Oregon Plan for Salmon and Watersheds

Water Quality Monitoring

Guidebook

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Understanding the status and trends in native fish populations and the stream and landscape conditions that affect them are essential to the success of the Oregon Plan for Salmon and Watersheds (OPSW). Having a standard tool that helps local groups, agency personnel and others determine these trends and conditions in a consistent and verifiable way is also essential. The use of standard monitoring techniques provides the public with such a tool.

The data collected through monitoring can be useful for developing plans to restore and protect a stream's biological capacity, as well as determining whether completed restoration projects achieved their intended goals. Watershed councils and other local groups play a critical role in identifying the causes of decline in a stream's ability to support salmon and trout populations and other beneficial uses, as well as documenting results of restoration projects. The purpose of this guidebook is to provide technical guidance so watershed councils and other volunteers may achieve their restoration goals as partners in the OPSW.

Many different agencies, volunteer groups, and private citizens are involved in data collection, so having a consistent method is important. To assist in collecting consistent and accurate data, the OPSW Water Quality Monitoring Team has prepared guidelines to measure water quality. These guidelines are designed for use by individual landowners, watershed councils, other citizen groups, and agency personnel. These guidelines complement the GWEB Watershed Assessment Manual (NES, 1999).

The Oregon Watershed Assessment Manual provides a guide for characterizing conditions in local watersheds and provides a strong base for identifying specific restoration and protection opportunities and monitoring needs. The monitoring techniques, or "protocols," presented in this guide describe the steps used for obtaining specific, field-based data about water quality. The Watershed Assessment Manual serves as a broad diagnostic tool. The Water Quality Monitoring Guidebook is a verification tool that can be used to refine the public's understanding and diagnosis of watershed and water quality conditions.

The initial chapters provide background information, monitoring strategies and ways to develop a monitoring plan. Also explained in these chapters are criteria for selecting monitoring sites, data quality guidelines, and methods to store and analyze water quality data. References and contacts are provided in each chapter to obtain more detailed or up-to-date information. The subsequent chapters provide protocols for monitoring:

- *stream temperature*
- *dissolved oxygen*
- *pH*
- *conductivity*
- *nitrogen/phosphorus concentration*
- *turbidity*
- *macroinvertebrates*
- *pesticides and toxic chemicals*

Each of these protocol chapters is designed to be a stand-alone document that provides basic monitoring techniques for that protocol. Information on additional references is also provided in each chapter. How each individual, group, or agency works through these protocols will depend on their technical background, experience, and what results they hope to accomplish. However, these protocols work best when integrated with the water quality, physical habitat, watershed assessment, and other monitoring protocols developed as part of the OPSW. They may also be useful in assessing water quality in watersheds where Senate Bill 1010 plans, Total Maximum Daily Load (TMDL) assessments or source area assessments under the Safe Drinking Water Act are developed.

An additional benefit in following the manual's recommendations is providing credible data for a state-wide database. Techniques for calibrating instruments, selecting appropriate sites, and managing data are included in the guidebook and, if used, will help agency personnel develop such a database. The database would eventually support the OPSW's effort to restore and protect fish habitat and watershed health throughout Oregon. But the real value in using the monitoring techniques described in this manual is providing watershed councils and other local volunteers with reliable methods for monitoring water quality in nearby streams which they can then use to make their own assessments. Accurate monitoring data can help inform local decisions about how to best manage for fish and watersheds.

The participation of local citizens in this effort is essential. Correctly collected data is useful to landowners, concerned citizens, and agency personnel. Poorly collected data of unknown quality can result in loss of time and money. It is the intent of this guidebook to share data collection techniques that will help everyone work toward a solution to restore fish populations. While contacts for equipment manufacturers and models of instruments are discussed in this guidebook, these references do not constitute an endorsement of any product.

Credits

This set of protocols was developed by a Water Quality Monitoring Team formed during the OPSW Monitoring Plan Scoping Sessions (January 1997). The work group was made up of representatives from the United States Environmental Protection Agency (EPA), United States Bureau of Land Management (BLM), Oregon Department of Agriculture (ODA), Oregon Department of Environmental Quality (DEQ), Oregon Department of Forestry (ODF), National Council of the Paper Industry for Air and Stream Improvement (NCASI), Boise Cascade Corporation, and the Mid-Coast Watershed Council. Key contributors to these guidelines included: Dr. George Ice, Liz Dent, Jenny Walsh, Rick Hafele, Dave Wilkinson, Lana Brodziak, Larry Caton, Travis Hunt, Ellen Hammond, and Paul Measeles. The protocol relies heavily on protocols developed by the Oregon Department of Environmental Quality (DEQ 1996) and the Oregon Department of Forestry (ODF). Valuable review comments on earlier drafts were received from Ken Bierly, Dr. Bob Beschta, Dr Sherri Johnson, Dr.

Bill Braumworth, Dr. Alan Herlihy, Sue Mauger, Stephanie Gunckel, Kristopher Wright, Andrew Talabere, Geoffrey Habron, Christian Torgerson, Dana Hicks and others. Their recognition in no way indicates an endorsement of this guidebook.

WATER QUALITY MONITORING GUIDEBOOK

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Chapter 1

Background

Many factors influence the health of aquatic ecosystems and the plant and animal life that depend on them. These factors include physical habitat, riparian function, water quantity, watershed health, and **water quality**. This guidebook focuses on methods for monitoring water quality.

Monitoring involves a series of observations, measurements, or samples collected and analyzed over time. Water quality varies naturally with location and time. For example: the headwaters of streams at high elevation tend to be cooler than wide streams at lower elevations; solar radiation influences stream temperature fluctuations throughout the day; natural differences in climate and the riparian vegetative cover cause differences in stream temperature. Disturbances such as fires, windthrow or even debris torrents can influence stream temperature, turbidity, and other water quality parameters. Geology, geomorphology, and climate also influence water quality.

Pollution can be defined as the fouling or making unclean air or water which harms beneficial uses. Water pollution is generally characterized as originating from either "point" or "nonpoint" sources. Point source pollution is associated with a particular site on a stream and typically involves a known quantity and type of pollutant that can be controlled at the site. An example of point source pollution is effluent from a factory outlet (an end-of-pipe discharge) delivered directly to a stream. Point sources are regulated under the Clean Water Act with National Pollution Discharge Elimination System (NPDES) permits.

Nonpoint source pollution is more difficult to manage and monitor than point source pollution. Nonpoint source pollution typically results from multiple contaminant sources in the vicinity where water quality has been impaired. The volume or "load" from individual sources is difficult to measure and often water quality may not be degraded at the source site. Instead, the accumulated impacts of multiple sources of pollution can cause the water quality problem. An example of nonpoint source pollution is fine sediment deposition in a stream bed. The stream may flow through a new housing development, agricultural operations, and forested areas with roads. All of these activities contribute various quantities of sediment to the stream channel in addition to the natural level of sediment the stream contains.

Emphasis has increased on controlling nonpoint source pollution because water quality cannot be protected or restored by focusing on point sources alone. Monitoring is an essential component of this effort. The strategy for controlling nonpoint source pollution includes the development of Best Management Practices (BMPs) to achieve water quality criteria and meet non-degradation requirements.

BMPs are defined as practices selected by an agency that are practical and effective in reducing pollution from nonpoint sources to levels compatible with water quality goals. Once an agency's BMPs are approved by the state water quality regulatory agency, they may become a part of the water quality management plan (WQMP) for those landowners that implement them.

An approved WQMP includes descriptions of the actions or activities that will allow a landowner to achieve acceptable water quality. For example, the Oregon Department of

Environmental Quality (DEQ) approved the Oregon State Forest Practices Act as an acceptable BMP program. It is the responsibility of the Oregon Department of Forestry (ODF) to monitor effectiveness of In Oregon, agricultural activities in watersheds with water quality limited waterbodies can come under the provisions of Senate Bill 1010. This bill requires the Oregon Department of Agriculture (ODA) to help reduce water pollution from agricultural sources. Under the guidance of the ODA, local committees develop a WQMP for the agricultural portion of the basin.

Water quality standards have been developed under the leadership of DEQ and can be used in assessing the effectiveness of BMPs. Water quality standards involve three elements: 1) a narrative that explains what the goals of the standards are; 2) the numeric criteria; 3) and a non-degradation policy.

The numeric criteria are set to protect the most sensitive beneficial uses. These standards are available on the web at <http://waterquality.deq.state.or.us/wq/wqrules.htm>. The non-degradation policy dictates that if a stream has better water quality than the defined standards, that stream shall not be degraded to a lower standard (unless there are compelling reasons).

these BMPs in achieving water quality standards.

While there are a number of water quality parameters regulated by DEQ, this guidebook focuses on those that have the greatest impact on fish and fish habitat or are important in the listing of water quality limited streams (streams identified on DEQ's 303d list). Parameters for Total Maximum Daily Load (TMDL)[1] assessments, or parameters that are part of source area assessments for municipal water supplies are also included. These include stream temperature, dissolved oxygen, pH, conductivity, nitrogen and phosphorus, sediment, macroinvertebrates, and pesticides and toxins.

Standards for each of these parameters have been established in order to protect a stream's beneficial uses. These standards have been developed after lengthy public review and involvement and are based on the latest scientific knowledge.

[1] Total Maximum Daily Load (TMDLs) is a tool used to meet water quality standards in those streams that do not meet such standards. TMDLs are based on a scientific method that uses extensive water quality data to identify locations and times of water quality impairment and the sources and volumes (loads) of the contributing pollutants. The TMDL process is rigorous enough that it can be duplicated by other parties using the same techniques.

Chapter 2

Monitoring Strategy and Plan

A monitoring plan describes the monitoring strategy that will be used. It is developed before starting a monitoring project. A monitoring plan provides a guide for why, how, when, and where to monitor water quality parameters. The monitoring plan can be referred to throughout the course of a monitoring project to help maintain consistency and provide documentation to others.

Why Monitor?

Many reasons exist for monitoring water quality. Monitoring can be used to identify areas where water quality standards are not being met and resources such as salmon and trout are being impaired. Monitoring can also be used to identify the sources and loads of pollutants that are causing these declines. Once the areas and causes of these water quality problems have been identified, then monitoring can be used to measure the overall effectiveness of the water quality protection efforts and individual practices. Monitoring is also important when knowledge of the effects from past restoration treatments or past management practices are desired in order to help design future management actions. Resource managers need monitoring data to improve practices and to better protect fish and fish habitat. The monitoring process and the data generated can also provide a valuable educational tool for a wide variety of user groups, such as watershed councils, school groups, researchers, and other interested people.

Monitoring without a defined purpose provides little benefit, so the first step to ask is, "What are the goals of the monitoring effort?" Typically, specific questions need to be answered. The questions vary depending on the aquatic resource(s) of interest. For example, asking if the stream meets the DEQ water quality standards for temperature and dissolved oxygen, or whether the BMPs are effectively reducing sediment inputs to the stream channel, leads to different monitoring approaches. Questions such as these will help focus the monitoring efforts and give a better idea of where and for how long monitoring is needed. Begin by listing all relevant questions about the aquatic system. Priorities can then be established in their order of importance and a timetable for the necessary monitoring projects developed.

In general, monitoring projects may provide information to address historical, current, or desired future conditions. Monitoring projects can also describe ecological trends that may or may not result from the effects of management practices. Monitoring can also describe the impacts from management activities, as well as interpret the effectiveness of management actions such as BMPs. Additionally, some problems cannot be addressed through monitoring water quality parameters and may need a research approach. Monitoring can help identify these problem areas, as well. The OPSW Monitoring Team has developed a Monitoring Framework that depicts these areas of monitoring (Appendix A).

Types of Monitoring

Monitoring strategies may be organized by different monitoring types. The type chosen depends on the project's objectives. Refer to Appendix B for an in-depth discussion on monitoring types. Identifying the monitoring type is useful when coordinating with other monitoring efforts and understanding how to interpret and apply results. However, identifying the type of monitoring is not as important as identifying the important resource questions and properly preparing a monitoring plan to answer them.

The Monitoring Plan

A plan usually consists of a few important sections. By using this guide as a template and inserting site-specific needs and objectives, a monitoring plan can be developed for an individual stream or stream reach. Stating the problem definition, goals, and objectives at the beginning of the monitoring plan structures it so that a reliable set of data can be developed which answers the initial set of questions. Without a monitoring plan to collect data that answers specific questions about the watershed, the data collected could be of limited value.

Monitoring Plan sections include the following:

Problem definition

This section defines the problem. For example: *People are concerned that temperatures in Dry Creek exceed water quality standards and are harming fish.*

Goal

The goal states the purpose for monitoring. What information and/or analysis is anticipated from monitoring? For example: *The goal of this Plan is to determine if temperatures are exceeding water quality standards in Dry Creek and if management practices are contributing to elevated temperatures*.

Objectives

Objectives usually are structured in the form of a specific question. For example: *Are stream temperatures above the state water quality standard of 64°F and does irrigation withdrawal from Dry Creek result in downstream temperatures that exceed that standard?* The kind of questions asked will determine the type of monitoring and amount of resources required.

Hypotheses

Identifying the objective leads to creating an "experimental hypothesis" that tests whether a relationship exists between an action or activity and the water quality parameter of concern. The experimental hypothesis for the Dry Creek example could be: *Irrigation withdrawal from Dry Creek results in downstream temperatures that are greater than 64°F.* This experimental hypothesis leads to designing an experiment or monitoring project to resolve whether the experimental hypothesis can be confirmed or refuted. Simply monitoring temperatures at different stations in Dry Creek may not answer this question because it does not demonstrate why the temperature pattern occurs. Patterns that can be tied to a cause-and-effect response support experimental hypotheses more strongly.

In the Dry Creek example, one approach might be to stop water withdrawals during periods when maximum temperatures are occurring and compare stream temperature with periods when withdrawals occur. The null hypotheses (a statement that assumes no direct relationship

exists) for the experimental design could be: *There is no difference in the hours that Dry Creek exceeds 64°F for days with or without water withdrawal.*

Natural variations in the temperature response of Dry Creek will exist because no day is exactly the same as another, but the experimental and monitoring design can test whether the null hypothesis is accurate or not (assuming that the quality and variations of the data are within acceptable tolerances). As the importance of these questions increases, collecting high quality data and a sufficient number of samples (for statistical credibility) may be needed both to have confidence in whether this null hypothesis can be accepted or rejected and to minimize differences in interpreting results.

Site Description

This section describes the physical characteristics of the sampling site(s) and places the monitoring site in the context of other watershed sites. For example, channel gradient, elevation, vegetative cover, landuse, region, soils, and geology can be described. Providing stream reach locations using latitude and longitude allows comparisons to be made to data sampled nearby or in other areas with similar site conditions, using a geographic information system (GIS).

Data Gathering Strategy

This section describes the physical location, date and time of data gathering, the types of data to be gathered and minimum and optimum data needs. The locations of data sites should include consideration of ecoregion, stream network, or other variables depending on the scale of the question to be answered (see Chapter 3, *Selecting Sites*). The timing for gathering data should reflect the hydrologic processes suspected of influencing water quality. For example, if the data to be gathered is related to storm events, low flows, or other seasonal variables, these should be identified. The need for monthly, daily, hourly or continuous data gathering should be identified both to determine the level of effort or equipment necessary and to establish the level of confidence in the data.

Methods

This section describes the technical portion of the monitoring project. It explains to readers the data collection techniques used, equipment calibration and use (see pages 16-18), what types of data were collected, and when. The methods section essentially creates a contract regarding how the data will be gathered, what types of data will be collected, and how the equipment's accuracy will be maintained for those conducting the monitoring and for others who may be depending on the data.

Data Quality

Quality Assurance and Quality Control (QA/QC) are essential elements of any monitoring plan. They provide evidence that the data is accurate and precise enough to address the questions being asked. These elements are addressed in detail in Chapter 4.

Data Storage and Analysis

Thinking through this section is critical early in the monitoring process in order to have the support necessary to store, transport, or analyze the data. If the data are to be used with the OPSW, knowing how to transport the data to local watershed councils, DEQ offices, or other public data repositories in the agreed-upon format is important. DEQ has

developed a data storage template that can be used to format data records (See Chapter 5, *Data Storage and Analysis*).The monitoring team will also want to establish its own database for the streams it is monitoring. Planning ahead can save time, money, and avoid the agony of lost data.

Timetable and Staff Requirements

Each monitoring project will have a unique schedule of activities which must occur for it to be successful. Planning and implementing these activities take time. Figure 2-1 and Table 2-1 are provided as general examples of the sequencing of steps and time requirements for a temperature monitoring project.

Confidentiality and Landowner Permission/Relations

Obtaining prior permission from private landowners for monitoring sites that could be located on their property is essential. The OPSW is based on cooperation, so all monitoring efforts need to maintain good will with the affected landowners. Creating an agreement with the landowner about how the data collected on his/her property will be used and reported is also important. In some cases, specific locations may not be reported to maintain confidentiality. It is also useful to provide landowners with previews of information collected. They may have insights about the data and are often interested in using the data to adjust their management decisions.

Table 2-1. Estimated personnel time for a stream temperature monitoring project.

Activity	Hours
Plan development *	40 hours
Temperature recorder calibration	
Pre-deployment	4 hours/batch
Post-deployment	4 hours/batch
Field site selection **	hour/site + travel time
Unit placement installation	0.5 hours/unit + travel time
Field audits	0.25 hours/unit + travel time
Ancillary data collection	1-2 hours/unit + travel time
Unit retrieval	0.5 hours/unit + travel time
Download data	0.25 hours/unit
Data storage ***	0.25 hours/unit
Data analysis/interpretation ****	4-8 hours/site
Total:	Minimum of 60 hours per project plus 10-20 hours per each study site.

