully expel the reagent by using a manual or mechanical plunger. The amount of reagent used is calculated by subtracting the original volume in the titrator from the volume left after the endpoint has been reached. Alkalinity is then calculated based on the amount used or is read directly from the titrator.
- *Digital titrators* have counters that display numbers. A plunger is forced into a cartridge containing the reagent by turning a knob on the titrator. As the knob turns, the counter changes in proportion to the amount of reagent used. Alkalinity is then calculated based on the amount used. Digital titrators cost approximately $90.

Digital titrators and burets allow for much more precision and uniformity in the amount of titrant that is used.

How to collect and analyze samples

The field procedures for collecting and analyzing samples for pH and total alkalinity consist of the following tasks:

TASK 1 **Prepare the sample containers**

Sample containers (and all glassware used in this procedure) must be cleaned and rinsed before the first run and after each sampling run by following the procedure described under Method A on page 128. Remember to wear latex gloves.

TASK 2 **Prepare before leaving for the sampling site**

Refer to pages 19-21 for details on confirming sampling date and time, safety considerations, checking supplies, and checking weather and directions. In addition to the standard sampling equipment and apparel, when sampling for pH and alkalinity include the following equipment:

- Digital titrator
- 100-mL graduated cylinder
- 250-mL beaker
- pH meter with combination temperature and reference electrode or pH "pocket pal"
- Sulfuric acid titration cartridge, 0.16 N
- Data sheet for pH and total alkalinity to record results
- Alkalinity voluette ampules standard, 0.500 N, for accuracy check
- Wash bottle with deionized water to rinse pH meter electrode
- Magnetic stirrer, if titrated in the lab

Be sure to calibrate the pH meter before you analyze a sample. The pH meter should be calibrated prior to sample analysis and after every 25 samples according to the instructions in the meter manual. Use two pH standard buffer solutions: 4.01 and 7.0. Following are notes regarding buffers:

- The buffer solutions should be at room temperature when you calibrate the meter.
- Do not use a buffer after its expiration date.
- Always cap the buffers during storage to prevent contamination.
- 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.
- Do not reuse buffer solutions!

Be sure to let someone know where you are going and when you expect to return.

TASK 3 **Collect the sample**

Refer to page 128 for details on how to collect water samples using screw-cap bottles or Whirl-pak® bags.

TASK 4 **Measure total alkalinity (field or lab)**

The following steps are for use of a digital titrator in the field or the lab. If you are using a buret, consult Standard Methods (APHA, 1992).

Alkalinity is usually measured using sulfuric acid with a digital titrator. Sulfuric acid is added to the water sample in measured amounts until the three main forms of alkalinity (bicarbonate, carbonate, and hydroxide) are converted to carbonic acid. At pH 10, hydroxide (if present) reacts to form water. At pH 8.3, carbonate is converted to bicarbonate. At pH 4.5, it is certain that all carbonate and bicarbonate are converted to carbonic acid. Below this pH, the water is unable to neutralize the sulfuric acid and there is a linear relationship between the amount of sulfuric acid added to the sample and the change in the pH of the sample. So, additional sulfuric acid is added to the sample to reduce the pH of 4.5 by exactly 0.3 pH units (which corresponds to an exact doubling of the pH) to a pH of 4.2. However, the exact pH at which the conversion of these bases might have happened, or total alkalinity, is still unknown. This procedure uses an equation derived from the slope of the line described above to extrapolate back to the amount of sulfuric acid that was added to actually convert all the bases to carbonic acid. The multiplier (0.1) then converts this to total alkalinity as mg/L $CaCO_3$. The following steps outline the procedures necessary to determine the alkalinity of your sample.

1. Insert a clean delivery tube into the 0.16 N sulfuric acid titration cartridge and attach the cartridge to the titrator body.
2. Hold the titrator, with the cartridge tip pointing up, over a sink. Turn the delivery knob to eject air and a few drops of titrant. Reset the counter to 0 and wipe the tip.
3. Measure the pH of the sample (see pH, section, 5.4). If it is less than 4.5, go to step 9 below.
4. Insert the delivery tube into the beaker containing the sample. Turn the delivery knob while magnetically stirring the beaker until the pH meter reads 4.5. Record the number of digits used to achieve this pH. Do not reset the counter.
5. Continue titrating to a pH of 4.2 and record the number of digits.
6. Apply the following equation:

Alkalinity (as mg/L $CaCO_3$) = (2a - b) x 0.1

Where:

a = digits of titrant to reach pH 4.5
b = digits of titrant to reach pH 4.2 (including digits required to get to pH 4.5)
0.1 = digit multiplier for a 0.16 titration cartridge and a 100-mL sample

Example:

Initial pH of sample is 6.5.
It takes 108 turns to get to a pH of 4.5.
It takes another 5 turns to get to pH 4.2, for a total of 113 turns.
Alkalinity = ((2 x 108) - 113) x 0.1
= 10.3 mg/L

7. Record the results as mg/L alkalinity on the lab sheet.
8. Rinse the beaker with distilled water before the next sample.
9. If the pH of your water sample, prior to titration, is less than 4.5, proceed as follows:
   - Insert the delivery tube into the beaker containing the sample.
   - Turn the delivery knob while swirling the beaker until the pH meter reads exactly 0.3 pH units less than the initial pH of the sample.
   - Record the number of digits used to achieve this pH.
   - Apply the equation as in step 6, but a = 0 and b = the number of digits required to reduce the initial pH exactly 0.3 pH units.

Example:

Initial pH of sample is 4.3.
Enter "0" in the 4.5 column on the lab sheet.
Titrate to a pH of 0.3 units less than the initial pH—in this case 4.0.
It takes 10 digits to get to 4.0.
Enter this in the 4.2 column on the lab sheet and note that the pH endpoint is 4.0.
Alkalinity = (0 - 10) x 0.1 = -1.0.

- Record the results as mg/L alkalinity on the lab sheet.

10. Perform an accuracy check on the first field sample, halfway through the run, and after analysis of the last sample as described below. Check the pH meter against pH 7.0 and 4.01 buffers after every 10 samples.

TASK 5 **Perform an accuracy check**

This accuracy check should be performed on the first field sample titrated, again about halfway through the field samples, and at the final field sample.

1. Snap the neck off an alkalinity voluette ampule standard, 0.500 N. Or if using a standard solution from a bottle, pour a few milliliters of the standard into a clean beaker.
2. Pipet 0.1 mL of the standard to the titrated sample (see above). Resume titration back to the pH 4.2 endpoint. Record the number of digits needed.
3. Repeat using two more additions of 0.1 mL of standard. Titrate to the pH 4.2 after each addition.
4. Each 0.1-mL addition of standard should require 250 additional digits of 0.16 N titrant.

TASK 6 **Return the field data sheets and samples to the lab or drop-off point**

Alkalinity samples must be analyzed within 24 hours of their collection. If the samples cannot be analyzed in the field, keep the samples on ice and take them to the lab or drop-off point as soon as possible.

References

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

Godfrey, P.J. 1988. *Acid rain in Massachusetts.* University of Massachusetts Water Resources Research Center, Amherst, MA.

River Watch Network. 1992. *Total alkalinity and pH field and laboratory procedures* (based on University of Massachusetts Acid Rain Monitoring Project). July 1.

5.11 Fecal Bacteria

What are fecal bacteria and why are they important?

Members of two bacteria groups, coliforms and fecal streptococci, are used as indicators of possible sewage contamination because they are commonly found in human and animal feces. Although they are generally not harmful themselves, they indicate the possible presence of pathogenic (disease-causing) bacteria, viruses, and protozoans that also live in human and animal digestive systems. Therefore, their presence in streams suggests that pathogenic microorganisms might also be present and that swimming and eating shellfish might be a health risk. Since it is difficult, time-consuming, and expensive to test directly for the presence of a large variety of pathogens, water is usually tested for coliforms and fecal streptococci instead. Sources of fecal contamination to surface waters include wastewater treatment plants, on-site septic systems, domestic and wild animal manure, and storm runoff.

In addition to the possible health risk associated with the presence of elevated levels of fecal bacteria, they can also cause cloudy water, unpleasant odors, and an increased oxygen demand. (Refer to the section on dissolved oxygen.)

Indicator bacteria types and what they can tell you

The most commonly tested fecal bacteria indicators are total coliforms, fecal coliforms, *Escherichia coli*, fecal streptococci, and enterococci. All but *E. coli* are composed of a number of species of bacteria that share common characteristics such as shape, habitat, or behavior; *E. coli* is a single species in the fecal coliform group.

Total coliforms are a group of bacteria that are widespread in nature. All members of the total coliform group can occur in human feces, but some can also be present in animal manure, soil, and submerged wood and in other places outside the human body. Thus, the usefulness of total coliforms as an indicator of fecal contamination depends on the extent to which the bacteria species found are fecal and human in origin. For recreational waters, total coliforms are no longer recommended as an indicator. For drinking water, total coliforms are still the standard test because their presence indicates contamination of a water supply by an outside source.

Fecal coliforms, a subset of total coliform bacteria, are more fecal-specific in origin. However, even this group contains a genus, *Klebsiella*, with species that are not necessarily fecal in origin. *Klebsiella* are commonly associated with textile and pulp and paper mill wastes. Therefore, if these sources discharge to your stream, you might wish to consider monitoring more fecal and human-specific bacteria. For recreational waters, this group was the primary bacteria indicator until relatively recently, when EPA began recommending *E. coli* and enterococci as better indicators of health risk from water contact. Fecal coliforms are still being used in many states as the indicator bacteria.

E. coli is a species of fecal coliform bacteria that is specific to fecal material from humans and other warm-blooded animals. EPA recommends *E. coli* as the best indicator of health risk from water contact in recreational waters; some states have changed their water quality standards and are monitoring accordingly.

Fecal streptococci generally occur in the digestive systems of humans and other warm-blooded animals. In the past, fecal streptococci were monitored together with fecal coliforms and a ratio of fecal coliforms to streptococci was calculated. This ratio was used to determine whether the contamination was of human or nonhuman origin. However, this is no longer recommended as a reliable test.

Enterococci are a subgroup within the fecal streptococcus group. Enterococci are distinguished by their ability to survive in salt water, and in this respect they more closely mimic many pathogens than do the other indicators. Enterococci are typically more human-specific than the larger fecal streptococcus group. EPA recommends enterococci as the best indicator of health risk in salt water used for recreation and as a useful indicator in fresh water as well.

Which Bacteria Should You Monitor?

Which bacteria you test for depends on what you want to know. Do you want to know whether swimming in your stream poses a health risk? Do you want to know whether your stream is meeting state water quality standards?

Studies conducted by EPA to determine the correlation between different bacterial indicators and the occurrence of digestive system illness at swimming beaches suggest that the best indicators of health risk from recreational water contact in fresh water are *E. coli* and enterococci. For salt water, enterococci are the best. Interestingly, fecal coliforms as a group were determined to be a poor indicator of the risk of digestive system illness. However, many states continue to use fecal coliforms as their primary health risk indicator.

If your state is still using total or fecal coliforms as the indicator bacteria and you want to know whether the water meets state water quality standards, you should monitor fecal coliforms. However, if you want to know the health risk from recreational water contact, the results of EPA studies suggest that you should consider switching to the *E. coli* or enterococci method for testing fresh water. In any case, it is best to consult with the water quality division of your state's environmental agency, especially if you expect them to use your data.

Sampling and equipment considerations

Bacteria can be difficult to sample and analyze, for many reasons. Natural bacteria levels in streams can vary significantly; bacteria conditions are strongly correlated with rainfall, and thus comparing wet and dry weather bacteria data can be a problem; many analytical methods have a low level of precision yet can be quite complex; and absolutely sterile conditions are required to collect and handle samples.

The primary equipment decision to make when sampling for bacteria is what type and size of sample container you will use. Once you have made that decision, the same, straightforward collection procedure is used regardless of the type of bacteria being monitored. Collection procedures are described under "How to Collect Samples" below.

It is critical when monitoring bacteria that all containers and surfaces with which the sample will come into contact be sterile. Containers made of either some form of plastic or Pyrex glass are acceptable to EPA. However, if the containers are to be reused, they must be sterilized using heat and pressure. The containers can be sterilized by using an autoclave, which is a machine that sterilizes containers with pressurized steam. If using an autoclave, the container material must be able to withstand high temperatures and pressure. Plastic containers—either high-density polyethylene or polypropylene—might be preferable to glass from a practical standpoint because they will better withstand breakage. In any case, be sure to check the manufacturer's specifications to see whether the container can withstand 15 minutes in an autoclave at a temperature of 121 °C without melting. (Extreme caution is advised when working with an autoclave.) Disposable, sterile, plastic Whirl-pak® bags are used by a number of programs. The size of the container will depend on the sample amount needed for the bacteria analysis method you choose and the amount needed for other analyses.

There are two basic methods for analyzing water samples for bacteria:

1. The membrane filtration method involves filtering several different-sized portions of the sample using filters with a standard diameter and pore size, placing each filter on a selective nutrient medium in a petri plate, incubating the plates at a specified temperature for a specified time period, and then counting the colonies that have grown on the filter. This method varies for different bacteria types (variations might include, for example, the nutrient medium type, the number and types of incubations, etc.).
2. The multiple-tube fermentation method involves adding specified quantities of the sample to tubes containing a nutrient broth, incubating the tubes at a specified temperature for a specified time period, and then looking for the development of gas and/or turbidity that the bacteria produce. The presence or absence of gas in each tube is used to calculate an index known as the Most Probable Number (MPN).

Given the complexity of the analysis procedures and the equipment required, field analysis of bacteria is not recommended. Bacteria can either be analyzed by the volunteer at a well-equipped lab or sent to a state-certified lab for analysis. If you send a bacteria sample to a private lab, make sure that it is certified by the state for bacteria analysis. Consider state water quality labs, university and college labs, private labs, wastewater treatment plant labs, and hospitals. You might need to pay these labs for analysis.

This manual does not address laboratory methods because several bacteria types are commonly monitored and the methods are different for each type. For more information on laboratory methods, refer to the references at the end of this section.

If you decide to analyze your samples in your own lab, be sure to carry out a quality assurance/quality control program. Specific procedures are recommended in the section below.

How to Collect Samples

The procedures for collecting and analyzing samples for bacteria consist of the following tasks:

TASK 1 **Prepare sample containers**

If factory-sealed, presterilized, disposable Whirl-pak® bags are used to sample, no preparation is needed. Any reused sample containers (and all glassware used in this procedure) must be rinsed and sterilized at 121 °C for 15 minutes using an autoclave before being used again for sampling.

TASK 2 **Prepare before leaving for the sampling site**

Refer to pages 19-21 of the introduction for details on confirming sampling data and time, picking up equipment, reviewing safety considerations, and checking weather and directions. In addition, to sample for coliforms you should check your equipment as follows:

- Whirl-pak® bags are factory-sealed and sterilized. Check to be sure that the seal has not been removed.
- Bottles should have tape over the cap or some seal or marking to indicate that they have been sterilized. If any of the sample bottles are not numbered, ask the lab coordinator how to number them. Unless sample containers are to be marked with the site number, do not number them yourself.

TASK 3 **Collect the sample**

Refer to page 128 for details on collecting a sample using screw-cap bottles or Whirl-pak® bags. Remember to wash your hands thoroughly after collecting samples suspected of containing fecal contamination. Also, be careful not to touch your eyes, ears, nose, or mouth until you've washed your hands.

Recommended field quality assurance/ quality control procedures include:

- *Field Blanks.* These should be collected at 10 percent of your sample sites along with the regular samples. Sterile water in sterilized containers should be sent out with selected samplers. At a predetermined sample site, the sampler fills the usual sample container with this sterile water. This is labeled as a regular sample, but with a special notation (such as a "B") that indicates it is a field blank. It is then analyzed with the regular samples. Lab analysis should result in "0" bacteria counts for all blanks. Blanks are used to identify errors or contamination in sample collection and analysis.
- *Internal Field Duplicates.* These should be collected at 10 percent of your sampling sites along with the regular samples. A field duplicate is a duplicate stream sample collected at the same time and at the same place either by the same sampler or by another sampler. This is labeled as a regular sample, but with a special notation (such as a "D") that indicates it is a duplicate. It is then analyzed with the regular samples. Lab analysis should result in comparable bacteria counts per 100 mL for duplicates and regular samples collected at the same site. Duplicates are used to estimate sampling and laboratory analysis precision.
- *External Field Duplicates.* An external field duplicate is a duplicate stream sample collected and processed by an independent (e.g., professional) sampler or team at the same place at the same time as regular stream samples. It is used to estimate sampling and laboratory analysis precision.

TASK 4 **Return the field data sheets and the samples to the lab or drop-off point**

Samples for bacteria must be analyzed within 6 hours of collection. Keep the samples on ice and take them to the lab or drop-off point as soon as possible.

TASK 5 **Analyze the samples in the lab**

This manual does not address laboratory analysis of water samples. Lab methods are described in the references below (APHA, 1992; River Watch Network, 1991; USEPA, 1985). However, the lab you work with should carry out the following recommended laboratory quality assurance/quality control procedures:

- *Negative Plates* result when the buffered rinse water (the water used to rinse down the sides of the filter funnel during filtration) has been filtered the same way as a sample. This is different from a field blank in that it contains reagents used in the rinse water. There should be no bacteria growth on the filter after incubation. It is used to detect laboratory bacteria contamination of the sample.
- *Positive Plates* result when water known to contain bacteria (such as wastewater treatment plant influent) is filtered the same way as a sample. There should be plenty of bacteria growth on the filter after incubation.

