Monitoring Water Quality

Acknowledgments

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Chapter 1
Introduction

1.1 - Manual Organization

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 V*olunteer 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 landuse 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 handinhand 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)
401 M Street, SW
Washington, DC 20460

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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

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Chapter 2
Elements of a Stream Study

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.

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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 as they flow from higher to lower elevations.

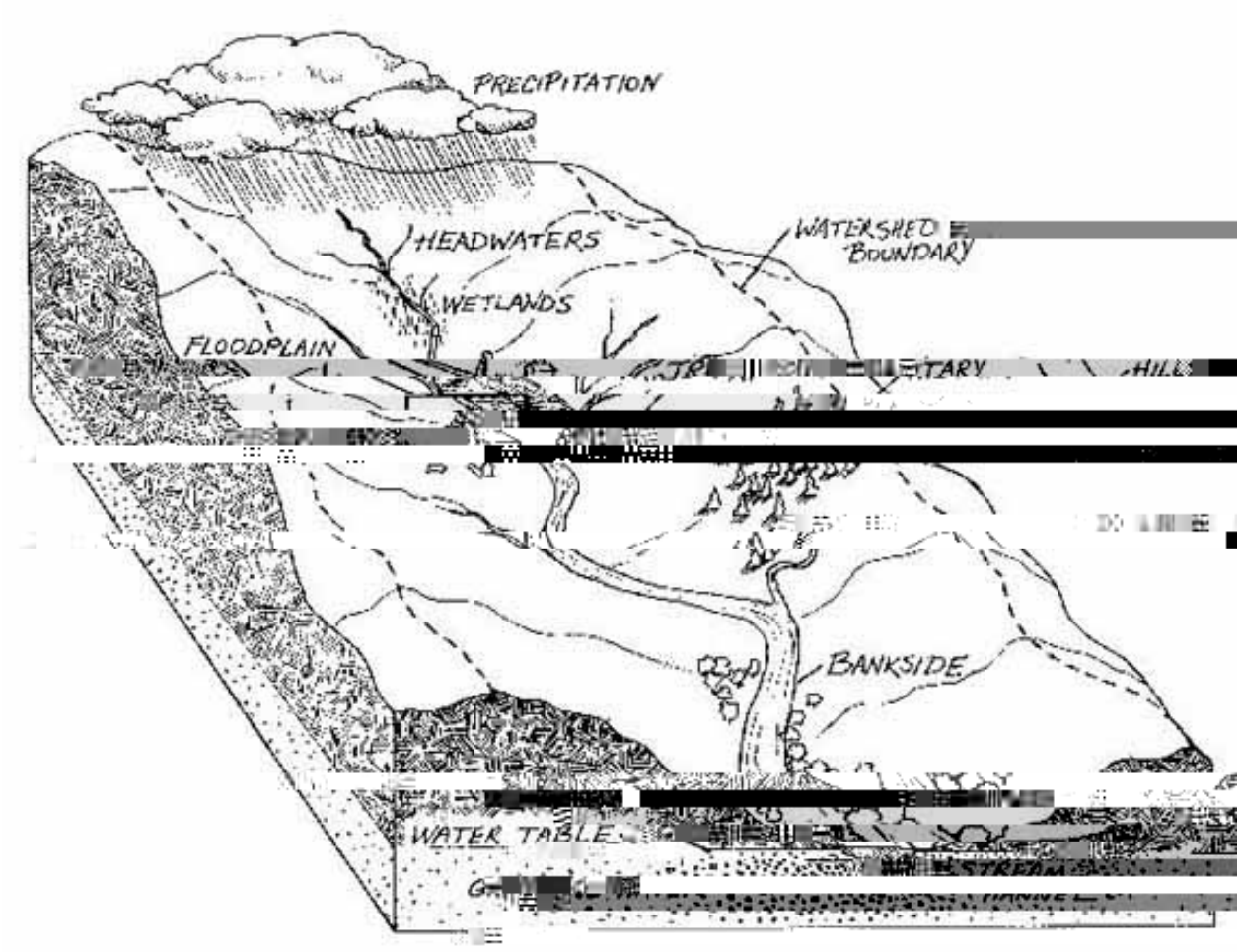


Figure 2.1

Cross section of a watershed
Volunteers should get to know the watersheds of their study streams.

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

to move water quickly away from developed areas and into a natural watercourse.

Figure 2.4

The fate of precipitation in undeveloped vs. developed watersheds
Survace runoff increases and ground water recharge decreases as watersheds become developed.

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 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

Figure 2.5

Streams losing and gaining water
The position of the water table sometimes plays a role in determinating the amount of streamflow.

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 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, slowmoving, 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, oxygendemanding 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.

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 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.

Type	Approach	Applications*

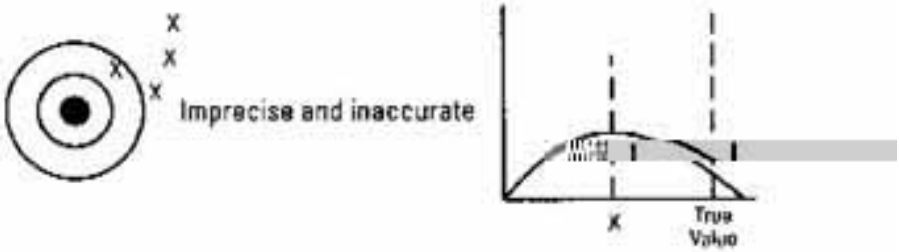
5. How good does the monitoring data need to be?

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).

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 paramter.
- ***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 a specified in the original sampling design. It is usually expressed as a percentage. For example, if 100 samples were scheduled but

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 timeconsuming. 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 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).

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.

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 19 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.)

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.)

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Training Volunteer Monitors

Back to Section 2.2 - Designing the Stream Study

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 postseason 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 illplanned, 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.

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

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 789equipm

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.

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

Dates, G. 1994. A Plan for Watershedwide Volunteer Monitoring. *The Volunteer Monitor*. 6(2):8.

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Weber, P. and F. Dowman. 1994. The Web of Water.

Chapter 3
Watershed Survey Methods

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 it0m(Pjl 05hleuct3Dn conduct is the wa may be dmond, or Tm two dmstingrap

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 AdoptA Stream Foundation. References are provided at the end of this chapter for further information on watershed surveys.

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3.1
How to Conduct a Watershed Survey

The Background Investigation

Researching the stream is generally a onetime 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

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.

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 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 schoolage volunteers.

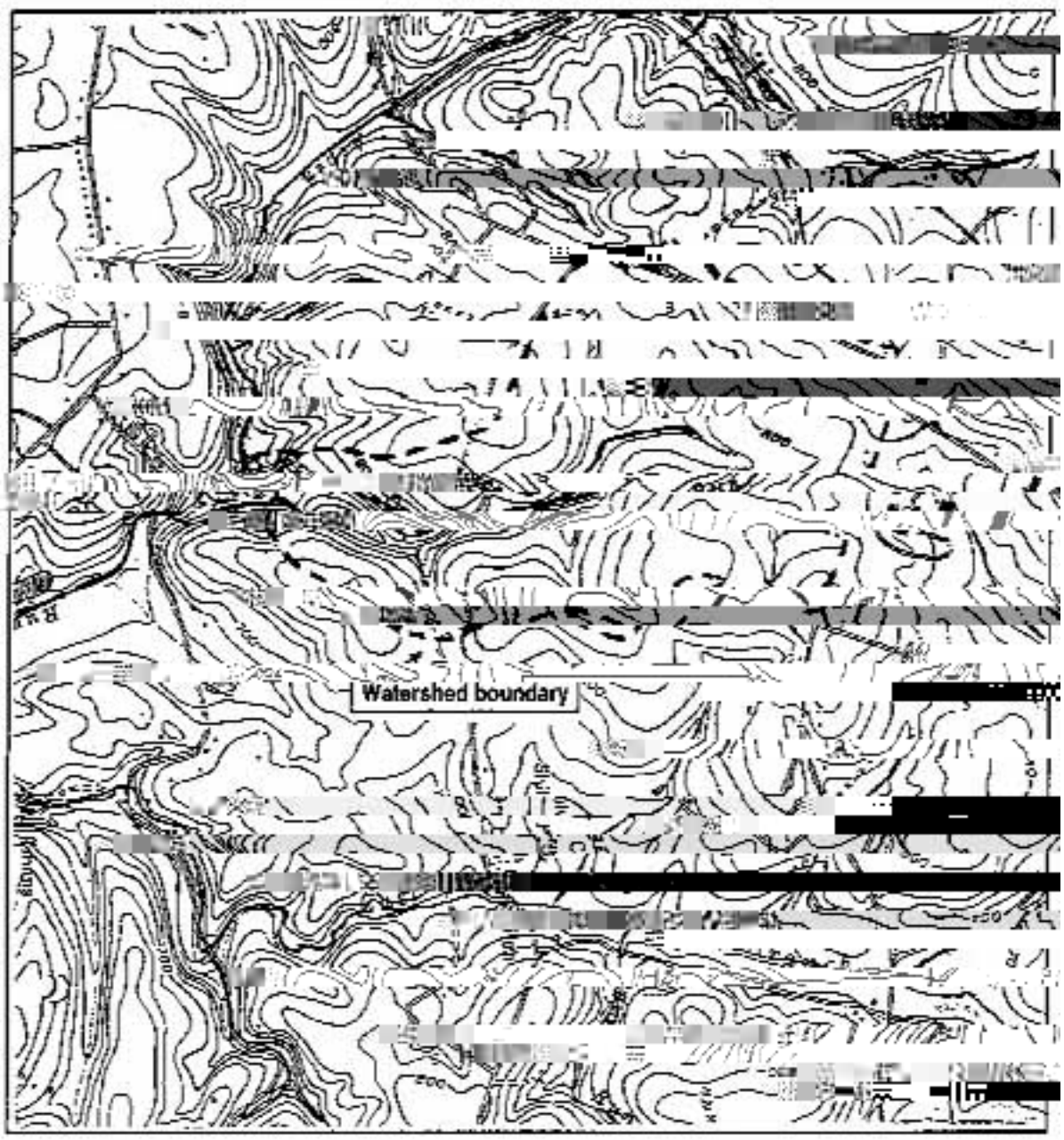

Figure 3.1

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.