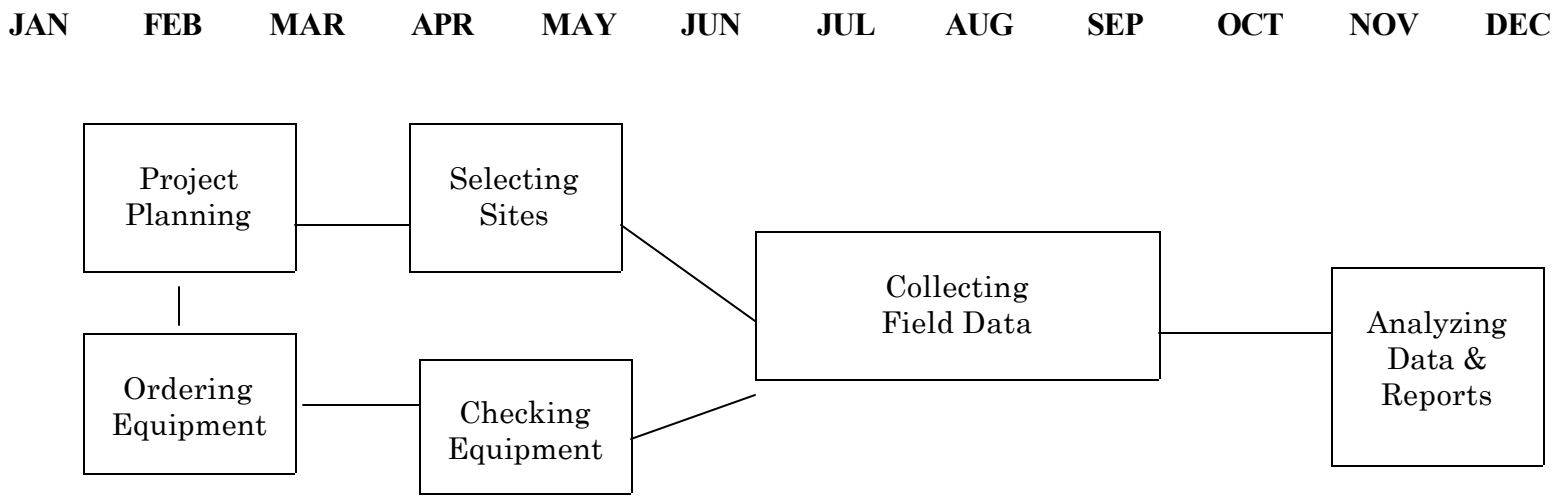

Figure 2-1. Stream temperature monitoring time line. The chart shown above depicts the steps one needs to complete during a typical season. Shaded boxes refer to steps which would normally be performed the first year and every succeeding year of a long-term study. Steps in unshaded boxes usually need to be completed only the first year of a long-term study.

* The time required to complete a plan will vary with the complexity of the project and experience of the personnel. Forty hours is a good estimate, but more or less time could easily be needed. The most important consideration is to allocate sufficient time to complete this step.

** Site selection begins with the project plan and preliminary identification of sites on maps. The field time involves walking planned study sites and finding a suitable location to install each temperature recorder.

*** Data storage can turn into a time draining task if it isn't planned at the beginning of the project. Determine the software to be used (one compatible with the temperature recorder's software), the data fields necessary, and the personnel responsible for both setting up the software and uploading the data. A suggested data format is shown in the data analysis section of this chapter and can be obtained from the cooperating state agencies (ODF & DEQ).

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**** Temperature recorders produce thousands of data points. The data must be summarized to provide a useful interpretation of the data. The time to complete this step will vary with the complexity of the project and level of experience of the personnel

This introduction to the basic structure of a monitoring plan is intended to help provide project volunteers with an understanding of a typical plan's components. As a plan is developed for a specific stream or stream reach, more detailed descriptions of the project's objectives will be needed. Please refer to the *Volunteer Monitor's Guide to Quality Assurance Project Plans* (1996) by EPA, the *National Handbook of Water Quality Monitoring* (1996) by NRCS, and other monitoring guides (Callaham 1990; Dissmeyer 1994; and MacDonald, Smart, and Wissmar 1991) for further help. For help or assistance at this stage, contact the monitoring mentor for the OPSW shown in each protocol chapter, the local ODFW office, or the regional DEQ monitoring coordinator shown below.

Statewide DEQ Volunteer Monitoring Coordinator: Karen Williams: (503) 229-5983 Email: williams.karen@deq.state.or.us Northwest Region: Larry Caton: (503) 229-5983. Email: caton.larry@deq.state.or.us	Western Region: Dennis Ades, (503) 229-5983 Email: ades.dennis@deq.state.or.us Eastern Region: Larry Marxer, (503) 229-5983 Email: marxer.larry@deq.state.or.us

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Chapter 3

Selecting Sites

Selecting the appropriate site or sites for monitoring water quality depends on the desired objectives. There are three geographic scales to consider in selecting the appropriate monitoring site: (1) the *sample point* provides representative[2] data at that spot, (2) the *reach approach* uses multiple sites to reflect conditions and trends for a segment of stream, and (3) the *basin scale* uses multiple reaches to reflect conditions and trends throughout a watershed.

In addition to the "scientific" considerations for monitoring sites (e.g. using standard data gathering techniques for consistency, maintaining data quality, etc.), there are also "practical" considerations. Easy access (such as road crossings) and landowner permission are two of these practical considerations. "Sampling stations should be accessible for all flow conditions that will be sampled" is a good working rule when selecting sites (Stednick 1991). If equipment is being installed for a long period of time, recognize that flow will change throughout the year. Equipment that was not designed to be submerged can be flooded. Conversely, equipment that needs to be submerged can be left "high and dry".

Precautions against vandalism, theft, and accidental disturbance should be considered when locating equipment. In areas frequented by the public, securing or camouflaging equipment is advisable. Visible tethers and equipment stations are not advisable since they attract attention. When equipment cannot be protected from disturbance, an alternative monitoring site should be considered. Access to electrical power can also be a consideration for some equipment.

Sample Point Considerations

The simplest and most specific geographic scale is a sampling point. Here, focus should be on selecting a location that will result in the most representative measure of the water quality parameter at that site.

When selecting a sample point, remember that if samples are collected where emerging groundwater or isolated eddies exist, the data will not represent the main portion of the stream. In order to collect representative data, sampling site selection must minimize the influence of potential confounding factors. Some examples of confounding factors include:

- the confluence of tributaries
- groundwater inflows
- channel structure or "morphology" (particularly conditions that create isolated segments or pools)
- springs, wetlands, water withdrawals, effluent discharges
- beaver ponds and other impoundments

By sampling in a section of a stream channel with good water mixing, the data will represent the site's average water quality condition. However, special cases can exist where monitoring should include sites containing these confounding factors. In these cases the objective of the monitoring may be to determine their influence on overall water quality.

[2] "Representative data" refers to the degree to which the data represents the actual environmental conditions at the time of monitoring. In this case, it should reflect the water quality integrated across and through the water column and not isolated elements.

Reach Scale

A monitoring project can be expanded to document water quality trends of a stream reach and/or effects of management practices on those trends. This is accomplished by monitoring the water quality parameter at multiple sample points. If the objective is to understand management impacts on water quality, or water quality treatment effects, then the most powerful and meaningful monitoring design will include a pre-project, or "baseline," data collection period.

For example, if the objective is to determine how a logging operation affects stream temperature, then multiple sample points will be needed. Ideally, these should be established prior to the logging activity over the same portion of the year when post-logging conditions will be monitored. Two, or preferably three, sample points should be placed slightly upstream and one slightly downstream from the harvest unit (Figure 3-1, points 2 and 3).

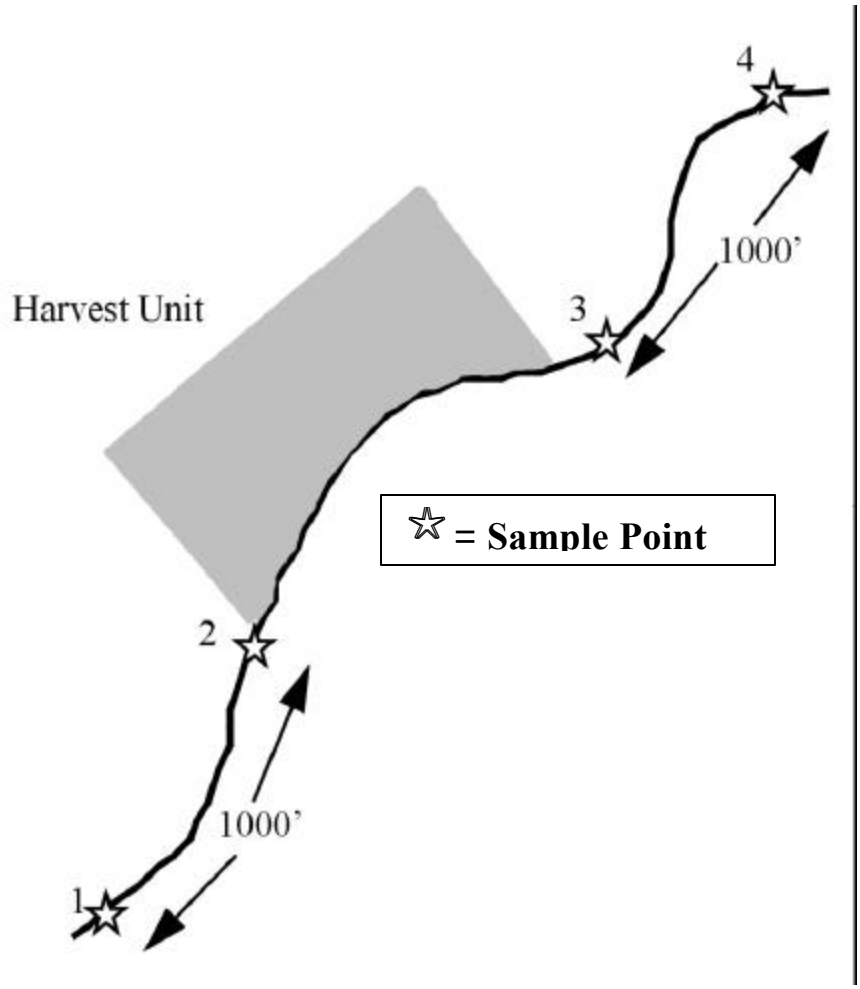

Figure 3-1. Sample point and reach-scale locations.

Furthermore, in order to understand the observed trends (e.g. any measured change in temperature) through the unit, sample points around "control" reaches will be needed. A control is designed to measure the parameter of concern at sites that are not impacted by management or other effects. These control sites are designed to help isolate the management or other effects from trends that may occur regardless of management or other impacts. In figure 2-1, the reaches between points 3 and 4 and between 1 and 2 can act as controls. If these reaches have intact riparian areas, then observed temperature trends through the harvest unit can be compared to these "control" reaches. These reaches should be located upstream and downstream of the harvest unit. *It is critical to recognize that without pre-treatment data, inferences about management effects can be weak*.

Many documents and protocols recommend establishing a "reference reach" to help provide comparisons and context between the stream reach of concern and a similar stream reach with less intensively managed conditions. (Dissmeyer 1994). Stream and riparian conditions for reference reaches represent the best available conditions. The reference reach for a forested area would most likely have good water quality, complex fish habitat, high quality spawning gravels, shade, cover, and rearing habitat for salmonids, ample large woody debris in the stream, and future supplies from the upstream adjacent riparian areas. In some cases, the reference stream is the "least impacted" reach available for monitoring (Plotnikoff 1992).

However, limitations to the reference-reach approach exist. For instance, a wide range of conditions result from "natural" disturbances. Fire, floods, and windstorms can cause major changes in streams and water quality. The occurrence or lack of occurrence of one of these events shapes stream characteristics. Therefore, caution is needed when comparing stream reaches with different disturbance histories. In addition, not all stream ecosystems *should* look alike. An estuary-influenced reach will not look like a headwater stream, and a high gradient, forested reach will not look like a meadow-dominated, low-gradient stream (see Oregon Watershed Assessment Manual discussion of channel habitat types).

Basin Scale Considerations

At the basin scale, landscape and stream patterns become the focus of monitoring. Basin-scale monitoring represents the major dilemma facing any sampling project—it is impossible to monitor everything, everywhere, all the time. While every location and stream reach in a watershed is unique, general patterns can be identified that help in understanding and managing watersheds. "Watershed analysis" is a process that resource professionals use based on identifying these patterns in the landscape and streams (NonPoint Source Solutions, 1999). This analysis involves developing hypotheses about how the watershed conditions and management activities on the landscape are linked to the riparian and stream response. Good basin-scale monitoring involves recognizing these linkages and developing monitoring that can be extended from a few sites to a more general representation of the watershed response.

A basin approach is more than merely a strung-together series of sites or reach-level monitoring activities. A limited number of monitoring sites must be identified whose information can represent conditions across the entire

watershed. Stratifying the basin into similar environmental and land-use conditions is one way of identifying candidate monitoring sites. Defining the basin by "ecoregion" is another classification that can be useful in identifying where factors such as geology or climate are relatively uniform. (Ecoregions are areas of relative ecosystem homogeneity containing essentially similar characteristics such as vegetation, geology, hydrology, soils, and climate).

Basin-scale monitoring programs should also consider the most sensitive or critical sites, both for sources of pollutant loads and water quality impacts. For example, roads built near streams on slopes with a high risk of landslides represent a potential source of sediment. Critical stream reaches, such as high value spawning or rearing habitat for salmon, may be identified as sensitive to sediment deposition. Again, these sites may have a high priority for monitoring to understand the watershed response.

An example of the value of basin-wide monitoring compared to an assessment from individual sampling points is a study of temperature patterns in the Steamboat Creek Watershed of Oregon by Holaday (1992). Holaday found that despite the recovery of riparian vegetation in Steamboat Creek from 1969 to 1990, no measurable change in the stream temperatures at the *mouth* of Steamboat Creek during summer extremes had occurred. Yet water temperature reductions of 1° to 11°F were measured for every major tributary to Steamboat Creek. The watershed-wide pattern, showing that increased shade was reducing maximum tributary temperatures, was clear. However, if temperature measurements at the mouth of Steamboat Creek were the only measurement taken, then it would appear that water temperature had not improved. Including tributary temperatures in the monitoring project more accurately reflected the watershed-wide temperature pattern..

Choosing Sites

Several types of sites may be selected for monitoring surveys:

- *Study sites* are selected to answer specific questions. These could include questions about the effects of certain land uses, improvement following restoration work, or the effectiveness of Best Management Practices, among others.
- *Reference sites* reflect the best available conditions present within a specific stream, watershed basin or ecoregion. An ideal reference site would be in a pristine, natural condition. A realistic reference site usually represents the best attainable conditions and has experienced some level of human effect. Ideally more than one reference site is used. Five to ten reference sites should be sampled for studies that include several streams over a range of habitats.
- *Randomly selected sites* are chosen completely at random, without regard to the level of human disturbance. In most cases, random sites are grouped, or stratified, according to certain factors such as stream order, land use, or ecoregion. Random site selection provides an unbiased assessment of the range of conditions present within a study area. (*Note: In Oregon, the EPA Research Lab in Corvallis can provide a list of randomly selected sites for specific projects. Contact Phil Larson at 541-754-4362.*)

Once potential sites have been identified, the actual locations where data will be collected need to be identified. Except for random sites, which are picked independent of other factors, sample sites should be representative of the larger study area. Physical and geographic

characteristics like vegetation, soils, geology, land use, gradient, riparian characteristics, and substrate type need to be considered to assure that sample sites are representative of the larger population. For example, sample sites should not be directly downstream from anomalies such as culverts, bridges, roads, landslides, or waterfalls unless these are the conditions that the monitoring program is evaluating.

Reference and study streams should be in the same ecoregion or ecologically similar area (watershed or basin) and be within an acceptable range of elevation, gradient, and stream order (Gallant, et al 1989). Similar streams in the same ecoregion would be expected to have similar water chemistry and habitat conditions, and support similar biological communities. Differences between well chosen reference and study sites should be due to human or natural disturbance and not due to natural differences between the streams.

Locating minimally impacted reference streams in the same ecoregion can sometimes prove difficult, especially at the lower elevation sections of streams. In cases where unimpaired reference sites are not available, one should select the least impaired areas possible. Generally, impacted and reference site selection is done in three stages:

- *Office Reconnaissance*: using maps, aerial photos, published reports, and other materials, the monitoring area is studied and likely streams are identified.
- *Consult the Experts*: federal and state resource management agency personnel are very knowledgeable of the natural characteristics and human impacts in the areas they administer. They can also provide information on work planned for the future in the basins being considered for study, such as proposed timber sales or stream improvement work. Local fisheries biologists are a particularly good resource.
- *Field Reconnaissance*: the streams identified in the previous two steps are visited and visually surveyed to verify the representation and similarity of the streams and to select specific stream reaches for sampling.

How Many Sites Per Stream?

The location and number of sites per stream depends on the objectives of the study, the type of impacts, and the resources available. Generally, program designs are of three types:

1) *Paired stream approach*, with several sites per stream. A study stream is paired with a nearby unimpacted (or *least* impacted) reference stream where several sites are also selected.
2) *Upstream/downstream approach*, with several sites along a single stream. Selected sites upstream of some disturbance, with the best available conditions, are used as the reference sites. Sites are then selected within and/or downstream from the area of concern.
3) *Ecoregion approach*. A number of least impacted reference sites within a single physiographic type or ecoregion are selected to determine the natural reference condition. A number of sites of concern are then selected within the same or a similar ecoregion.

Whichever approach is used, it is important to sample enough sites to determine the inherent variability within and between different sites, because water quality parameters vary in both space and time. Gathering additional data collected by other agencies or groups can improve the effectiveness of monitoring to detect differences between sites. The collection and analysis methods used by other studies need to be comparable, however.

References

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Chapter 4

Data Quality

Background

The goal of data gathering is to produce data of a known quality which is adequate for the intended use. Environmental monitoring often requires large investments of resources. Instituting techniques which protect that investment and insure that the data is valuable to other users is important.

The methods used to eliminate flaws and errors before they compromise the quality of the data collected are generally referred to as "quality assurance" (see next page). To insure that the data are credible, procedures must be documented, regular evaluations of precision and accuracy should be conducted, and regular, independent audits should also be conducted.

10 Steps To Quality Data

Proper planning is the key to producing high quality data. The ten steps described below are useful whether a project will sample two sites on a small creek or 200 sites in a statewide monitoring network.

1. Define the goals and objectives of the project. Why is the project needed? What question is being addressed? How will the data be used? Who will use the data?
2. Collect background information about the project area.
3. Refine the project goals based on the background information gathered.
4. Design the project's sampling, analytical, and data requirements. This is the "what, how, when, and where" of sampling.
5. Write an implementation plan that describes when tasks will be completed and who will complete them.
6. Write a draft project plan that includes sampling methods and project objectives.
7. Get feedback on the draft plan from other professionals such as state agency monitoring staff.
8. Revise the project plan based on review comments.
9. Implement monitoring work as described in the final monitoring plan.
10. Evaluate and refine the project over time as knowledge is acquired during the project.

Key Data Quality Concepts

Quality assurance (QA) and quality control (QC) are key components of any monitoring program. They are defined as:

<u>Quality Assurance</u>
The overall management system of a project including the organization, planning, data collection, quality control (QC), documentation, evaluation, and reporting activities. QA provides the information needed to determine the data's quality and whether it meets the project's requirements.