Positive plates are used to detect procedural errors or the presence of contaminants in the laboratory analysis that might inhibit bacteria growth.

- *Lab Replicates*. A lab replicate is a sample that is split into subsamples at the lab. Each subsample is then filtered and analyzed. Lab replicates are used to obtain an optimal number of bacteria colonies on filters for counting purposes. Usually, subsamples of 100, 10, and 1 milliliter (mL) are filtered to obtain bacteria colonies on the filter that can be reliably and accurately counted (usually between 20 and 80 colonies). The plate with the count between 20 and 80 colonies is selected for reporting the results, and the count is converted to colonies per 100 mL.
- *Knowns*. A predetermined quantity of dehydrated bacteria is added to the reagent water, which should result in a known result, within an acceptable margin of error.
- *Outside Lab Analysis of Duplicate Samples*. Either internal or external field duplicates can be analyzed at an independent lab. The results should be comparable to those obtained by the project lab.

References

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

Hogeboom, T. Microbiologist, Vermont Environmental Conservation Laboratory, Waterbury, VT. Personal communication.

River Watch Network. 1991. Escherichia coli (E. coli) membrane filter procedure (adapted from USEPA Method 1103.1, 1985). Montpelier, VT. October.

USEPA. 1985. *Test methods for Escherichia coli and enterococci in water by the membrane filter procedure (Method #1103.1).* EPA 600/4-85-076. U.S. Environmental Protection Agency, Environmental Monitoring and Support Laboratory, Cincinnati, OH.

USEPA. 1986. *Bacteriological ambient water quality criteria for marine and fresh recreational waters.* EPA 440/5-84-002. U.S. Environmental Protection Agency, Office of Research and Development, Cincinnati, OH.

It is hard to overemphasize the importance of having established methods of handling volunteer data, analyzing that data, and presenting results effectively to volunteers, the public, and water resource decision-makers. Without these tools and processes, the data that volunteers and program managers have labored hard to collect are virtually useless, and the program will surely fail to meet its goals.

This chapter addresses data management and data presentation. Members of the program planning committee will need to make many decisions on these issues before the first field data sheet is filled out by the program's first volunteer. In particular, they should consult any potential data users such as state water quality agencies or county planning boards regarding their own data needs. Data users will be particularly concerned about:

- Procedures used to verify and check the raw volunteer data.
- Databases and software used to manage the data.
- Analytical procedures used to convert the raw data into findings and conclusions.
- Reporting formats.

Data users may, for example, be able to offer concrete suggestions about databases and presentation formats that will make the data more accessible to them. To ensure that all questions about the validity of the data can be answered, the program planning committee should develop and implement a quality assurance/quality control plan designed to minimize data collection errors, weed out data that fail to meet the program's standards, and effectively analyze and present the results. This plan should identify key personnel with responsibilities for data management and data analysis and clearly indicate all the steps the program will take to handle the data.

Unfortunately, volunteers and program coordinators seldom recognize the importance of this aspect of a volunteer monitoring program. It tends to be considered "drudge" work assigned to one or two technically- inclined people. However, that attitude is seriously out of date. Program organizers should make every effort to involve a range of volunteers and program staff in all aspects of data management and presentation. Sufficient time should be budgeted to the tasks that are involved. People who produce the reports should be acknowledged. After all, it is the final reports that will be reviewed by stream management decision-makers, not the field data sheets. No other tasks are more important to the success of the volunteer stream monitoring program.

6.1 Managing Volunteer Data

The following steps will help ensure that the data collected by volunteers are well managed, credible, and of value to potential data users.

Review Field Data Sheets

The volunteer program coordinator or designated analyst should screen and review the field data sheets as they are received. This involves some basic "reality checks." Questions that should be kept in mind include the following:

- *Are the results as might be anticipated, or are they highly unexpected? If unexpected, are they still within the realm of possibility?*

  For example, can the kit or technique the volunteer used actually produce results like that? Does the volunteer offer any possible explanations for the results (e.g., a sewage treatment plant malfunction had been recently reported) or corollary information (e.g., a fish kill has been observed along with the extremely low dissolved oxygen readings)? Also check for consistency between similar parameters. For example, total dissolved solids and conductivity should track together—if one goes up, so should the other. So should total solids and turbidity.

- *Are there outliers? (Findings that differ radically from past data or other data from similar sites.)*

  Values that are off by a factor of 10 or 100 should be questioned. Follow up on any data that seems suspect. If you can't come up with an explanation for why the results are so unusual, but they are still within the realm of possibility, you may want to flag the data as questionable. Ask an experienced volunteer or program staffer to sample at that site as a backup until uncertainties are resolved, or work with the volunteer to verify that proper sampling and analytical protocols are being followed.

- *Are the field data sheets complete?*

  If a volunteer is consistently leaving a section of the sheet incomplete, follow up and ask why. Instructions may not always be easily understood. All sheets should include site location and identification, name of the volunteer, date, time, and weather conditions.

- *Are all measurements reported in the correct units?*

  You should minimize the chance for error by including on the data form itself any equations needed to convert measurements, and specify on the form what units should be used. Check the math. All field data sheets should be kept on file in the event that findings are brought into question at a later date.

Review Information in Your Database

Once volunteer data enters a computerized database, it can take on a life of its own. It is a phenomenon of human nature that data suddenly seem more believable once computerized. Therefore, be sure to carefully screen information as soon as you enter it into a database. Then review a printout (preferably with a fresh pair of eyes) against the original field data sheets. One way to minimize transcription errors is to design the computer input screens to look like the field data forms.

As a further check, you can run some simple calculations like determining medians and means to make sure no errors have slipped through. (If the median and the mean are very different, an outlier may be skewing the results.) Again, if you uncover unusual data points that cannot be explained by backup information on the field data sheets or the comment field in the database, flag the data as questionable until it can be verified.

Review Your Final Results

Once volunteer monitoring data has been entered into a database, the next step is to generate reports on the findings of the data. Even at this stage you should continue to look for inconsistencies and problems. For example, you should:

- Review findings against previous years' data.
- Look for outliers on graphs and maps.
- Not remove data just because you don't like it, but do investigate findings that are unusual or can't be explained.

By the time you present your final results to your volunteers or other data users, you should feel fully confident that you have assembled the best possible picture of water quality conditions in your study streams.

Develop a Coding System

A coding system will help simplify the tracking and recording of data. Make sure, however, that the system you create is easily understood and simple to use. Codes developed for sample sites, parameters, and other information on field and lab sheets should parallel the codes you use in your database. If you will be sharing your information with a state or local natural resource agency, you may want your coding system to match or complement the agency system.

Sample Sites: Because sample sites tend to change over time, it is important to have a site numbering system that accommodates change. A good convention to follow is to use a site coding system that includes an abbreviation of the waterbody and a site number (e.g., CtR020 for a site on the Connecticut River). For consistency, you might choose to start the site numbers at the downstream end of the stream and increase them as you move upstream (e.g., the first Connecticut River site would be CtR010, the second CtR020, etc.). Leave extra numbers between sites to allow for your program's future expansion.

Water Quality Parameters: It is also important to develop a coding system for each of the water quality parameters you are testing. These are the codes you will use in the database to identify and extract results. To keep the amount of clerical work to a minimum, abbreviate without losing the ability to distinguish parameters from one another. For example, EC could represent *E. coli* bacteria and FC fecal coliform bacteria.

Spreadsheets, Databases, and Mapping Software

Today's computer software includes a variety of spreadsheet and database packages that allow you to sort, manipulate, and perform statistical analyses on the data you have entered into the computer. For most applications, spreadsheets are adequate and have the advantage of being relatively simple to use. Most spreadsheet packages have graphics capabilities that will allow you to plot your data onto a graph of your choice (i.e., bar, line, or pie chart). Examples of common spreadsheet software packages are Lotus 1-2-3, Excel, and Quattro Pro.

Database software may be more difficult to master and usually lack the graphics capabilities of spreadsheet software. If you manage large amounts of data, however, a database is almost a necessity.

Using a database, you can store and manipulate very large data sets without sacrificing speed. The database can also relate records in one file to records in another file. This allows you to break your data up into smaller, more easily managed files that can work together as though they were one.

If you use database software for storage and retrieval, you may still want to use a spreadsheet or other program with graphics capabilities. Many spreadsheet and database software packages are compatible and will allow you to transport sets of data back and forth with relative ease. Very large data sets can be organized and manipulated in a database. Specific parts of the data (such as results for a particular metric from all stations and all sampling events) can then be transported into the spreadsheet, statistically analyzed, and graphically displayed. Examples of popular database software packages are dBase, FileMaker Pro, and FoxPro.

An effective way to display your data is on a map of the stream or watershed. This clearly illustrates the relationship between land uses and the quality of water, habitat, and biological communities. This type of graphic display can be used to effectively show the correlation between specific activities or land uses and the impacts they have on the ecosystem. Simple personal computer-based mapping packages are available. They allow you to enter layers of data and conduct spatial analysis of that data.

Systems that allow you to map and manipulate various layers of information (such as water quality data, land use information, county boundaries, or geologic conditions) are known as Geographic Information Systems (GIS). They can vary from simple systems run on personal computers to sophisticated and very powerful systems that run on large main frames. For any GIS application, you need to know the coordinates of your sample sites—either their latitude and longitude, or some alternate system such as an EPA River Reach File identifier. You can also locate your sites on a topographic map that can be digitized on to an electronic map of the watershed. Once these points have been established, you can link your database to the points on the map, query your database, and create graphic displays of the data.

Powerful GIS applications typically require expensive hardware, software, and technical training. Any volunteer program interested in GIS applications should consider working in partnership with other organizations such as universities, natural resource agencies, or large nonprofit groups that can provide access to a GIS.

Many people are capable of writing their own programs to manipulate and display data. The disadvantage of using a "homegrown" software program, however, is that if its author leaves, so too does all knowledge about how the program works. Commercial software, on the other hand, comes with consumer services that provide over-the-phone help and instructions, user's guides, replacement guarantees, and updates as the company improves its product. Also, most commercial programs are developed to easily import and export data in standard formats. This feature is important if you want to share data with other programs or organizations—all you need are compatible software programs.

STORET

EPA's national water and biological data storage and retrieval system, STORET, is being modernized and will be available in 1998-1999. Volunteer programs are encouraged to enter their data into the modernized STORET. Individual systems will "feed" data to a centralized file server which will permit national data analyses and through which data can be shared among organizations. A specific set of quality control measures will be required for any data entered into the system to aid in data sharing. For more information, see the EPA web page at www.epa.gov/owow/STORET.

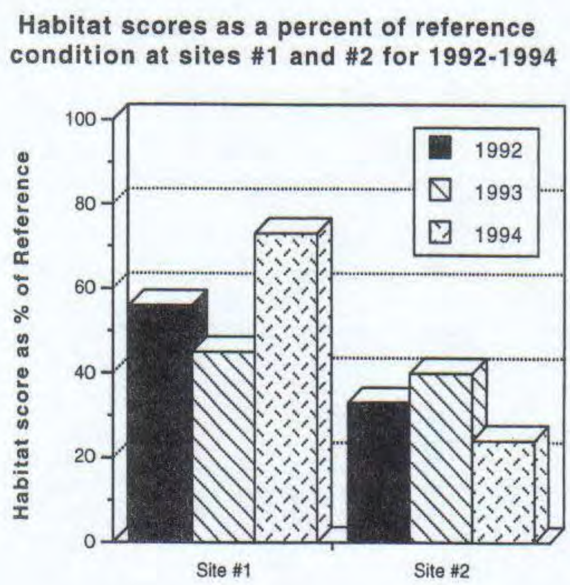
6.2 Presenting the Data

When presenting numerical data, one of your chief goals should be to maintain the attention and interest of your audience. This is very difficult using tables filled with numbers. Most people will not be interested in the absolute values of each parameter at each sampling site. Rather, they will want to know the bottom line for each site (e.g., is it good or bad) and seasonal and year to year trends.

Graphs and charts, therefore, are typically the best way to present volunteer data. Take care, however, that your graphs "fit" your audience and are neither too technical nor too simplistic.

Fig. 6.1

Example of a bar graph displaying biological data



Graphs and Charts

Graphs can be used to display the summarized results of large data sets and to simplify complicated issues and findings. The three basic types of graphs that are typically used to present volunteer monitoring data are:

- Bar graph
- Line graph
- Pie chart

Bar and line graphs are typically used to show results, such as bioassessment scores, along a vertical or y-axis for a corresponding variable (such as sampling date or site) which is marked along the horizontal or x-axis. These types of graphs can also have two vertical axes, one on each side, with two sets of results shown in relation to each other and to the variable along the x-axis.

Bar Graph

A bar graph uses columns with heights that represent the value of the data point for the parameter being plotted. Fig. 6.1 is an example using fictional data from Volunteer Creek.

Line graph

A line graph is constructed by connecting the data points with a line. It can be effectively used for depicting changes over time or space. This type of graph places more emphasis on trends and the relationship among data points and less emphasis on any particular data point.

Fig. 6.2 is an example of a line graph again using fictional data from Volunteer Creek.

Pie chart

Pie charts are used to compare categories within the data set to the whole. The proportion of each category is represented by the size of the wedge. Pie charts are popular due to their simplicity and clarity. (See Fig. 6.3)

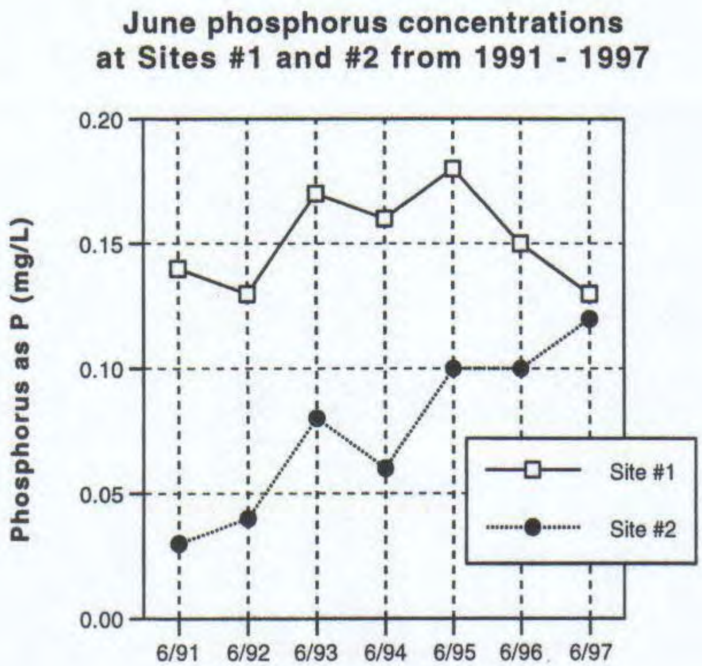


Fig. 6.2

Example of a line graph depicting trends in phosphorus data

Graphing Tips

Regardless of which graphic style you choose, follow these rules to ensure you use them most effectively.

- *Each graph should have a clear purpose.* The graph should be easy to interpret and should relate directly to the content of the text of a document or the script of a presentation.
- *The data points on a graph should be proportional to the actual values so as not to distort the meaning of the graph.* Labeling should be clear and accurate and the data values should be easily interpreted from the scales. Do not overcrowd the points or values along the axes. If there is a possibility of misinterpretation, accompany the graph with a table of the data.
- *Keep it simple.* The more complex the graph, the greater the possibility for misinterpretation.

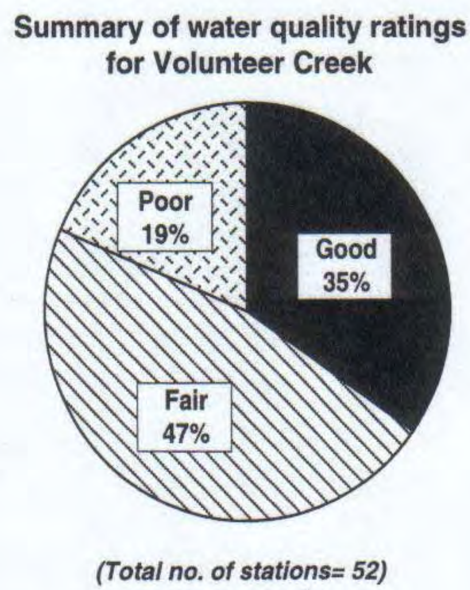


Fig. 6.3

Example of a pie chart summarizing water quality ratings

- *Limit the number of elements.* Pie charts should be limited to five or six wedges, the bars in a bar graph should fit easily, and the lines in a line graph should be limited to three or less.
- *Consider the proportions of the graph and expand the elements to fill the dimensions, thereby creating a balanced effect.* Often, a horizontal format is more visually appealing and makes labeling easier. Try not to use abbreviations that are not obvious to someone who is unfamiliar with the program.
- *Create titles that are simple, yet adequately describe the information portrayed in the graph.*
- *Use a legend if one is necessary to describe the categories within the graph.* Accompanying captions may also be needed to provide an adequate description of the elements.

Summary Statistics

Summary statistics can reduce a very large data set to a few numerical values that can then be easily described and analyzed. Such statistics include the mean and standard deviation—two of the most frequently used descriptors of environmental data.

Textbook statistics commonly assume that if a parameter is measured many times under the same conditions, then the measurement values will be randomly distributed around the average with more values clustering near the average than further away. In this ideal situation, a graph of the frequency of each measure plotted against its magnitude should yield a bell-shaped or normal curve. The *mean* and the *standard deviation* determine the height and breadth of this curve, respectively.