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.

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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

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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 spotcheck 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

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***

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.

References and Further Reading

Delaware Nature Education Center. 1996. *Delaware Stream Watch Guide.* July.

Ely, E. 1994. Delineating a Watershed. *The Volunteer Monitor.* 6(2):3.

Ely, E. 1994. LandUse Surveys. *The Volunteer Monitor.*

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 databases.

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

Watershed Survey Visual Assessment (PDF, 15.0 KB)

Adobe Acrobat Reader is required to view PDF documents. The most recent version of the Adobe Acrobat Reader is available as a free download. An Adobe Acrobat plug-in for assisted technologies is also available.

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Chapter 4
Macroinvertebrates and Habitat

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underoxygenated 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

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.

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 folows.

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

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.

- *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 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 Mary-land 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 indentification/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-streaming 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	• Periodic workshops and streamside training sessions	• Formal lab and field training with experienced team leaders before all assessments

Table 4.1

Tiered framwork for volunteer biological monitoring programs
Program designers might choose simple or complex approaches according to program goals and resources.

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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 primar ily 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, l ate 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 i s 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 cri tical 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 c hoose 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.

data sheet, base your re sponses 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 t hem 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-y ard 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 dat a sheets because regular ink will run if wet.

Figure 4.3

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

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. Eve n 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 volu nteer 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 th e lat/long coordinates of the user. In 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 an d 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 "Macroinverte-brate 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 ma croinvertebrates. 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.

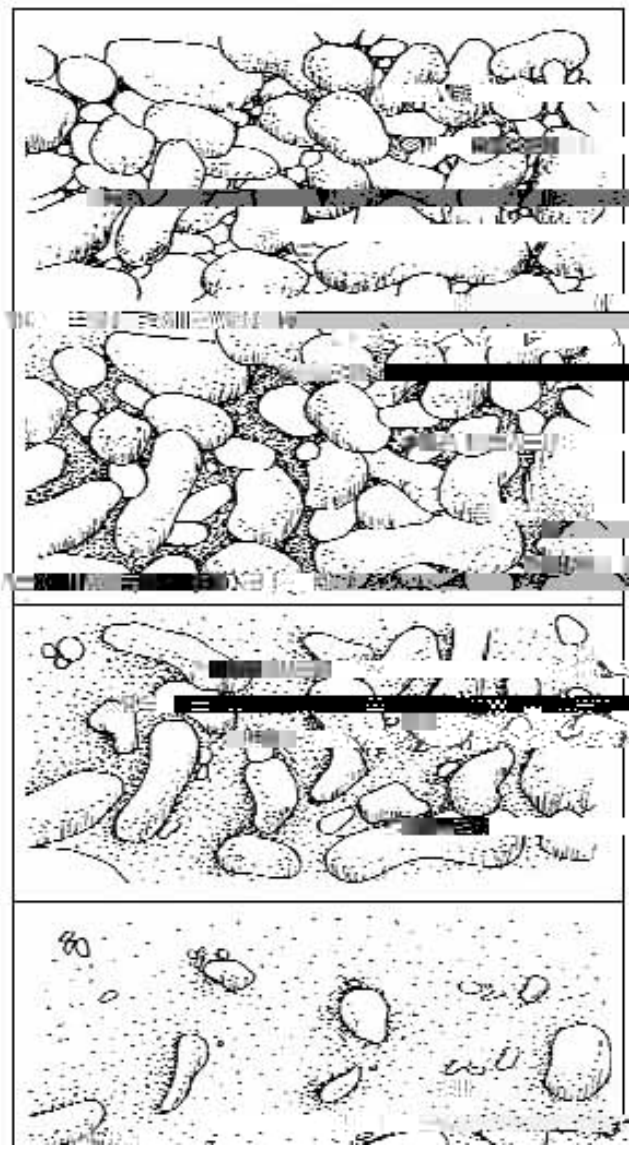
Step 4--Check data forms for completeness and return forms to program coordinator

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 da ta 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 disc repancies in the data. **If you have identified an urgent problem, such as**

In-stream Characteristics

1. *Pools, riffles, and runs.* A mixture of flows and depth 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 ladybug 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).
    - *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 embedded-ness. 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.
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



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.

- *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 gr ass 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.

Figure 4.6

Types of streambank shapes
Undercut banks provide good cover for fish and macroinvertebrates.

- *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.
- *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* - anwmen5.4545rs or pavdg araes, incluwingpathes,roades, ridgnes,huouse s, etc.

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,

- *Crayfish* look like lobsters or shrimp. They are generally somewhat tolerant of pollution.
- *Snail-like organisms* include snails and clam-like organisms. They range from somewhat tolerant of pollution to somewhat intolerant.
- *Insects* include a wide variety of organisms that generally have distinct legs, head, bodies, and tails and often move quickly over rocks or sticks. They come in many sizes and shapes as well as a wide range of pollution-tolerance levels.

When finished, return all organisms to the stream.

Stream Habitat Walk (PDF, 137.0 KB)

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Monitoring Water Quality

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.

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.

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--Figure 4.7).
- *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, in Section 4.1 - Stream Habitat Walk).

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.)

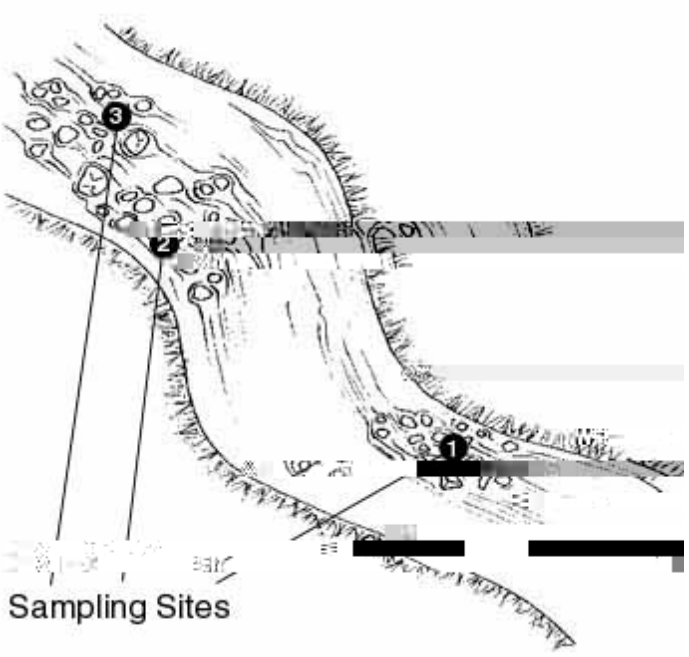

Figure 4.8

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 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 stream-bed 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. 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

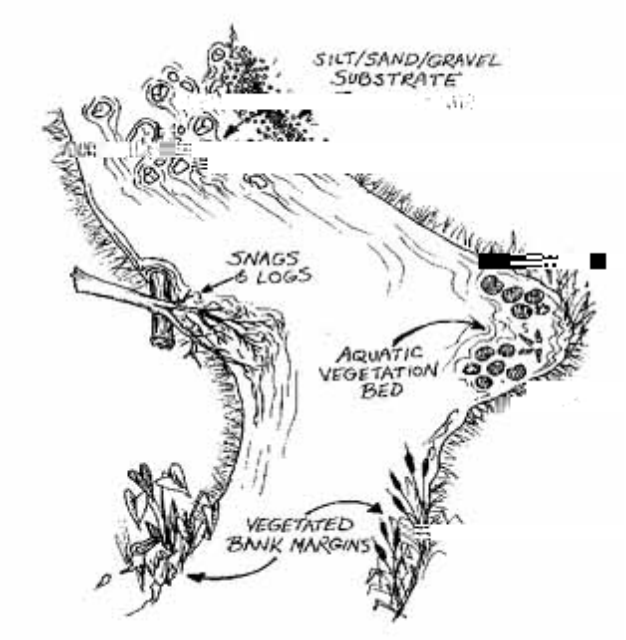

Figure 4.10

Four habitats found in muddy-bottom streams
Volunteers will likely find the most macroinvertebrates in vegetated habitats and snags and logs.

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.

- 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.

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.

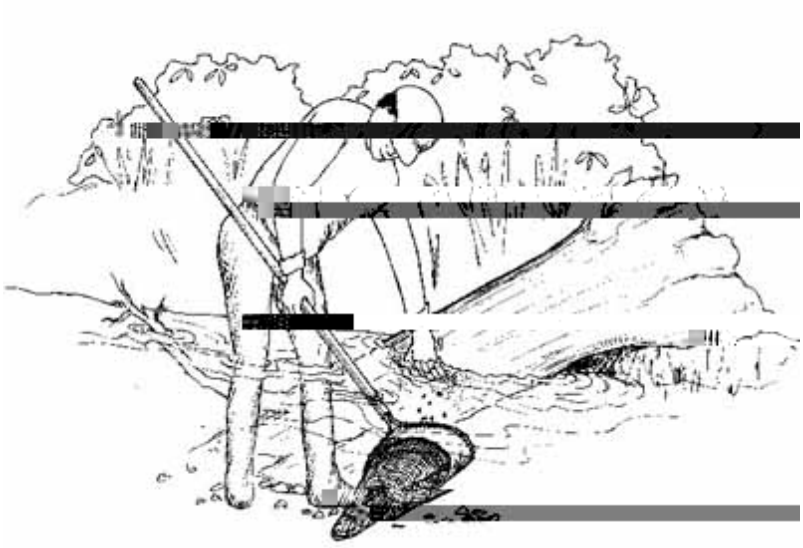

Figure 4.11

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

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.