<u>Quality Control</u>
The routine technical activities intended primarily to control errors. Since errors can occur in either the field, the laboratory, or in the office, QC must be part of each of these activities.

As part of QA/QC planning, certain data quality objectives need to be defined. These

relate to *the precision, accuracy, representation, completeness*, and *comparability* of the data.

Precision

Precision refers to the amount of agreement among repeated measurements of the same parameter. To determine precision, duplicate samples must be collected at a number of sample sites. As an example, volunteers may wish to collect one duplicate sample per trip or duplicate samples for 10% of the total samples collected, whichever is greater. Duplicate samples should be collected during each sampling trip. The actual number of duplicates depends on the variability of the data and how precise the data must be to estimate the actual water quality (EPA 1996).

Accuracy

Accuracy measures how close the results are to a true or expected value. This is normally determined by measuring a standard or reference sample of a known amount and comparing how far the results at the monitoring site are from the reference value.

Representation

To what extent do the field samples actually represent the true environmental condition(s) or population(s) at the time a sample was collected? Representation is largely determined by the selection of the sample sites. Do these sites accurately reflect (or represent) the conditions of the waterbody being studied?

Completeness

The comparison between the amount of valid, or usable, data originally planned for collection, versus the amount actually collected.

Comparability

The degree to which different methods and data sets agree or are similar. For instance, the Winkler titration method for dissolved oxygen (a method for measuring the concentration of dissolved oxygen in water. See Chapter 7) and a polarographic probe (a different method for measuring dissolved oxygen) may not provide highly comparable data. This is particularly important to determine when using data from other studies.

The level of accuracy and precision will not be the same for each parameter measured, and may not be the same for each project. Precision and accuracy will depend on the study objectives (i.e. how precise and accurate the data must be to answer the questions of concern), the amount of money available for equipment purchases and data analysis, and the level of training of the people collecting samples. The original data quality objectives may not be met in a monitoring project because funding can be cut (reducing the level of analysis), the equipment fails, or project personnel don't perform as expected. If this occurs it is critical to report the data quality level attained and explain why.

Data Quality Matrix

Determining the level of accuracy and precision desired at the project's beginning is important. Table 4-1 has been developed to help determine the data quality objectives. The table identifies three data quality levels for six commonly collected water quality parameters. The purpose of the water quality data matrix is to help collectors select the level of data quality that meets their objectives, experience, level of expertise, and budget. Data quality levels depend on the methods used and the QA/QC protocol followed.

Level A

Level A is the highest level of data quality. It can be used to assess compliance with water quality standards, permitting requirements, or other regulatory activities.

Level B

Level B is the next highest level. It is typically easier and less expensive to collect. Level B

data can be used as an early warning of potential problems or for screening information.

Level C

Level C is the lowest data quality level and is normally the easiest to collect. Because of its lower accuracy and precision, Level C data is best used for educational purposes.

Not all field parameters will need to be at Level A, or even Level B, data quality. A principal decision for data collectors is to decide how the data will be used.

Depending on the data collection objectives, equipment available, collector training and adherence to QA/QC procedures, data quality levels may vary for different parameters. The procedures and instruments described in the specific protocol chapters are generally designed to meet Level A data quality with appropriate QA/QC. But, remember what the data will be used for and determine what is the appropriate data quality level.

References

USEPA. 1996. *The volunteer monitor's guide to quality assurance project plans.* EPA 841-B-96-003. U.S. Environmental Protection Agency, Office of Wetlands, Oceans, and Watersheds: Washington, D.C. Chapter 2.

Table 4-1. Water quality parameters by data quality level. Data quality level depends on a combination of quality control and method selection.

DATA QUALITY MATRIX
Water Quality Parameters by Data Quality Level
Data Quality Level depends on a combination of quality control and method selection.

Data Quality Level	Quality Assurance Plan	Water Temperature Methods	PH Methods	Dissolved Oxygen Methods	Turbidity Methods	Conductivity Methods	E. coli Bacteria Methods	Potential Data Uses
A	QAPP approved *QA criteria met.*	Thermometer or datalogger. Accuracy checked with NIST standard. A=+/-0.5 C P=+/-1.0 C	Calibrated pH Electrode A=+/-0.2 P=+/-0.3	Winkler Titration or calibrated Oxygen Meter A=+/-0.3mg/1 P=+/-0.5	Mephlometric Turbidity Meter A=+/-5% of std. value. P=+/-5%	Meter. Temperature correction to 25C. A=+/-7% of std. value. P=+/-2%	DEQ Approved Methods Split Sample P=+/-0.5 log	Regulatory. Permitting. Compliance with water quality standards.
B	Meets DEQ Data Acceptance Criteria	Thermometer or datalogger on NIST accuracy check. A=+/-2.0 C P=+/-1.0 C	Any method with: A=+/-0.5 P=+/-0.5	Winkler Titration or calibrated Oxygen Meter A=+/-1 mg/1 P=+/-1 mg/l	Any method with: A=+/-30% P=+/-30%	Meter. Temperature correction to 25C. A=+/-10% Of std. Value. P=+/-5%	DEQ Approved Methods Split Sample P>+/-0.5 log	Screening level info. – Red flag or early warning
C	Meets DEQ Data Acceptance Criteria	Un-calibrated thermometer	Any method +/- 1 pH unit	Any method +/-> 1 mg/1	Observations clear, muddy, etc.	Meter without routine calibration.	Presence – Absence test kits	Education

NOTE: In "Methods" boxes, A = Accuracy and P = Precision

Chapter 5

Data Storage And Analysis

Chapter 4 emphasized the importance of insuring data quality. If the steps described in Chapter 4 have been taken to achieve the desired level of data quality, then the method of storing and analyzing that data is equally important. Data properly stored and analyzed is essential if the goal is to gather credible data for use by volunteers, landowners and agency personnel for monitoring, management or regulatory purposes

Further, the level of precision and accuracy desired (see Table 4-1, Chapter 4) will influence the ability to detect meaningful differences in the data. For example, if a calibrated thermometer is used in temperature monitoring with a precision of ±1 degree, then it will not be useful in detecting temperature changes of 0.5 degree (the criteria for Level A accuracy). Data collectors, therefore, need to be aware of the level of data quality they want to achieve as they develop their monitoring plan, purchase or acquire equipment, and analyze the data.

Data should be stored and backed up on both the computer hard drive and disks. Data files should be clearly labeled for quick identification of what the file contains.

What basic data should be included in files will vary depending on the water quality parameter. In general, include the sampling point name and number, latitude and longitude of the site, stream name, and when the data was collected by date, month and year. Attempting to remember the particulars about how data was collected months later can be difficult; therefore, enter the data as soon as possible. Some of the equipment used in the following protocols (temperature monitoring probes) may actually create data files. It is important to make sure that the dates and times recorded in those files are correct.

Data Analysis

Making generalized comments about data analysis is difficult because such analyses will vary greatly depending on the particular question(s) asked and what parameters are measured. Different levels of analysis can be appropriate for most parameters.

Graphical Techniques

Graphing data is very useful and important for understanding the characteristics of the "data set" (i.e. the total amount of data collected for a particular monitoring site or project) and identifying any potential relationships. Examples include bar charts, XY graphs, frequency distributions, or pie charts.

For example, by graphing stream temperature versus distance from a divide, an understanding of basin trends can develop. By graphing stream temperature versus time, an understanding of when the highest temperatures occurred can be gained. This also provides a means to check the data for accuracy.

Descriptive Statistics

These are the very basic statistics that describe a data set (for more information on statistical analysis, refer to the monitoring mentors listed on page 7 in Chapter 2). Commonly reported statistics are: median, average, maximum, minimum, and standard deviation. By graphing the average plus and minus the standard deviation, data collectors begin to understand the distribution of their data.

Statistical Methods

The presentation of data in a valid scientific manner requires that a statement of the investigator's confidence in that data be included. Statistical methods are the tools used to show what levels of confidence, or the amount of error, investigators have in the data. A number of statistical methods or models are available for analyzing data. However, it is critical to understand the assumptions of these models prior to using them. For example, many natural resource data sets may not be normally distributed (i.e. the sets don't reflect a normal "bell shaped" curve on a graph) and therefore standard analytical methods may result in analyses that are flawed. These problems can often be addressed by logarithmic or power transformations of the data. Non-parametric methods are also available (Hirsch et al. 1992). Some statistical analyses include: ANOVA, multiple and linear regression, multivariate analyses, and correlation analyses. Some user-friendly software packages are available to aid statistical analyses. Without familiarity or training in statistical analysis, however, help in developing statistical models will be needed. Contact one of the regional monitoring coordinators listed on page 7 in Chapter 2 for further assistance.

Water Quality Criteria

Oregon water quality criteria are provided on the web at <http://waterquality.deq.state.or.us/wq/wqrules/wqrules.html>. These criteria may be in terms of a seven-day moving average of the daily maximum or minimum temperatures. Special conditions may also be recognized which naturally cause water quality to exceed the standards. For example extreme low streamflows or prolonged warm periods can cause streams to exceed state temperature standards. It is useful to analyze the data collected and compare the results to the water quality criteria.

- Maximums, minimums, averages

Depositing Data

The OPSW Monitoring Team is currently exploring options for storage of the monitoring data collected for the OPSW. Some of the attached protocols contain example data sheets. These sheets provide a template for organizing the data collected by volunteers into a format compatible with the OPSW database. In general, some important components include:

- Global Positioning Data Point or latitude and longitude
- Date of data collection
- Stream name
- Responsible party
- Project objective
- Site description
- Monitoring question
- Parameters measured

.

These protocols will conform to the recommendations for data storage that are being developed and will, in the future, provide guidelines for transporting and delivering the data to the OPSW database. At a minimum, guidelines for the data storage format will be developed. Those wishing for additional information on progress with data storage issues should contact Kelly Moore with the Oregon Department of Fish and Wildlife (541-737-7623)

References

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Chapter 6

Stream Temperature Protocol

Background

Water temperature is a key factor affecting the growth and survival of all aquatic organisms. The effect of stream temperature on fish, amphibians, macroinvertebrates, etc. varies between species and within the life cycle of a given species (Armour 1991; Beschta et al. 1987; Bjornn and Reiser 1991; Lantz 1971; DEQ 1995). Preferred temperature ranges for major fish species and their particular life stages are shown in Table 6-1.

Increases in stream temperature cause an increase in an organism's metabolic rate (Warren 1971). If enough food is available, growth rates can actually increase with some increase in temperature. For salmonids, temperature ranges of 40-66°F support healthy growth. Outside this temperature range, salmon and trout generally don't grow in size, and extreme temperatures can be lethal. Research has found that elevated stream temperatures often result in increased competition for a limited food supply, with young salmonids forced into habitat areas where they are easier prey (Reeves, Everest and Hall 1987). As food availability goes down, so does the growth rate. In addition, elevated stream temperatures increase the risk of disease-related mortality.

As stream temperatures increase, the amount of dissolved oxygen (DO)[3] available to aquatic biota decreases. As a result, even if food is abundant at higher temperatures, decreases in DO may metabolically stress salmonids, further increasing their susceptibility to disease.

When temperatures reach stressful levels, pockets of cool water provide "refugia" for fish and amphibian species that are sensitive to high stream temperature. Cool water refugia can sustain populations of sensitive species (Sedell et al. 1990). Cool water habitat can be sustained in deep pools, cold springs, areas of groundwater inflow, and at the junction of cooler tributary streams.

Stream temperature has been heavily researched and monitored (DEQ 1996; Dissmeyer 1994). Studies have investigated the effects of land management on stream temperature, developed models to predict stream temperature, and evaluated the effects of elevated temperature on aquatic biota. What follows is a detailed description of how to monitor stream temperature at multiple scales. *Please refer to previous chapters and Appendix B for information on developing a monitoring plan, selecting sites, and storing data.*

[3] The term "dissolved oxygen" (DO) refers to the amount of oxygen that is dissolved in water at a given temperature and atmospheric pressure. The amount of dissolved oxygen available in the stream is important for the respiratory and other metabolic functions of water borne organisms.

Table 6-1. Optimum and lethal limit temperature ranges for coho, chinook, and bull trout.

Fish Species	DEQ Standard	Preferred Juvenile Temperature Range	Adult migration, holding, or spawning	Lethal Limit
Coho	64°F	54-57°F	45-60°F	77°F
Chinook	64°F	50-60°F	46-55°F	77°F
Bull Trout	50 °F	39-50°F	39-59°F	---------

Mentors

As with any monitoring project, questions will come up that are not answered or covered sufficiently in this protocol. Therefore, a group of mentors that are agency experts in monitoring have been identified. These mentors may be contacted with specific questions about a particular monitoring effort.

Statewide DEQ Volunteer Monitoring Coordinator
Karen Williams: (503) 229-5983
E-mail: williams.karen@deq.state.or.us

North Coast
Larry Caton (503)229-5983;
E-mail: caton.larry@deq.state.or.us

South Coast & Willamette
Dennis Ades (503) 229-5983;
Email: ades.dennis@deq.state.or.us

Eastern Oregon
Larry Marxer (503) 229-5983;
E-mail: marxer.larry@deq.state.or.us

Equipment

Temperature Recorders
Temperature recorders include maximum/minimum thermometers, mechanical thermographs, and digital thermographs or temperature data loggers.

Max/min recording thermometers designed for total immersion may be used, but require daily site visits during the entire sampling period. Use of max/min data is also limited because it lacks information about the length of time when temperatures were at or near the maximum. They are therefore not preferred in most watershed studies.

Mechanical thermographs have been used successfully in watershed studies. Reliability can be a problem for some mechanical thermographs and the data needs to be transferred from the instrument to a database.

Temperature data loggers are the preferred temperature recorder for watershed monitoring. These data loggers have temperature recorders that can be set to record at regular intervals (usually hourly). This allows them to capture the shape of the water temperature over a day. Shorter time intervals will more closely approximate the maximum for any day. These types of instruments continuously record data for weeks or even months. This makes it more likely to detect the maximum daily temperature during the critical stream temperature season. Data loggers also significantly reduce the work load of the person or group conducting the monitoring because data can be directly downloaded to a computer database. The cost of temperature data loggers continues to decline while their reliability and ease of use continues to improve. A list of manufacturers who sell temperature recorders and their phone numbers is provided in Table 6-2.

Table 6-2. Temperature recorder manufacturers and their telephone numbers.

Company	Telephone
Vemco	(902) 852-3047
Onset	(508) 759-9500
Ryan	(206) 883-7926

Temperature recorders must have a temperature range which is appropriate for the environment that will be monitored. Water temperatures do not vary as greatly as air temperatures, but they can change 10 to 15ºC over a 24-hour period. Instruments with a measurement range of –5ºC to 35ºC are suitable for monitoring all stream systems.

Temperature recorders should have a an accuracy of approximately 0.3ºC or better for Level A quality data (See Chapter 4). This information will be available from the manufacturer.

Listed below are several useful materials and pieces of equipment that should be taken to the field to install or service temperature probes.

- Securing devices such as rebar, aircraft cables, locks, and/or diver's weights
- Surveyors marking tape
- 2-pound sledge hammer
- Wire cutters or pocket knife
- Temperature recording equipment requirements (silicone rings, submersible cases, silicone grease, silica packets)
- Portable computer and interface as needed by the temperature recorder if downloading and launching will be completed in the field
- Backup batteries and temperature recorders
- Timepiece
- Field book
- Waders
- Camera and film
- Machete or other brushing equipment
- Maps and aerial photos
- Wood or metal stakes or spikes Global Positioning System Device
- First aid kit and personal ID

Calibration Vs. Accuracy Check

Checking the temperature logger against a known temperature is often referred to as "calibrating" the instrument. This is a misnomer, however, since the temperature readings of continuous temperature loggers cannot be changed to agree with a known standard (i.e., calibrated). Their reading is simply checked against a known temperature, and any deviation from the known temperature is recorded. We refer to this procedure as an accuracy check.

Table 6-3. Estimated equipment costs.

Equipment	Required Costs *	Possible Shared Costs
NIST** thermometer		$180
Audit thermometer	$60	
Computer (laptop if field downloads are planned)		$2500
Waders	$100	
Rebar, cables, tubing, etc.	$100	
Surveyors tape	$2 / unit	
Sledge Hammer	$15	
Wire cutters	$10	
Camera and film		$100
Maps	$3 /each	
Compass	$30	
*** Global Positioning System (GPS)		$500
Field notebook	$10	
Watch	$20	
Backup batteries	$10/each	
Backup temperature recorders		$135/each
TOTAL	$360	$3415

* Required costs are those expenses each study will incur. Actual total cost will depend on the number of study sites and temperature logging units required. Shared costs are for items used infrequently and could be shared between different groups or projects.

** National Institute of Standards and Technology

*** Accurate location of study sites on a map and latitude and longitude information is necessary. A GPS unit is one simple way of collecting this information, but it can also be obtained from good maps. A GPS unit is not required. Excellent map location information is also available on CD-ROMs for about $20.

Equipment Costs

Estimates of equipment costs are based on 1997 prices (Table 6-3). It may be possible to share some equipment with others doing similar monitoring or to receive funding from the DEQ Healthy Streams Partnership program for equipment

Equipment Set-Up

Hardware and Software Checks

Prior to going to the field, make sure the operator is familiar with the software for the computer and data logger. The clock on the computer should be synchronized with the user's watch. Knowing the quality of the data being collected is necessary for any monitoring effort. The following procedures describe

methods for documenting the accuracy of the temperature recorders before and after they are deployed in the field, and testing for proper function during the sampling period. Temperature recorders not properly tested may result in data showing streams cooler or warmer than actual temperatures.

Pre- and Post-deployment Accuracy Checks

The accuracy of temperature recorders needs to be tested before and after field deployment to insure that they are operating within their designed range of accuracy. Monitoring equipment with detachable sensors should be marked in order to match sensors with temperature recorders. This allows an instrument and sensor to be consistently tested together, and also makes malfunctions easier to diagnose and correct. A logbook is helpful to document each unit's accuracy, check dates and test results. (An example of a temperature audit form is shown in Table 6-4.)