The mean is simply the sum of all the measurement values divided by the number of measurements. This statistic is a measure of location and in a normal curve marks the highest point at the center of the bell.

The standard deviation, on the other hand, describes the variability of the data points around the mean. Very similar measurement values will have a small standard deviation while widely scattered data will have a much larger standard deviation.

While both the mean and standard deviation are quite useful in describing stream data, often the actual measures do not fit a normal distribution. Other statistics often come into play to describe the data. Some data are skewed in one direction or the other. Other data may have a flattened bell shape.

It is important to note that biological information often does not follow normal, bell-shaped distribution. This is because biological communities are dynamic, complex, and interdependent systems; many factors influence them, and these cannot be statistically predicted. For example, bioassessment scores plotted against habitat assessment scores will be at their best when habitat quality is at its best. For data that is nonnormally distributed, the mean and the standard deviation are not appropriate summary statistics.

For describing nonnormally distributed data, it is best to use statistics that can convey the information for a variety of conditions and which are not overly influenced by the data points at the extremes of the distribution. The median and the interquartile range are two statistics that are commonly used to describe the central tendency and the spread around the median, respectively. These statistics are derived by placing the data points in order of value from lowest to highest. The median is simply the value that is in the middle of the data set. The interquartile range is the difference between the value at the 75 percent level and the value at the 25 percent level.

The best method for presenting this type of data is called a box and whisker plot. One simple box and whisker plot will graphically display the following information:

- Median
- Variability of the data around the median
- Skew of the data
- Range of the data
- Size of the data set

Statistical software packages for computers will easily construct box and whisker plots. You can construct these plots by following procedure shown below:

1. Order the data from the lowest to the highest.
2. Plot the lowest and highest values on the graph as short horizontal lines. These are the extreme values of the data set and represent the data range.
3. Determine the 75 percent value and the 25 percent value of the data set. These values define the interquartile range and are represented by the location of the top and bottom lines of the box.
4. The horizontal length of the lines that define the top and bottom lines of the box (the box width) can be used as a relative indication of the size of the data set. For example, the box width that describes a data set of 20 values can be displayed twice as wide as a data set of 10 values. Any proportional scheme can be used as long as it is consistently applied.
5. Close the box by drawing vertical lines that connect to the ends of the horizontal lines.
6. Plot the median inside the box.

Fig. 6.4 is an example depicting the extreme values, interquartile range, and median of biosurvey metric scores from 52 sites sampled in Volunteer Creek in June, 1995.

Maps

Displaying the results of your monitoring data on a map can be a very effective way of showing the data and helping people understand what it means. A map shows the location of sample sites in relation to land features, such as cities, wastewater treatment plants, farmland, and tributaries that may have an effect on water quality. Because a map also displays the stream's relationship to neighborhoods, parks and recreational areas, it can help to develop concern for the stream and strengthens interest in protecting it.

Choosing a Map

It is best to have two types of maps. One should be a working map with a lot of detail. The other should be used for display

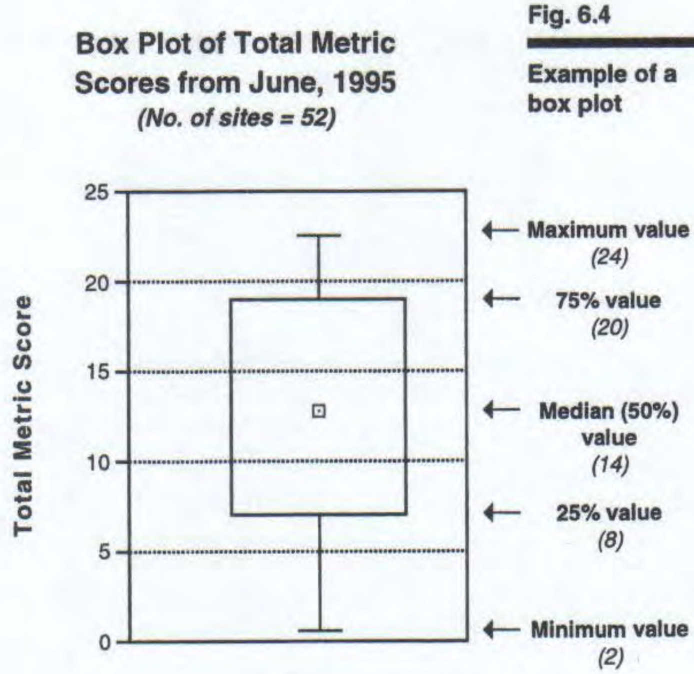


Fig. 6.4

Example of a box plot

purposes. The working map should include important features such as:

- Stream and its tributaries
- Wetlands
- Lakes and ponds
- Cultural features such as roads
- Rail and power lines; municipal boundaries
- Some indication of land use patterns and vegetation.

The map should be of a scale large enough to add the location of sample sites.

U.S. Geological Survey (USGS) 7.5 minute quads (scale of 1:24,000; 1 in. = 2,000 ft) are available with and without topographic contours (elevation markings). These maps are available for the most of the United States.

The USGS maps are particularly useful if your information will be incorporated into a geographic information system (GIS), since many of these systems use the USGS maps as base maps. For your data to be used in a GIS, it is likely that you will have to provide the latitude and longitude of your sample sites, which can be obtained by using the grid markings on the USGS topographic maps. Several different coordinate systems are marked, including standard latitude/longitude and the Universal Transmercator coordinates. For assistance in learning how to use these coordinate markings, talk to the local USGS office or someone in the geography department at a university. It may also be possible for the GIS office you work with you to "digitize" the maps, thus saving you the trouble of trying to calculate the coordinates.

The display map is best used to illustrate your program results at public meetings or in reports. This map should be simpler than the detailed map and show only principal features such as roads, municipal boundaries, and waterways. It should have sufficient detail and scale to show the location of sample sites, and have space for summary information about each of the sample sites. Commercial road atlases and county or town road maps available from state transportation departments are examples of the types of maps that can be used for display purposes (See Fig. 6.5).

Creating a Display Map

Some suggestions for using a map to display your data include:

- Keep the amount of information presented on each map to a minimum. Do not try to put so much on one map that it becomes visually complicated and difficult to read or understand. Use another map to display a different layer or "view" of the data. For example, if there are several dates for which you wish to display sampling results, use one map for each date.
- Clearly label the map and provide an explanation of how to interpret it. If you need a long and complicated explanation, you may want to present the data differently. If you have reached a clear conclusion, state the conclusion on the map. For example, if a map shows that tributaries are cleaner than the mainstem, use that information as the subtitle of the map.
- Provide a key to the symbols that are used on the map.
- Rather than packing lots of information into a small area of the map, use a "blow-up" or enlargement of the area elsewhere on the map to adequately display the information.
- Use symbols that vary in size and pattern to represent the magnitude of results. For example, a site with a fecal coliform level of 10 per 100 milliliters could be a light gray circle one-sixteenth inch diameter while a site with a level of 200 per 100

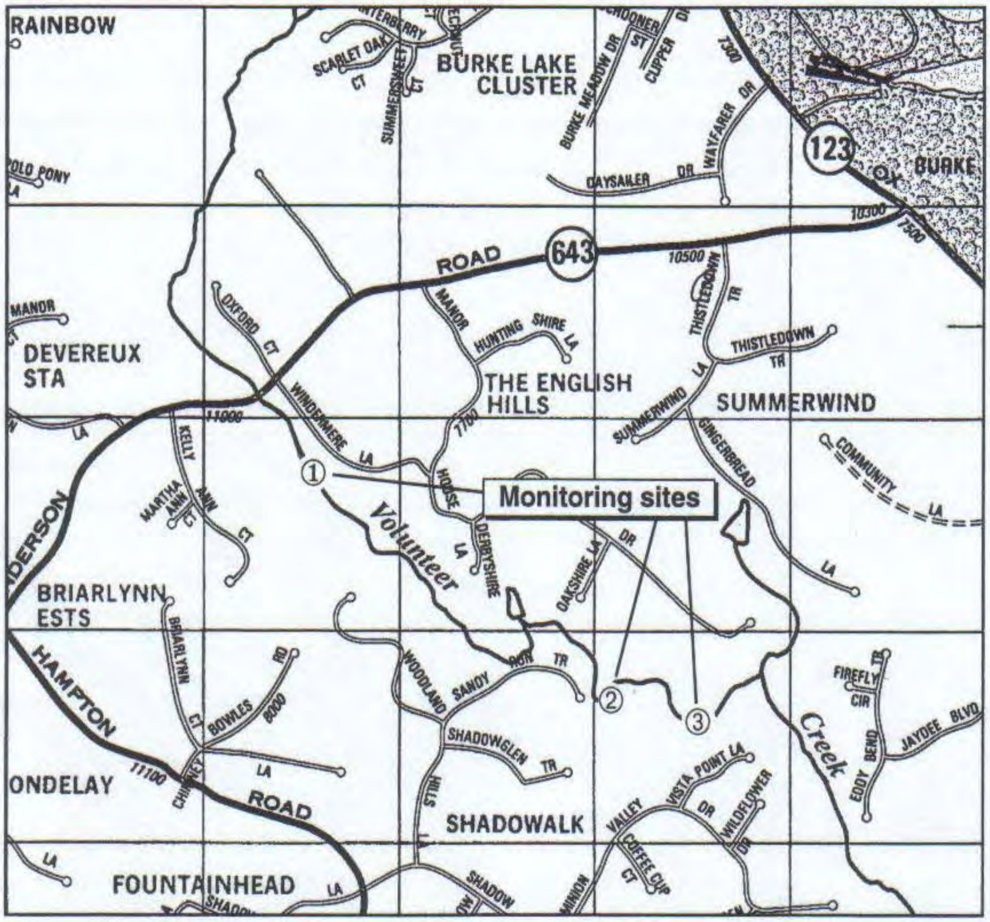


Figure 6.5

A road map is useful for displaying station locations.

milliliters would be a dark gray circle one-quarter inch diameter. Start by finding the highest and lowest values, assign diameters and patterns to those and then fill in steps along the way. For the above example you might have four ranges: 0 to 99, 100 to 199, 200 to 500 and 500 +.

Maps on Demand

EPA provides a World Wide Web service known as Maps on Demand that allows users to generate maps displaying environmental information for anywhere in the U.S. (except Hawaii, Puerto Rico, and the Virgin Islands). Types of information that can be mapped include EPA-regulated facilities, demographic information, roads, streams, and drinking water sources. Maps of varying scales can be generated on the site (latitude and longitude), zip code, county, and basin levels. Submit your request and email address, and after a brief wait, you will be able to view your map on-line or download it. Maps on Demand can be reached through EPA's *Surf Your Watershed* homepage at www.epa.gov/surf/info.html.

6.3 Producing Reports

On a regular basis, a successful stream volunteer monitoring program should produce reports that summarize key findings to volunteers; data users such as state water quality agencies, and local planning boards; and/or the general public, including the media. State water quality agencies will require detailed reports, whereas shorter and less technical summaries are more appropriate for the general public. All reports should be subjected to the review process prescribed by your Quality Assurance Project Plan.

Professional Report

In a report designed for water quality or planning professionals, you should go into detail about:

- The purpose of the study
- Who conducted it
- How it was funded
- The methods used
- The quality control measures taken
- Your interpretation of the results
- Your conclusions and recommendations
- Further questions that have arisen as a result of the study.

Graphics, tables and maps may be fairly sophisticated. Be sure to include the raw data in an appendix and note any problems encountered.

Lay Report

A report for the general public should be short and direct. It is very important to write in a non-technical style and to include definitions for terms and concepts that may be unfamiliar to the lay person. Simple charts, summary tables, and maps with accompanying explanations can be especially useful. This type of report should include a brief description of the program, the purpose of the monitoring, an explanation of the parameters that were monitored, the location of sample sites, a summary of the results, and any recommendations that may have been made.

Both types of reports should acknowledge the volunteers and the sources of funding.

Publicizing the Report

Develop a strategy for distributing and publicizing your report before it is completed. Be sure the planning committee is confident about the data and comfortable with the statements and conclusions that have been included in the document. When the report is released to the public, you will need to be prepared to respond to questions regarding the data and your interpretation of that data.

Some ideas for distributing the results and informing the public include the following:

- *Mailing the report.* If you have access to a mailing list of people who are interested in your stream, mail the report with a cover letter that summarizes the major findings of the study. The cover letter should be brief and enticing so that the recipient will be curious enough to read the report. If you want people to take some kind of action, such as supporting the expenditure of public funds to upgrade a sewage treatment plant, you may want to ask for their support in the cover letter. If you do not have an extensive mailing list, perhaps other organizations that share your goals would be willing to supply you with their list. Be sure to also send the report to the newspapers, radio and television stations, and state and federal agencies.

- *Speaking tour.* You may also want to develop an oral presentation (with slides, overheads, etc.) that could be offered to groups such as the Chamber of Commerce, Rotary clubs, conservation organizations, schools, and government entities. Your presentation could even be videotaped for distribution to a wider audience.
- *Public meetings.* You may want to schedule a series of public meetings that highlight the program and its findings and recommendations. At the meetings, distribute the report, answer questions and tell your audience how they can get involved. These meetings can also help you recruit more volunteers.

  Be sure to schedule the meetings at times when people are more likely to attend (i.e., weekday evenings, weekend days) and avoid periods when people are normally busy or on vacation. Invite the media and publicize the meetings in newspaper calendars, send press releases to newspapers, radio and television stations and other organizations, and ask volunteers to distribute flyers at grocery stores, city hall, etc.
- *News releases.* Writing and distributing a news release is a cost effective means of informing the public about the results and accomplishments of your program. Develop a mailing list of newspapers, radio and television stations, and organizations that solicit articles for publication. Send the news release to volunteers and others who are interested in publicizing the monitoring program.

  The first page of your news release should feature the sponsoring organization's name and logo to clearly designate the source of the news. Include a headline, the date, a contact name and number, and whether the story is for release immediately or a later date. The first paragraph should begin with a dateline (the city of origin for the event or story described in the release) and include the essentials: who, what, where, when, and why and a synopsis of the most important elements of the story. The second paragraph should contain the second most important facts, the third paragraph the third most important points and so on. Editors tend to chop off the last paragraphs if short on space. Therefore, be sure to state your major points early in the press release.
- *News conferences.* If your report contains some real news, or if it has led to a significant event, (e.g., the mayor or city council has recognized the value of the report and issued a statement of support) hold a news conference. Timing and location are important. Early in the day, but after 10 a.m. is good (most camera crews start their workday at 9 a.m.) because it allows plenty of time to edit the tape before the noon news broadcast. You may want to consider timing the conference so that a TV station could broadcast it live at the noon or the evening news show. For the conference, choose a place that has good visuals, such as location along the river or water body that you have been studying, at your headquarters where volunteers can be shown working in the background or at a recognition gathering for volunteers.
- *Other publicity.* Be creative in getting your report and message out. Try writing op-ed articles for local or statewide papers, writing letters to the editor, producing radio feeds (a

recording of the group's leader played over the phone to a radio station), issuing media advisories, and even advertising in publications. For more help on getting your message across, consult the references cited below.

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Water Science Glossary of Terms

Here's a list of water-related terms that might help you understand our site better. It is compiled from a number of sources and should not be considered an "official" U.S. Geological Survey water glossary. A detailed water glossary is kept by the Water Quality Association, and an extremely detailed water dictionary is offered by the Nevada Division of Water Resources.

A | B | C | D | E | F | G | H | I | K | L | M | N | O | P | R | S | T | U | W | X | Y

A

Acequia: gravity-driven waterways, similar in concept to a flume. Most are simple ditches with dirt banks, but they can be lined with concrete. They were important forms of irrigation in the development of agriculture in the American Southwest. The proliferation of cotton, pecans and green chile as major agricultural staples owe their progress to the acequia system.

Acid: a substance that has a pH of less than 7, which is neutral. Specifically, an acid has more free hydrogen ions (H^+) than hydroxyl ions (OH^-).

Acre-foot (acre-ft): the volume of water required to cover 1 acre of land (43,560 square feet) to a depth of 1 foot. Equal to 325,851 gallons or 1,233 cubic meters.

Alkaline: sometimes water or soils contain an amount of alkali (strongly basic) substances sufficient to raise the pH value above 7.0 and be harmful to the growth of crops.

Alkalinity: the capacity of water for neutralizing an acid solution.

Alluvium: deposits of clay, silt, sand, gravel, or other particulate material that has been deposited by a stream or other body of running water in a streambed, on a flood plain, on a delta, or at the base of a mountain.

Appropriation doctrine: the system for allocating water to private individuals used in most Western states. The doctrine of Prior Appropriation was in common use throughout the arid west as early settlers and miners began to develop the land. The prior appropriation doctrine is based on the concept of "First in Time, First in Right." The first person to take a quantity of water and put it to Beneficial Use has a higher priority of right than a subsequent user. Under drought conditions, higher priority users are satisfied before junior users receive water. Appropriative rights can be lost through nonuse; they can also be sold or transferred apart from the land. Contrasts with Riparian Water Rights.

Aquaculture: farming of plants and animals that live in water, such as fish, shellfish, and algae.

Aqueduct: a pipe, conduit, or channel designed to transport water from a remote source, usually by gravity.

Aquifer: a geologic formation(s) that is water bearing. A geological formation or structure that stores and/or transmits water, such as to wells and springs. Use of the term is usually restricted to those water-bearing formations capable of yielding water in sufficient quantity to constitute a usable supply for people's uses.