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 Macroinverte-brates 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.

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.**

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Note:

In-stream Characteristics

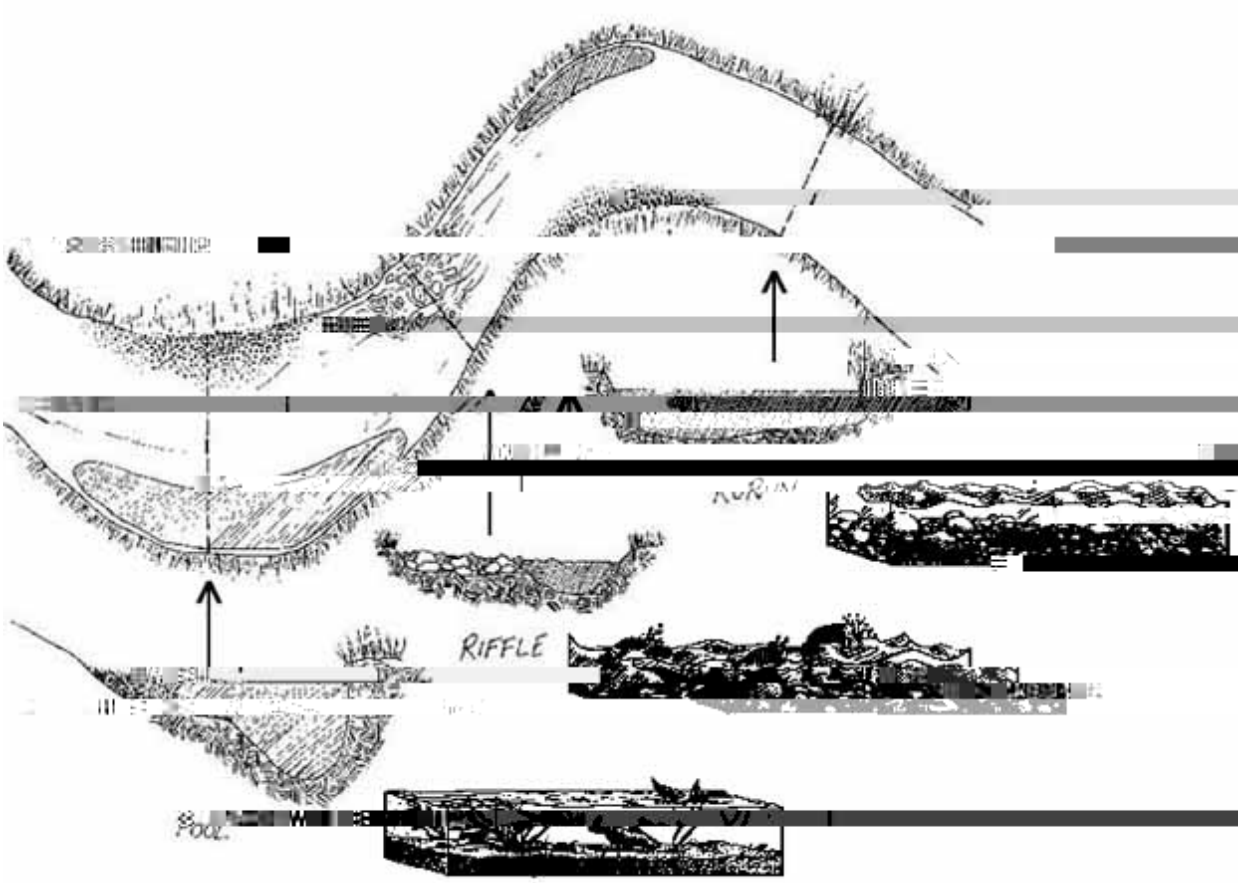

Figure 4.13

Overview and cross sections of a pool, riffle, and run
Varying flows and depths create a variety of habitats for macroinvertebrates.

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).
    - *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.

   Water appearance can be a physical indicator of water pollution.

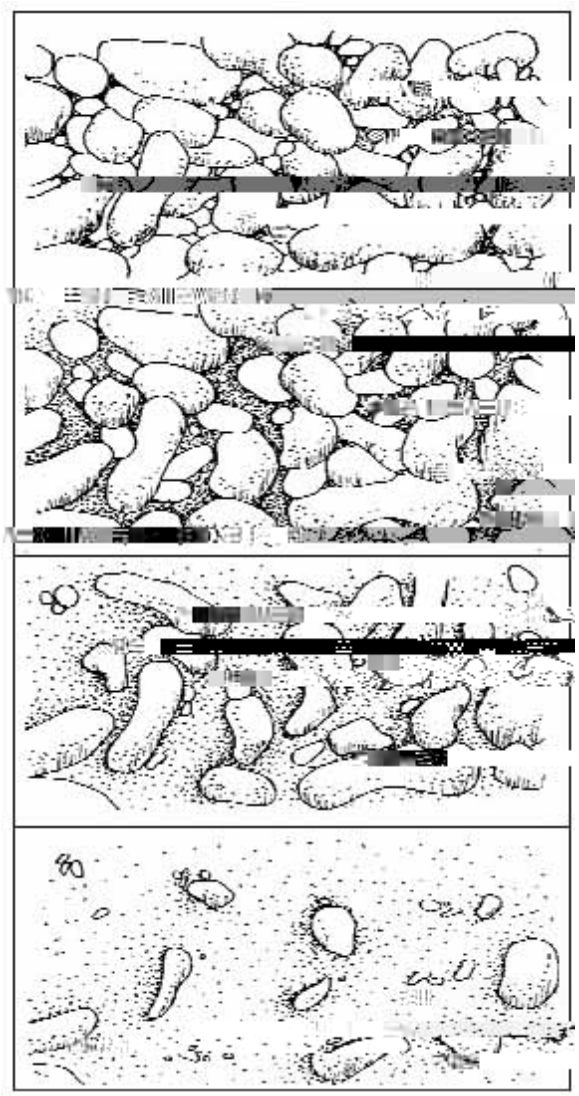

Figure 4.14

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.

stream at the site.

Step 4 Complete all the field data sheets

4.3
Intensive Stream Biosurvey

Selecting Metrics to Determine Stream Health

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 Maryl and 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 macroinve rtebrate 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 an alysis.

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 train ing, 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 co mposite 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 107pact

sample of macroinvertebrates is preserved and returned to a laboratory. A portion, o r 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 th e 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 biologi st 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 mi croscopes) 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

Sieve Buckets

Most professional biological monitoring programs

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.

Rocky-Bottom Streams
Part1: 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, Step 2 in Section 4.1 - Stream Habitat Walk)

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 sub strate. 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 macroinvertebrat es and alter your sampling results.

sampling area. Hold the net handles at a 45 degree angle to the water's surface. Be sure th at 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. 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 organis ms 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 wi th 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 bucke t. 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 emp ty 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 ja r.

Figure 4.16

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.

Pouring sample water through the net

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 m embers of the collecting crew. Place the ID tag into the sample container writing side facing out, so that identification can be seen clearly.

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

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)

formation of islands, shoals, or point bars (sediments that build up in the stream, usually a t 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 easi ly 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 tim e. 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 combi nations 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. Wh en 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.

1. *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 ero sion. 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. Veg etative 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.

2. *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 potenti al. Signs of erosion include crumbling, unvegetated banks, exposed tree roots, and exposed soil.

   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 nea r 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 measu red 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 _____

- *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.

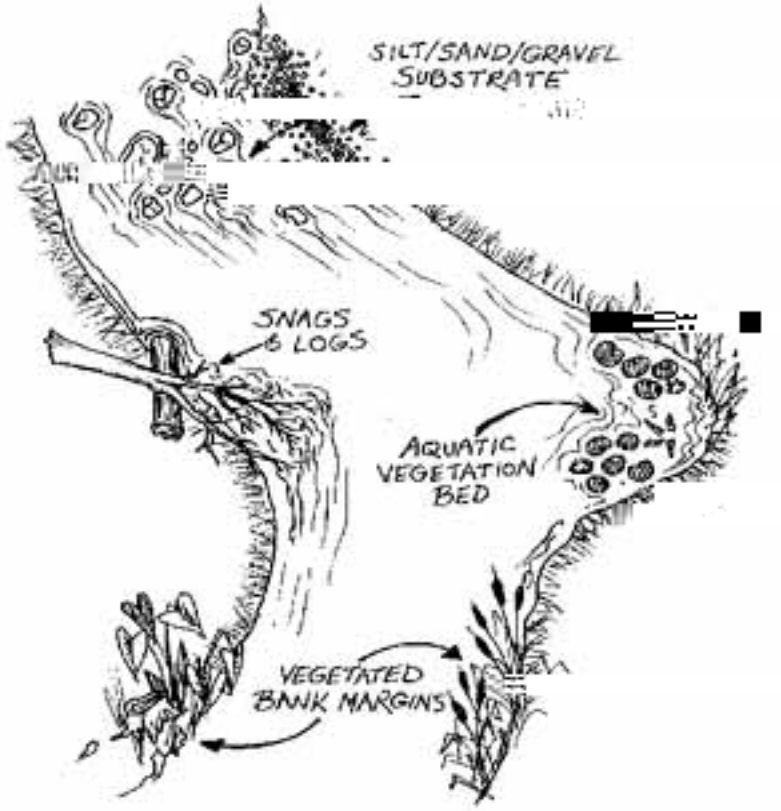

Figure 4.17

Four habitats found in muddy-bottom streams
Volunteers will likely find the most macroinvertebrates in vegetated habitats and snags and logs.

- *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 productiv e 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.

- 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 fi ndings.

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 strea m 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 n et 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.