Accuracy checks should be made at one or more temperatures, preferably two; one between 5-15ºC (42-62ºF) and one between 15-25ºC (62-82ºF). Testing is done using a stable thermal mass, such as a water-filled thermos bottle or cooler. Procedures for determining temperature recorder accuracy are as follows:

Needed Equipment

- NIST (National Institute of Standards and Technology) traceable (calibrated and maintained) thermometer accurate to ±0.2ºC or a field audit thermometer accurate to ±0.2ºC or better, that has been checked against an NIST traceable thermometer. (NIST temperatures are given in Celsius. Please refer to the table in Appendix G).
- 1 or 2 medium sized coolers
- Temperature audit forms
- Small weights (bags of sand, diver weights, lead weights, etc.)
- Temperature recorders. Note: If using HOBOs, **do not** use their sealed plastic cases.
- 2 bags of ice

Accuracy Check Procedure

1. For the 20ºC calibration test, pour room temperature water into a cooler. Adjust temperature in the ice chest with ice, cold water, or warm water to the desired temperature near 20ºC. If ice is used make sure it all melts. Close lid. Repeat procedure for the 10º calibration test but start with cold water.
2. Insert the NIST thermometer probe through a hole in the cooler lid. Pull it through enough so that when the lid is closed, the probe will be suspended midway (or slightly higher) in the waterbath.
3. Use accompanying software and a laptop computer to set the recorders to a 1-minute time interval.
4. Most temperature probes can be placed directly into the waterbath. If the temperature recorders are either internal or external sensor HOBOs, place the temperature recorders without their cases into a plastic pint-size Ziplock® bag. Place this bag inside a gallon-size plastic bag along with the small weight(s). The weight(s) should be sufficient in mass to hold down the combined lifting force of the temperature recorders and the air trapped inside the Ziplock® bags while allowing the temperature recorders to be suspended in the water column. Place the double bagged HOBOs into the waterbath.
5. Wait approximately an hour or until the waterbath temperature has stabilized before recording the NIST temperatures in a

logbook. Record temperatures every minute for five minutes (a total of six readings). More readings may be necessary if there is suspicion that the waterbath temperature has not reached an equilibrium.

6. Download temperature results from temperature recorders and record logger results and audit thermometer results with time of record on an audit form. Water temperatures should not vary more than ±0.5ºC between the NIST recorded temperature and the data logger's temperature. Units not passing the accuracy test should not be used.

NIST thermometers are available at DEQ offices in Coos Bay, Astoria, and Medford, and at the Hatfield Marine Science center in Newport. DEQ has 60 NIST thermometers available for use by watershed councils. Contact the temperature mentor with additional questions about accuracy checks. Remember, accuracy checks should be made before units are deployed and after they are retrieved at the end of the sample period..

Table 6-4. Temperature logger audit form.

Project Name:

Temperature Logger ID:
Data File Name:
Date of Battery Installation:

Start Date:
Interval:
Duration:

Site Name:
Site STORET #:
USGS Quad Name & #:
Site Latitude:
Site Longitude:
Site Description:

Pre- Deployment Temperature Check
Date of Check:
Master thermometer ID: DEO

Low Temp TIME	TEMP MASTER	TEMP UNIT	Difference	STATUS

Pre- Deployment Temperature Check
Date of Check:
Master thermometer ID: DEO

Room Temp TIME	TEMP MASTER	TEMP UNIT	Difference	STATUS

AUDIT VALUES		Water Temperature		Air Temperature		**Audit Thermometer ID**		
Date	**Time**	**Audit**	**Logger**	**Audit**	**Logger**		**Comments**	**STATUS**

COMMENT:

Post- Deployment Temperature Check
Date of Check:
Master thermometer ID: DEO

Low Temp TIME	TEMP MASTER	TEMP UNIT	Difference	STATUS
	-	-	-	-
			0.0	
			0.0	
			0.0	
			0.0	
			0.0	
			0.0	

Post- Deployment Temperature Check
Date of Check:
Master thermometer ID: DEO

RoomTemp TIME	TEMP MASTER	TEMP UNIT	Difference	STATUS
	-	-	-	-
			0.0	
			0.0	
			0.0	
			0.0	
			0.0	
			0.0	

Alternative Method

To achieve Level A data quality (the highest level as described in Chapter 4, *Data Quality*) the accuracy check procedure using an NIST thermometer described above must be used. If a lower level of data quality (Level B or C—see page Chapter 4 *Data Quality)* is acceptable for a project, an alternate accuracy check procedure can be used that does not require a NIST thermometer.

For this method, create an ice-water slurry in a large insulated cooler by mixing cold water with a large amount of ice. Temperature recorders can be placed into the cooler to check that they are performing accurately. They should read 0°C (±0.5ºC). Multiple probes can be placed in the cooler at the same time to provide cross-checks. This method only assures accuracy at 0°C. If the data will be used for regulatory purposes, the described NIST method must be used instead

Field Checking Instrument Performance

In addition to pre- and post-deployment checks, check temperature recorders during the field measurement period. A field check compares the continuous temperature recorder reading with the reading on a field audit thermometer. The purpose in conducting field checks is to insure data accuracy.

Attempt to obtain at least two field temperature audits for three months of sampling—one after deployment when the instrument has reached thermal equilibrium with the stream (approximately 30 minutes to 1 hour after placement), and one just before temperature units are removed from the stream. Additional field checks, while not critical, are useful as they can minimize loss of data in case loggers malfunction during the sample period. Field audit thermometers used for field checks should have an accuracy of ±0.5ºC (±1.0ºF) and resolution of ±0.2ºC (±0.4ºF).

Check the temperature by placing the audit thermometer next to the continuous monitoring instrument's sensor. The temperature is recorded when a stable reading is obtained. A stable reading is usually achieved within 10 "thermal response times". For example, a thermometer with a 10-second response time (refer to manufacturer specifications) should give a stable reading within 100 seconds.

Most temperature recorders interrupt data collection when the unit is connected to a computer. With this type of unit, field checking data can only be applied by "post-processing" (i.e., after the units are retrieved and the stored data are offloaded). For this reason, field audit times should be scheduled close to the temperature recorder's logging time. Otherwise, rapidly changing water temperatures may cause the audit thermometer to record a different temperature than the logger.

Temperature recorders typically set date and time based on the set-up computer's clock. Field personnel should synchronize watches to this time, otherwise a poorly timed check could cause valid data to be rejected.
Post-processing audit accuracy should be within ± 1.5ºC (±3ºF) as well.

Field Methods

Site Selection

Chapter 3 addresses site selection criteria. Some additional considerations unique to stream temperature (Figure 6-1) include:

Install temperature recorders at sites with turbulence and mixing, such as riffles, runs, or cascades (high stream energy, fast moving stream reaches).

Install temperature recorders toward the lowest point of the channel bed (the"thalweg") of the channel where possible.

Consider that flow will decrease throughout the summer and a location that appears adequate in June may leave the temperature recorder exposed in August.

Some researchers have recommended shading temperature recorders from direct sunlight. Discuss this with a temperature mentor.

Do not place monitoring equipment in pools, areas where stream temperatures can be stratified by depth or channel width, or other confounding variables (See Chapter 3), unless the specific purpose is to evaluate these areas for temperature refugia.

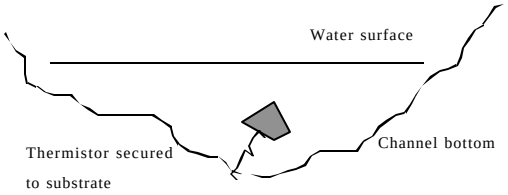

Figure 6-1. Illustration of temperature recorder installation and site locations.

Adequate Mixing

If uncertain whether a selected site has adequate mixing, a hand-held thermometer can be used to evaluate the degree of mixing. Make frequent measurements horizontally and vertically across the stream cross-section. If stream temperatures are relatively homogeneous throughout the cross-section during summer low-flow conditions, then sufficient mixing exists.

Installation

Installation of the sensor[4] or probe at the monitoring site can be an important consideration. Monitoring equipment must be installed so that the:

- temperature sensor is completely submerged
- temperature sensor is not in contact with the bottom or other mass that could serve as a heat sink/source
- where possible, the sensor is set about half-way in the water column

For non-wadeable streams, the sensor should be placed one meter below the surface, but not in contact with a large thermal mass like a bridge abutment or boulder. If volunteers are working in a large, non-wadeable stream, contact the temperature mentor for guidance. Field checks during the monitoring should confirm that the temperature sensor has remained submerged, that it is not buried in the substrate, and that it has not been damaged by changing flows, animals or vandals.

Temperature recorders frequently become coated with algae or silt and can be difficult to locate when one returns to retrieve the unit or check the temperature. A photograph of the monitoring site can be useful for locating equipment. The Governor's Watershed Enhancement Board (1993) provides guidance for photo documentation of monitoring sites.

Installing, maintaining, and retrieving the temperature recorder is fairly simple. The general procedure for field work is as follows:

1. Start the temperature recorder either prior to going to the field or in the field with a laptop computer. Follow instructions for the specific logging device. Many temperature loggers have a delayed start function which allows them to be initialized prior to going to the field. It also allows recorders to be synchronized to have the same starting time throughout the watershed.
2. Secure the temperature recorder with rebar, cable, or weights depending on the streambed characteristics, in a section of stream channel with adequate mixing and flow.
3. Record in a fieldbook the time of deployment and how long the monitor will record measurements. Check the stream temperature with an audit thermometer. Record site conditions, weather conditions, and site location using latitude and longitude.
4. Collect any additional environmental parameters of interest such as riparian shade, flow, channel width and depth, substrate composition, and riparian vegetation characteristics. For more information on these measures, check the following references: EPA 1993; EPA 1996; Bjornn and Reiser 1991; ODF 1994; and Appendix D.
5. Photograph the site location for future reference. Write a description of the site

[4] The sensor is the thermistor or other temperature detector and is a part of the temperature recorder. The sensor or probe must be submerged in the water column, not the temperature recorder.

and sketch the exact location of the temperature recorder. Record the serial number of the logger with each site description.

6. If possible, permanently mark the site location. Vandalism, theft, and landowner permission should be considered.

Attaching and Securing the Temperature Recorder

DEQ uses aircraft cable to attach temperature monitors for security purposes and stabilization in large river systems. Other securing devices such as rebar and hose clamps or diver's weights also can be used.

Retrieval and/or Mid-Season Redeployment

1. Locate the temperature recorder and check stream temperature with audit thermometer before removing it from the stream.
2. Offload the data using a laptop computer and the temperature recorder's specific software. Back up the data files on both the hard drive and a disk.
3. Record the time of downloading, site conditions (changes in streamflow, riparian vegetation, etc.), and weather conditions.

Monitoring Timing

For assessing maximum stream temperature, continuous temperature monitoring is generally conducted from June through September when solar angles are high and streamflow is low. Where this is not possible, monitoring can be conducted during a three-month period including July and August when stream temperatures are generally the highest. Depending on study objectives, temperature data may be of interest during fish spawning seasons also. This typically occurs in the fall, winter, or spring. Ideally, at least two weeks of data should be collected on either side of the period of maximum temperature.

Monitoring Frequency

The monitoring frequency should be adequate to provide a realistic estimate of the maximum temperature. If monitoring data are collected infrequently, the maximum temperature may be missed. *The Monitoring Team recommends that the monitoring frequency should be set for continuous temperature recorders at one hour intervals.* More frequent monitoring can more precisely determine the duration of daily maximum temperatures. The disadvantage to more frequent readings is fewer days of data collection are possible and more data points for the same period of time must be stored and analyzed.

Data Analysis

Data Quality

Reviewing data for errors prior to analysis is important. Viewing data graphically as soon as possible is a good way of checking for errors. Some data logging software actually graph the data while it is offloaded from the temperature recorder. Graphing the data provides a view of the entire period of record. The collected data set can then be scrutinized for illogical or incorrect segments. For example, extremely high or low blips and sustained periods of little or no change in temperature shown by flat portions on the graph are areas of concern. Often these areas exist at the beginning or end of the data file and can result from starting the temperature recorder long before it is placed in the stream. Areas of concern in the middle of the data period may have occurred when the temperature recorder was exposed to the air because of low flow or because of removal by animals, or vandals. These areas of concern must be deleted from the dataset. However, it is valuable to keep a backup file of the complete unaltered dataset in case the data quality comes into question.

DEQ will provide an electronic spreadsheet for reporting temperature data. Contact one of the mentors listed at the beginning of this chapter for a copy, or for further information on data reporting. An example of a data summary sheet is provided in Table 6-5. What follows are some examples of statistical parameters for summarizing stream temperature data.

Seven-Day Moving Mean of Daily Maximum
The "seven-day moving mean of daily maximum" smoothes out some of the daily fluctuations in the temperature profile and also provides a picture of the average temperature affecting fish over a longer period of time than daily maximum. It is also the basis of the DEQ water quality standard for stream temperature.

Before calculating the seven-day moving mean of daily maximums, the daily maximum temperatures must be determined. Using a spreadsheet, query the maximum reading for each 24-hour period of measurement. Store these temperature readings in a separate file or column accompanied by their date. The seven-day moving mean is calculated as the average of the 24-hour maximum temperature for the day and the maximum temperatures for the proceeding three days and following three days

Daily Fluctuation
Daily fluctuations are also often used in stream temperature analysis. This is the difference between the daily maximum and daily minimum temperatures at a station.

Spatial Trends & Rate of Change
With two or more temperature recorders available, changes in temperature between multiple stations on a stream can be analyzed. This is calculated by subtracting the temperature (maximum, minimum, or seven-day moving mean of maximum) at one station from the other station. The change is reported as an increase (positive value) or an decrease (negative value) in temperature.

Temperature change can also be reported in terms of rate of change. This is commonly reported as change in temperature per linear distance (i.e. 2°C/1000 feet). Two or more probes are needed, and the distance between stations must be measured.

Basin Trends

Stream temperature generally increases in a downstream direction. If stream temperatures are monitored throughout a basin (i.e. 5-20 probes) the basin trend from the divide can be analyzed by distance. Graph the highest 7-day maximum temperature for each station versus its distance from the ridge or watershed divide. Then answer the following questions: How does the rate of change (calculated above) vary from upstream locations to downstream locations? Is there a point in the basin where stream temperatures stop increasing and level off? What is the maximum stream temperature and where does it occur? How does tributary input affect the basin trend?

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Table 6-5. Examples for stream temperature data summary.

Template for stream temperature data management.

Stream Name	Station Number	GPS * Location	Calibration Method	Monitoring Period		Highest 7-day Max	Date of Occurrence	Absolute Maximum	Date of Occurrence	Diurnal Fluctuation	Rate of Change **	Landuse
			(NIST or Alternative)	(beginning date)	(end date)	(°F)	(m/d/yr)	(°F)	(m/d/yr)	(°F)	(°F/1000 ft)	(AG/ Forestry/ Urban)
Deer Creek	1		NIST	6/15/98	9/15/98	58.3	8/4/98	60.2	8/3/98	2.0		Forestry
Deer Creek	2		NIST	6/15/98	9/15/98	60.3	8/4/98	62.9	8/5/98	2.5	3.0	Forestry
Deer Creek	3		NIST	6/15/98	9/15/98	62.2	8/4/98	65.7	8/3/98	3.0	2.5	Forestry
Deer Creek	4		NIST	6/15/98	9/15/98	62.1	8/4/98	63.3	8/2/98	2.5	–1.5	Forestry
Clear Creek	1		NIST	7/4/98	9/20/98	62.5	7/21/98	63.7	7/19/98	4.1		For. & Ag
Clear Creek	2		NIST	7/4/98	9/20/98	58.1	7/21/98	59.2	7/18/98	4.5	–0.8	For. & Ag
Clear Creek	3		NIST	7/4/98	9/20/98	58.9	7/22/98	60.1	7/18/98	4.6	0.5	For. & Ag

* If a Global Positioning Station was not used, then latitude and longitude will suffice.

** Rate of change can only be calculated when more than one station is established.

NOTE: Other useful data that were not described in this protocol include: elevation, distance from divide, shade, channel gradient, substrate, channel width, and depth, and riparian buffer width. These could be added as columns to this template.

Chapter 7

Dissolved Oxygen Protocol

Background

The term "dissolved oxygen" (DO) refers to the amount of oxygen that is dissolved in water at a given temperature and a given atmospheric pressure. DO is critical to the entire biological community in surface waters and is a key element of healthy salmon habitat. DO is one of the principal parameters used to measure water quality. In Oregon, water quality criteria have been developed for DO based on the life history requirements of aquatic species, particularly salmonids (DEQ 1994).

DO is usually measured in parts per million (ppm) or the equivalent of milligrams per liter (mg/l). Water can hold more dissolved oxygen (DO saturation) at low temperatures than at high temperatures. For example, at 08C and 1 atmosphere of pressure, the maximum concentration of DO (100% saturation) is 14.6 mg/l; at 308C the same water sample would contain only 7.55 mg/l (Hitchman 1978).

In waters supporting salmonids, the necessary DO levels range from 11 mg/l in spawning and rearing waters (in order to support embryo and larval production stages with no impairment) to 6 mg/l in non-spawning waters (the absolute minimum to avoid acute mortality).

In addition to temperature, various supplies and demands influence the concentration of DO in water. The primary *sources* for dissolved oxygen are photosynthetic activities of aquatic plants and reaeration (as water spills and splashes downstream, atmospheric oxygen is trapped and dissolved in the water). The major *demands* on DO concentration come from plant respiration and the biological breakdown (or decomposition) of organic material by bacteria and other microorganisms.

The DO protocol described here is for sampling surface water DO concentration (DEQ 1995; DEQ 1997; EPA 1996; MacDonald, Smart, and Wissmar 1991). Intergravel DO concentration is also an important measure of stream habitat for salmon (McCullough and Espinosa 1996; MacDonald, Smart, and Wissmar 1991). Intergravel DO samples can be collected by pumping a water sample from the gravel near potential redds. However, DEQ does not recommend that these types of samples be collected by watershed councils. Additional information of intergravel DO sample collection should be referred to the mentor.

Mentors

As with any monitoring project, questions will come up that are not answered or covered sufficiently in this protocol. Therefore, a group of mentors that are agency experts in monitoring have been identified . These mentors may be contacted with specific questions about a particular monitoring effort.

For more information on dissolved oxygen, contact:

Dr. George Ice
E-mail: gice@wcrc-ncasi.org

OR

Statewide DEQ Volunteer Monitoring Coordinator

Karen Williams: (503) 229-5983
E-mail: williams.karen@deq.state.or.us

North Coast
Larry Caton (503)229-5983;
E-mail: caton.larry@deq.state.or.us

South Coast & Willamette
Dennis Ades (503)-229-5983;
E-mail: ades.dennis@deq.state.or.us

Eastern Oregon
Larry Marxer (503) 229-5983;
E-mail: marxer.larry@deq.state.or.us

Ordering Equipment

The sampling method for measuring DO concentration outlined in this protocol is known as the *Winkler Titration Method* (APHA 1998). The Winkler Method can be done with liquid or dry chemical reagents. Approximate costs for equipment and reagents are shown in Table 7-1. For information about the chemical reagents and the other equipment required for this method, contact:

The HACH Company
P.O. Box 608
Loveland, CO 80539-0608
1-800-227-4224

NOTE: *The chemicals, liquid or dry, used in the Winkler Titration method for measuring DO concentrations are hazardous*. Material Safety Data Sheets are provided with each purchase, and all safety precautions and procedures should be employed during use

.