Aquifer (confined): soil or rock below the land surface that is saturated with water. There are layers of impermeable material both above and below it and it is under pressure so that when the aquifer is penetrated by a well, the water will rise above the top of the aquifer.

Aquifer (unconfined): an aquifer whose upper water surface (water table) is at atmospheric pressure, and thus is able to rise and fall.

Artesian water: ground water that is under pressure when tapped by a well and is able to rise above the level at which it is first encountered. It may or may not flow out at ground level. The pressure in such an aquifer commonly is called artesian pressure, and the formation containing artesian water is an artesian aquifer or confined aquifer. See flowing well

Artificial recharge: a process where water is put back into ground-water storage from surface-water supplies such as irrigation, or induced infiltration from streams or wells.

B

Base flow: sustained flow of a stream in the absence of direct runoff. It includes natural and human-induced stream flows. Natural base flow is sustained largely by ground-water discharges.

Base: a substance that has a pH of more than 7, which is neutral. A base has less free hydrogen ions (H^+) than hydroxyl ions (OH^-).

Bedrock: the solid rock beneath the soil and superficial rock. A general term for solid rock that lies beneath soil, loose sediments, or other unconsolidated material.

C

Capillary action: the means by which liquid moves through the porous spaces in a solid, such as soil, plant roots, and the capillary blood vessels in our bodies due to the forces of adhesion, cohesion, and surface tension. Capillary action is essential in carrying substances and nutrients from one place to another in plants and animals.

Commercial water use: water used for motels, hotels, restaurants, office buildings, other commercial facilities, and institutions. Water for commercial uses comes both from public-supplied sources, such as a county water department, and self-supplied sources, such as local wells.

Condensation: the process of water vapor in the air turning into liquid water. Water drops on the outside of a cold glass of water are condensed water. Condensation is the opposite process of evaporation.

Consumptive use: that part of water withdrawn that is evaporated, transpired by plants, incorporated into products or crops, consumed by humans or livestock, or otherwise removed from the immediate water environment. Also referred to as water consumed.

Conveyance loss: water that is lost in transit from a pipe, canal, or ditch by leakage or evaporation. Generally, the water is not available for further use; however, leakage from an irrigation ditch, for example, may percolate to a ground-water source and be available for further use.

Cubic feet per second (cfs): a rate of the flow, in streams and rivers, for example. It is equal to a volume of water one foot high and one foot wide flowing a distance of one foot in one second. One "cfs" is equal to 7.48 gallons of water flowing each second. As an example, if your car's gas tank is 2 feet by 1 foot by 1 foot (2 cubic feet), then gas flowing at a rate of 1 cubic foot/second would fill the tank in two seconds.

D

Desalination: the removal of salts from saline water to provide freshwater. This method is becoming a more popular way of providing freshwater to populations.

Discharge: the volume of water that passes a given location within a given period of time. Usually expressed in cubic feet per second.

Domestic water use: water used for household purposes, such as drinking, food preparation, bathing, washing clothes, dishes, and dogs, flushing toilets, and watering lawns and gardens. About 85% of domestic water is delivered to homes by a public-supply facility, such as a county water department. About 15% of the Nation's population supplies their own water, mainly from wells.

Drainage basin: land area where precipitation runs off into streams, rivers, lakes, and reservoirs. It is a land feature that can be identified by tracing a line along the highest elevations between two areas on a map, often a ridge. Large drainage basins, like the area that drains into the Mississippi River contain thousands of smaller drainage basins. Also called a "watershed."

Drip irrigation: a common irrigation method where pipes or tubes filled with water slowly drip onto crops. Drip irrigation is a low-pressure method of irrigation and less water is lost to evaporation than high-pressure spray irrigation.

Drawdown: a lowering of the ground-water surface caused by pumping.

E

Effluent: water that flows from a sewage treatment plant after it has been treated.

Erosion: the process in which a material is worn away by a stream of liquid (water) or air, often due to the presence of abrasive particles in the stream.

Estuary: a place where fresh and salt water mix, such as a bay, salt marsh, or where a river enters an ocean.

Evaporation: the process of liquid water becoming water vapor, including vaporization from water surfaces, land surfaces, and snow fields, but not from leaf surfaces. See transpiration

Evapotranspiration: the sum of evaporation and transpiration.

F

Flood: an overflow of water onto lands that are used or usable by man and not normally covered by water. Floods have two essential characteristics: The inundation of land is temporary; and the land is adjacent to and inundated by overflow from a river, stream, lake, or ocean.

Flood, 100-year: the100-year flood does not refer to a flood that occurs once every 100 years, but to a flood level with a 1 percent chance of being equaled or exceeded in any given year.

Flood plain: a strip of relatively flat and normally dry land alongside a stream, river, or lake that is covered by water during a flood.

Flood stage: the elevation at which overflow of the natural banks of a stream or body of water begins in the reach or area in which the elevation is measured.

Flowing well/spring: a well or spring that taps ground water under pressure so that water rises without pumping. If the water rises above the surface, it is known as a flowing well.

Freshwater: water that contains less than 1,000 milligrams per liter (mg/L) of dissolved solids; generally, more than 500 mg/L of dissolved solids is undesirable for drinking and many industrial uses.

G

Gage height: the height of the water surface above the gage datum (zero point). Gage height is often used interchangeably with the more general term, stage, although gage height is more appropriate when used with a gage reading.

Gaging station: a site on a stream, lake, reservoir or other body of water where observations and hydrologic data are obtained. The U.S. Geological Survey measures stream discharge at gaging stations.

Geyser: a geothermal feature of the Earth where there is an opening in the surface that contains superheated water that periodically erupts in a shower of water and steam.

Giardiasis: a disease that results from an infection by the protozoan parasite Giardia intestinalis, caused by drinking water that is either not filtered or not chlorinated. The disorder is more prevalent in children than in adults and is characterized by abdominal discomfort, nausea, and alternating constipation and diarrhea.

Glacier: a huge mass of ice, formed on land by the compaction and recrystallization of snow that moves very slowly down slope or outward due to its own weight.

Grey water: wastewater from clothes washing machines, showers, bathtubs, hand washing, lavatories and sinks.

Ground water: (1) water that flows or seeps downward and saturates soil or rock, supplying springs and wells. The upper surface of the saturate zone is called the water table. (2) Water stored underground in rock crevices and in the pores of geologic materials that make up the Earth's crust.

Ground water, confined: ground water under pressure significantly greater than atmospheric, with its upper limit the bottom of a bed with hydraulic conductivity distinctly lower than that of the material in which the confined water occurs.

Ground-water recharge: inflow of water to a ground-water reservoir from the surface. Infiltration of precipitation and its movement to the water table is one form of natural recharge. Also, the volume of water added by this process.

Ground water, unconfined: water in an aquifer that has a water table that is exposed to the atmosphere.

H

Hardness: a water-quality indication of the concentration of alkaline salts in water, mainly calcium and magnesium. If the water you use is "hard" then more soap, detergent or shampoo is necessary to raise lather.

Headwater: (1) the source and upper reaches of a stream; also the upper reaches of a reservoir. (2) the water upstream from a structure or point on a stream. (3) the small streams that come together to form a river. Also may be thought of as any and all parts of a river basin except the mainstream river and main tributaries.

Hydroelectric power water use: the use of water in the generation of electricity at plants where the turbine generators are driven by falling water.

Hydrologic cycle: the cyclic transfer of water vapor from the Earth's surface via evapotranspiration into the atmosphere, from the atmosphere via precipitation back to earth, and through runoff into streams, rivers, and lakes, and ultimately into the oceans.

I

Impermeable layer: a layer of solid material, such as rock or clay, which does not allow water to pass through.

Industrial water use: water used for industrial purposes in such industries as steel, chemical, paper, and petroleum refining. Nationally, water for industrial uses comes mainly (80%) from self-supplied sources, such as a local wells or withdrawal points in a river, but some water comes from public-supplied sources, such as the county/city water department.

Infiltration: flow of water from the land surface into the subsurface.

Injection well: refers to a well constructed for the purpose of injecting treated wastewater directly into the ground. Wastewater is generally forced (pumped) into the well for dispersal or storage into a designated aquifer. Injection wells are generally drilled into aquifers that don't deliver drinking water, unused aquifers, or below freshwater levels.

Irrigation: the controlled application of water for agricultural purposes through manmade systems to supply water requirements not satisfied by rainfall. Here's a quick look at some types of irrigation systems.

Irrigation water use: water application on lands to assist in the growing of crops and pastures or to maintain vegetative growth in recreational lands, such as parks and golf courses.

K

Kilogram: one thousand grams.

Kilowatt-hour (KWH): a power demand of 1,000 watts for one hour. Power company utility rates are typically expressed in cents per kilowatt-hour.

L

Leaching: the process by which soluble materials in the soil, such as salts, nutrients, pesticide chemicals or contaminants, are washed into a lower layer of soil or are dissolved and carried away by water.

Lentic waters: ponds or lakes (standing water).

Levee: a natural or manmade earthen barrier along the edge of a stream, lake, or river. Land alongside rivers can be protected from flooding by levees.

Livestock water use: water used for livestock watering, feed lots, dairy operations, fish farming, and other on-farm needs.

Lotic waters: flowing waters, as in streams and rivers.

M

Maximum contaminant level (MCL): the designation given by the U.S. Environmental Protection Agency (EPA) to water-quality standards promulgated under the Safe Drinking Water Act. The MCL is the greatest amount of a contaminant that can be present in drinking water without causing a risk to human health.

Milligram (mg): one-thousandth of a gram.

Milligrams per liter (mg/l): a unit of the concentration of a constituent in water or wastewater. It represents 0.001 gram of a constituent in 1 liter of water. It is approximately equal to one part per million (PPM).

Million gallons per day (mgd): a rate of flow of water equal to 133,680.56 cubic feet per day, or 1.5472 cubic feet per second, or 3.0689 acre-feet per day. A flow of one million gallons per day for one year equals 1,120 acre-feet (365 million gallons).

Mining water use: water use during quarrying rocks and extracting minerals from the land.

Municipal water system: a water system that has at least five service connections or which regularly serves 25 individuals for 60 days; also called a public water system

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N **Nephelometric turbidity unit (NTU)**: unit of measure for the turbidity of water. Essentially, a measure of the cloudiness of water as measured by a nephelometer. Turbidity is based on the amount of light that is reflected off particles in the water.

NGVD: National Geodetic Vertical Datum. (1) As corrected in 1929, a vertical control measure used as a reference for establishing varying elevations. (2) Elevation datum plane previously used by the Federal Emergency Management Agency (FEMA) for the determination of flood elevations. FEMA current uses the North American Vertical Datum Plane.

NGVD of 1929: National Geodetic Vertical Datum of 1929. A geodetic datum derived from a general adjustment of the first order level nets of the United States and Canada. It was formerly called "Sea Level Datum of 1929" or "mean sea level" in the USGS series of reports. Although the datum was derived from the average sea level over a period of many years at 26 tide stations along the Atlantic, Gulf of Mexico, and Pacific Coasts, it does not necessarily represent local mean sea level at any particular place.

Non-point source (NPS) pollution: pollution discharged over a wide land area, not from one specific location. These are forms of diffuse pollution caused by sediment, nutrients, organic and toxic substances originating from land-use activities, which are carried to lakes and streams by surface runoff. Non-point source pollution is contamination that occurs when rainwater, snowmelt, or irrigation washes off plowed fields, city streets, or suburban backyards. As this runoff moves across the land surface, it picks up soil particles and pollutants, such as nutrients and pesticides.

O **Organic matter**: plant and animal residues, or substances made by living organisms. All are based upon carbon compounds.

Osmosis: the movement of water molecules through a thin membrane. The osmosis process occurs in our bodies and is also one method of desalinating saline water.

Outfall: the place where a sewer, drain, or stream discharges; the outlet or structure through which reclaimed water or treated effluent is finally discharged to a receiving water body.

Oxygen demand: the need for molecular oxygen to meet the needs of biological and chemical processes in water. Even though very little oxygen will dissolve in water, it is extremely important in biological and chemical processes.

P **pH** a measure of the relative acidity or alkalinity of water. Water with a pH of 7 is neutral; lower pH levels indicate increasing acidity, while pH levels higher than 7 indicate increasingly basic solutions. View a diagram about pH.

Particle size: the diameter, in millimeters, of suspended sediment or bed material. Particle-size classifications are: (1) Clay—0.00024-0.004 millimeters (mm); (2) Silt—0.004-0.062 mm; (3) Sand—0.062-2.0 mm; (4) Gravel—2.0-64.0 mm, etc.

Parts per billion: the number of "parts" by weight of a substance per billion parts of water.

Used to measure extremely small concentrations.

Parts per million: the number of "parts" by weight of a substance per million parts of water. This unit is commonly used to represent pollutant concentrations.

Pathogen: a disease-producing agent; usually applied to a living organism. Generally, any viruses, bacteria, or fungi that cause disease.

Peak flow: the maximum instantaneous discharge of a stream or river at a given location. It usually occurs at or near the time of maximum stage.

Per capita use: the average amount of water used per person during a standard time period, generally per day.

Percolation: (1) The movement of water through the openings in rock or soil. (2) the entrance of a portion of the stream flow into the channel materials to contribute to ground water replenishment.

Permeability: the ability of a material to allow the passage of a liquid, such as water through rocks. Permeable materials, such as gravel and sand, allow water to move quickly through them, whereas unpermeable material, such as clay, doesn’t allow water to flow freely.

Point-source pollution: water pollution coming from a single point, such as a sewage-outflow pipe.

Polychlorinated biphenyls (PCBs): a group of synthetic, toxic industrial chemical compounds once used in making paint and electrical transformers, which are chemically inert and not biodegradable. PCBs were frequently found in industrial wastes, and subsequently found their way into surface and ground waters. As a result of their persistence, they tend to accumulate in the environment. In terms of streams and rivers, PCBs are drawn to sediment, to which they attach and can remain virtually indefinitely. Although virtually banned in 1979 with the passage of the Toxic Substances Control Act, they continue to appear in the flesh of fish and other animals.

Porosity: a measure of the water-bearing capacity of subsurface rock. With respect to water movement, it is not just the total magnitude of porosity that is important, but the size of the voids and the extent to which they are interconnected, as the pores in a formation may be open, or interconnected, or closed and isolated. For example, clay may have a very high porosity with respect to potential water content, but it constitutes a poor medium as an aquifer because the pores are usually so small.

Potable water: water of a quality suitable for drinking.

Precipitation: rain, snow, hail, sleet, dew, and frost.

Primary wastewater treatment: the first stage of the wastewater-treatment process where mechanical methods, such as filters and scrapers, are used to remove pollutants. Solid material in sewage also settles out in this process.

Prior appropriation doctrine: the system for allocating water to private individuals used in most Western states. The doctrine of Prior Appropriation was in common use throughout the arid West as early settlers and miners began to develop the land. The prior appropriation doctrine is based on the concept of "First in Time, First in Right." The first person to take a quantity of water and put it to beneficial use has a higher priority of right than a subsequent user. The rights can be lost through nonuse; they can also be sold or transferred apart from the land. Contrasts with riparian water rights.

Public supply: water withdrawn by public governments and agencies, such as a county water department, and by private companies that is then delivered to users. Public suppliers provide water for domestic, commercial, thermoelectric power, industrial and public water users. Most people's household water is delivered by a public water supplier. The systems have at least 15 service connections (such as households, businesses, or schools) or regularly serve at least 25 individuals daily for at least 60 days out of the year.

Public water use: water supplied from a public-water supply and used for such purposes as firefighting, street washing, and municipal parks and swimming pools.

R

Rating curve: a drawn curve showing the relation between gage height and discharge of a stream at a given gaging station.

Recharge: water added to an aquifer. For instance, rainfall that seeps into the ground.

Reclaimed wastewater: treated wastewater that can be used for beneficial purposes, such as irrigating certain plants.

Recycled water: water that is used more than one time before it passes back into the natural hydrologic system.

Reservoir: a pond, lake, or basin, either natural or artificial, for the storage, regulation, and control of water.

Return flow: (1) That part of a diverted flow that is not consumptively used and returned to its original source or another body of water. (2) (Irrigation) Drainage water from irrigated farmlands that re-enters the water system to be used further downstream.

Return flow (irrigation): irrigation water that is applied to an area and which is not consumed in evaporation or transpiration and returns to a surface stream or aquifer.

Reverse osmosis: (1) Desalination- the process of removing salts from water using a membrane. With reverse osmosis, the product water passes through a fine membrane that the salts are unable to pass through, while the salt waste (brine) is removed and disposed. This process differs from electro-dialysis, where the salts are extracted from the feed water by using a membrane with an electrical current to separate the ions. The positive ions go through one membrane, while the negative ions flow through a different membrane, leaving the end product of freshwater. (2) Water quality - an advanced method of water or wastewater treatment that relies on a semi-permeable membrane to separate waters from pollutants. An external force is used to reverse the normal osmotic process resulting in the solvent moving from a solution of higher concentration to one of lower concentration.

Riparian water rights: the rights of an owner whose land abuts water. They differ from state to state and often depend on whether the water is a river, lake, or ocean. The doctrine of riparian rights is an old one, having its origins in English common law. Specifically, persons who own land adjacent to a stream have the right to make reasonable use of the stream. Riparian users of a stream share the stream flow among themselves, and the concept of priority of use (Prior Appropriation Doctrine) is not applicable. Riparian rights cannot be sold or transferred for use on non-riparian land.

River: a natural stream of water of considerable volume, larger than a brook or creek.