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 thorough ly 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 m aterial 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 com pletely 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 h ands 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,

with less diverse samples. Refresher workshops for e xperienced 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)

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 randomizi ng 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.

   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 tran sferred. Keep a written count of the number of organisms you have transferred. The objective is have at least 100 individual organisms in your tray,

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

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

SUBSAMPLE ID TAG

Station #: _____

Stream: _____

Location: _____

Date/Time: _____

Subsample team members:

_____

_____

_____

Figure 4.21

Example of a Subsample ID tag
To prevent wubsamples from being mixed up, volunteers should place the ID tag *inside* the subsample jar.

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 a dvisor 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 recycled). 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

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.	score.
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.	

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 interpretati on 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 organism s 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). T hese 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 va riety 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.

Sensitive taxa index = $E(X_i t)/n$

where:

E = 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 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 t o 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 c alculate 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 fic tional 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. Choo se 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< /a>).

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Intensive Biosurvey: Macroinvertebrate Assessment (PDF, 90.5 KB)

Intensive Biosurvey: Habitat Assessment (PDF, 80.8)

Selecting Metrics to Determine Stream Health

Back to Section 4.3 - Intensive Stream Biosurvey

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).

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Figure 4.1

Back to Chapter 4 - Macroinvertebrates and Habitat

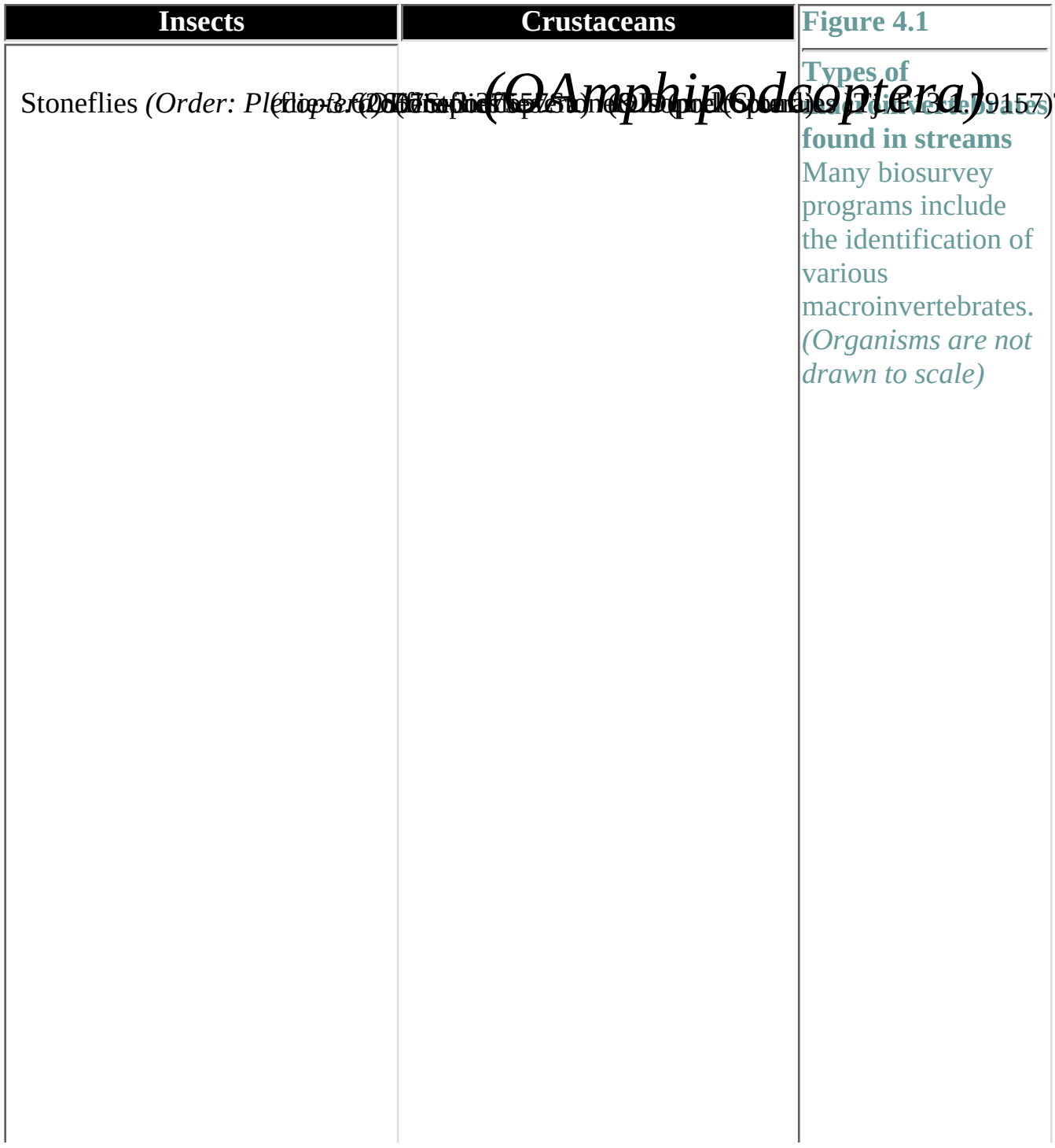
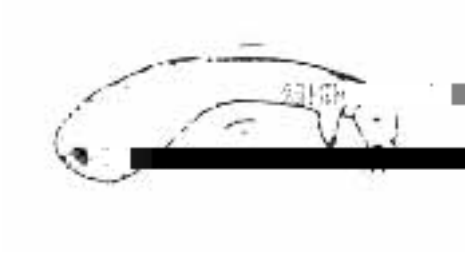


Figure 4.1

Types of macroinvertebrates found in streams

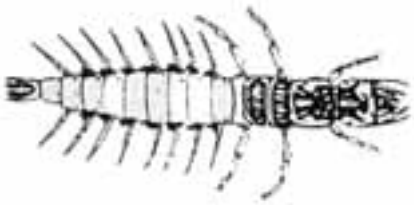
Many biosurvey programs include the identification of various macroinvertebrates. *(Organisms are not drawn to scale)*

Flies & Midgets *(Order: Diptera)*

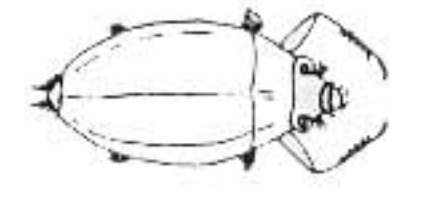


Water Bugs *(Order: Hemiptera)*

Dobsonfly *(Order: Megaloptera)*



Beetles *(Order: Coleoptera)*



Snails

Mussels & Clams

Worms

Leeches

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Figure 4.7

Back to Chapter 4.2 - Streamside Biosurvey

Back to Chapter 4.3 - Intensive Stream Biosurvey

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.)

Monitoring Water Quality

3. Dislodge macroinvertebrates by rubbing rocks thoroughly. 	6. Flush out the net with clean stream water.	

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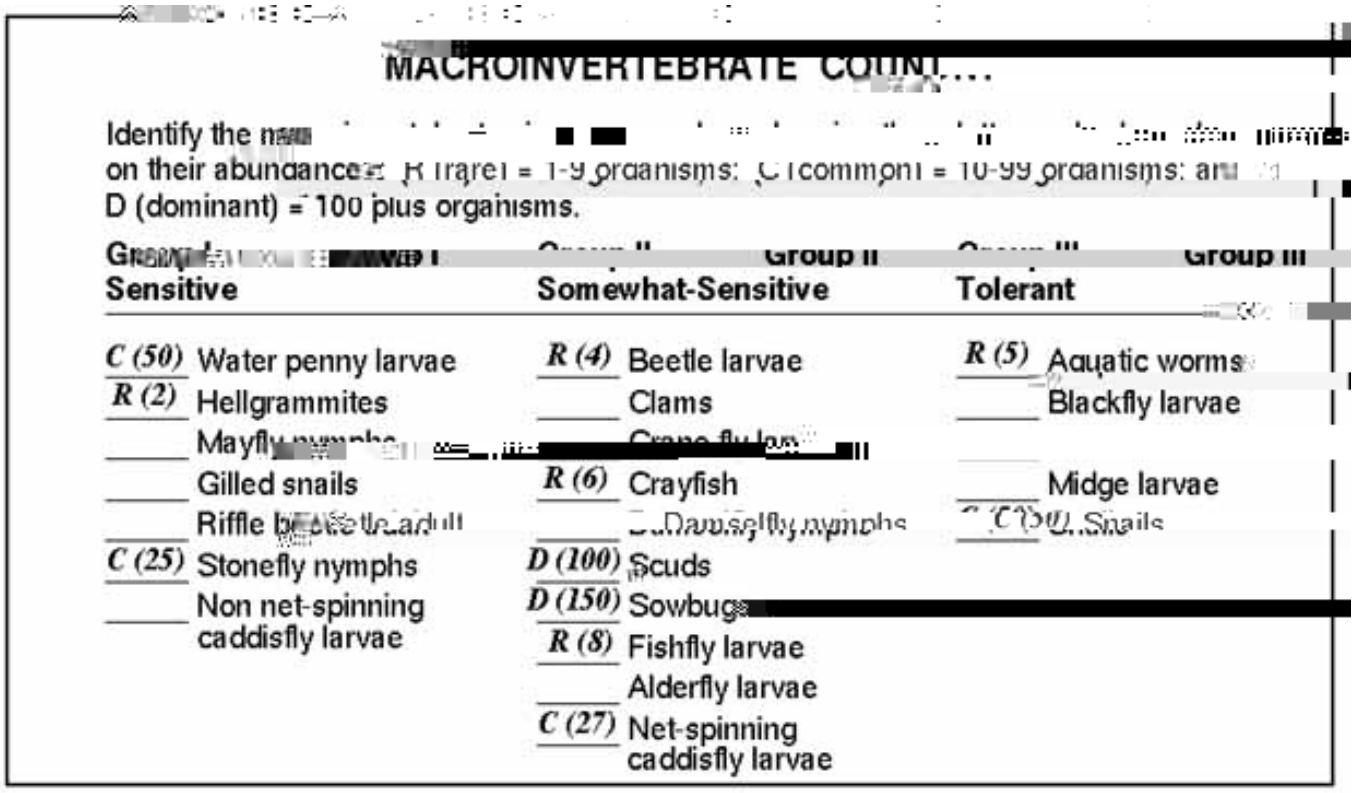
Figure 4.12

Back to Section 4.2 - Streamside Biosurvey



MACROINVERTEBRATE COUNT....