Table 7-1. Equipment costs.

	Estimated Costs
Winkler Method Field Sampling Equip.:	
Hach Digital Titrator DO Test Kit	$190.00 (50 tests)
Additional reagents	$43.00 (50 samples)
200 ml Volumetric Flask	$15.00
Field Audit Thermometer	$60.00
Field Notebook (Data Sheets)	$10.00

Field Protocol

The Winkler Titration Method is *the most accurate chemical method for measuring DO concentration*. It is based on the oxidation of manganese, the liberation of iodine in proportion to the DO present in the sample, and then the "titration" of the iodine with

sodium thiosulfate. [5]

Monitoring Frequency & Timing

Monitoring frequency depends on the objectives of the project plan (MacDonald, Smart, and Wissmar 1991). The goal of DO monitoring is to provide a realistic estimate of the stream's typical DO conditions, therefore the frequency and total number of monitoring samples should be based on the attainment of that goal. The concentration of DO in surface waters will vary throughout the day due to oxygen production by aquatic plants, respiration, and changes in water temperature. If DO samples are not collected frequently enough, the degree of daily DO fluctuation may be missed, the immediate or potential problems may not be identified, or the long-term trends may not be determined.

The timing of collecting samples also depends on the objectives of the project, which may target a particular time of day. Low DO concentrations usually occur in the early morning because plants stop producing oxygen at nightfall and don't begin again until sunrise. DO concentrations build up throughout the day following the pattern of photosynthesis. Concentrations usually peak in the afternoon, and then decline as respiration exceeds photosynthesis (Ricklefs 1979; Willers 1991).

The timing of sample collection may also be influenced by other oxygen sinks and sources that occur at a specific time of day or season of the year.

For example, large inputs of organic material may result in a significant drop in oxygen concentration due to an increase in biochemical oxygen demand (BOD)[6].

The dissolved oxygen needs of salmonids vary with their life history stages, from embryo development to growth and sexual maturity. Having experience with regional weather patterns and knowing the timing and length of salmonid spawning seasons is important if monitoring duration must be limited. For example, west of the Cascades, the majority of salmon and steelhead spawning takes place during the fall, winter, and spring months when water levels are elevated and water temperatures are at a minimum.

Recommended Sample Collection & Analysis

Field Collection

The sample containers used for collecting water samples for DO measurements are clean 300 ml glass BOD bottles with glass stoppers (these bottles come with the HACH monitoring kits). Field staff can fill the sample bottle by: a) submersing it directly into the stream; or, b) collecting the water sample from a bridge or similar structure using a suitable grab sample collection method.

1. Fill the sample bottle to overflowing to ensure that no air bubbles are trapped in the bottle. Replace the glass stopper. Invert to check for air bubbles. This minimizes the risk of additional aeration of the sample.
2. Remove the glass stopper and add the contents of 1 powder pillow of Manganous

[5] "titration" is a standard analytical method that measures the amount of one chemical or solution needed to react with another chemical or solution. In this case, the amount of sodium thiosulfate needed to react with the iodine present in the water sample.

[6] B.O.D. is a measure of the amount of oxygen consumed in the biological processes that break down organic matter in water. The greater the BOD, the less oxygen is available for other biological uses (including salmonid respiration).

Sulfate (Winkler Reagent 1), and 1 powder pillow of Alkaline Azide (Winkler Reagent 2). Replace stopper and repeatedly invert the sample bottle so that the contents mix **vigorously** for 15-20 seconds. A flocculent precipitate (a cloudy substance created from the chemical reaction) will form in the sample (brownish-orange if oxygen is present, white if oxygen is absent).

3. Allow the sample to stand until the "floc" has settled approximately half way to the bottom of the bottle. **REPEAT** the vigorous mixing for 15-20 seconds. Allow the sample to stand until the floc settles a second time. **Note**: Vigorous mixing is critical to dissolving the powdered reagents and allowing the chemical reactions to occur. Not all of the reagents will dissolve, but large chunks of the reagents should not be visible after mixing. Inadequate mixing in steps 2 and 3 is a common error.

4. After the floc has settled again, remove the glass stopper and add 1 powder pillow of Sulfamic Acid. Replace the glass stopper and invert the bottle several times, mixing vigorously 15-20 seconds. The sample should turn a clear amber color. (Some of the powdered reagents may not have completely dissolved – this is normal.)

After the initial water sample has been chemically preserved, it can be held for up to eight hours, in the dark at 4°C, before the titration step is performed.

Sample Titration

Note: The sample can be titrated with either a Hach "Digital Titrator" or a standard burette.

<u>Option 1: Hach Digital Titrator Method</u>

Use Hach Method 8215 as described below, using 0.2N sodium thiosulfate titrant.

1. Remove the glass stopper and fill a 200 ml volumetric flask with a sample. Transfer this 200 ml portion to a 500-ml Erlenmeyer flask. Insert sodium thiosulfate cartridge into the titrator.[7]

2. Insert a clean delivery tube (approximately 1/16th diameter "hook") into the titration cartridge (that comes with the monitoring kit). Attach the cartridge to the titrator body. Lower the plunger for the titrator gently until it contacts the sodium thiosulfate cartridge.

3. Turn the delivery knob to eject a few drops of titrant. Reset the counter to zero and wipe the tip of the delivery tube.

4. Place the delivery tube tip into the sample. Turn the delivery knob clockwise to add titrant to the sample. **NOTE**: *swirl the flask while adding titrant to make sure it mixes*. The sample will gradually turn a pale yellow color.
5. Once the sample is pale yellow, the endpoint of the titration is approaching. Add a 1 ml dropper of Starch Indicator Solution (also a part of the kit) and swirl to mix. **Note***: A dark blue color will develop.*

6. Continue the titration until the sample turns from blue to colorless. This is the endpoint! Record the number of digits on the Digital Titrator's counter.

7. Multiply the number on the counter by 0.01. The result is the sample DO in mg/l.

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[7] Contact the DO monitors listed in this chapter for more information about equipment or techniques needed in the titration process.

Option 2: Burette Titration Method

1. Remove the glass stopper and fill a 200 ml volumetric flask with a sample. Transfer this 200 ml portion to a 500-ml Erlenmeyer flask.
2. Fill a 10 ml burette with standard sodium thiosulfate titrant (0.025N). Slowly add titrant to the sample drop by drop.
3. Swirl the flask while titrating to ensure good mixing. The sample will turn a pale yellow color, which means the endpoint is approaching.
4. Add 1 ml of Starch Indicator Solution. **Note***: A dark blue color will develop.*.
5. Continue addition of titrant drop by drop until the sample changes from dark blue to clear. **NOTE**: *Take care not to overrun the end point.*
6. Read the amount of titrant used to reach the endpoint. 1 ml of titrant = 1 mg/l DO.

Oxygen Percent Saturation

Atmospheric pressure varies with weather and elevation. At sea level, it averages near 760 mm Hg. As elevation increases, pressure decreases. The average pressure at Burns (4,200 feet) is 750 mm Hg. Extremes observed in Oregon average from a low of 632 to a high of 780 mm Hg.

Determine the percent saturation of DO using Table 7-2 and the following calculation method:

Table 7-2. Oxygen solubility (saturation) in fresh water (mg/L)

Percent Saturation Table				Elevation Correction	
Water Temperature C	DO 100% Saturation	Water Temperature C	DO 100% Saturation	Elevation in feet	Elevation Factor
0.0	14.60	20.5	9.10	0	1.00
0.5	14.40	21.0	9.00	500	1.02
1.0	14.20	21.5	8.90	750	1.03
1.5	14.00	22.0	8.80	1000	1.04
2.0	13.80	22.5	8.75	1250	1.05
2.5	13.65	23.0	8.70	1500	1.05
3.0	13.50	23.5	8.60	1750	1.06
3.5	13.30	24.0	8.50	2000	1.07
4.0	13.10	24.5	8.45	2250	1.08
4.5	12.95	25.0	8.40	2500	1.09
5.0	12.80	25.5	8.30	2750	1.10
5.5	12.65	26.0	8.20	3000	1.11
6.0	12.50	26.5	8.15	3250	1.12
6.5	12.35	27.0	8.10	3500	1.13
7.0	12.20	27.5	8.00	3750	1.14
7.5	12.05	28.0	7.90	4000	1.15
8.0	11.90	28.5	7.85	4250	1.16
8.5	11.75	29.0	7.80	4500	1.17
9.0	11.60	29.5	7.70	4750	1.19
9.5	11.45	30.0	7.60	5000	1.20
10.0	11.30	30.5	7.55	5250	1.21
10.5	11.20	31.0	7.50	5500	1.22
11.0	11.10	31.5	7.45	5750	1.23
11.5	10.95	32.0	7.40	6000	1.24
12.0	10.80	32.5	7.35	6250	1.25
12.5	10.70	33.0	7.30	6500	1.26
13.0	10.60	33.5	7.25	6750	1.27
13.5	10.50	34.0	7.20	7000	1.29
14.0	10.40	34.5	7.15	7250	1.30
14.5	10.30	35.0	7.10	7500	1.31
15.0	10.20	35.5	7.05	7750	1.32
15.5	10.10			8000	1.34
16.0	10.00				
16.5	9.85				
17.0	9.70				
17.5	9.60				
18.0	9.50				
18.5	9.45				
19.0	9.40				
19.5	9.30				
20.0	9.20				

Alternative Method Using DO Probes

DO meters are an alternative method for collecting data on water temperature and DO. However, DO meters are not as accurate as the Winkler Titration Method and may be subject to "drift," thus requiring frequent re-calibration. A useable meter will be in the $1200 to $2000 price range. For these and other reasons, using the Wrinkler Titration method is probably more reliable for volunteer groups collecting DO data in a cost effective, credible and timely manner. For more information about DO meters, contact the mentors listed in this chapter.

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Chapter 8

pH Protocol

Background

Just as "degree" is a measure of temperature, pH is a measure of how *acidic* or *basic* the water is[8]. Water pH is critical to fish habitat because it can affect fish egg production and survival, aquatic insect survival and emergence, and the toxicity of other pollutants such as heavy metals or ammonia. Like water temperature, pH naturally varies both daily and seasonally.

Most daily cycles in pH occur as a result of the photosynthesis of aquatic plants. Through photosynthesis, plants convert the sun's energy into chemical products they need to live and grow. During daylight hours, aquatic plants consume carbon dioxide (an acid), and produce hydroxide (a base). As a result, water becomes more basic during the day (pH values get higher) and usually peaks in mid-to-late afternoon. Virtually all aquatic organisms produce carbon dioxide (acid) through their normal metabolism of food (respiration). As a result, water becomes more acidic during the night (pH values drop) and usually is lowest just before sunrise. A similar daily pattern occurs in dissolved oxygen concentrations as a result of photosynthesis (see Chapter 7).

When acids dissolve in water, hydrogen ions (H^+) are produced. Hydrogen ion concentrations in water usually comprise very small fractions—1/10,000,000, for example. For convenience, these concentrations are converted to a pH scale—a logarithmic numerical scale that ranges from 0 to 14. Pure water has a pH of 7, and is the neutral point—neither *acidic* nor *basic*. Water is acidic when the pH value is below 7 and basic when the pH value is above 7. Note that a unit change in pH is a tenfold change in hydrogen ion concentration. Thus, a solution of pH 7 is ten times as acidic as one of pH 8, and one hundred times as acidic as one of pH 9 (McCutcheon, Martin, and Barnwell 1992; Sawyer and McCarty 1967).

Water pollution can cause changes in pH through the direct addition of acids or bases such as acid mine drainage, acid rain, or chemical spills. More commonly, pH is altered by excessive plant growth that results from the addition of fertilizers. Fertilizers end up in our waterways from sewage or industrial discharges, failing septic systems, and agricultural and urban runoff.

The most accurate way to measure pH is with a calibrated meter and pH electrode. The pH electrode is sensitive to the concentration of H^+ ions in the water. Measuring pH with an electrode requires a very small electrical current to flow through the water sample. When immersed in water, the electrode develops an electrical potential that is related to the pH of the solution. A "reference" electrode completes the circuit and provides a stable electrical reference potential. For convenience, "combination" pH electrodes are designed with the reference electrode built in. The reference electrode makes electrical contact with the water sample through a small opening (called the junction) that allows slow leakage of a salt solution or gel into the sample. A clogged or

[8] This measure differs from acidity and alkalinity. Acidity and Alkalinity are measures of the capacity of water to neutralize added base or acid, respectively.

dirty junction is a common source of measurement error. Electrodes with a fouled junction may not calibrate properly and typically show a slow response when immersed in a sample or distilled water.

When a pH electrode is connected to a meter, the voltage developed at the electrode is amplified, then converted to the pH scale, and displayed as a digital readout. The meter is calibrated with solutions of known pH called "buffers" (pre-packaged and available through scientific supply outlets). Each buffer has a specific pH at a specific temperature.

Temperature also influences the electrical potential of the pH electrode. This potential source of error is eliminated by using meters and electrodes equipped with automatic temperature compensation, commonly abbreviated as ATC.

Buffer solutions that are used to calibrate pH meters are relatively salty solutions. Most of Oregon's surface waters have relatively low concentrations of dissolved salts. These waters have what is called "low ionic strength." As noted above, pH measurement with an electrode requires a very small electrical current to flow through the water sample, and dissolved salts carry the electrical charge through the water. Sometimes a pH electrode will appear to be working during calibration in the buffer, but will give inaccurate readings in low ionic strength water. These problems can be avoided. First, start by using the right equipment—pH probes specifically designed for use in low ionic strength solutions. Second, once the meter is calibrated, test the electrode response in distilled water—it should read between pH 5.4 to 6.0 within 10 minutes.

Some companies advertise low ionic strength buffers, but tests at the DEQ Lab have found that these buffers are still too high in salt concentration for Oregon surface water measurements.

One way to overcome the problem of measuring pH in low ionic strength water is to add a small amount of potassium chloride (KCl) salt solution to the sample: 1 ml per 100 ml of sample (see the section on *Measuring Water Sample pH* below). These "pH ionic strength adjuster" solutions are commercially available from scientific supply companies. **NOTE***: Ionic strength adjustment is the recommended pH procedure—but it is* <u>*not*</u> *a substitute for using good quality equipment with careful calibration and maintenance*.

Other possible problems with pH measurement include damage to the electrode and chemical interference.

<u>Glass adhering materials</u>

Substances that adhere to glass can interfere with the response of the sensing glass bulb on the electrode. The problem can be solved by cleaning the probe according to the manufacturer's instructions.

<u>Abrasives and physical damage</u>

If the sensing glass bulb becomes scratched or damaged in any way, it may not be able to establish a proper potential with the sample solution. Damaged electrodes should be discarded. Care in handling the electrode should minimize this problem.

Ordering Equipment

<u>pH Meters</u>

pH meters can be purchased from a variety of scientific supply companies. Important features include digital readout and accuracy to ±0.1 pH units, two-point calibration, automatic temperature compensation, and the ability to use standard pH electrodes.

pH Electrodes
pH electrodes must be designed for use in low ionic strength water. Gel filled electrodes should be avoided. An example of a good electrode is the Orion Ross model 81-02 or equivalent.

Ionic Strength Adjuster
The Orion brand "pHix" is recommended. Orion catalog number 700003.
Orion meters are also available from scientific suppliers such as VWR (800-932-5000).

pH Field Protocol

Calibrating Equipment
One calibration per day is required. Follow the pH meter manufacturer's calibration procedure for a 2-point calibration using pH 7 and 10 buffers.

A proper calibration will include the following steps:

1. Slide the plastic sleeve on the pH probe up so that it no longer covers the filling solution opening venthole near the top of the probe.
2. Check the level of the filling solution in the probe. If it is 1 inch or more below the opening, add more filling solution.
3. Check for crystal formation in the electrode body. Small amounts of crystal are tolerated. However, if more than one-half inch of crystals is observed, empty out the electrode, rinse with warm distilled water to dissolve the crystals, then refill with the appropriate filling solution.
4. Observe the electrode-sensing bulb for any dirt or damage. Clean with de-ionized water if dirty and replace if damaged.
5. Rinse the probe thoroughly with distilled water.
6. Insert the probe into pH 7 buffer and stir moderately for 30 seconds. The electrode must be inserted to a depth of at least one-half inch in the solution. Do not allow the probe to contact the walls of the container.
7. Set the meter to "pH" and begin the calibration period. Stop stirring, wait for the reading to stabilize. Instruct the meter to accept the pH 7 calibration.
8. Remove the electrode from the pH 7 buffer and rinse thoroughly with distilled water.
9. Put the probe in the pH 10 buffer. Stir for 30 seconds. Stop stirring and wait for the reading to stabilize. Instruct the meter to accept the pH 10 calibration.

Some meters display a "slope" value when the calibration is complete. The slope is expected to be >95%. If the slope is <90%, use another meter or electrode.

Once the meter is calibrated, re-check its readings on the calibration buffers. Record the calibration information in the meter's logbook. Be sure to include the date and time of calibration with the monitor's name, the buffer temperature, the buffer value, and the meter's pH reading. When recording the buffer value, be aware that the buffer's pH changes with temperature—look on the bottle label and record the buffer pH at the temperature just measured.

Sample Handling and Preservation
Collect a water sample in a clean container. The DEQ routinely measures pH in the field, but when this is not possible, samples are analyzed at the lab. If pH can't be measured in the field, samples are collected in clean, tightly sealed 500 ml (1 pint) plastic containers. The desirable quantity of a sample for analysis is 100 ml of solution. To preserve the samples keep them cool at 4ºC. Acceptable storage time for laboratory pH measurements is 36 hours after collection (including transport time).

Samples are moderately agitated before pouring into a beaker—it's important to avoid mixing air into the sample because it could change the pH. Air intrusion is also avoided when stirring during pH measurements.