Runoff: (1) That part of the precipitation, snow melt, or irrigation water that appears in uncontrolled surface streams, rivers, drains or sewers. Runoff may be classified according to speed of appearance after rainfall or melting snow as direct runoff or base runoff, and according to source as surface runoff, storm interflow, or ground-water runoff. (2) The total discharge described in (1), above, during a specified period of time. (3) Also defined as the depth to which a drainage area would be covered if all of the runoff for a given period of time were uniformly distributed over it.

S

Saline water: water that contains significant amounts of dissolved solids.

Here are our parameters for saline water:
Fresh water - Less than 1,000 parts per million (ppm)
Slightly saline water - From 1,000 ppm to 3,000 ppm
Moderately saline water - From 3,000 ppm to 10,000 ppm
Highly saline water - From 10,000 ppm to 35,000 ppm

Secondary wastewater treatment: treatment (following primary wastewater treatment) involving the biological process of reducing suspended, colloidal, and dissolved organic matter in effluent from primary treatment systems and which generally removes 80 to 95 percent of the Biochemical Oxygen Demand (BOD) and suspended matter. Secondary wastewater treatment may be accomplished by biological or chemical-physical methods. Activated sludge and trickling filters are two of the most common means of secondary treatment. It is accomplished by bringing together waste, bacteria, and oxygen in trickling filters or in the activated sludge process. This treatment removes floating and settleable solids and about 90 percent of the oxygen-demanding substances and suspended solids. Disinfection is the final stage of secondary treatment.

Sediment: usually applied to material in suspension in water or recently deposited from suspension. In the plural the word is applied to all kinds of deposits from the waters of streams, lakes, or seas.

Sedimentary rock: rock formed of sediment, and specifically: (1) sandstone and shale, formed of fragments of other rock transported from their sources and deposited in water; and (2) rocks formed by or from secretions of organisms, such as most limestone. Many sedimentary rocks show distinct layering, which is the result of different types of sediment being deposited in succession.

Sedimentation tanks: wastewater tanks in which floating wastes are skimmed off and settled solids are removed for disposal.

Self-supplied water: water withdrawn from a surface- or ground-water source by a user rather than being obtained from a public supply. An example would be homeowners getting their water from their own well.

Seepage: (1) The slow movement of water through small cracks, pores, Interstices, etc., of a material into or out of a body of surface or subsurface water. (2) The loss of water by infiltration into the soil from a canal, ditches, laterals, watercourse, reservoir, storage facilities, or other body of water, or from a field.

Septic tank: a tank used to detain domestic wastes to allow the settling of solids prior to distribution to a leach field for soil absorption. Septic tanks are used when a sewer line is not available to carry them to a treatment plant. A settling tank in which settled sludge is in immediate contact with sewage flowing through the tank, and wherein solids are decomposed by anaerobic bacteria action.

Settling pond: an open lagoon into which wastewater contaminated with solid pollutants is placed and allowed to stand. The solid pollutants suspended in the water sink to the bottom of the lagoon and the liquid is allowed to overflow out of the enclosure.

Sewage treatment plant: a facility designed to receive the wastewater from domestic sources and to remove materials that damage water quality and threaten public health and safety when discharged into receiving streams or bodies of water. The substances removed are classified into four basic areas: (1) greases and fats; (2) solids from human waste and other sources; (3) dissolved pollutants from human waste and decomposition products; and (4) dangerous microorganisms. Most facilities employ a combination of mechanical removal steps and bacterial decomposition to achieve the desired results. Chlorine is often added to discharges from the plants to reduce the danger of spreading disease by the release of pathogenic bacteria.

Sewer: a system of underground pipes that collect and deliver wastewater to treatment facilities or streams.

Sinkhole: a depression in the Earth's surface caused by dissolving of underlying limestone, salt, or gypsum. Drainage is provided through underground channels that may be enlarged by the collapse of a cavern roof.

Solute: a substance that is dissolved in another substance, thus forming a solution.

Solution: a mixture of a solvent and a solute. In some solutions, such as sugar water, the substances mix so thoroughly that the solute cannot be seen. But in other solutions, such as water mixed with dye, the solution is visibly changed.

Solvent: a substance that dissolves other substances, thus forming a solution. Water dissolves more substances than any other, and is known as the "universal solvent".

Specific conductance: a measure of the ability of water to conduct an electrical current as measured using a 1-cm cell and expressed in units of electrical conductance, i.e., Siemens per centimeter at 25 degrees Celsius. Specific conductance can be used for approximating the total dissolved solids content of water by testing its capacity to carry an electrical current. In water quality, specific conductance is used in ground water monitoring as an indication of the presence of ions of chemical substances that may have been released by a leaking landfill or other waste storage or disposal facility. A higher specific conductance in water drawn from down gradient wells when compared to up gradient wells indicates possible contamination from the facility.

Spray irrigation: a common irrigation method where water is shot from high-pressure sprayers onto crops. Because water is shot high into the air onto crops, some water is lost to evaporation.

Storm sewer: a sewer that carries only surface runoff, street wash, and snow melt from the land. In a separate sewer system, storm sewers are completely separate from those that carry domestic and commercial wastewater (sanitary sewers).

Stream: a general term for a body of flowing water; natural water course containing water at least part of the year. In hydrology, it is generally applied to the water flowing in a natural channel as distinct from a canal.

Stream flow: the water discharge that occurs in a natural channel. A more general term than runoff, stream flow may be applied to discharge whether or not it is affected by diversion or regulation.

Subsidence: a dropping of the land surface as a result of ground water being pumped. Cracks and fissures can appear in the land. Subsidence is virtually an irreversible process.

Surface tension: the attraction of molecules to each other on a liquid's surface. Thus, a barrier is created between the air and the liquid.

Surface water: water that is on the Earth's surface, such as in a stream, river, lake, or reservoir.

Suspended sediment: very fine soil particles that remain in suspension in water for a considerable period of time without contact with the bottom. Such material remains in suspension due to the upward components of turbulence and currents and/or by suspension.

Suspended-sediment concentration: the ratio of the mass of dry sediment in a water-sediment mixture to the mass of the water-sediment mixture. Typically expressed in milligrams of dry sediment per liter of water-sediment mixture.

Suspended-sediment discharge: the quantity of suspended sediment passing a point in a stream over a specified period of time. When expressed in tons per day, it is computed by multiplying water discharge (in cubic feet per second) by the suspended-sediment concentration (in milligrams per liter) and by the factor 0.0027.

Suspended solids: solids that are not in true solution and that can be removed by filtration. Such suspended solids usually contribute directly to turbidity. Defined in waste management, these are small particles of solid pollutants that resist separation by conventional methods.

T

Tertiary wastewater treatment: selected biological, physical, and chemical separation processes to remove organic and inorganic substances that resist conventional treatment practices; the additional treatment of effluent beyond that of primary and secondary treatment methods to obtain a very high quality of effluent. The complete wastewater treatment process typically involves a three-phase process: (1) First, in the primary wastewater treatment process, which incorporates physical aspects, untreated water is passed through a series of screens to remove solid wastes; (2) Second, in the secondary wastewater treatment process, typically involving biological and chemical processes, screened wastewater is then passed a series of holding and aeration tanks and ponds; and (3) Third, the tertiary wastewater treatment process consists of flocculation basins, clarifiers, filters, and chlorine basins or ozone or ultraviolet radiation processes.

Thermal pollution: a reduction in water quality caused by increasing its temperature, often due to disposal of waste heat from industrial or power generation processes. Thermally polluted water can harm the environment because plants and animals can have a hard time adapting to it.

Thermoelectric power water use: water used in the process of the generation of thermoelectric power. Power plants that burn coal and oil are examples of thermoelectric-power facilities.

Transmissibility (ground water): the capacity of a rock to transmit water under pressure. The coefficient of transmissibility is the rate of flow of water, at the prevailing water temperature, in gallons per day, through a vertical strip of the aquifer one foot wide, extending the full saturated height of the aquifer under a hydraulic gradient of 100-percent. A hydraulic gradient of 100-percent means a one foot drop in head in one foot of flow distance.

Transpiration: process by which water that is absorbed by plants, usually through the roots, is evaporated into the atmosphere from the plant surface, such as leaf pores. See evapotranspiration.

Tributary: a smaller river or stream that flows into a larger river or stream. Usually, a number of smaller tributaries merge to form a river.

Turbidity: the amount of solid particles that are suspended in water and that cause light rays shining through the water to scatter. Thus, turbidity makes the water cloudy or even opaque in extreme cases. Turbidity is measured in nephelometric turbidity units (NTU).

U

Unsaturated zone: the zone immediately below the land surface where the pores contain both water and air, but are not totally saturated with water. These zones differ from an aquifer, where the pores are saturated with water.

W **Wastewater**: water that has been used in homes, industries, and businesses that is not for reuse unless it is treated.

Wastewater-treatment return flow: water returned to the environment by wastewater-treatment facilities.

Water cycle: the circuit of water movement from the oceans to the atmosphere and to the Earth and return to the atmosphere through various stages or processes such as precipitation, interception, runoff, infiltration, percolation, storage, evaporation, and transportation.

Water quality: a term used to describe the chemical, physical, and biological characteristics of water, usually in respect to its suitability for a particular purpose.

Water table: the top of the water surface in the saturated part of an aquifer.

Water use: water that is used for a specific purpose, such as for domestic use, irrigation, or industrial processing. Water use pertains to human's interaction with and influence on the hydrologic cycle, and includes elements, such as water withdrawal from surface- and ground-water sources, water delivery to homes and businesses, consumptive use of water, water released from wastewater-treatment plants, water returned to the environment, and instream uses, such as using water to produce hydroelectric power.

Watershed: the land area that drains water to a particular stream, river, or lake. It is a land feature that can be identified by tracing a line along the highest elevations between two areas on a map, often a ridge. Large watersheds, like the Mississippi River basin contain thousands of smaller watersheds.

Watt hour (Wh): an electrical energy unit of measure equal to one watt of power supplied to, or taken from, an electrical circuit steadily for one hour.

Well (water): an artificial excavation put down by any method for the purposes of withdrawing water from the underground aquifers. A bored, drilled, or driven shaft, or a dug hole whose depth is greater than the largest surface dimension and whose purpose is to reach underground water supplies or oil, or to store or bury fluids below ground.

Withdrawal: water removed from a ground- or surface-water source for use.

X **Xeriscaping**: a method of landscaping that uses plants that are well adapted to the local area and are drought-resistant. Xeriscaping is becoming more popular as a way of saving water at home. More on xeriscaping: Colorado WaterWise Council

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Y **Yield**: mass per unit time per unit area

Some of this information is courtesy of the Nevada Division of Water Resources.

U.S. Department of the Interior | U.S. Geological Survey

http://ga.water.usgs.gov/edu/dictionary.html

Top

This is a partial list of a variety of scientific supply companies from which to purchase equipment for volunteer monitoring.

Acorn Naturalist
155 El Camino Real
Tustin, CA, 92780
800-422-8886
http://www.acornnaturalists.com/

Ben Meadows
PO Box 5277
Janesville, WI 53547-5277
800-241-6401 (7 AM – 7 PM)
http://www.benmeadows.com/

Carolina Biological Supply
2700 York Road
Burlington, NC 27215-3398
800-334-5551
http://www.carolina.com/

Cole Palmer Scientific Instruments
625 East Bunker Court
Vernon Hills, IL 60061
800-323-4340
http://www.coleparmer.com/

Dazor Manufacturing Company
2079 Congressional
St. Louis, MO
63146 800-345-9103
http://store.dazor.com/

Forestry Suppliers, Inc.
205 West Rankin Street
Jackson, MS 39284-8397
800-647-5368
http://www.forestry-suppliers.com/

Hanna Instruments
584 Park East Drive
Woonsocket, RI 02895
800-426-6287
http://www.hannainst.com/usa/index.cfm

Magnifier.com
2181 N Powerline Rd
Pompano Beach FL 33069
800-779-2801
http://www.magnifier.com/

Aquatic Research Instruments
PO Box 98 (620 Wellington Place)
Hope, ID 83836-0098
800-320-9482
http://www.aquaticresearch.com/default.htm

BioQuip Products
2321 Gladwick Street
Rancho Dominguez, CA 90220
310-667-8800
http://www.bioquip.com/default.asp

CheMetrics
4295 Catlett Road
Calverton, VA 20138
800.356.3072
http://www.chemetrics.com/home.html

Consolidated Plastics
4700 Prosper Dr.
Stow, OH 44224
800.362.1000
http://www.consolidatedplastics.com/

Fisher Scientific
711 Forbes Avenue
Pittsburgh, PA 15219-4785
800-766-7000
http://www.fishersci.com/wps/portal/HOME

HACH Company
PO Box 389
Loveland, CO 80539
800-227-4224
http://www.hach.com/

LaMotte
PO Box 329 (802 Washington Avenue)
Chestertown, MD 21620
800-344-3100
http://www.lamotte.com/

Millipore Corporation
290 Concord Road
Billerica, MA 01821
800-645-5476
http://www.millipore.com/

Nalgene Labware
75 Panorama Creek Drive
Rochester, NY 14625
800-625-4327
http://nalgenelab.nalgenunc.com/default.asp

Nichols Net and Twin Company
220 Highway 111
Granite City, IL 62040
800-878-NETS (6387)
http://www.nicholsnetandtwine.com/

The Lab Depot
PO. Box 1300
Dawsonville, GA 30534
800-733-2522
http://www.labdepotinc.com/

Thomas Scientific
1654 High Hill Road
Swedesboro, NJ 08085
800-345-2100
http://www.thomassci.com/

US Plastics Corporation
1390 Neubrecht Rd.
Lima, OH 45801-3196
800-809-4217
http://www.usplastic.com/

VMR Scientific Products
1310 Goshen Parkway West
Chester, PA 19380
800-932-5000
https://www.vwrsp.com/

Wards Natural Science
PO Box 92912
Rochester, NY 14692-9012
800-926-2600
http://wardsci.com/

Water Monitoring Equipment and Supply
PO Box 344
Seal Harbor, ME 04675
207-276-5746
http://www.watermonitoringequip.com/pages/home.html

Wilco Wildlife Supply Company
Gene Lasserre Blvd
Yulee, FL 32097
800-799-8301 86475
http://www.wildco.com/

YSI Incorporated
1700 Brannum Lane
Yellow Springs, OH 45387
800-765-4974
http://www.ysi.com/

Information about WV Save Our Streams bioassessment equipment

Appendix C: Determining Latitude and Longitude

There are many ways that monitoring groups identify and describe the location of sampling sites. Commonly, monitoring sites are described by stream name and geographic location, such as *Volunteer Creek behind the picnic area in Volunteer Park*. Often these descriptions are accompanied by an assigned station number (e.g. VC001, VC002). Some programs use river miles—the distance from the sampling station to the stream's mouth—as an additional identifier.

The most accurate way to identify a sampling location is by determining its latitude and longitude. Latitude and longitude are defined in degrees, minutes, and seconds. The symbols are:

° = degrees ' = minutes " = seconds

Any volunteer program that wishes to have its data used by state, local, or federal agencies, or that plans to enter its data into a Geographic Information Systems (GIS) either now or in the future, must provide latitudes and longitudes for its sampling locations. USEPA's STORET water quality database, for example, requires latitude/longitude information before any data can be entered.

Section 4.1 in Chapter 4, *Macroinvertebrates and Habitat*, briefly describes using a global positioning system (GPS) to determine latitude and longitude. This hand-held tool is used in the field and receives signals from orbiting satellites to calculate lat/long coordinates.

New tools are continually being developed to help locate sites. For example, USEPA's *Surf Your Watershed* web page ties in with the U.S. Geological Survey's Names Information System to provide latitude and longitude information for locations throughout the U.S. These locations include bridges, schools, rivers, parks, and more. Visit this feature of *Surf Your Watershed* at *www.epa.gov/surf/surf_search.html* for more information.

Section 3.1 in Chapter 3, *Watershed Survey Methods*, discusses the various types of maps used by monitoring programs. In addition to the commonly used U.S. Geological Survey (USGS) 7.5 and 15 minute topographic map series, volunteers often use other types of topographic maps including countywide topographic maps available from state Geological Surveys.

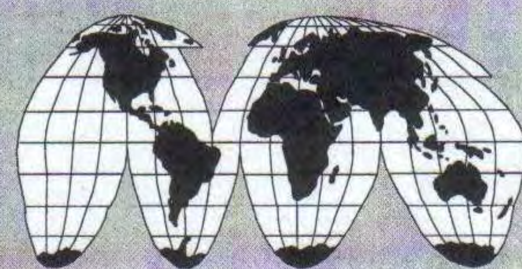
A worksheet is presented on the following pages which can be used for calculating the latitude and longitude of a site using a topographic map. This protocol was adapted from USEPA's 1992 Streamwalk Manual (USEPA Seattle, WA) with revisions by Michael Goodrich of GeoQuest Publications, P.O. Box 1665, Lake Oswego, OR 97034.

Latitude and Longitude

Latitude and longitude are defined and measured in degrees (°), minutes ('), and seconds ("). There are 60 seconds in a minute and 60 minutes in a degree of latitude and longitude.