Identify the n... 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		
	Gilled snails	R (6)	Crayfish		Midge larvae
	Riffle beetle adult		Damselfly nymphs	C (30)	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		

Figure 4.12

Sample macroinvertebrate count for (hypothetical) Volunteer Creek

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Tables 4.6 - 4.9

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Table 4.6

Metric worksheet for rocky-bottom streams

Primary Metrics	(M) Monitored Site Values	Biosurvey Score (Circle the appropriate range for each metric) 6	3	0
No. of Taxa		>15	15-8	<8
No. of EPT Taxa		>8	8-4	<4
% Dominance		<34%	34-67%	>6.4
Optional Metrics				
% Abundance of Scrapers		>18%	18-10%	<10
% Abundance of Shredders		>9%	9-5%	<5%
COLUMN SCORE *(Multiply no. of circled values by the biosurvey score)*				
TOTAL SCORE *(Sum all the column scores)*				

Notes: If fewer than 60 individuals in the monitored site, don't calculate metrics for any of the sites. Biosurvey scoring ranges determined for the summer index period.

Table 4.7

Metric worksheet for muddy-bottom streams

Primary Metrics	(M) Monitored Site Values	Biosurvey Score (Circle the appropriate range for each metric) 6	3	0
No. of Taxa		>19	19-10	<10
No. of EPT Taxa		>7	7-4	<4
% Dominance				

Sensitive Taxa Index		<5.0	5.0-6.8	>6.8
Optional Metrics				
% Abundance of EPT		>39%	39-20%	<20
% Abundance of Midge Larvae		>24%	24-60%	<60%
COLUMN SCORE *(Multiply no. of circled values by the biosurvey score)*				
TOTAL SCORE *(Sum all the column scores)*				
Notes: If fewer than 60 individuals in the monitored site, don't calculate metrics for any of the sites. Biosurvey scoring ranges determined for the summer index period.				

Table 4.8

Sample metric worksheet for Volunteer Creek (hypothetical rocky-bottom stream)
There were 119 macroinvertebrates in this sample.

Primary Metrics	**(M) Monitored Site Values**	**Biosurvey Score** (Circle the appropriate range for each metric)		
		6	**3**	**0**
No. of Taxa		>15	***15-8***	<8
No. of EPT Taxa		>8	***8-4***	<4
% Dominance		<34%	***34-67%***	>67%
Sensitive Taxa Index		***<4.8***	4.8-6.4	>6.4
COLUMN SCORE *(Multiply no. of circled values by the biosurvey score)*		6	9	0
TOTAL SCORE *(Sum all the column scores)*		**Biosurvey Score for this site is 15 This site scores in the Fair range, 9-15**		

Table 4.9

Biosurvey Scoring Guide
This guide is based on the four primary metrics. If your score

Total Score From Metrics	**Condition Category**	**Attributes**

Chapter 5
Water Quality Conditions

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 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.

Figure 5.1

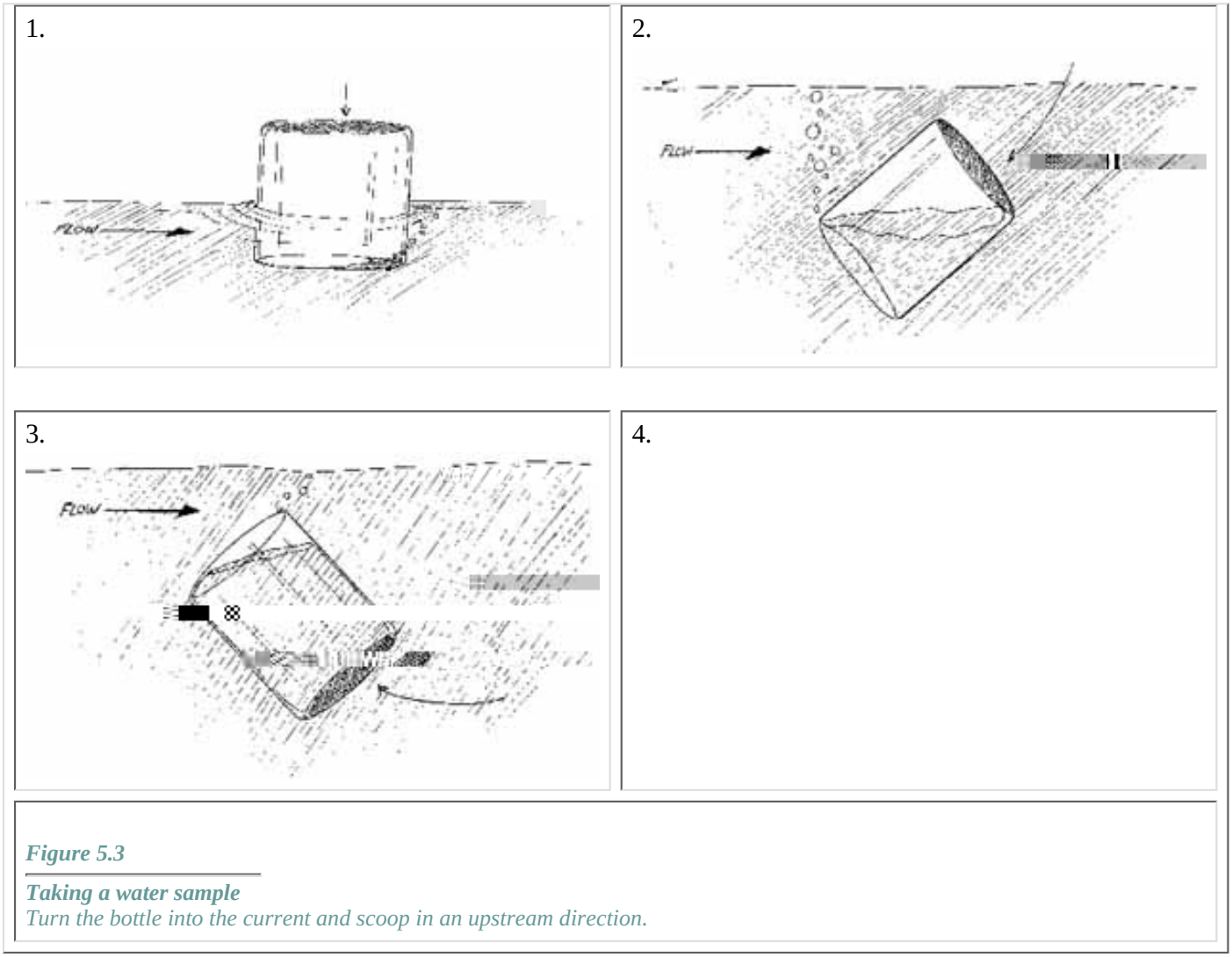

Figure 5.3

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.

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QUALITY ASSURANCE, QUALITY CONTROL, and QUALITY ASSESSMENT MEASURES

Quality Control And Assessment Measures: External Checks

External checks are performed by nonvolunteer 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 problemidentification 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 your can monitor.

References

APHA. 1992. *Standard Methods for the Examination of Water and Wastewater*. 18th ed. American Public Health Association, Washington, DC.

Intergovernmental Task Force on Monitoring Water Quality. 1994. *Water quality monitoring in the United States.* 1993 report and technical appendixes. Washington, DC.

Mattson, M. 1992. The basics of quality control. *The Volunteer Monitor.* 4(2) Fall 1992.

USEPA. 1983. *Methods for chemical analysis of water and wastes.* EPA600/479020. U.S. Environmental Protection Agency, Environmental Monitoring and Support Laboratory, Cincinnati, OH. March.

USEPA. 1984. *Guidance for preparation of combined work/quality assurance project plans for environmental monitoring.* ORWS QA1, U.S. Environmental Protection Agency, Office of Water Regulations and Standards. Washington DC, May.

USEPA. 1996. *The Volunteer Monitor's Guide to Quality Assurance Project Plans.* EPA841-B-96-003. Environmental Protection Agency, Office of Water, Washington, DC.

Common Quality Control Measures

	Dissolved Oxygen	Temperature	pH	Turbidity	Phosphorus	Nitrates	Total Solids	Conductivity	Total Alkalinity	Fecal Bacteria
Internal Checks										
Field blanks										

Lab replicates	•		•	•	•	•	•	•	•	•b
Positive plates										•
Negative plates										•
Spike samples		•			•	•			•	•
Calibration blank				•	•	•		•		
Calibration standard	•a		•	•	•	•		•		
External Checks										
External field duplicates			•	•	•	•	•	•	•	•
Split samples			•	•	•	•	•	•	•	
Outside lab analysis	•			•	•	•	•	•	•	•
Verification										•
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:

$$\text{Flow} = ALC / T$$

Where:

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

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

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

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

How to Measure and Calculate Stream Flow

Task 1 Prepare before leaving for the sampling site

Refer to section 2.3 - Safety Considerations 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

flow would be:

Where:

A = 5.42 ft2
L = 20 ft
C = 0.8 (coefficient for a rocky-bottom stream)
T = 15 seconds

Flow = 15 seconds (5.42 ft^2) (20 ft) (0.8) / 15 sec.
Flow = 86.72 ft^3/ 15 sec.
Flow = 5.78 ft3/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.