Measuring Water Sample pH

1. Calibrate the pH meter as described above. If the meter was calibrated earlier in the day, be sure to remove the probe's filling solution vent plug before making any pH measurements.
2. Thoroughly rinse the probe with distilled water. Put the probe in a beaker of distilled water (DW) while preparing the water sample.
3. Gently shake the sample container and pour approximately 100 ml of the sample into a clean beaker. Remove the probe from DW and insert it into the sample water. Rinse probes in the sample for at least 30 seconds.
4. Dump out the sample, rinse, and pour another 100 ml of fresh sample into the beaker. Add 1 ml of pH Ionic Strength Adjuster to the sample using a 1 ml syringe.
5. Stir for a full 30 seconds (moderate to rapid stir is required). Stirring should not be so vigorous that air bubbles are entrapped in the sample.
6. Stop stirring and wait for the pH reading to stabilize. Up to 10 minutes may be required for this to occur. If more than 10 minutes are required for readings to stabilize, the probe should be cleaned or replaced. A reading may be considered stable when it changes at a rate of less than .03 units/min. Even after a reading has "stabilized" it will often fluctuate by ±0.04 units.
7. Record pH to the nearest 0.1 units.
8. Remove the probe from the sample, rinse thoroughly with DW, and insert it into the next sample. Check the pH 7 buffer every 10 samples or at the end of the day to confirm stability of the calibration. The pH should not have changed by more than 0.2 units. If necessary, re-calibrate before continuing with the analysis.

When sample testing is completed, replace the probe's vent plug and store the probe in the manufacturer's recommended storage solution.

Analyzing Data

Verify that the data is meeting the data quality objectives of the Quality Assurance Project Plan. Accuracy is verified by checking meter calibration records. Precision is verified by comparing the results of duplicate measurements on the same sample.

Data which passes the accuracy and precision objectives can be compared to the water quality standards for the entire basin.

Water Quality Standards

The DEQ has adopted pH standards that are intended to protect aquatic life. These standards differ slightly from basin to basin because waters in some parts of the state have naturally higher pHs. In general, aquatic life suffers when pH drops below 6.5 or goes above 8.5. Check the Oregon Administrative Rules for the pH standards that apply specifically to the respective basin.

Mentors

As with any monitoring project, questions will come up that are not answered or covered sufficiently in this protocol. Therefore, a group of mentors that are agency experts in monitoring have been identified . These mentors may be

contacted with specific questions about a particular monitoring effort.

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References

McCutcheon, S.C., Martin, S.L., and Barnwell, T.O. 1992. Water quality. Chapter 11 in *Handbook of hydrology*. Maidmont, D.R. [Ed.]. McGraw-Hill, Inc.: New York, NY.

Sawyer, C.N., and McCarty, P.L. 1967. *Chemistry for sanitary engineers*. McGraw-Hill Book Company: New York, NY.

Chapter 9

Conductivity Protocol

Background

Conductivity (or specific conductance) is a measure of water's ability to conduct an electrical current (Sawyer and McCarty 1967). The conductivity of a water sample depends on the water temperature and on the concentration of dissolved salts or other substances that can carry an electrical charge.

There is no water quality standard for conductivity, but conductivity can be a useful diagnostic tool for interpreting other water quality information. For example, domestic and industrial wastewater, stormwater, and irrigation return water often have higher conductivities than the receiving streams. Groundwater inflows also typically have higher conductivities than surface runoff. Measuring conductivity is also informative in estuaries and coastal rivers that may be influenced by salt water as a result of ocean tides.

Conductivity is measured with a meter and is reported in units called micromhos/centimeter (mhos/cm) or microsiemens.cm (s/cm).

Conductivity meters are usually factory calibrated, but need to be periodically tested for accuracy in a standard salt solution. Conductivity meters in general require much less maintenance than pH meters or dissolved oxygen meters. Problems associated with a conductivity meter are usually due to dead batteries, a cracked or damaged cable, or a damaged or defective electrode.

Fresh surface waters in Oregon range in conductivity from about 20 to 500 mhos/cm. In the Willamette Valley and Coast Range conductivities are typically 150 mhos/cm or less. Distilled or de-ionized water that has been in contact with the air usually has a conductivity of about 1 mho/cm.

Ordering Equipment

Conductivity meters can be purchased from a variety of scientific supply companies. Important features include digital readout, automatic temperature compensation, and accuracy of ±0.5%.

Standard salt solutions for testing instrument accuracy are also available from scientific suppliers. The recommended standard solution for fresh surface water measurements is potassium chloride (KCl) with a conductivity of 147 mmhos/cm.[9]

Field Protocol

Calibrating Equipment

Most conductivity meters are calibrated at the factory, but it is necessary to check the accuracy against a standard solution. For surface water monitoring the DEQ uses a standard solution of potassium chloride which has a conductivity of 147 mhos/cm.

1. Turn on the meter and rinse the probe in distilled or deionized water.
2. Rinse the probe with the Conductivity Standard Solution.
3. Pour about 200 ml of Standard Solution into a clean beaker and immerse the probe. Make sure the temperature and conductivity sensors are fully submerged.
4. Set the meter to display temperature.

[9] Note: some meters display an equivalent unit to µmhos/cm called micro-siemens/cm (µS/cm). 1 µmhos/cm = 1 µS/cm Other meters display in *milli-siemens/m* (mS/m). Be careful not to confuse units! 10 µmhos/cm = 1 mS/m

5. Agitate the probe in the solution, but do not allow probe to contact the walls of the container.
6. Record the solution temperature when the reading is stable.
7. Set the meter to display conductivity in mhos/cm with the temperature compensation.
8. Agitate the probe as in step 5 and record the conductivity to the nearest whole number.
9. Calculate the Relative Percent Difference (RPD) between the instrument reading and the Standard Solution's value.
10. The Relative Percent Difference should be within 7% for Data Quality Level "A" or within 10% for Data Quality Level "B" (refer to Chapter 4, *Data Quality*). If the RPD is greater than the expected value, repeat the accuracy test with fresh solutions. If that doesn't fix the problem, the meter or probe needs service.

Sample Handling and Preservation

Collect a water sample in a clean container or lower the probe directly into the water. The DEQ routinely measures conductivity in the field, but when this is not possible samples are analyzed at the lab. If conductivity can't be measured in the field, samples are collected in clean, tightly sealed 500 ml (1 pint) plastic containers.
Sample preservation—samples are kept cool at 4ºC on ice or in a refrigerator. Storage time for samples is up to 28 days after collection.

Measuring Conductivity of a Water Sample

1. Turn on the meter and rinse the probe in distilled or deionized water.
2. Rinse the probe with the water sample.
3. Put the probe directly in the waterbody or pour about 200 ml of sample into a clean beaker and immerse the probe. Make sure the temperature and conductivity sensors are fully submerged.
4. Set the meter to display temperature.
5. Agitate the probe in the sample, but do not allow the probe to hit bottom or contact the walls of the container.
6. Record the sample's temperature when the reading is stable.
7. Set the meter to display conductivity in mhos/cm. Make sure the meter is also set for automatic temperature compensation to $25^{o}C$.
8. Agitate the probe as in step 5 and record the conductivity to the nearest whole number.

Analyzing Data

Verify that the data are meeting the data quality objectives of the Quality Assurance Project Plan. Accuracy is verified by checking the meter calibration records. Precision is verified by comparing the results of duplicate measurements on the same sample.

Mentors

As with any monitoring project, questions will come up that are not answered or covered sufficiently in this protocol. Therefore, a group of mentors that are agency experts in monitoring have been identified . These mentors may be contacted with specific questions about a particular monitoring effort.

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References

Sawyer, C.N., and McCarty, P.L. 1967. *Chemistry for sanitary engineers*. McGraw-Hill Book Company: New York, NY.

Chapter 10

Nitrogen and Phosphorus Protocols

Background

The two primary nutrients of concern for water quality are nitrogen (N) and phosphorus (P). Excess ammonia and nitrate can be toxic to stream organisms and humans. Of particular concern is blue baby syndrome (infant methemoglobinemia) caused by excess nitrate/nitrite. These concerns are usually associated with concentrated loads from municipal sites such as sewage treatment outfalls or they may result from repeated heavy applications of nitrogen fertilizer or animal waste.

The most common basin-wide concern with nutrients is *eutrophication* of streams and lakes. Eutrophication is an excessive growth of aquatic plants. Eutrophication occurs when concentrations of nitrogen, phosphorus and other environmental conditions favorable to aquatic plant growth (temperature, light, flow velocity) are elevated and available.

This excessive plant growth causes daily fluctuations in dissolved oxygen and pH and can impart undesirable tastes and odors to water. These water quality impacts can adversely affect the uses of water for fish habitat, recreation, and drinking water. The US Environmental Protection Agency is currently developing new nationwide water quality criteria[10] for nutrients. By the year 2000, numeric criteria for nitrogen and phosphorus will be developed which will reflect different types of water bodies and ecoregions.

The protocols described here are designed for collecting and preserving a sample for analysis by a laboratory. Levels of concern are often below the detection limits of field sampling kits and are difficult to record accurately with specific ion electrodes. Therefore these samples are best analyzed at a commercial analytical laboratory. The first step is to determine what forms of nitrogen and phosphorus should be analyzed so that the appropriate preservation protocols can be selected (Stednick 1991).

Nitrogen can occur in many forms in the environment. Nitrogen can also cycle between these different forms (Figure 10-1.) The nitrogen forms that are most commonly tested are those which are the most biologically available: soluble nitrate/nitrite nitrogen (NO_3^-, NO_2^-), ammonia (NH_3), and total Kjehldahl nitrogen (the sum of the free ammonia and organic nitrogen). Nitrate is especially important because it is relatively soluble in water compared to other nitrogen forms. The Monitoring Team for the OPSW recommends that samples be analyzed for nitrate/nitrite and total Kjehldahl nitrogen.

[10] http://www.epa.gov/cleanwater/action/overview.html

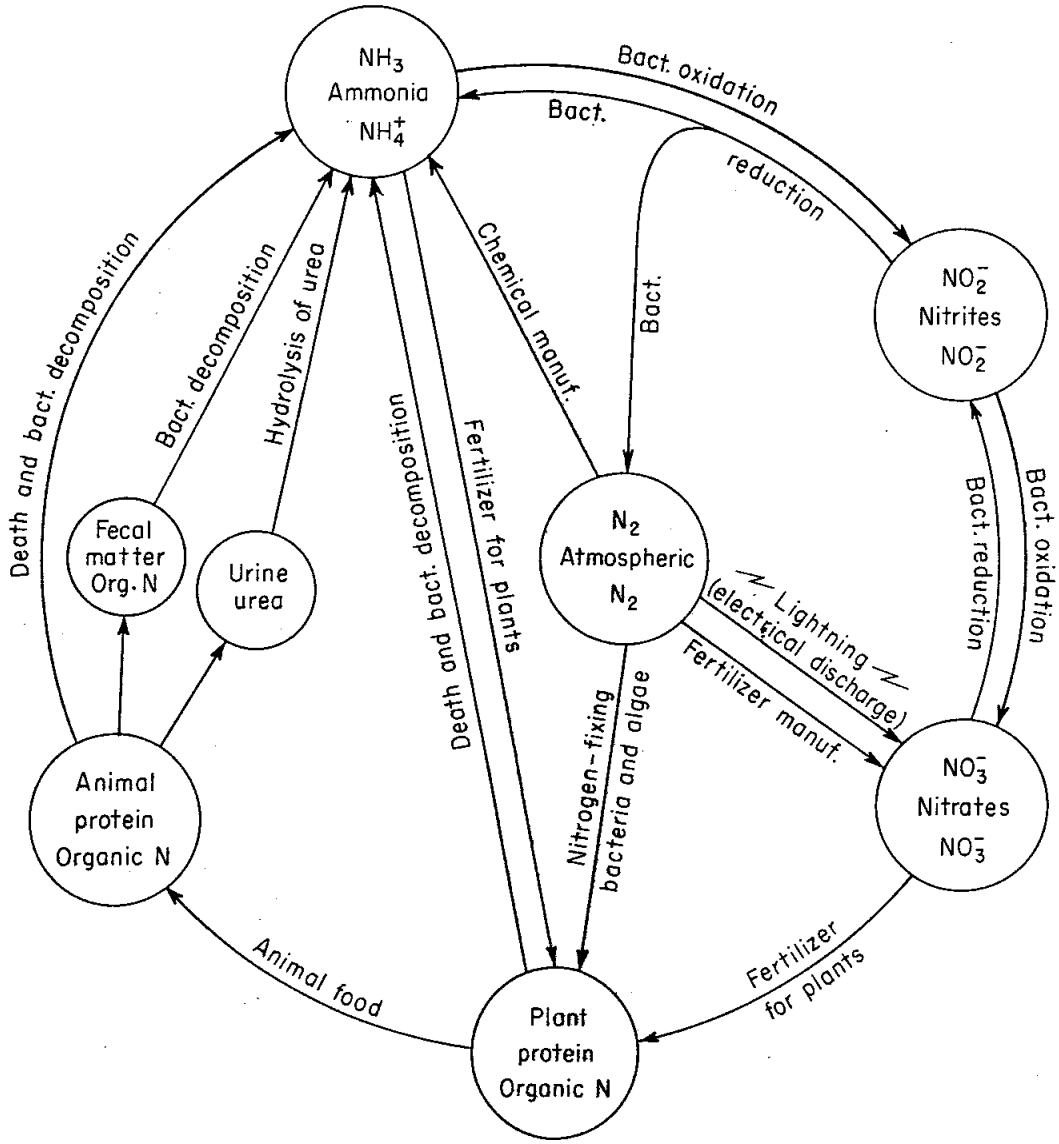

Figure 10-1. The nitrogen cycle (from Sawyer and McCarty 1967)

Phosphorus also comes in many different forms (Figure 10-2). The two forms for which analyses are the most common are orthophosphate and total phosphorus. Orthophosphates include $H_2PO_4^-$, and HPO_4^{2-}, and PO_4^{3-}. These are dissolved forms of phosphorus which are available for aquatic plant use. Total phosphorus includes dissolved and particulate, organic, and inorganic forms.

Equipment

A list of equipment needed to sample for nitrogen and phosphorous is shown in Table 10-1. Sample bottles need to be clean and made of a material that does not leach or react with the sample. Cleaning of reusable sample

bottles should be done according to instructions from the laboratory receiving the sample because some detergents can contaminate samples.

Table 10-1. Materials needed to collect samples for nitrate/nitrite, kjehldahl nitrogen, orthophosphate, and total phosphorous.

500 ml clean, reusable polyethylene bottle
250 ml clean, reusable polyethylene bottle
0.45 µm filter discs and syringe
Cooler and ice or blue ice
Concentrated H_2SO_4
Marking pen and labels

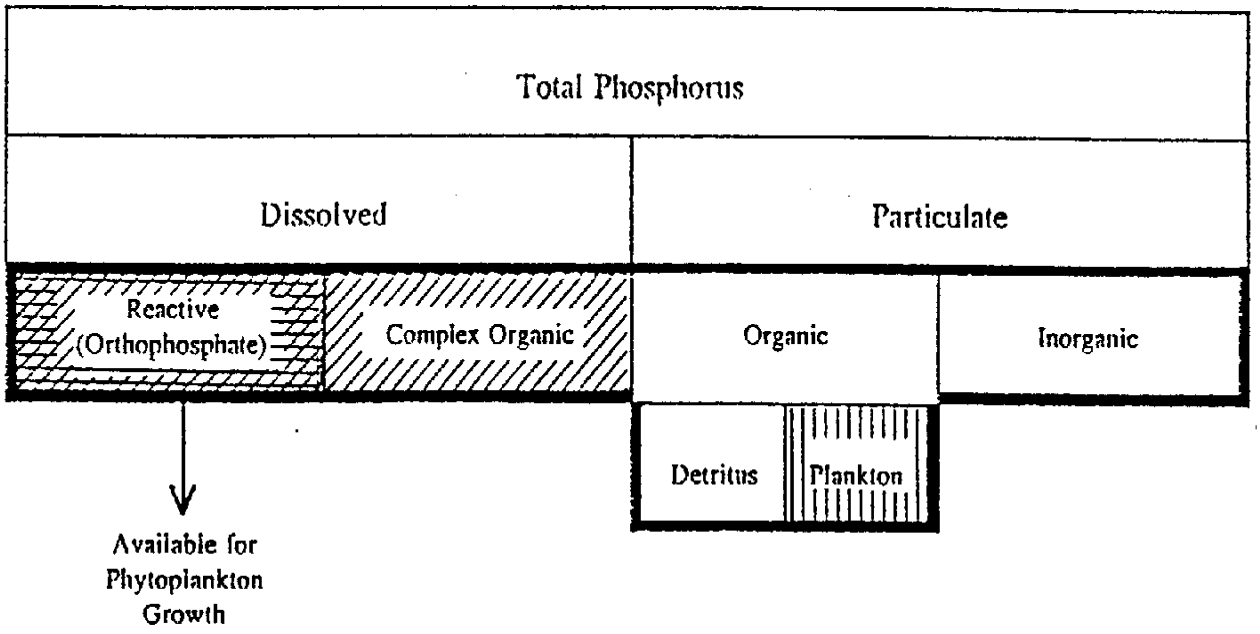

Figure 10-2. Forms of phosphorus in water (from McCutcheon, Martin, and Barnwell 1993).

Field Method

This sample will be analyzed for *nitrate/nitrite, kjehldahl nitrogen, and total phosphorous*. *The sample must be delivered to the lab for analysis within 28 days.*

1. Rinse the 500 ml bottle in the water body to be sampled, discarding initial water collected.
2. Fill the 500 ml bottle, leaving about an inch at the top of the bottle.
3. Add 12 drops of concentrated H_2SO_4 (sulfuric acid).
4. Label and place in the cooler with ice to keep the sample preserved.

This sample will be analyzed for *orthophosphates*. *The sample must be delivered to the lab for analysis within 48 hours.*

1. Using a 45 µm filter disc and syringe, filter about 200 mls into the bottle.

2. Label and store in the cooler

Take samples directly to the laboratory or ship them immediately. Keep samples cool and refrigerated (4°C) until analyzed.

Mentors

As with any monitoring project, questions will come up that are not answered or covered sufficiently in this protocol. Therefore, a group of mentors that are agency experts in monitoring have been identified . These mentors may be contacted with specific questions about a particular monitoring effort.

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Chapter 11

Turbidity Protocol

Sediment Characteristics and Effects on Stream Ecosystems

Sediment is an essential component of healthy salmon and trout streams. Channel features such as point bars, riffles, and floodplains are all products of sediment inputs. Sediments provide substrate and habitat for algae and macroinvertebrates, plus spawning gravels and rearing habitats for fish. Yet, sediment is frequently identified as a factor contributing to the impairment of aquatic productivity in salmon and trout streams throughout the Pacific Northwest because too much fine sediment in the water column or streambed can be detrimental to aquatic insects and the fish that feed upon them.