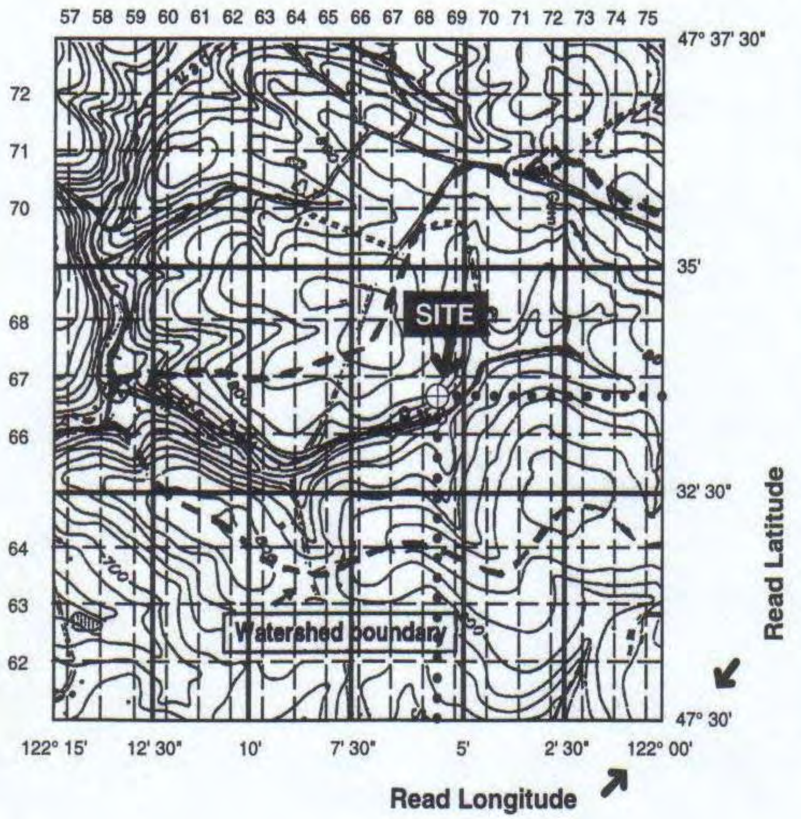
Latitude (lat) is the angular distance of a particular location north or south from the equator. Latitude lines are called *parallels*.

Longitude (long) is the angular distance of a particular location east or west of some prime meridian (usually Greenwich, England). Longitude lines are called *meridians*.



Worksheet for Calculating Latitude and Longitude

(With an example using a 7.5 x 15 Minute Series USGS topographic map)



IMPORTANT NOTE!

When working with longitude and latitude you must remember that there are 60 seconds in a minute and 60 minutes in a degree. This system works like time. When you add or subtract, 59 is the highest number you can enter in the seconds or minutes "column."

For example, if you add 20 seconds to your already established line of latitude of 38° 58' 49", your new latitude would be 38° 59' 09".

Step 8 in the worksheet requires you to add or subtract latitude and longitude using this system.

Mathematical determination of LONGITUDE:

	Your Site	Example
1. Find and record the numbers of two lines of longitude that exist on either side of your site. Identify which of these lines is closest to your monitoring site.		
a. Record the closest line of longitude east of your site.	___° ___' ___"	122° 05' 00"
b. Record the closest line of longitude west of your site.	___° ___' ___"	122° 07' 30"

(In this example, the line east of the site is the closest)

	Your Site	Example
2. Subtract #1b from #1a. Record that number in minutes and seconds.	___' ___"	2' 30"
3. Convert the answer to #2 to seconds by multiplying the minutes by 60 and adding the remaining seconds.	___"	150"
4. Measure and record the distance in millimeters (mm) between the two lines of longitude that were identified in #1.	___ mm	16 mm
5. Measure and record the distance from the closest line of longitude to your site. *(In the example, 122° 05' 00" is the closest line of longitude to the site.)*	___ mm	4 mm
6. Convert this distance into seconds by multiplying the distance (#5) by the number of seconds (#3) and dividing that sum by the distance between the two longitude lines (#4). $\frac{\#5 \times \#3}{\#4}$		$\frac{4 \text{ mm} \times 150''}{16 \text{ mm}}$
Round to the answer to the nearest whole number. *(The distance units (mm) cancel each other out and the answer is in seconds.)*	___"	38"
7. If your answer to #6 is less than 60, record it as your answer to #7. If you answer is 60 or more, convert your response into minutes and seconds by dividing by 60. Do not use a calculator for this section because you will need to use the remainder, which would not be displayed on a calculator. Record to the nearest whole number. The whole digits are entered in the minutes column. The remainder is entered in the seconds column. *(In the example, the answer to #6 is less than 60. Therefore it is placed directly in the worksheet)*	___' ___"	0' 38"
8. The site's longitude is calculated using one of the following:		122° 05' 00"
a. If the line of longitude you identified in #1 as closest to the site is east of the site, add #7 to that longitude.	___° ___' ___"	+ 0' 38" = 122° 05' 38"
b. If the line of longitude you identified in #1 as closest to the site is west of the site, subtract #7 from that longitude.	___° ___' ___"	
(In the example, the closest line of longitude is east of the site. Therefore #7 and that longitude were added together.)		

Mathematical determination of LATITUDE:

	Your Site	***Example***
1. Find and record the numbers of two lines of latitude that exist on either side of your site. Identify which of these lines is closest to your monitoring site.		
a. Record the closest line of latitude north of your site.	____° ____' ____"	47° 35' 00"
b. Record the closest line of latitude south of your site.	____° ____' ____"	47° 32' 30"
(In this example, the line south of the site is the closest)		
2. Subtract #1b from #1a. Record that number in minutes and seconds.	____' ____"	2' 30"
3. Convert the answer to #2 to seconds by multiplying the minutes by 60 and adding the remaining seconds.	____"	150"
4. Measure and record the distance in millimeters (mm) between the two lines of latitude that were identified in #1.	____ mm	31 mm
5. Measure and record the distance from the closest line of latitude to your site. *(In the example, 47° 32' 30" is the closest line of latitude to the site.)*	____ mm	14 mm
6. Convert this distance into seconds by multiplying the distance (#5) by the number of seconds (#3) and dividing that sum by the distance between the two latitude lines (#4). $\frac{\#5 \text{ X } \#3}{\#4}$		$\frac{14 \text{ mm X } 150''}{31 \text{ mm}}$
Round to the answer to the nearest whole number. *(The distance units (mm) cancel each other out and the answer is in seconds.)*	____"	68"
7. If your answer to #6 is less than 60, record it as your answer to #7. If you answer is 60 or more, convert your response into minutes and seconds by dividing by 60. Do not use a calculator for this section because you will need to use the remainder, which would not be displayed on a calculator. Record to the nearest whole number. The whole digits are entered in the minutes column. The remainder is entered in the seconds column. *(In the example, the answer to #6 is 68". Dividing by 60 equals 1 with a remainder of 8)*	____' ____"	68" ÷ 60 = 1' 08"

	Your Site	Example
8. The site's latitude is calculated using one of the following:		
a. If the line of latitude you identified in #1 as closest to the site is north of the site, subtract #7 from that latitude.	___° ___' ___"	47° 32' 30"
		+ 01' 08"
b. If the line of latitude you identified in #1 as closest to the site is south of the site, add #7 to that latitude.	___° ___' ___"	47° 33' 38"

(In the example, the closest line of latitude is south of the site. Therefore #7 and that latitude were added together.)

The purpose of this section is to assist volunteer monitors with the identification of many aquatic invertebrates found in our rivers, streams and wetlands. General information is included about the distinguishing features of the aquatic stage that aid in identification. Also included are the organisms, habit, feeding group, tolerance rating, size range, and habitat preferences. For images or more information, simply click-on the links provided; [Adult] images are provided for many families. Additionally, an explanation of functional feeding groups, a glossary and a reference section is included.

Note: Many of the words that may be unfamiliar are defined using on-line dictionaries. The credibility of the web pages included here has not been thoroughly investigated. Apply the normal standards of Internet research to your investigation of each website in order to determine its veracity. Most of the illustrations are courtesy of the Cacapon Institute and artist Jennifer Gillies (JG). **Not all families are included here**.

Habitat preferences

- (**F**) Fast-moving waters with rocky substrate (i.e. riffles and runs of streams and rivers)
- (**S**) Slow-moving or still waters with soft substrate and vegetation (i.e. pools and backwater areas of streams and rivers; wetlands and springs)

Size classes (mm)

> 50	50 - 30	29 - 15	14 - 5	< 5
(VL)	(L)	(M)	(S)	(VS)

General	1.	Kingdom
	2.	Phylum
	3.	Class
	4.	Order
	5.	Family
	6.	Genus
Specific	7.	Species

Classification: Plants and animals are classified according to a hierarchal system that arranges the organisms into groups based upon their similarities. These groups are arranged from general to very specific. The science of classification is known as taxonomy. The table provides the basic taxonomic groups. In certain situations these major groups are sub-divided (i.e. sub-phylum, sub-class, sub-order etc.). This occurs when a group of organism is different enough to be noted but not different enough to place them in a separate classification. Genus and species names are included together (e.g. Homo sapiens).

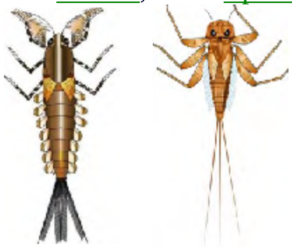
Stress (pollution) tolerance is the organism's ability to withstand a certain amount of anthropogenic influences. The general stress categories of low, moderate and high are described below. The index range for each category is based upon a (0-10) scale, which is based mostly on the invertebrate's ability to withstand varying levels of dissolved oxygen and other chemical and physical disturbances. For example, invertebrates with a low tolerance need adequate dissolved oxygen and chemical and physical stability, while those with a high tolerance can survive for a period of time when dissolved oxygen levels are less than adequate or other disturbances may be present.

Low (**L**): Occur in environments with little or no disturbance to moderately disturbed conditions. Certain kinds may occur in slightly elevated numbers under certain conditions (0-3). Moderate (**M**): Occur in environments from moderately to highly disturbed conditions but can also occur in less disturbed conditions. The over abundance or dominance of moderate tolerant organisms are often good indications of disturbance (4-6). High (**H**): Occur most often under disturbed conditions. In these environments, only one or two groups may dominate the entire community. They are also found in good conditions, but usually in low numbers (7-10). In some cases tolerance values are undetermined (**U**).

Contents

Arthropod Groups

Class Insecta; order Ephemeroptera (**Mayflies**) JG



Wing pads may be present on the thorax; three pairs of segmented legs attach to the thorax; one claw occurs on the end of the segmented legs; gills occur on the abdominal segments and are attached mainly to the sides of the abdomen, but sometimes extend over the top and bottom of the abdomen; gills consist of either flat plates or filaments; three long thin caudal (tails filaments) usually occur at the end of the abdomen, but there may only be two in some kinds. (Hemimetabolism)

Swimming mayflies

1. Ameletidae (Comb-mouth minnow mayfly): Comb of stiff spines on the mouthparts; gills have a dark, sclerotized (hard) band along the outside edge; antennae are shorter than twice the width of the head; usually have dark bands on the tail and alternating dark and light on the abdomen. Swimmer, clinger; Collector, gatherer; (**L**); VS-M (**F**) [Adult]
2. Baetidae (Small minnow mayfly): Antennae two times longer than the width of the head; gills variable in shape and attached at abdominal segments one through seven; two or three caudal (tail) filaments. Swimmer; Collector, gatherer; (**M**); VS-M (**F**/**S**) [Adult]
3. Isonychiidae (Brush-legged mayfly): Forelegs have a double row of setae (hairs); gills oval shaped and present on abdominal segments one through seven; long hairs on the margins of the caudal filaments. Swimmer, crawler; Collector, gatherer; (**L**); S-M (**F**) [Adult]
4. Siphlonuridae (Primitive minnow mayfly): Antennae less than two-time the width of the head; gills usually oval shaped and present on abdominal segments one through seven; long setae on the caudal filaments. Swimmer; Collector, gatherer; (**L**); S-M (**F**/**S**)

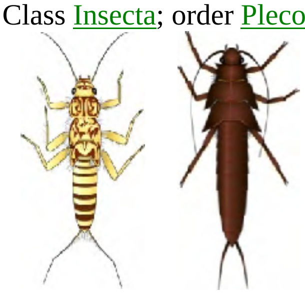
Clinging/crawling mayflies

5. Caenidae (Square-gilled mayfly): Gills on the first abdominal segment very small; gills on the second segment operculate (plate-like) covering much of the remaining gills. Clinger, crawler, burrower; Collector, gatherer; (**M**); VS-S (**F**/**S**)
6. Ephemerellidae (Spiny-crawler mayfly): Gills present of the first abdominal segment but absent from the second; gills usually present on the remaining segments; two or three caudal filaments. Clinger, crawler; Collector, gatherer; (**L**); VS-M (**F**) [Adult]
7. Heptageniidae (Flatheaded mayfly): Body, head and legs are flattened (femora); gills present on abdominal segments one through seven; usually three caudal filaments, but some may have two. Clinger; Collector, scraper; (**L**); S-M (**F**) [Adult]
8. Leptophlebiidae (Prong-gilled mayfly): Gills on abdominal segments two through seven forked and variable in shape; gills on the first segment finger-like; short hairs usually cover the caudal filaments. Clinger, crawler; Collector, gatherer; (**L**); VS-M (**F**) [Adult]
9. Tricorythidae (Stout-crawler mayfly): Gills absent from abdominal segment one; gills on segment two are (operculate), plate-like triangular or oval shaped and conceals gills on segments three through six. This family is similar in appearance to Caenidae. Clinger, crawler, burrower; Collector, gatherer; (**M**); VS-M (**F**/**S**) [Adult]

Burrowing mayflies

10. Beatiscidae (Armored mayfly): Top portion of the thorax is fused and coves most of the abdomen concealing the gills; caudal filaments are short and fringed with hairs. Burrower, crawler; Collector, gatherer; (**M**); VS-M (**F**)
11. Ephemeridae (Burrowing mayfly): Has upturned mandibular tusks; head and front legs slightly widened and are used for burrowing; gills on the upper abdominal segments are small and the remaining gills are forked with fringed margins (feathered) and held over the top and sides of the abdomen. Burrower; Collector, gatherer; (**M**); M-L (**S**/**F**) [Adult]
12. Potamanthidae (Hackle-gilled mayfly): Mandibular tusks present; front legs slender, not modified for burrowing; gills on segment one small, gills on remaining abdominal segments are feathery. Burrower; Collector, gatherer; (**M**); S-M (**S**/**F**)

Class Insecta; order Plecoptera (**Stoneflies**) JG



Long thin antenna project in front of the head; wing pads usually present on the thorax but may only be visible in older larvae; three pairs of segmented legs attach to the thorax; two claws are located at the end of the segmented legs; gills occur on the thorax region, usually on the legs or bottom of the thorax, or there may be no visible gills (usually there are none or very few gills on the abdomen); gills are either single or branched filaments; two long thin tails project from the rear of the abdomen. Stoneflies have very low tolerance to many insults; however, several families are tolerant of slightly acidic conditions. (Hemimetabolism)

Winter stoneflies

1. Capniidae (Small winter stonefly): Slender elongated body; front of thorax slightly wider than the abdomen; wing pads not divergent from the midline; abdominal segments separated by a membranous fold. Clinger, crawler; Shredder; (**L**); S-M (**F**) [Adult]
2. Leuctridae (Rolled-wing stonefly): Slender elongated body; front of thorax slightly wider than the abdomen; wing pads not divergent from the midline; abdominal segments not separated by a membranous fold. Very similar characteristics to Capniidae. Clinger, crawler; Shredder; (**L**); S-M (**F**) [Adult]
3. Taeniopterygidae (Large winter stonefly): Stout bodies with pronotum much wider than the abdomen; wing pads greatly divergent from the midline. Clinger, crawler; Shredder (**L**); S-M (**F**) [Adult]

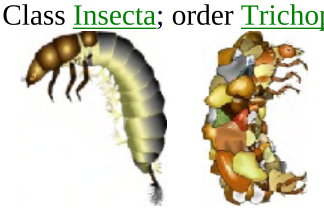
Perlid stoneflies

4. Chloroperlidae (Green stonefly) Body elongated, front of the thorax slightly wider than the abdomen; wing pads not divergent from the midline; tails (cerci) shorter than the abdomen. Will sometimes have patterns similar to Perlodidae. Clinger, crawler; Shedder, predator; (**L**); M (**F**) [Adult]
5. Peltoperlidae (Roach-like stonefly): Small stout body; rear divergent wing pads; thoracic segments are oval or triangular shaped and cover much of the upper body; some have fine gills on the front legs. Clinger, crawler; Shredder; (**L**); S-M (**F**) [Adult]
6. Perlidae (Common stonefly): Usually a large strikingly patterned and often having a golden color; finely branched gills present on all thoracic segments; wing pads diverge slightly from the midline. Sometimes called the golden stonefly. Clinger, crawler; Predator; (**L**); M-L (**F**) [Adult]
7. Perlodidae (Patterned stonefly): Strikingly patterned and colored similar in appearance to Perlidae; hind wing pads divergent; no gills on the thoracic segments. Clinger, crawler; Shredder; (**L**); M-L (**F**) [Adult]

Other stoneflies

8. Nemouridae (Little brown stonefly): Very small, often hairy appearance; wing pads diverge greatly from the midline; hind legs as long as the abdomen; gills often present between the head and thorax. Clinger, crawler; Shredder; (**L**); S-M (**F**/**S**) [Adult]
9. Pteronarcyidae (Giant stonefly): Very large and usually dark brown in color; finely branched gills on all thoracic segments plus the first two abdominal segments. Clinger, crawler; Shredder; (**L**); M-VL (**F**) [Adult]