Data Form for Calculating Flow (PDF, 82.8 KB)

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.

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			

Table 5.3
Maximum dissolved oxygen concentrates vary with temperature

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.

   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

Figure 5.7

Taking a water sample for DO analysis
Point the bottle downstream and fill gradually. Cap underwater when full.

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 th e 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 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. 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.

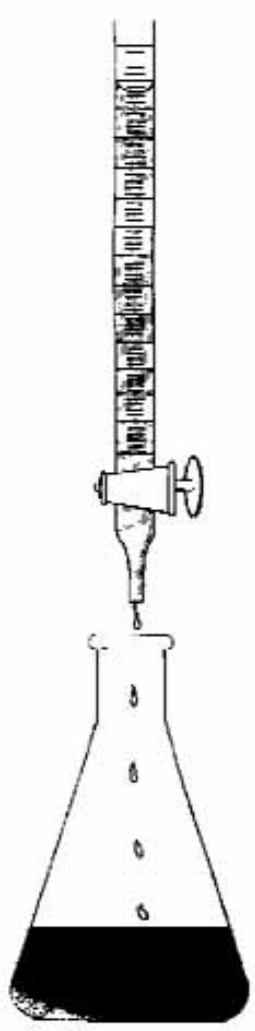

Figure 5.8

Titrating a DO sample using a buret

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.
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

Expected Range	Sample Volume	Titration Cartridge	Digit Multiplier
1-5 mg/L	200 mL	0.2 N	0.01

Table 5.4

Sample volume selection and

2-10 mg/L	100 mL	0.2 N	0.02	**corresponding values for Winkler titration**
10+ mg/L	200 mL	2.0 N	0.10	

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 nu mber 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 samize ts nm tg cc an1 nm tg cc3S4p6mt an1 nn

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 i s 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.

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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 de-grees Fahrenheit (F) or degrees Celsius (C).

Channel catfish	32 °C (90 °F)	35 °C (95 °F)	27 °C (81 °F)	29 °C (84 °F)
Largemouth bass	32 °C (90 °F)	34 °C (93 °F)	21 °C (70 °F)	27 °C (81 °F)
Rainbow trout	19 °C (66 °F)	24 °C (75 °F)	9 °C (48 °F)	

TASK 1 Prepare before leaving for the sampling site

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.

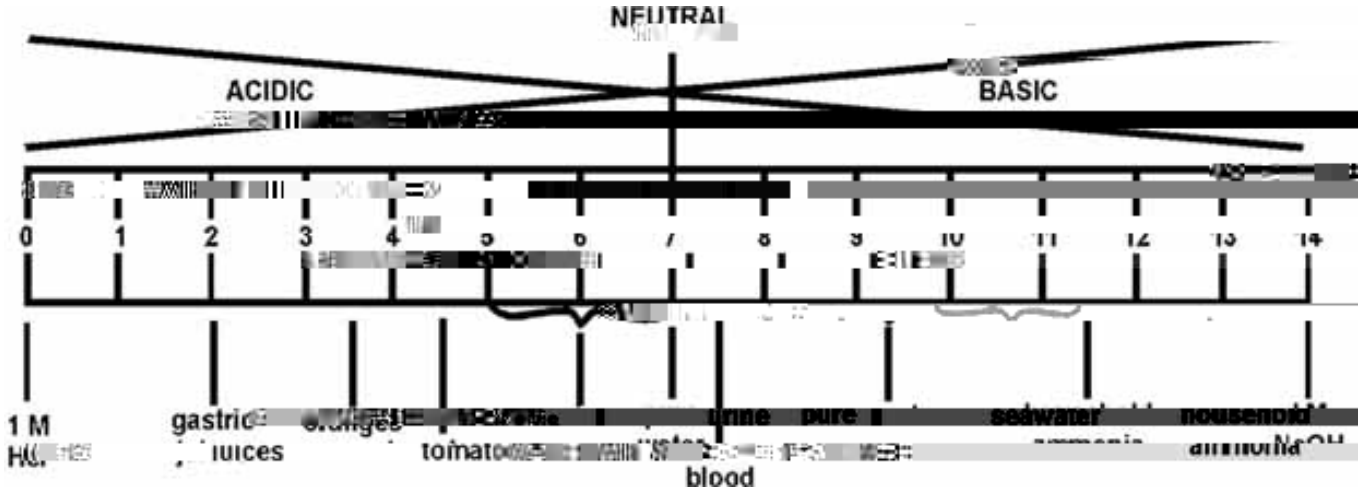

Figure 5.9

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

Analytical and equipment considerations

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

the water, which will bring the pH toward 7. If your program requires a high degree of accuracy and precision in

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 Task 2 in Chapter 5 - Water Quality Conditions 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.

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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 - Conductivity).

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 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

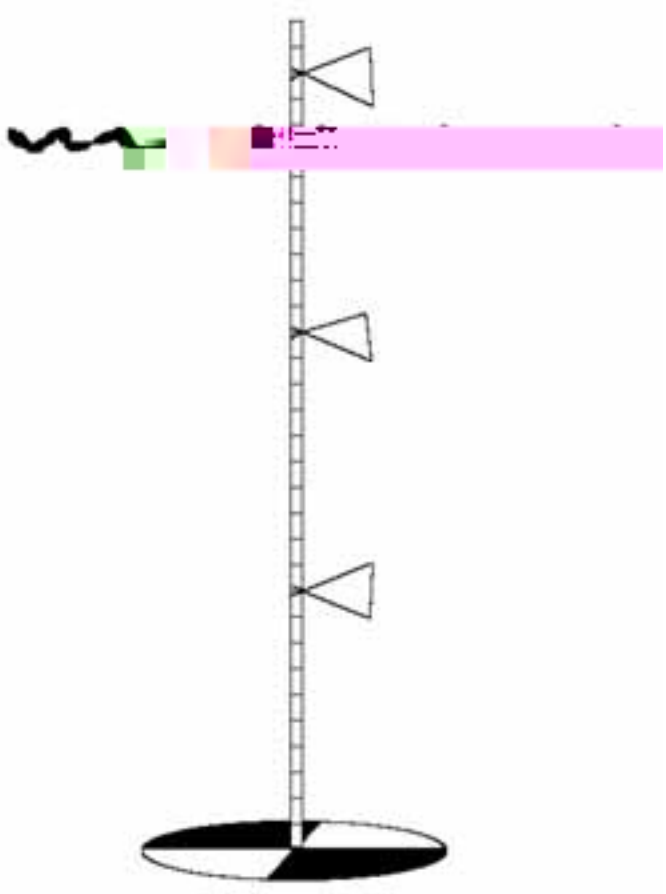

Figure 5.10

Using a Secchi disk to measure transparency

cleaned before the first run and after each sampling run by following Method A described in Chapter 5 - Water Quality Conditions.

TASK 2 Prepare before leaving for the sampling site

Refer to section 2.3 - Safety Considerations 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 Task 2 in Chapter 5 - Water Quality Conditions 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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Mitchell, M.K., and W. Stapp. *ed.*

Back to Section 5.5 - Turbidity

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:

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).

5.6
Phosphorus

Phosphorus changes form as it cycles through the aquatic environment.

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.

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

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 (PO4). 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 reporti gth s 1 Tf 1horu5Lhue ases are pecommoe4drolow conc do the aloric rccurate detea 7pc.7272 re ano c. F8571ostll (set for a wavers tha

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 x C) ö 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 x 25) ö 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 section 2.3 - Safety Considerations 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 Task 2 in the Introduction to Chapter 5 for details on how to collect water samples using screw-cap

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.

Results can either be reported "as P" or "as PO4." Remember that your results are reported as milligrams per liter weight per unit of volume. Since the PO4 molecule is three times as heavy as the P atom, results reported as PO4 are three times the concentration of those reported as P. For example, if you measure 0.06 mg/L as PO4, that's equivalent to 0.02 mg/L as P. To convert PO4 to P, divide by 3. To convert P to PO4, 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.

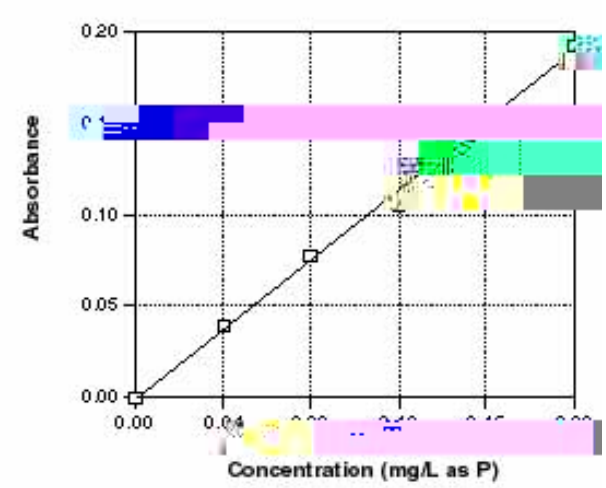

Figure 5.13

Absorbance of standard concentrations, when plotted, should result in a straight line

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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 (NH3), nitrates (NO3), and nitrites (NO2). 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 (NO2) and then to nitrate (NO3). Under such circumstances, it might be necessary to also monitor for nitrites or ammonia,

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 m e ooah a 0.4.Brator is thea 0.4n muld ster Aasgnn againsmBratoerr co 3ingalsocurvevar

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.

- 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 Task 2 in Chapter 5 - Water Quality Conditions 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.

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 (NO3-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

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.

6. 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 ñ 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.

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

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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

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 in Task 1 in Chapter 5 - Water Quality Conditions.

TASK 2 Prepare before leaving for the sampling site

Refer to section 2.3 - Safety Considerations 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 Task 2 in Chapter 5 - Water Quality Conditions for details on how to collect water samples using screw-cap bottles or Whirl-pak® bags.