Large inputs of fine sediment to the stream can degrade aquatic invertebrate and fish habitats and alter the structure and width of stream channels and adjacent riparian zones (MacDonald et al. 1991). Increased sediment input may elevate suspended sediment concentrations and turbidity. Excess fine sediments fill intergravel spaces used by aquatic insects and young fish. Pool frequency and depth may diminish and channel *sinuosity*[11] and other channel characteristics can be appreciably changed. Land management activities can contribute to these impacts by affecting watershed processes and altering sediment delivery to a stream network.

Background

Sediment is the product of erosional and fluvial processes. Erosion involves the processes of *detaching* sediment particles, *transporting* them from the original site and eventually *depositing* those particles. Site characteristics such as geology, soils, slope steepness and length, vegetation, precipitation regime, channel and streamflow characteristics all influence natural erosion rates. In addition land management activities can cause increased rates of erosion. Erosion and the delivery of sediment to stream systems are complex and naturally occurring processes in all watersheds.

Monitoring the sources of sediment, its transportation by streams, and deposition trends can often provide important information for better management decisions. Monitoring turbidity addresses one component of the erosional cycle—the *transport* of fine sediment. Other components of the erosional process include *sources* of sediment and *deposition* of sediment.
If monitors are interested in identifying road-related sources of sediment, they should refer to **Appendix D** for the *Road Hazard Risk Inventory*. Other sources of sediment within a watershed may also need to be addressed (e.g. urban or residential development, agricultural run-off, logging, etc.).

For methods to monitor the *deposition* of fine sediment in stream reaches please refer to **Appendix E** for the *Sediment Deposition Protocol*. These protocols are designed to complement watershed analysis activities identified in the Oregon Watershed Assessment Manual.

Turbidity

Sediment particles are characterized by their size. They range from the finest clays and silt particles to sand, pebbles, gravels, and boulders. Once sediment particles have been introduced to a stream system, the smaller

[11] Sinuosity is the amount that a stream channel curves or meanders laterally across the land surface.

particles (silts and clays) are typically transported as suspended sediment in the water column before eventually settling out and depositing.

Processing and analyzing suspended sediment samples and data can be complex and expensive. A frequently used substitute for measuring suspended sediment is turbidity. *Turbidity is relatively easy and inexpensive to measure and is often the basis for water quality standards and can be correlated with suspended sediment on a site-specific basis*. Monitoring turbidity can provide valuable information to help understand baseline trends over time as well as the effects of a specific project on water quality. The nature and attributes of turbidity are described below to aid in data interpretation.

Turbidity varies with the number and size of particles present in the water column. Turbidity is defined as the optical property of a sample that causes light to be scattered and absorbed.

Since water-borne particles other than sediment can scatter light (e.g., fine organic matter, plankton, microscopic organisms), *turbidity is not a direct measure of sediment in the water column*. The relationship between suspended sediment and turbidity can vary greatly between sites. For example, a watershed with coarse soils may have great fluxations in suspended sediment, but turbidity may remain fairly stable. A watershed with fine clay soils may have consistently high turbidity, but low concentrations of sediment (MacDonald et al. 1991).

Turbidity levels are influenced by the same factors as suspended sediment with the additional complication of turbidity's sensitivity to water-borne particles other than sediment (Brown 1983). In general, turbidity can be expected to increase during high stream flow events, but this will vary within a given storm and between storms. For example, the first storm of the year may produce higher turbidity levels than a storm of the same magnitude that occurs later in the season. Likewise, as stream flow initially rises during a storm event (referred to as the "rising limb" of a storm hydrograph), turbidities may be high. The equivalent flow as the stream recedes (the "falling limb" of a storm hydrograph) may produce lower turbidity levels. Because of these characteristics, the relationship between suspended sediment and turbidity must be determined for each site (Beschta 1980) and a range of flow conditions (Brown 1983).

The variability in turbidity between sites and over time can make it very difficult to establish a natural or background level. Measurement errors can increase this variability as well. So it is important to use caution when drawing conclusions with the monitoring data about effects of management.

Turbidity measurements may be most useful for project monitoring. In this case samples should be collected upstream and downstream of a planned project, before, during and after the project commences.

The most commonly used measurement method for turbidity is the nephlometric turbidity method (Stednick 1991). Nephlometric methods measure the scatter of light and perform better for high and low turbidities (measured in Nephlometric Turbidity Units or NTUs).

Mentor Contacts

As with any monitoring project, questions will come up that are not answered or covered sufficiently in this protocol. Therefore, a group of mentors that are agency experts in monitoring have been identified. These mentors may be contacted with specific questions about particular monitoring goals and efforts.

Questions about turbidity monitoring should be directed to one of the following:

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Equipment

The following equipment will be needed to sample turbidity:

- A portable turbidimeter (available from DEQ's Volunteer Monitoring Program or scientific supply houses). These instruments are calibrated on the nephlometric turbidity method (see above) and meets the criteria established by EPA. The HACH 2100P (portable) Turbidimeter is available to local watershed groups through DEQ's Volunteer Monitoring Program and is referenced in this protocol. The HACH Turbidimeter kit includes the Gelex Secondary Standards (for checking the accuracy of the turbidimeter in the field), and small sample bottles for testing turbidity with the turbidimeter.
- Stabilized Formazin Primary Standard Kit (available from the DEQ Volunteer Monitoring Program) for a more thorough, periodic, turbidimeter calibration. (see the "Calibration" section below)
- Any clean container for taking grab samples.

Site Selection

Site selection procedures described in Chapter 3 (*Selecting Sites*) apply to turbidity monitoring. All water quality samples collected to measure turbidity must be representative of the environmental conditions being investigated. For example, if the monitoring objective is to determine the effects of a grazing activity on turbidity, the sample must be collected in a location directly affected by the grazing activity (immediately downstream of the activity). The easiest place to obtain the sample may be a few hundred feet downstream of the grazing site at a road crossing. However, this would not provide a representative sample because the likelihood of capturing other turbidity-generating activities (a dirt road, development site, etc) increases and the sample is no longer representative of the grazing activity.

Grab Sample

Materials that cause turbidity tend to be evenly distributed in the water column and across the stream cross-section. Therefore a "grab sample" sufficiently represents the sample location. The sample can be collected at any point in the stream (either near the bank or the

deepest part of the channel) by lowering the lip of the sample bottle below the surface of the water.

Sample Timing

Timing of the grab sample is just as critical as site selection. Stream flow greatly influences turbidity. Therefore, it is important to sample for turbidity during similar flow conditions unless the objective is to monitor the differences between low flow and peak flow turbidity. For example, it would be inappropriate to compare a pre-project sample collected during a storm event with a post-project sample that was collected during lower flow conditions. Given the above discussion regarding site selection and sample timing, consider the following guidelines when designing a sample:

- Clearly define the project objectives and monitoring questions. This will help identify sampling location and timing.
- Clearly identify the source, project, or activity being monitored and locate sample points to the closest proximity of these activities.
- Clearly identify the time period or flow conditions of concern and consistently monitor during those times and conditions.
- If the objective is to monitor a specific activity, then obtain turbidity samples upstream of the project site during the activity as a control to monitor background turbidity conditions. These samples should be collected in addition to the samples obtained immediately downstream of the project site.
- If the objective is to collect baseline data on turbidity, the sample frequency and number of locations must be large enough to capture the range of flow conditions and turbidity-generating activities that are occurring in the reach or basin.

Accuracy Check

Field check the turbidimeter against the Gelex Secondary Standards at the start of each set of measurements. If numerous samples are to be processed, periodically check the instrument against the calibration standards and adjust accordingly.

- Place the first Gelex Standard (0 to 10 range) in the cell compartment of the meter with the white diamond on the vial aligning with the orientation mark on the meter. Close the lid.
- Press **"POWER"**, and when 0.00 shows in the display window, press **"READ."** If the reading is not within 5% of the Standard, recalibrate the instrument with the primary Formazin Standard (see below).
- Repeat this procedure with the remaining two Gelex Standards (0 to 100 and 01 to 1000 ranges).

Duplicate Samples

Obtaining duplicate samples is important for documenting the variability from sample to sample. Duplicate samples should be collected at a rate of one duplicate sample for every ten regular samples collected. All samples must be identified on a field data sheet by:

- Description of sampling point
- Identification (or Lat/Long.) of sampling site
- Date and time of collection
- Name of collector

Calibration

The Model 2100P Turbidimeter is calibrated with Formazin Primary Standard at the factory

and *does not require recalibration before use.* With steady field use, however, the HACH Company recommends recalibration every three months, or as often as experience dictates. Refer to the Instrument Manual for complete instructions.

Field Turbidity Measurement Procedure.

Data collection can begin after following the procedures described above for instrument preparation and site selection.

1. Collect a representative sample in a clean container. Fill one of the sample bottles (included with the turbidimeter kit) to the line (approx. 15-ml), taking care to handle the sample bottle by the top to avoid fingerprints and dirt on the bottle. Cap the bottle.
2. Wipe the bottle with a soft, lint-free cloth to remove water spots and fingerprints.
3. Press the **"I/O"** button to turn the instrument on. Place the instrument on a flat, steady surface.
4. Put the sample bottle in the instrument cell compartment so the diamond mark on the bottle aligns with the orientation mark on the instrument.
5. Select the manual or automatic range by pressing the **"RANGE"** key. "**AUTO RNG**" is recommended and will be displayed. Press **"READ."** The display will show "**-------NTU**" then the turbidity reading in **NTU**. Record the turbidity after the lamp symbol turns off.

Notes: on taking Measurements

- Always cap the sample bottle to prevent spillage of sample water into the instrument.
- Always place the instrument on a level, stationary surface.
- Always close the cell compartment lid during measurement and storage.
- Do not leave the sample bottle in the cell compartment for long periods of time.
- Always use clean sample bottles.
- Avoid operating instrument in direct sunlight.
- Make sure that a cold water sample does not fog the sample bottle.
- Avoid allowing the water sample's contents to settle prior to taking a turbidity measurement.
- Always take turbidity measurements within 24 hours of collecting the samples.

A permanent record of each sampling event should be maintained and should include:

- Significant observations related to the sample
- Other ancillary environmental measurements (see below)
- Weather and other physical conditions
- Sample date
- Sample ID

Ancillary Data

Once a site is selected, other important descriptive information should be recorded such as general flow conditions and depths, and references to landmarks such as tributary names, river mile, roads, and bridges. The latitude and longitude of the site is an important piece of information that can be obtained from a topographic map or from a global position device (GPS).

Information about the landowner and contacts (telephone, address, E-mail) should be

recorded. *Document a landowner's granted permission for access to private lands.*
A photograph of the monitoring site can be useful for locating equipment. Guidance for photo documentation of monitoring sites is provided by the Governor's Watershed Enhancement Board (1993).

Turbidity Data Analysis

Once the data has been stored on a computer and on backup disks, data analyses can begin. Analysis of turbidity data depends on the specific objectives.

Project Monitoring
If the collected data will be used to determine whether management activities are increasing turbidities above a given level, then the following information should be included:

- Information on the activity or project.
- Turbidity data upstream and downstream of the activity, and, depending on the objectives, possibly within the reach affected by the activity.
- Collecting data at these same locations before the activity starts is also advisable.

With this information, an investigation of trends between turbidity and the management activity can begin.

Relationships between turbidity and other stream characteristics.

Suspended Sediment
If the turbidity data will be used to determine suspended sediment characteristics, the relationship between suspended sediment and turbidity for the particular sites must be established. Contact the mentor for specific sampling procedures that must be followed for suspended sediment.

Streamflow
Streamflow information can be collected at the same sites as the turbidity data. Monitors should investigate the relationships between increases and decreases in streamflow and turbidity. The Oregon Water Resources Department is developing a protocol for measuring stream flow. Contact a turbidity mentor for more information on this.

References

Beschta, R.L., Jackson, W.L. 1980. Turbidity and suspended sediment relationship. P 271-282 in *Proceedings, symposium on Watershed management*. Amer. Soc. Of Civ. Engineers.

Brown, G. 1983. *Forestry and water quality.* OSU Bookstores Inc., Corvallis, OR. 142 pp.

Macdonald, LH., Smart, A.W., and Wissmar, R.C. 1991. *Monitoring guidelines to evaluate effects of forestry activities on streams in the Pacific Northwest and Alaska.* EPA region 10, 910/9-91-001. 166 pp.

Stednick, J.D. 1991. *Wildland water quality sampling and analysis.* Academic Press, Inc., San Diego, CA.
The project coordinator is directed also to the EPA Volunteer Monitor's Guide to Quality Assurance Project Plans (EPA 1996).

Chapter 12

Stream Macroinvertebrate Protocol

Background: Why Monitor Macroinvertebrates?

Evaluating the biological community of a stream through assessments of algae, macroinvertebrates, and fish provides a sensitive and cost effective means of determining stream condition. Such evaluations are particularly effective when stream impacts are from nonpoint sources, sporadic events, or cumulative low level pollution. Each biological community has its own advantages and disadvantages for assessing stream conditions, and they all have published protocols (Plafkin et al. 1989; EPA 1990). The protocols described here are for macroinvertebrates--invertebrates large enough to see with the naked eye.

Macroinvertebrates are fairly stationary, easy to collect, and are responsive to human disturbance. In addition, the relative sensitivity or tolerance of many macroinvertebrates to stream conditions is well known. In general, they provide a simple "hands-on" approach to understanding and measuring stream health without the problems often encountered when measuring fish communities impacted by sport fishing, stocking sport fish, and the introduction of exotic fish species.

In order to adequately evaluate the overall ecological integrity of aquatic systems, a monitoring program that encompasses chemical, physical, and biological integrity should be developed (EPA 1990). The macroinvertebrate bioassessment protocol described here is part of a comprehensive approach that involves analyzing the stream habitat conditions, its physical and chemical parameters, and the biological community. The physical and chemical water quality parameters routinely measured are listed in Appendix A. The biological community evaluation methods described in this manual are adapted from the EPA Bioassessment Protocols (EPA 1996) and other referenced sources.

Types of Methods

Three different levels of macroinvertebrate sampling procedures are described in this protocol. They have unique objectives and require different levels of expertise.

Level 1

Level 1 methods are the simplest to use and require the least experience. They also provide the least amount of information about the health of the macroinvertebrate community. Education is the main goal for Level 1. If the monitoring objective is to inform citizens or students about the various animals that live in streams, and only a very basic assessment of stream conditions is needed, Level 1 methods will be appropriate.

Level 2

The Level 2 protocol is designed to provide a screening level assessment of stream conditions. Sites can be classed as *heavily disturbed, slightly disturbed,* or *non-disturbed.* Finer levels of impairment will be difficult to detect. If the objective is to screen the condition of a variety of sites for prioritizing more in-depth studies, or if the budget or expertise to complete Level 3 studies is unavailable, then the Level 2 protocol will be appropriate.

Level 3
The Level 3 protocol provides a sensitive measure of stream condition using macroinvertebrate communities as the primary indicator. Four classes of stream conditions can be determined: *no disturbance*, *slight disturbance*, *significant disturbance*, and *severe disturbance*. Applied correctly, studies following this protocol can be used for a variety of objectives such as identifying levels of stream disturbance within a watershed or region, effectiveness monitoring of restoration projects, trend assessments, and evaluating whether the state's standards for protecting aquatic life (fish, macroinvertebrates, algae, amphibians, etc) are met.

Mentor Contacts

As with any monitoring project, questions will come up that are not answered or covered sufficiently in this protocol. Therefore, a group of mentors that are agency experts in monitoring have been identified. These mentors may be contacted with specific questions about particular monitoring goals and efforts. Questions about macroinvertebratre monitoring should be directed to one of the following:

Rick Hafele (503) 229-5983
E-mail: hafele.rick@deq.state.or.us

or

Mike Mulvey (503)229-5983
E-mail: mulvey.mike@deq.state.or.us

Oregon DEQ Laboratory
1712 SW 11th
Portland, OR 97201

Statewide DEQ Volunteer Monitoring Coordinator
Karen Williams: (503) 229-5983
E-mail: williams.karen@deq.state.or.us

North Coast
Larry Caton (503)229-5983
E-mail: larry.caton@deq.state.or.us

South Coast & Willamette
Dennis Ades (503) 229-5983
E-mail: ades.dennis@deq.state.or.us

Eastern Regional Monitoring Coordinator
Larry Marxer (503) 229-5983
E-mail: marxer.larry@deq.state.or.us

Selecting Sites

For an overview of the process used for selecting sites, please review Chapter 3, (*Selecting Sites*.) The concepts presented here apply to any of the bioassessment Levels (1, 2, or 3). Level 1 studies, designed primarily for education, don't require the same consideration as studies designed to assess stream conditions within or between different streams. A site with easy access and a good diversity of invertebrates will be adequate for most educational (i.e. Level 1) projects.

For Level 2 or Level 3 studies, remember that stream habitats are complex and change over distance and time. Different communities can inhabit different portions of the same stream, due to natural and human-caused factors. Also, the composition and abundance of the macroinvertebrate species present can change dramatically between seasons due to life-cycle patterns of the different species.

Careful site selection and monitoring timing is critical to insure that the data collected are not biased, and that the differences noted between

sites are not due to some artifact of the monitoring program design.

Selecting Specific Sample Locations

Streams with flowing water can generally be divided into several habitat types: pools, runs, glides, riffles, bends, undercuts, etc. Within the major habitat types other habitat categories can be created. Examples would be inorganic substrate like rocks and gravel, or organic substrate like submerged logs and leaf packs. Since each habitat type can have a different macroinvertebrate assemblage, deciding what habitat(s) to sample is necessary.

Two approaches to habitat selection are commonly followed: *multiple* and *single habitat* assessments.
Assessing *multiple habitats* involves a sample design that evaluates two or more habitat types. Each habitat type is sampled, processed, and evaluated separately. Pools and riffles are the most common habitat types sampled in a multiple habitat design, but other habitats might be included. The habitats most typical of the study stream should be chosen.

Riffles are usually the only habitat sampled in a *single habitat* assessment. Riffles tend to contain the most diverse and sensitive invertebrate assemblage compared to other habitats (Plafkin et al. 1989). In most cases, a single habitat assessment of riffles will be adequate when sampling streams. However, sampling only riffles may not always be adequate. Defining the questions in the sampling plan will help determine whether single or multiple habitats should be collected.

Note: The analysis procedures presented in this chapter apply to "riffle" habitat only. If monitors plan on sampling other habitat types, they should contact one of the monitoring mentors to determine the best sampling and assessment methods.

When Are Sites Sampled?