Class Insecta; order Trichoptera (**Caddisflies**) JG



Head has a thick hardened skin; antennae are very short, usually not visible; no wing pads occur on the thorax; top of the first thorax always has a hardened plate and in several families the second and third section of the thorax have a hardened plate; three pairs of segmented legs attach to the thorax; abdomen has a thin soft skin; single or branched gills on the abdomen in many families, but some have no visible gills; pair of prolegs with one claw on each, is situated at the end of the abdomen; many families construct various kinds of retreats, which consists of a wide variety of materials collected from the streambed. (Holometabolism)

Netspinning caddisflies

1. Hydropsychidae (Common netspinner): Top of all thoracic segments hardened; most abdominal segments have tufts of finely branched gills; anal prolegs terminate into a brush of hairs. Do not make cases but instead creates a retreat (net) made of a variety of materials held together by fine strands of silk. Clinger, crawler; Collector, filterer; (**M**); M-L (**F**/**S**) [Adult]
2. Philopotamidae (Finger-net caddisfly): Labrum (structure between the mouthparts) is t-shaped and membranous; head capsule large usually orange in color; only first thoracic segment is hardened; abdominal gills usually absent. Builds a long tube or finger-like net. Clinger, crawler; Collector, filterer; (**L**); M (**F**) [Adult]
3. Polycentropodidae (Tube-net caddisfly): Labrum is rounded and hardened; only first thoracic segment is hardened; no plates or gills on the abdominal segments. Does not build cases but instead constructs a net that is often in the shape of a long tube. Clinger, crawler; Collector, filterer, predator; (**M**); S-L (**F**/**S**) [Adult]
4. Psychomiidae (Trumpet-net caddisfly): Bottom of thorax is hardened with black edges; middle thoracic segment is swollen and usually larger than the others; has an enlarged hatchet shaped leg segment on the upper legs. Clinger, crawler; Collector; (**M**); M (**F**)

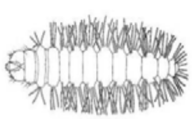
Free-living caddisfly

5. Rhyacophilidae (Free-living caddisfly): First thoracic segment is hardened; abdominal gills variable; hardened plate on top of abdominal segment nine; distinctive anal prolegs with large claws; is often green in color; referred to by trout fisherpersons as the green sedge. This family does not build a case or net, but often uses silk strands to attach itself to substrates. Clinger, crawler; Predator; (**L**); M-L (**F**) [Adult]

Case-building caddisflies

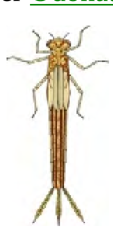
6. Brachycentridae (Humpless-case caddisfly): Antennae close to the margins of the head capsule; first two thoracic segments with hardened plates; no humps on abdominal segments; gills simple or lacking. Case is elongated and made of strips of materials, resembles a log cabin. Clinger; Collector, gatherer, shredder; (**L**); M (**F**) [Adult]
7. Glossosomatidae (Saddle-case caddisfly): First thoracic segment is hardened; hardened plate on top of abdominal segments nine. Case resembles a tortoise shell or saddle. Clinger; Scraper, shredder; (**L**); VS-M (**F**) [Adult]
8. Goeridae (Goerid-case caddisfly): Hardened head, yellow to reddish brown colored; large horn-like structure on the thorax. Case is constructed with sand-grains and small pebbles, usually is slightly curved. Clinger; Scraper (**L**) (VS-M) (**F**/**S**) [Adult]
9. Helicopsychidae (Snail-case caddisfly): Body is curled; all three thoracic segments are hardened; stout hairs at the end of the third thoracic segment; gills present on anterior abdominal segments. Case resembles a snail shell. Clinger; Scraper; (**L**); VS-S (**F**)
10. Hydroptilidae (Purse-case caddisfly): All three of their thoracic segments have sclerotized dorsal plates; no gills on the abdomen; most commonly build cases with sand, algae, silk or detritus, but the shapes vary considerably. Clinger, crawler; Scraper; (**M**); VS-S (**F**/**S**) [Adult]
11. Lepidostomatidae (Lepidostomid-case caddisfly): Antennae located close to the eyes; lateral hump on abdominal segment one; first two thoracic segments hardened; gills simple or lacking; hardened plate on top of abdominal segment nine. Case is usually four-sided built with square pieces of barks and leaves. Clinger, crawler; Shredder; (**L**); S-M (**F**) [Adult]
12. Leptoceridae (Longhorn-case caddisfly): Antennae prominent; first two thoracic segments hardened; hind legs are usually longer than the front legs; abdominal gills variable. Cases are built from a variety of materials and vary considerably; the most common is a stone/sand case resembling a long tube. Clinger, crawler; Collector, predator; (**L**); S-M (**F**/**S**) [Adult]
13. Limnephilidae (Northern-case caddisfly): Antennae between the eyes and the mouth; first two thoracic segments hardened; dorsal and lateral humps on first abdominal segment; hardened plate on the top of abdominal segment nine; abdominal gills variable. The cases are built from many kinds of bottom materials and exhibit a wide variety of shapes and sizes. Clinger, crawler; Shredder; (**L**); S-L (**F**) [Adult]
14. Molannidae (Hooded-case caddisfly): Two-thirds of the top of the thorax is hardened; tarsal claws on the hind legs smaller than the rest and are covered with fine hairs; gills along the abdomen are simple or branched; a hardened plate sits atop abdominal segment nine. The cases are constructed mostly with sand and are shaped like a flattened tube with a hood that extends over the opening of the case. Clinger, crawler; Collector, shredder; (**M**); S-M (**S**) [Adult]
15. Phryganeidae (Giant-case caddisfly): Head and portions of the thorax marked with prominent stripes; front part of the thorax hardened; dorsal and lateral humps on abdominal segment one; hardened plate on top of abdominal segment nine. Builds elongated cases out of plant fragments. Clinger, crawler; Collector, predator; (**M**); M-L (**S**) [Adult]
16. Uenoidae (Uenoid-case caddisfly): The first two thoracic segments are hardened and there are some small plates present on the third; abdominal segment one has a hump, and the anterior margin of their mesonotum is notched on either side of the midline; cases are variable but usually always constructed with small stones and sand. Clinger, crawler; Scraper; (**L**); S-M (**F**) [Adult]

Class Insecta; order Lepidoptera (**Aquatic moth**)



Head hardened; a few families have elongated lateral gills; three pairs of segmented legs attach to the thorax; abdomen with prolegs that end in tiny hooks. They often look very similar to terrestrial caterpillars; closely related to Trichoptera. (Holometabolism) Crawler, burrower; Shredder; (**M**); 3-35 (**S**/**F**) [Adult]

Class Insecta; order Odonata; sub-orders Anisoptera (**Dragonflies**) and Zygoptera (**Damselflies**) JG



Dragonflies: Lower lip (labium) is long and elbowed to fold back against the head when not feeding, thus concealing other mouthparts; wing pads are present on the thorax; three pairs of segmented legs attach to the thorax; no gills on the sides of the abdomen; Dragonflies have three pointed structures may occur at the end of the abdomen forming a pyramid shaped opening; bodies are long and stout or somewhat oval. Damselflies have three flat gills at the end of the abdomen forming a tail-like structure and their bodies are long and slender. (Hemimetabolism)

Dragonflies

1. Aeshnidae (Darner dragonfly): Prementum and papal lobes are flattened; six or seven antennal segments present all of a similar size. Clinger, crawler; Predator; (**L**); M-VL (**S**) [Adult]

2. Cordulegastridae (Spiketail dragonfly): Often appear hairy; prementum large, covering much of the underside of the head, usually triangular shaped. Clinger, crawler; Predator; (**L**); M-L (**S**/**F**) [Adult]
3. Gomphidae (Clubtail dragonfly): Body shape variable from long cylindrical to oval and flattened; prementum flattened; third antennal segment large and different from the rest. Clinger, crawler; Predator; (**M**); M-L (**S**/**F**) [Adult]
4. Libellulidae (Skimmer dragonfly): Antennal segments similar in shape and size; prementum and palpal lobes spoon shaped or small and rounded. Crawler, burrower; Predator; (**H**); M-L (**S**) [Adult]

Damselflies

5. Calopterygidae (Broad-wing damselfly): Lower portion of labium is diamond shaped; first antennal segment longer than all the others together; middle gills shorter than the lateral two; no visible veins on the gills. Clinger, crawler; Predator; (**M**); M-L (**S**/**F**) [Adult]
6. Coenagrionidae (Narrow-wing damselfly): Slender but slightly more stout bodied than most damselflies; labium triangular shaped; antennal segments same length; gills same length, veins radiate diagonally. Clinger, crawler; Predator; (**H**); M-L (**S**) [Adult]
7. Lestidae (Spread-wing damselfly): Long and slender bodied; labium stalked and spoon shaped; all gills similar in shape with perpendicular veins. Clinger, crawler; Predator; (**H**); M-L (**S**) [Adult]

Class Insecta; order Coleoptera (**Beetles**) JG

Head has thick hardened skin; thorax and abdomen of most adult families have moderately hardened skin, several larvae have a soft-skinned abdomen; no wing pads on the thorax in most larvae, but wing pads are usually visible on adults; three pairs of segmented legs attach to the thorax; no structures or projections extent from the sides of the abdomen in most adult families, but some larval stages have flat plates or filaments; no prolegs or long tapering filaments at the end of the abdomen. Beetles are one of the most diverse and abundant of the insect groups but are not as common in aquatic environments. (Holometabolism)

1. Chrysomelidae (Reed beetle): The body is soft; three-pairs of segmented legs attached to the thorax and two hooks on the lower end of the abdomen. Crawler; collector/gatherer; (**H**); S-M (**S**) [Adult]
2. Dryopidae (Long-toed beetle): Adults are hard bodied with very short comb-like antennae. The family is similar in appearance to the adult riffle beetle. The larva of this beetle is not aquatic but may be found in the splash zone. Clinger, crawler; Shredder; (**M**); VS-S (**F**/**S**)
3. Dytiscidae (Predacious diving beetle): Legs have five-segments and two-claws on the end; abdomen terminates into a pair of filaments. [Adult] slender antennae; hind coxae extends posterior dividing the first abdominal segment into two sections. Swimmer, crawler; Predator; (**M**); VS-VL (**S**)
4. Elmidae (Riffle beetle): Legs with four segments and a single claw; nine abdominal segments some with a cavity that protect the hind gills. [Adult] hard bodied, slender sometimes clubbed antennae; the forewings have numerous rows of indentations; legs long compared to body. Clinger, crawler; Scraper, shredder; (**M**); VS-S (**F**)
5. Gyrinidae (Whirligig beetle): Two claws of each leg, legs with five segments; ten abdominal segments with pairs of lateral filaments. [Adult] compound eyes, which appear divided into pairs; antennae clubbed; mid and hind legs paddle-like. Swimmer, crawler; Predator; (**M**); S-L (**S**/**F**)
6. Haliplidae (Crawling water beetle): Legs with five segments and a single claw; abdomen terminates into long filaments; some have many long slender filaments along the entire length of the body. [Adult] antennae long and slender; forewings have many indentations; legs lined with small hairs for swimming. Swimmer, crawler; Shredder; (**H**); S-M (**S**)
7. Hydrophilidae (Water scavenger beetle): Large mandibles; legs with four segments and a single claw; end of the abdomen usually blunt. [Adult] antennae clubbed with cup-like segments at the base; hind coxae (joined base) do not extend or divide the abdomen. Swimmer, crawler; Predator; (**H**); VS-VL (**S**)
8. Ptilodactylidae (Toe-wing beetle): Legs with four segments and a single claw; abdomen has ventral gills. Very similar in appearance to the riffle beetle larva. The adult is not aquatic but is sometimes found near the stream. Clinger, crawler; Scraper, shredder; (**M**); VS-S (**F**/**S**) [Adult]
9. Psephenidae (Water penny): Body flattened with thoracic and abdominal segments expanded so that the legs and head are obscured from above; legs terminate into a single claw. The adult is semi-aquatic, sometimes encountered near the stream. Clinger, crawler; Scraper; (**L**); VS-M (**F**)

Class Insecta; order Hemiptera (**True bugs**)

The most distinguishing characteristic of the order is the mouthparts that are modified into an elongated, sucking beak. Most adults have hemelytra, which are modified leathery forewings. Some adults and all larvae lack wings; both most mature larvae posses wing pads. Both adults and larvae have three-pairs of segmented legs with two tarsal claws at the end of each leg. Many families are able to also utilize atmospheric oxygen. This order is generally not used for the biological assessment of flowing waters, due to their ability to use atmospheric oxygen. Several families are described. (Hemimetabolism)

Surface

1. Gerridae/Veliidae (Water striders): Variable body shape; cylindrical beak; rear legs extend well beyond the tip of the abdomen. Swimmer; Predator; (**H**); VS-M (**S**)
2. Corixidae (Water boatman): Broad triangular beak; forelegs are scoop-like and fringed with hairs; antennae are short and concealed beneath the eyes. Swimmer; Predator; (**H**); VS-M (**S**)
3. Notonectidae (Backswimmer): Body cylindrical; antennae are short and concealed beneath the eyes; hind legs are oar-like; hind tarsal claws inconspicuous. Swimmer; Predator; (**H**); M (**S**)

Sub-surface

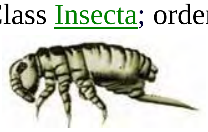
4. Belostomatidae (Giant water bug): Large oval body; antennae are short and concealed beneath the eyes; raptorial front legs. Swimmer/clinger; Predator; (**H**); M-VL (**S**)
5. Hydrometridae (Water measurer): Body slender and elongated; antennae longer than the head; head usually longer than the thorax; legs are long and slender, each with two claws. Clinger, crawler; Predator; (**H**); S-M (**S**)
6. Nepidae (Water scorpion): Body long and cylindrical; antennae are small and concealed beneath the eyes; forelegs are raptorial other legs are long and slender; abdomen terminates into a long breathing appendage. Clinger, crawler; Predator; (**H**); M-L (**S**)

Class Insecta; order Megaloptera (**Hellgrammites** and **Alderflies**) JG

Head and thorax has thick hardened skin, while the abdomen has thin soft skin; prominent chewing mouthparts project in front of the head; no wing pads on the thorax; three pairs of segmented legs attach to the thorax; seven or eight pairs of stout tapering filaments extend from the abdomen; end of the abdomen has either a pair of prolegs with two claws on each proleg, or a single long tapering filament with no prolegs. (Holometabolism)

1. Corydalidae (Hellgrammite/Fishfly): Elongate dorsally flattened body; large jaws on the head, projecting forward; first eight abdominal segments and segment ten with paired lateral filaments; abdomen terminates in fleshy appendages bearing hooks. Clinger, crawler; Predator; (**L**); M-VL (**F**) [Adult]
2. Sialidae (Alderfly): Elongate dorsally flattened body; large jaws on the head, projecting forward; first seven abdominal segments and segment ten with paired lateral filaments; abdomen terminates into a single long hairy filament. Crawler, burrower; Predator; (**M**); S-L (**S**/**F**) [Adult]

Class Insecta; order Collembola (**Springtails**)



Abdomen consisting of six segments, the first having collophores; abdomen terminates into a forked appendage. Has a habit of jumping on the surface of the water. Swimmer, crawler; Collector, gatherer; (**M**); VS (**S**) (Holometabolism)

Class Insecta; order Diptera (**True flies**) JG

Head may be a capsule-like structure with thick hard skin; head may be partially reduced so that it appears to be part of the thorax, or it may be greatly reduced with only the mouthparts visible; no wing pads occur on the thorax; false-legs (pseudo-legs) may extend from various sections of the thorax and abdomen in some families; no segmented legs in the larval forms; thorax and abdomen composed of entirely soft skin, but some families have hardened plates scattered on various body features. Note: the larval stages do not have segmented legs (Holometabolism)

Midges/mosquitoes

1. Blephariceridae (Net-wing midge): Head fused with thorax and first abdominal segment; six abdominal segments with deep constrictions between segments; gill tufts present ventrally. Clinger; Scraper; (**L**); VS-S (**F**) [Adult]
2. Ceratopogonidae (Biting midge): Variable characteristics occur in this family, often similar in appearance to Chironomidae; usually a distinct head is visible with small mandibles. Crawler, burrower; Predator; (**H**); VS-M (**F**/**S**)

3. Chironomidae (Non-biting midge); Hardened clearly visible head; long worm-like body; two pairs of prolegs with terminal hooks. Some families may be red in color due to a hemoglobin-like compound. Crawler, burrower; Collector, gatherer, predator; (**H**); VS-L (**F**/**S**) [Adult]
4. Culicidae (Mosquito): Head hardened and separate from the thorax; brush-like setae near the labrum (upper-lip); thorax is fused and swollen and wider than the abdomen. Swimmer; Collector, gatherer, filterer; (**H**); VS-M (**S**) [Adult]
5. Dixidae (Dixid midge): Head hardened and rounded; prolegs terminate in hooks on abdominal segment one and two; abdomen terminates into two lobes fringed with hairs. Crawler, burrower; Collector, gatherer; (**M**); VS-M (**S**/**F**) [Adult]
6. Syrphidae (Rat-tailed maggot): Head blunt or reduced and withdrawn into the thorax; 7 prolegs; abdomen terminates into a very long respiratory tube. Crawler, burrower; Collector, gatherer; (**H**); VS-M (**S**) [Adult]