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

TASK 2 Prepare before leaving for the sampling site

Refer to section 2.3 - Safety Considerations 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 Task 2 in Chapter 5 - Water Quality Conditions 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 solutio n.
3. Rinse the probe with distilled or deionized water.

   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 ex ceeds the range of the instrument, you may dilute the sample. Be sure to perforna8te

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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 and 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). Th e 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 t he 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 in Chapter 5 - Water Quality Conditions. Remember to wear latex gloves.

TASK 2 Prepare before leaving for the sampling site

Refer to section 2.3 - Safety Considerations 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:

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 cas 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.

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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 1 5 minutes using an autoclave before being used again for sampling.Consiparatnces

acan enlysinytyIf factory-rinsed and ster. Cr helsed lab,

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

that might inhibit bacteria growth.

Figure 5.6

Back to Section 5.1 - Stream Flow

Determining Average Cross-Sectional Area (A)

	Transect #1 (upstream)			Transect #2 (downstream)		
	Interal width (feet)	Depth (feet)		Interal width (feet)	Depth (feet)	
A to B	= 2.0	1.0	(at B)	= 2.5	1.1	(at B)
B to C	= 2.0	0.8	(at C)	= 2.5	1.0	(at C)
C to D	= 2.0	0.5	(at D)	= 2.5	0.4	(at D)
D to E	= 2.0	0.0	(shoreline)	= 2.5	0.0	(shoreline)
	----	----		----	----	
Totals	8.0	2.3		10.0	2.5	

Average depth = 2.3/4 = 0.575 feet

Average depth = 2.5/4 = 0.625 feet

Cross-sectional area of Transect #1

= Total width X Average depth
= 8.0 ft X 0.575
= 4.60 ft^2

Cross-sectional area of Transect #2

= Total width X Average depth
= 10.0 ft X 0.625
= 6.25 ft^2

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

= (4.60 ft^2 + 6.25 ft^2)/2
=5.42 ft^2

Figure 5.6

A sample calculation of average cross-sectional area

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.

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 ASAP 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.

Chapter 6
Managing and Presenting Monitoring Data

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.

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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 informatio n (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 f lag 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 foll owed.

- *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, an d 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 f indings 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 yo u 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 shoul d parallel the codes you use in your database. Ime yonrill he skharelomyckin your n

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 s ite 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 wor k 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.

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 r un on personal computers to sophisticated and very powerful systems that run on large mainframes. 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 data base, 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 a gencies, 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 because 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

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.

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 yaxis for a corresponding variable (such as sampling date or site) which is marked along the horizontal or xaxis. 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 xaxis.

Habitat scores as a percent of reference condition at sites #1 and #2 for 1992-1994

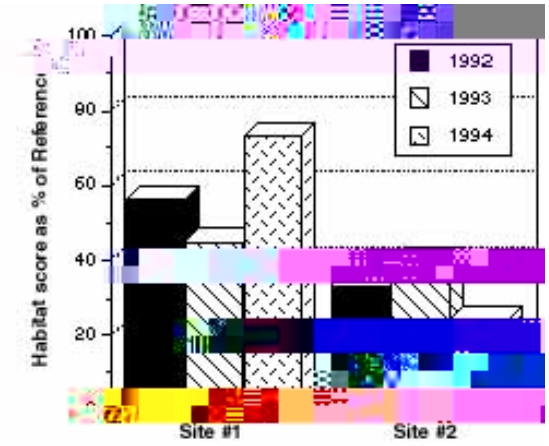

Figure 6.1

Example of a bar graph displaying biological data

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 p articular 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)

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 t he 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.
- *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.

June phosphorus concentrations at Sites #1 and #2 from 1991-1997

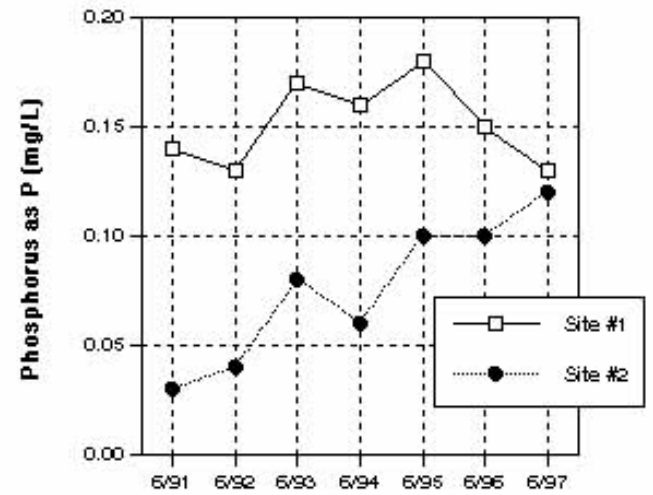

Figure 6.2

Example of a line graph depicting trends in phosphorus data

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 i deal 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

Summary of water quality ratings for Volunteer Creek

(total no. of stations=52)

Figure 6.3

statistic is a measure of location and in a normal curve marks the highest point at the center of the bell.

Example of a pie chart summarizing water quality ratings

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 oth er. 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 statistica lly 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 non-normally distributed, the mean

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 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 most of the United States.

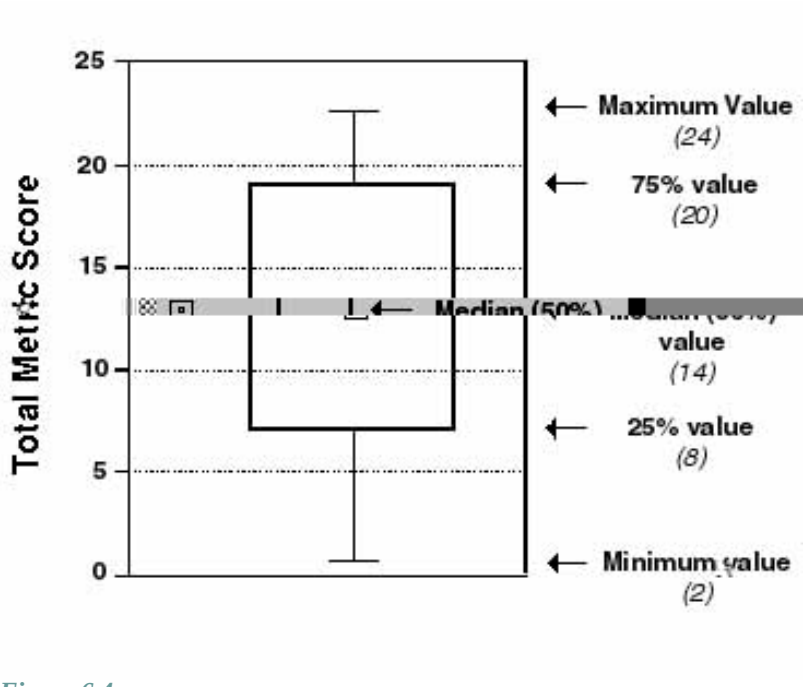

Figure 6.4

Example of a box plot

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).

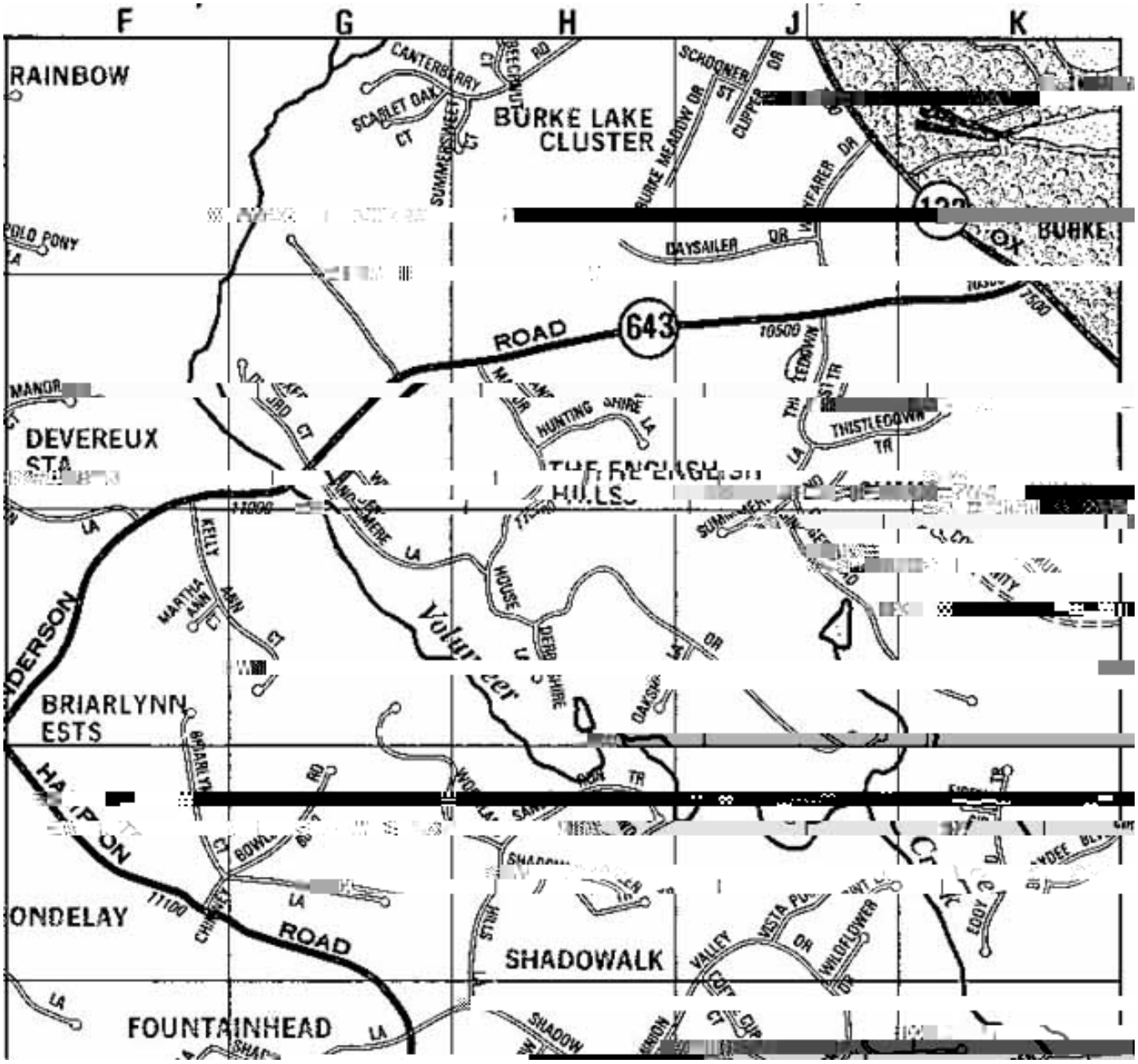

Figure 6.5

A road map is useful for displaying station locations

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 "blowup" or enlargement of the area elsewhere on the map to adequately display the information.

Monitoring Water Quality

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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Appendix A: Glossary

accuracy - a measure of how close repeated trials are to the desired target.

acidity - a measure of the number of free hydrogen ions (H^+) in a solution that can chemically react with other substances.

alkalinity - a measure of the negative ions that are available to react and neutralize free hydrogen ions. Some of most common of these include hydroxide (OH), sulfate (SO_4), phosphate (PO_4), bicarbonate (HCO_3) and carbonate (CO_3)

ambient - pertaining to the current environmental condition.

assemblage - the set of related organisms that represent a portion of a biological community (e.g., benthic macroinvertebrates).

benthic ambient

discharge from CSOs is typically untreated.

community - the whole of the plant and animal population inhabiting a given area.

culvert - man-made construction that diverts the natural flow of water.

dframe net - a fine mesh net that is attached to a pole and used for sampling. It resembles a butterfly net.

deionized water - water that has had all of the ions (atoms or molecules) other than hydrogen and oxygen removed.

designated uses - state-established desirable uses that waters should support, such as fishing, swimming, and aquatic life. Listed in state water quality standards.

dissolved oxygen (DO) - oxygen dissolved in water and available for living organisms to use for respiration.

distilled water - water that has had most of its impurities removed.

effluent - wastewater discharge.

dredge - to remove sediments from the stream bed to deepen or widen the channel.

ecoregion - geographic areas that are distinguished from others by ecological characteristics such as climate, soils, geology, and vegetation.

embeddedness - the degree to which rocks in the streambed are surrounded by sediment.

emergent plants - plants rooted underwater, but with their tops extending above the water.

Erlenmeyer flask - a flask having a wide bottom and a smaller neck and mouth that is used to mix liquids.

eutrophication - the natural and artificial addition of nutrients to a waterbody, which may lead to depleted oxygen concentrations. Eutrophication is a natural process that is frequently accelerated and intensified by human activities.

floating plants - plants that grow free floating, rather than being attached to the stream bed.

flocculent (floc) - a mass of particles that form into a clump as a result of a chemical reaction.

glide/run - section of a stream with a relatively high velocity and with little or no turbulence on the surface of the water.

graduated cylinder - a cylinder used to measure liquids that is marked in units.

gross morphological features - large obvious identifying physical characteristics of an organism.

headwaters - the origins of a stream.

hypoxia - depletion of dissolved oxygen in an aquatic system.

impairment - degradation.

impoundment - a body of water contained by a barrier, such as a dam.

inert - not chemically or physically active.

kick net - a fine mesh net used to collect organisms. Kick nets vary in size, but generally are about three feet long and are attached to two wooden poles at each end.

land uses - activities that take place on the land, such as construction, farming, or tree clearing.

macroinvertebrate - organisms that lack a backbone and can be seen with the naked eye.

NPDES- National Pollutant Discharge Elimination System, a national program in which pollution dischargers such as factories and sewage treatment plants are given permits to discharge. These permits contain limits on the pollutants they are allowed to discharge.

orthophosphate - inorganic phosphorus dissolved in water.

outfall - the pipe through which industrial facilities and wastewater treatment plants discharge their effluent (wastewater) into a waterbody.

permeable - porous.

pH - a numerical measure of the hydrogen ion concentration used to indicate the alkalinity or acidity of a substance. Measured on a scale of 1.0 (acidic) to 14.0 (basic); 7.0 is neutral.

phosphorus - a nutrient that is essential for plants and animals.

photosynthesis - the chemical reaction in plants that utilizes light energy from the sun to convert water and carbon dioxide into simple sugars. This reaction is facilitated by chlorophyll.

pipet - an eyedropper-like instrument that can measure very small amounts of a liquid.

pool - deeper portion of a stream where water flows slower than in neighboring, shallower portions.

precision - a measure of how close repeated trials are to each other.

protocol - defined procedure.

reagent - a substance or chemical used to indicate the presence of a chemical or to induce a chemical reaction to determine the chemical characteristics of a solution.

riffle - shallow area in a stream where water flows swiftly over gravel and rock.

riparian - of or pertaining to the banks of a body of water.

riparian zone - the vegetative area on each bank of a body of water.

riprap - rocks used on an embankment to protect against bank erosion.

run/glide - see glide/run.

saturated - inundated; filled to the point of capacity or beyond.

sheen - the glimmering effect that oil has on water as light is reflected more sharply off the surface.

sieve bucket - a bucket with a screen bottom that is used to wash macroinvertebrate samples and to remove excess silt and mud.

silviculture - forestry and the commercial farming of trees.

submergent plants - plants that live and grow fully submerged under the water.

substrate - refers to a surface. This includes the material comprising the stream bed or the surfaces to which plants or animals may attach or live upon.

taxon (plural taxa) - a level of classification within a scientific system that categorizes living organisms based on their physical characteristics.

taxonomic key

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Appendix B:
Scientific Supply Houses

This is a partial list of chemical and scientific equipment supply companies from which to purchase equipment for a volunteer monitoring program.

Aquatic Research Instruments
P.O. Box 2214
Seattle, WA 98111
(206) 789-0138
Water samplers, plankton nets, Surber samplers, Hess samplers, drift nets, calibrated lines, armored thermometers, BOD bottles.

Ben Meadows
3589 Broad Street
Atlanta, GA 30341
(800) 241-6401
Waders, rubber boots, field water test equipment, kick nets, dip nets, wash buckets, forceps.

Carolina Biological Supply Company
2700 York Court
Burlington, NC 272153398
(800) 3345551
Flexible arm magnifiers, hand lenses, forceps, kick nets, microscopes, reagents, educational materials, live and mounted specimens for instruction.

Cole Palmer Instruments, Inc.
625 East Bunker Court
Vernon Hills, IL 60061
(800) 323-4340
Lab equipment, field water test equipment, microscopes.

Chemetrics
Route 28
Calverton, VA 22016-0214
(800) 356-3072

Water testing mini-kits for field analysis of dissolved oxygen, nitrate, nitrite, ammonia, phosphates, chlorine, sulfur, manganese, etc.

Consolidated Plastics
8181 Darrow Road
Twinsburg, OH 44087
(800) 362-1000
Sampling trays, buckets, nalgene bottles, garbage bags, Whirl Paks ®.

Dazor Manufacturing Corp.
4483 Duncan Ave.
St. Louis, MO 63110
(800) 245-9103
Illuminated magnifiers.

Fisher Scientific
711 Forbes Ave.
Pittsburgh, PA 152194785
(800) 7667000
Lab equipment, sample bottles, sieves, reagents, incubators, water test equipment, Whirl Paks ®.

Hach Equipment Company
P.O. Box 329
Loveland, CO 80539-0389
(800) 227-4224
Field and lab water testing equipment, spectrophotometers, incubators, water sampling kits, fecal coliform sampling supplies, reagents, educational materials.

Hydrolab Corporation
P.O. Box 50116
Austin, TX 78763
(800) 949-3766
Water monitoring equipment and supplies.

LaMotte
P.O. Box 329
Chestertown, MD 21620
(800) 3443100
Water sampling kits, field and lab water testing equipment, Secchi disks, water samplers, armored thermometers, calibrated lines, plankton nets, kicknets, educational materials.

Lawrence Enterprises
P.O. Box 344
Seal Harbor, ME 04675
(207) 276-5746
Transparency tubes, view scopes, Secchi disks, water samplers, kick nets, sieve buckets.

Millipore Corporation
397 Williams Street
Marlborough, MA 01752
(800) 645-5476
Fecal coliform testing supplies (complete sterile water filtration system), membrane filters, sterile pipette, petri dishes, sterile media, other water sampling equipment and lab supplies, incubators, Whirl Paks ®.

Nalge Company
P.O. Box 20365
Rochester, NY 14602
Fecal coliform testing supplies, disposal fecal coliform filtration systems, membrane filters, sterile pipettes, petri dishes, incubators, Whirl Paks ®.

Nichols Net and Twine, Inc.
200 Highway 111
Granite City, IL 62040
(618) 797-0211
Kick nets.

Ohmicron
375 Pheasant Run
Newtown, PA 18940
(800) 544-8881
Immunoassay kits for pesticides, other contaminants.

Thomas Scientific Company
99 High Hill Road at I295
P.O. Box 99
Swedesboro, NJ 080850099
(609) 345-2100
Lab equipment, sample bottles, sieves, reagents, incubators, water test equipment, Whirl Paks ®.

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educational materials, live and mounted specimens for instruction.

Wildco Wildlife Supply Company
301 Cass Street
Saginaw, MI 48602
(517) 7998100
Kick nets, wash buckets, field biological sampling equipment.

YSI Incorporated
1725 Brannum Lane
Yellow Springs, Ohio 45387
(513) 7677241
Water quality monitoring equipment supplies.

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