Flies

7. Athericidae (Watersnipe fly): Body long (caterpillar-like); head reduced but may be visible; prolegs on most abdominal segments; abdomen ends in a fringed tail. The family is often green in color. Clinger, crawler; Predator; (**L**); S-M (**F**) [Adult]
8. Empididae (Dance fly): Body elongated; head reduced or pulled into the thorax; prolegs present on most abdominal segments; prolegs longer on segment eight; abdomen is blunt on the end or terminates in welts. Crawler, burrower; Predator; (**H**); VS-M (**S**/**F**) [Adult]
9. Muscidae (Muscid fly): Anterior portion of the body tapered, posterior is blunt; head reduced or withdrawn into thorax; whelps on abdominal segments; abdomen terminates into a pair of respiratory tubes. Predator; (**H**); (S-M); (**S**/**F**) [Adult]
10. Psychodidae (Moth fly): Head hardened, rounded and separate from the thorax; body segments with two to three secondary divisions (annuli) often having hardened plates. Burrower; Collector, gatherer; (**H**); VS-S (**S**) [Adult]
11. Ptychopteridae (Phantom cranefly): Head hardened and rounded; prolegs present on abdominal segments one through three, terminating with claws; abdomen terminates into a long respiratory tube. Crawler, burrower; Collector, gatherer; (**H**); M (**S**) [Adult]
12. Simuliidae (Black fly): Head hardened and rounded bearing a pair of labral fans (mouth brushes); prolegs on lower thorax; lower third of the abdomen is swollen (vase-like) and terminates in a ring of hooks. Clinger; Collector, filterer; (**M**); VS-M (**F**) [Adult]
13. Stratiomyidae (Solider fly): Body is flattened dorsally with a leathery feel; head is reduced but visible; thorax is broader than the head; spiracles at the end of the abdomen for breathing. Swimmer, burrower; Collector, gatherer; (**H**); S-L (**S**) [Adult]
14. Tabanidae (Horse fly): Body spindle shape both ends tapered; head reduced usually not visible; creeping welts with small hooks present on abdominal segments one through seven; no prolegs. Crawler, burrower; Predator; (**H**); M-VL (**S**/**F**) [Adult]
15. Tipulidae (Crane fly): Rounded head capsule, often reduced and barely visible; ventral welts on some abdominal segments; abdomen terminates into a disc surrounded by lobes or tentacle-like projections of varying shapes. Crawler, burrower; Shredder, predator; (**M**); VS-VL (**F**/**S**) [Adult]

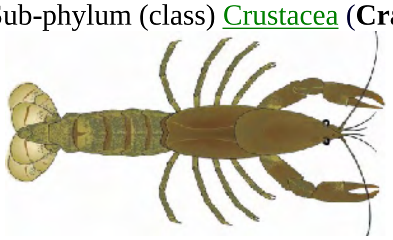
Class Insecta; order Neuroptera (**Spongillaflies**)

Antennae long and multi-segmented; jaws long and needle-like; body covered in tiny hairs (seta); has a pair of hardened plates on the thorax and each abdominal segment. They are associated with freshwater sponges, found on the outside or in the canals of the sponge. (Holometabolism) Clinger; Piercer; (**U**); VS-S (**S**)

Class Arachnida; order Trombidiformes; family Hydrachnidae (**Water mites**)

Four-pairs of segmented legs; one-pair of pedipalps; body is rounded and appears to consist entirely of an abdomen without segments. When captured will move very rapidly in a circular pattern. Click-here to view the life cycle. Swimmer, crawler; Predator; (**M**); S-M (**F**/**S**)

Sub-phylum (class) Crustacea (**Crayfish**, **Shrimp**, **Scuds** and **Sowbugs**) JG



More than three pairs of legs (> 6) attached to the thorax; the first several pairs of legs may have a hinged claw, which is often enlarged as in the order Decapoda; bodies strongly flattened from top to bottom or from side to side; abdomen consists of individual segments or the segments may be fused to form a thoracic shield; some kinds have a broad flipper on the end of the abdomen.

Order **Amphipoda**

1. Gammaridae (Sideswimmer/Scud): Having a shrimp-like appearance; body flattened from side to side; one pairs of antennae of equal length; seven-pairs of walking legs, first two are claw-like the remaining legs are simple. Has a habit of swimming sideways. Crawler, swimmer; Collector, gatherer; (**M**); S-M (**F**)

Order **Decapoda**

2. Cambaridae (Crayfish): Body mostly dorsally flattened; two-pairs of antennae one longer than the other; five-pairs of legs, first three-pairs with hinged claws and the first pair of claws are greatly enlarged; abdomen terminates in a flipper-like structure. Crawler, burrower; Collector, gatherer; (**M**); M-VL (**S**/**F**)
3. Palaemonidae (Freshwater shrimp): Cephalothorax and abdomen cylindrical with some side-to-side flattening; 5-pairs of walking legs the first two have claws, which are not enlarged; abdomen terminates in a flipper-like structure. Crawler, swimmer; Scraper, collector; (**M**); M-VL (**S**)

Order **Isopoda**

4. Asellidae (Aquatic sowbug): Body dorsally flattened; two-pairs of antennae one longer than the other; seven-pairs of legs, the first is claw-like and slightly enlarged, and the others have a simple pointed claw. Looks similar in appearance to its terrestrial cousin, the pill bug. Crawler, burrower; Collector, gatherer; (**H**); S-M (**S**/**F**)

Non-Arthropod Groups

Class Gastropoda; sub-classes Prosobranchia (**Operculate snails**) and Pulmonata (**Non-operculate snails**) JG

Operculate snails have a flat lid-like structure called an operculum that can seal the body of the snail inside the shell; the whorls of the shell bulge out distinctively to the sides (inflated); most have their opening on the right when the narrow (dextral) end is held up; shells often extended into a spiral shape. Non-operculate snails have no operculum; the whorls of the shell do not distinctly bulge out to the sides; often the shells of most kinds are shaped like a low flat cone or coiled flat instead of being extended in a spiral shape. For more information and images visit Marshall University's Aquatic Snails of West Virginia website.

Operculate snails

1. Bithyniidae (Bithynid snail): Shell is whorled and bulges out to the side (inflated); opens to the right when the narrow end is held up; usually a small shell, sometimes colored or having a mottled pattern; gills sometimes visible. Clinger, crawler; Scraper; (**M**) VS-M (**F**/**S**)
2. Hydrobiidae (Pebble snail): Shell is whorled and bulges out to the side (inflated); opens to the right when the narrow end is held up. The family is very diverse in shell size and shape; shell shape can range from conical (cone-like) to spherical (rounded). Clinger, crawler; Scraper; (**L**) S-L (**F**)
3. Pleuroceridae (Rock snail): Shell is spiraled and whorled but does not budge out to the side (flattened); opens to the right when the narrow end is held up; operculum is smaller than most others and can be pulled into the shell. Clinger, crawler; Scraper; (**L**) S-L (**F**)
4. Viviparidae (Viviparid snail): Shell is whorled and bulges out to the side (inflated); opens to the right when the narrow end is held up; operculum has concentric lines, which are slightly off-center. Clinger, crawler; Scraper; (**M**) S-L (**S**)

Non-operculate snails

5. Ancylidae (Limpet): Shell shaped like a low flat cone; no operculum. Clinger, burrower; Scraper; (**H**) VS-M (**F**)
6. Planorbidae (Orb snail): Shell is coiled flat instead of extended in a spiral; no operculum. Clinger, burrower; Scraper; (**M**) S-L (**S**/**F**)
7. Physidae (Left-handed snail): Shell is high, spiraled, with a slight bulge; opens to the left when the narrow end is held up; no operculum. Clinger, crawler; Scraper; (**H**) S-L (**S**/**F**)

Class Bivalvia (**Clams** and **Mussels**) JG

Two shells opposite of each other and strongly connected by a hinged ligament; the shell is thick and strong or thin and fragile in some kinds; growth rings on the shell are either far apart and are distinctly raised, or very close together and hardly raised at all; the foot usually consists of two tubular structures that can often be seen protruding from the shell; the body is soft tissue, often pinkish or gray in color.

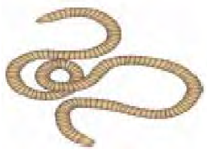
Clams

1. Corbiculidae (Asian clam): Shell is rounded; brown in color usually lighter than mussels; raised separated ridges along the top and sides of the shell. Clinger, burrower; Collector, filterer; (**M**); VS-VL (**S**/**F**)
2. Sphaeriidae (Pea clam): Shell is very small and rounded; light colored; ridges spaced close together, not raised. Clinger, burrower; Collector, filterer; (**M**); VS-M (**S**/**F**)

Mussels

3. Dreissenidae (Zebra mussel): Zebra mussels get their name from the striped pattern of their shells, though not all shells bear this pattern; usually about fingernail size but can grow to a maximum length of nearly two inches; often they can be found in large colonies attached to a variety of objects (Invasive species). (M); VS-M (**S**)
4. Unionidae (Mussel): Largest of the bivalves; shell usually dark in color, variable in shape but maybe somewhat oblong; has many indentations and ridges on the tops and sides of the shell. Clinger, burrower; Collector, filterer; (**L**); VS-VL (**S**/**F**)

Phylum Annelida (**Leeches** and **Worms**) JG



Body is soft, muscular and cylindrical in shape; body consists of many similar, round ring-like segments arranged in rows; numerous segments along the entire length, number often depends upon the order or family. Leeches have distinct suckers situated on the bottom of the body, one at the front and one at the rear.

Leeches

1. Hirudinea (Leech): Body dorsally flattened with 34 segments, which are divided so there appears to be more; suction disks present on one or both ends; eyespots may be present. Crawler, burrower; Predator, parasite; (**H**); S-VL (**S**/**F**)

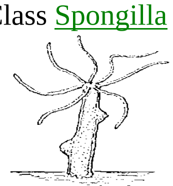
Worms

2. Oligochaeta (Aquatic worm): Body elongated (worm-like); divided into many segments most having bundles of small hairs; no eyespots or suckers present. Aquatic earthworms are common in riffles; however wetlands have a much wider variety from this group (i.e. Naidid worms, Tubiflex worms etc.) Crawler, burrower; Collector, gatherer; (**H**); VS-VL (**S**/**F**)
3. Nematoda (Round worm): Worm-like; no segmentation; body usually translucent or having a pale coloration. Crawler, burrower; Collector, gatherer; (**H**); VS-M (**S**)
4. Nematomorpha (Horsehair worm): Body very long and slender; no segments. Burrower; Parasite, predator; (**H**); VL (**S**)

Class Turbellaria (**Flatworms**) JG

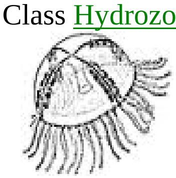
Soft-elongated body flattened from top to bottom; no individual segments; digestive track with only one opening which functions both as the mouth and anus; mouth usually on the bottom side positioned about one-fifth to the length of the body; sides of the body constricted towards the front forming a head that is often somewhat triangular shaped; two eyespots situated on top of the head gives the animal a cross-eyed appearance. Most families can withstand high nutrient and organic enrichment, but some are very sensitive to toxics. Crawler, burrower; Collector, gatherer; (**H**); VS-L (**S**/**F**)

Class Spongilla (**Freshwater sponge**)



Sponges are delicate in structure, growing as encrusting or branching masses; usually appear greenish because of the algae that live on them. Freshwater sponges may attain a volume of more than 2,500 cubic centimeters (150 cubic inches). The larva of the Spongilla fly lives as a parasite on freshwater sponges. (**U**); S-L (**S**)

Class Hydrozoa (**Freshwater jellyfish**)

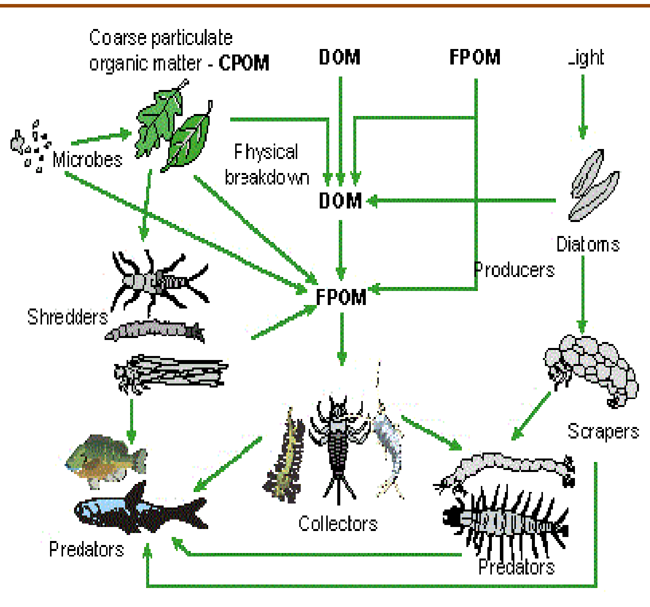


Usually small and bell-shaped; tentacles of varying lengths protrude from the margins of the velum; is often translucent with a whitish or greenish tinge. Swimmer, clinger; Collector, predator; (**U**); VS-S (**S**)

Glossary: This glossary includes select terminology used here, and it also includes other terms often associated with the description of aquatic invertebrates.

1. Abdomen: The third main division of the body; behind the head and thorax.
2. Anterior: In front (before).
3. Apical: Near or pertaining to the end of any structure, part of the structure that is farthest from the body.
4. Basal: Pertaining to the end of any structure that is nearest to the body.
5. Burrower: Animal that uses a variety of structures designed for moving and burrowing into sand and silt, or building tubes within loose substrate.
6. Carapace: The hardened part of some arthropods that spreads like a shield over several segments of the head and thorax.

7. Caudal filament: Threadlike projection at the end of the abdomen, like a tail.
8. Clinger: Animal that uses claws or hooks to cling to the surfaces or rocks, plants or other hard surfaces and often moves slowly along these surfaces.
9. Concentric: A growth pattern on the opercula of some gastropods, marked by a series of circles that lie entirely within each other; compare multi-spiral and pauci-spiral.
10. Crawler: An animal, whose main means of locomotion is moving slowly along the bottom, usually has some type of hooks, claws or specially designed feet to help hold them to surfaces.
11. Detritus: Disintegrated or broken up mineral or organic material.
12. Dextral: The curvature of a gastropod shell where the opening is visible on the right when the spire is pointed up.
13. Distal: Near or toward the free end of any appendage; that part farthest from the body.
14. Dorsal: Pertaining to, or situated on the back or top, especially of the thorax and abdomen.
15. Elytra: Hardened shell-like mesothoraic wings of adult beetles (Coleoptera).
16. Femur: The leg section between the tibia and coxa of Arthropoda, comparable to an upper arm or thigh.
17. Flagellum: A small fingerlike or whip-like projection.
18. Gill: Any structure especially adapted for the exchange of dissolved gases between animal and a surrounding liquid.
19. Glossae: A lobe or lobes front and center on the labium; in Plecoptera, the lobes are between the paraglossae.
20. Hemimetabolism: incomplete metamorphosis.
21. Holometabolism: complete metamorphosis.
22. Labium: Lower mouthpart of an arthropod, like a jaw or lip.
23. Labrum: Upper mouthpart of an arthropod consisting of a single usually hinged plate above the mandibles.
24. Lateral: Feature or marking located on the side of a body or other structure.
25. Ligula: Forming the ventral wall of an arthropod's oral cavity.
26. Lobe: A rounded projection or protuberance.
27. Mandibles: The first pair of jaws in insects.
28. Maxillae: The second pair of jaws in insects.
29. Multi spiral: A growth pattern on the opercula of some gastropods marked by several turns from the center to the edge.
30. Operculum: A lid or covering structure, like a door to an opening.
31. Palpal lobes: The grasping pinchers at the end of the Odonata lower jaw.
32. Pauci-spiral: A growth pattern on the opercula of some gastropods marked by few turns from the center to the edge.
33. Periphyton: Algae and associated organisms that live attached to underwater surfaces.
34. Posterior: Behind; opposite of anterior.
35. Proleg: Any projection appendage that serves for support locomotion or attachment.
36. Prothorax: The first thoracic segment closest to the head.
37. Rostrum: A beak or beak-like mouthpart.
38. Sclerite: A hardened area of an insect body wall, usually surrounded by softer membranes.
39. Seta (pl. setae): Hair like projection.
40. Sinistral: The curvature of a gastropod shell where the opening is seen on the left when the spire is pointed up.



Functional feeding groups are a classification approach that is based on behavioral mechanisms of food acquisition rather than taxonomic group. The same general behavioral mechanisms in different species can result in the ingestion of a wide range of food items. The benefit of this method is that instead of hundreds of different taxa to be studied, a small number of groups of organisms can be studied collectively based on the way they function and process energy in the stream ecosystem. Individuals are categorized based on their mechanisms for obtaining food and the particle size of the food, and not specifically on what they are eating. This method of analysis avoids the relatively non- informative necessity to classify the majority of aquatic insect taxa as omnivores and it establishes linkages to basic aquatic food resource categories, coarse particulate organic matter (CPOM), and fine particulate organic matter (FPOM), which require different adaptations for their exploitation. The major functional feeding groups are: (1) Scrapers (grazers) consume algae and associated material; (2) Shredders consume leaf litter or other CPOM, including wood;

(3) Collectors and gatherers collect FPOM from the stream bottom; (4) Filterers collect FPOM from the water column using a variety of filters; and (5) Predators feed on other consumers. A sixth category includes species that do not fit neatly into the other categories such as parasites and piercers. It is important to keep in mind that many kinds of invertebrates use a variety of food acquisition methods.

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16. WV Save Our Streams – Macroinvertebrate sub-sampling protocols

http://www.dep.wv.gov/WWE/getinvolved/sos/Documents/Macroinvertebrates/WVSOS_MacroinvertebrateGuide.htm

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