Stream habitats will have different macroinvertebrate communities, habitat conditions, and chemical water quality at different times of the year. Bioassessment surveys are typically done over the course of several years, so it is important to repeat sampling at the same time of year to make year-to-year comparisons possible. Sampling several times per year may be desirable to describe the seasonal variability of the stream and to determine the best time of the year to evaluate a specific type of impact. Once the seasonality of a stream has been adequately characterized, it may be possible to reduce the sampling to a single critical season that best indicates impacts.

Effective periods for macroinvertebrate sampling in Oregon include:

Winter: December, January, February
Spring: March, April, May, June
Summer: July, August, September
Fall: October, early November.

Depending on a stream's elevation or region in the state, the months of May/June and October/November can be transition months between seasons, and invertebrate communities may be changing faster than at other times. Most macroinvertebate studies in Oregon are done during summer low flow conditions in July, August and September. Whatever sampling period is selected*, sampling should be avoided during or immediately after high water*, because high flows can significantly effect the ability to collect representative samples**.**

Equipment

The following equipment, listed separately for Level 1 and Level 2-3 assessments, will be needed to sample macroinvertebrate populations:

Table 12-1. Level 1 assessments.

Equipment:	Costs:
• Collection net - Kick screen, or D-frame kick net are the most versatile. If these are not available a large fish aquarium net with fine mesh netting could also be used. Simply picking up stones from the stream bottom is also an option.	• $10 - $50
• Small buckets	• $20
• Waterproof boots or waders	
• Waterproof, insulated, elbow-length gloves (if working in polluted or very cold water).	• $35
• Shallow white plastic tray (ex. 12" x 16" or larger, 1 to 3 inches deep).	• $5
• 2 to 4 white ice cube trays	• $5
• Tweezers	• $5 - $10
• Sample vials	• $10
• Hand lens	• $5 - $30
•Macroinvertebrate field guides	• $10 – 50
• Pencils and paper	• $5
• Denatured ethanol (80-90%)	• $20
	Total Costs: $100 - $200

Table 12-2. Level 2 and 3 assessments.

Equipment:	Costs:
• Sub-sampling sorting tray (Caton Tray)	• $150
• Tripod for field sorting (optional)	• ($50) - optional
• Random number table, or other random # generator	
• D-frame Kick net, 30 cm. wide D-shaped hoop net with 500 micrometer mesh opening	• $50
• Plastic sieve bucket with a 500 micrometer mesh bottom (optional)	• $50
• Plastic jars with tight fitting lids or zip-lock bags, 0.5 to 1.0 liter	• $10 - optional
• Denatured ethanol (80-90%)	• $20
• Shallow white plastic tray (ex. 12" x 16" or larger, 1 to 3 inches deep).	• $5
• Waterproof, insulated, elbow-length gloves (if working in polluted or very cold water).	• $35
• Labeling tape and alcohol-resistant marking pens (ethanol dissolves most inks)	• $10
• Small vegetable scrub brush	• $5
• Tweezers	• $10
• Sample vials	• $20
• Hand lens	• $30
• Macroinvertebrate field guides	• $50
• Paper and pencils	• $5
	Total Costs: $450

Field Sampling Methods

Level 1 Assessments

Field procedures for Level 1 assessments can follow a variety of techniques using simple, inexpensive equipment. The main objective is to collect a representative variety of species from the selected area.

Procedure

- If possible, select a shallow area having a gravel/cobble bottom with a fairly fast current (make sure the current is not too fast for safe wading). Other habitats may also be sampled; for example, wood and leaf debris, pools, and stream margins.
- If using a kick screen or D-frame net, place the bottom of the net firmly against the stream bottom and disturb the area upstream of the net by picking up pieces of large gravel and cobble and rubbing their surfaces by hand or with a small vegetable brush upstream of the net. After most of the cobble-sized pieces have been moved, continue disturbing the stream bottom immediately upstream of the net with hands or feet to a depth of several inches. Repeat this process at two or three locations in the same habitat type and combine the contents from each net into a single sample.
- Remove the net from the stream and wash its contents into a small bucket. Clean and discard large pieces of gravel, leaves, twigs, etc. from the sample.
- If no net is used, pick up pieces of large gravel or cobble and hold over the bucket while rubbing the surfaces clean. Pieces of wood and leaf packs can also be gently washed in the bucket
- Pour the material in the bucket into the white plastic tray, and remove all the invertebrates found.
- Turn to *Sample Processing Methods* section (below) for final processing steps.

Level 2 and 3 Assessments

Both Level 2 & 3 assessments follow the same field sampling methods.

Method Overview

The goal of the field sampling technique is to collect an unbiased, representative sample of macroinvertebrates. First, a "representative" stream reach approximately 40 times longer than the average (mean) wet surface channel width should be selected. From within this sample reach choose two riffles (e.g. if pools will be sampled, select two pools). Two 0.18 square meter (2 square feet) kick samples are randomly selected in each riffle or pool. The four kick samples from each habitat type (riffle and pool) are combined, resulting in one composite riffle sample and one composite pool sample to process in either the field or the lab.

Procedure

- Randomly select two kick-net sites within the downstream riffle or pool. Random numbers in the table used by DEQ have four digits. The first two identify the percent up from the downstream end of the riffle or pool, and the second two are the percent of stream width across the channel. For example, a random number of 3225 would place the sample at 32 percent up from the downstream end and one quarter across the stream width. These percentages are determined by visual estimates.
- After locating the random sample site, place the net into the stream with the flat part of the hoop perpendicular to the stream flow and resting on the bottom. Collect the macroinvertebrate sample by disturbing a 30 by 60 centimeter area (1 ft x 2 ft) of

stream bottom directly upstream of the net so that the current carries the macroinvertebrates and debris into the net.

- Carefully rub by hand, or with a small scrub brush, all substrate larger than five centimeters (golf ball size and larger) in front of the net to dislodge any clinging macroinvertebrates. After rubbing, place the substrate outside of the sample plot.
- Thoroughly disturb the remaining substrate to a depth of five to ten centimeters with the hands or feet. This usually takes between 30 seconds and a minute.
- After the sample is collected and the net removed, the large substrate is returned to the sample plot.
- The contents of the net are placed in a sieve bucket and the sampling procedure is repeated at three more plots for that habitat type. The preferred order for sampling is from downstream to upstream to minimize influences of disturbance to each sample plot.
- All four samples for the same habitat type are combined in the sieve bucket. Large organic material and rocks are rinsed, carefully inspected for clinging macroinvertebrates, and removed. As much fine sediment as possible is washed away. Leaf packs from pool samples may require considerable rinsing and removal of debris before preserving the composite sample.
- For lab sorting and analysis the composite sample is placed in a labeled jar or double Ziplock® bag and preserved with 90% ethanol for sorting and subsampling in the lab. Change the alcohol in the sample with fresh alcohol within one week to ensure adequate preservation. Place a label inside the jar (using paper and pencil), as well as an exterior label.
- For field sorting, do not preserve the specimens. Keep them alive and follow the subsorting procedures described in the next section. Field sorting is faster since live, moving specimens are easier to see. Field sorted macroinvertebrates also tend to be in better condition than lab sorted specimens, making identification easier.

NOTE: The disadvantage to field sorting is that it adds one to three hours to the field time per site. This is especially true for low productivity streams that may require sorting most, if not all, of the sample to get the minimum number of specimens required for analysis.

Sample site:_____

Location:_____Date:_____

Habitat sampled: riffle_____ pool _____ other _____

Collected by:_____

Sampler type: D-net _____ Other: _____

# of kicks composited:_____

# of squares sorted:_____

Figure 12-1. Field sample label information.

Sample Processing Methods

Level 1 Assessments

Level 1 assessments follow a simplified sample processing procedure compared to Level 2 or 3 assessments. For example, Level 1 assessments do not utilize a specific subsorting method or require a minimum number of invertebrates for identification. The main objective is to group the invertebrates by order and determine the number of sensitive or tolerant taxa present (see the *Analysis & Evaluation* Section below for a discussion of "sensitive" and "tolerant" taxa). As a result, Level 1 studies help volunteers recognize the importance of the invertebrate community as indicators of a stream's conditions and provide a general indication of disturbance.

Key Elements

1. Remove all invertebrates from samples collected within the same habitat at the same reach.
2. Sort specimens into individual containers (ice cube trays are often used) by order: Mayflies, Stoneflies, Caddisflies, etc.
3. Visually estimate the number of different types of taxa within each order. For example, how many different looking mayflies are there?
4. Record the number of different taxa within each order and count how many are present.

Based on the numbers recorded, a general water quality rating can be calculated as described in the *Analysis and Evaluation* Section.

Level 2 and 3 Assessments

The goal of the sample processing procedures for Level 2 and 3 studies is to create an unbiased, random representative subsample of macroinvertebrates from the composited stream bottom sample of debris.

The size of the subsample is a minimum of 300 individuals. The same size subsample should be used for all sites for effective comparisons.

Equipment

- Subsampling tray (see Caton, 1991) and associated sorting equipment
- Tripod with sorting tray platform for field sorting (optional)
- Random number table, or other random number generator
- Denatured ethanol
- Vials, approximately 20 mls.
- Labeling tape and alcohol-resistant marking pens

- Forceps
- Squirt Bottle & plastic spoon
- Tally counter (optional)

Procedure

- To sort the sample, place the composited sample into the mesh bottomed sorting tray. DEQ uses the equipment described by Caton (1991).
- Place the mesh bottomed tray into the plastic outer tray and add approximately 3 cm of water to facilitate the even distribution of debris. In the field, place the tray on a level surface or tripod platform.
- Evenly distribute the material in the tray and lift the mesh bottom tray out of the water.
- The sorting tray is divided into thirty separate 6 X 6 cm squares. Use the random number table to select a minimum of four of these squares. Use the 6 X 6 square sorting device (included in subsampling tray kit recommended by Caton) to isolate the four square and remove the selected material.
- Distribute the contents of the four squares into a separate white plastic tray with a small quantity of clean water. All the macroinvertebrates are removed with forceps and placed in a labeled vial of alcohol. An inside paper and pencil label is recommended as well as an exterior label.
- A minimum of 300 specimens and four squares are sorted. If necessary, an additional one or more squares must be sorted to attain the 300 organism minimum sample size. All organisms are completely removed from all sub-sampled squares to avoid biasing the macroinvertebrate sample toward the larger, more visible species. Use a tally counter for best results. Keep track of the number of squares subsampled in order to estimate the original macroinvertebrate density in the stream.
- The Caton sorting tray has thirty squares, each six centimeters square. When four D-frame kick samples are composited, each square represents approximately sixty square centimeters of stream bottom.

Identifying Invertebrates

<u>Method overview</u>

Three different levels of "taxonomic identification" can be used after specimens are sorted: *order*, *family* or *genus/species* level. The level of taxonomic identification is important in determining the cost and expertise needed for the analysis, as well as the resolution and sensitivity of the data to detect environmental impacts.

Level 1 assessments do not identify organisms beyond the *order* level (Ephemeropter, Plecoptera, Diptera, etc.). Within each *order* organisms are simply lumped into similar looking groups. This approach is useful for demonstrating the variety of organisms living in a stream reach, but has limited value in assessing differences between sites. In general a rough approximation of the invertebrate community can be determined and sample sites categorized as having either an adequate or limited invertebrate community. Further sampling and more detailed analysis should be performed using Level 2 or Level 3 assessment methods if concerns about a stream's condition exist.

Level 2 assessments rely on *family* level identification for assessing the invertebrate community. *Family* level identification is faster and requires less expertise than *genus/species* level, but is less sensitive. Three levels of biological conditions may be determined from *family* level identification: *non-impaired*, *moderately impaired*, and *severely impaired*.

L*evel 3* assessments rely on *genus/species* identification for most *orders*. This is the most effective level for evaluating stream conditions and evaluating differences between sites. It also requires the most time and expertise. Because of the identification skills required, contracting specimen identification to a qualified taxonomist for Level 3 assessments is often the most effective approach (costs are typically $50 to $75 per sample). Four impairment categories may be discerned at this level: *non- impaired*, *slightly- impaired*, *moderately- impaired*, and *severely-impaired*. Table 12-3 shows the recommended level of taxonomy for each order.

Level 2 and 3 Identification Methods

Equipment

- Dissecting microscope (10X-60X zoom)
- Light source
- Forceps
- Petri dish
- Macroinvertebrate taxonomic keys. See references for recommended keys (keys in **bold** type are the most important)
- Data recording form

Procedure

- If the sample was not sorted in the field, then lab sort according to the procedure described in the *Sample Processing Methods* section (above).
- Identify the macroinvertebrates to the taxonomic level desired. Table 12-3 lists the level of taxonomic identification for different macroinvertebrate groups recommended for Level 3 assessments.
- Identification to *genus/species* should be performed by experienced entomologists using current taxonomic keys (see *Taxonomic References*) under the supervision of a senior aquatic entomologist. *Family* level identification is possible by less experienced staff, but sufficient taxonomic training is still critical.
- The number of each taxon is noted on a tally sheet along with other site identifier information (see Data Recording Forms).
- Quality control procedures described in the *Quality Assurance* section (see below and Chapter 4 *Data Quality*) should be completed to evaluate the quality of the sample identification.
- The biometrics and biological condition assessments used to analyze the macroinvertebrate data are outlined in the *Analysis and Evaluation* section.

Table 12-3. Level of macroinvertebrate identification for Level III analysis.

	Level of Identification				
Taxon	Order	Family	Sub-family	Genus	Species
Amphipoda (scuds)				X	X
Arachnida (spider and water mites)	X				
Coleoptera (most beetles)		X			
Elmidae (riffle beetles)				X	X
Diptera (most true flies)				X	
Chironomidae (midges)			X		
Ephemeroptera (mayflies)				X	X
Gastropoda (snails)		Some		X	
Hemiptera (true bugs e.g. water boatmen)				X	
Lepidoptera (butterflies & moths)				X	
Megaloptera (hellgarmmites & alder flies)				X	
Odonata (dragonflies & damselflies)		Some		X	
Oligochaeta (worms)	X				
Ostracoda (seed shrimp)	X				
Pelecypoda (clams)	X				
Plecoptera (stoneflies)		Some		X	X
Trichoptera (caddis flies)				X	
Turbellaria (flatworms)	X				
Hirudinea (leeches)	X				

Analysis & Evaluation

Overview

Data analysis and evaluation of stream conditions are often based on assessing the characteristics of the macroinvertebrate community. This is often accomplished through the use of "metrics." Metrics are measures of community characteristics based on single or multiple taxa. The metrics used in this manual have been selected because they are known to change as a result of anthropogenic (human caused) disturbance. Examples include total taxa richness, mayfly richness, % dominant taxa, etc. Each metric is scored (usually 1, 3, or 5) based on scoring criteria. All the individual metric scores are then summed together for an overall "Biotic Index" score for the site. The final biotic index falls within a known range indicating different levels of impairment.

Criteria for the individual metric scores and the impairment categories for the biotic index scores are based on data collected from reference sites in regions similar to the study sites being evaluated. The metric values presented here are based on reference site data collected by the Department of Environment Quality (DEQ) in the Oregon Coast Range. These criteria will work for assessing riffle samples from other Oregon coastal streams, but should not be used to assess other habitat types or streams from other areas of the state. The mentors listed at the beginning of this section should be contacted for assessing streams outside the coast range for the most appropriate metric criteria.

Level 1 Assessments

To develop a general evaluation of a site with Level 1 data the invertebrates are first separated by *order*, then the number of different "looking" organisms in each order are recorded and counted. The different orders of invertebrates can be generally classed as "sensitive," or "tolerant."

Sensitive organisms are those most sensitive to pollution and are first to disappear from the invertebrate community as a result of disturbance or pollution. Those considered sensitive include the following:

- Mayflies (Ephemeroptera)
- Stoneflies (Plecoptera)
- Caddisflies (Trichoptera)

Tolerant organisms are those that tolerate high levels of disturbance and pollution, and remain present after other groups have disappeared. This includes the following orders:

- Aquatic worms (Oligocheata)
- Leeches (Hirudinea)
- Blackflies (Diptera)
- Midges (Diptera)
- Snails (Gastropoda)

Since Level 1 assessments are primarily an educational level, different levels of stream impairment cannot be calculated. The generalized data only provide enough information to determine whether the macrovinvertebrate community appears to be adequate or limited. Sites where each of the three sensitive *orders* (mayflies, stoneflies, and caddisflies) are present and tolerant organisms such as worms, leeches and blackflies make up less than 50% of the total organisms counted from the sample are considered adequate. If any one of the three sensitive *orders* are absent and/or tolerant organisms equal more than 50% of the total in the sample, the site has a limited invertebrate community. Level 2 or 3 assessments are then necessary to evaluate the sites further.

Level 2 Assessments

Level 2 site assessments are based on *family* level identifications. The number of organisms in each *family* are counted and recorded. These values are then used to determine metric values or scores. Metric scores are summed to determine the overall rating for the site. The following table outlines the family level metrics and scoring criteria.

Taxa Richness

This is the total number of invertebrate families identified from the sample.

Mayfly Richness

This is the total number of mayfly families identified from the sample.

Stonefly Richness

This is the total number of stonefly families identified from the sample.

Caddisfly Richness

This is the total number of caddisfly families identified from the sample.

% Chironomidae

This is the total number of chironomids (midges) in the sample divided by the total number of organisms sorted from the sample, multiplied by 100.

% Dominance (top 3 taxa)

This is the total number of the three most abundant organisms divided by the total number sorted from the sample, multiplied by 100.

Add up the scores for each metric to determine the total site score or biotic index. The total scores are used to determine three levels of impairment as indicated below.

Table 12-4. Family level metrics and scoring criteria.

		Scoring Criteria			
Metric	Raw Value	5	3	1	Score (Circle)
Taxa Richness		>18	10-18	<10	5 3 1
Mayfly Richness		>4	2-4	<2	5 3 1
Stonefly Richness		>3	1-3	0	5 3 1
Caddisfly Richness		>4	2-4	<2	5 3 1
% Chironomidae		<15	15-30	>30	5 3 1
% Dominance (Top 3 Taxa)		<30	30-50	>50	5 3 1

Score Range	Stream Condition
>23	No impairment: passes Level 2 assessment. Indicates good diversity of invertebrates and stream conditions with little disturbance. Further sampling will help confirm the site's condition as unimpaired.
17-23	Moderate Impairment: evidence of some impairment exists. Requires further study and more detailed analysis.
<17	Severe Impairment: fails Level 2 assessment. Evidence of stream disturbance exists. Further study may be warranted to confirm level of impairment and potential causes.

Level 3 Assessments

Level 3 assessments are based on *genus/species* level identifications, which provides a more sensitive measure of the invertebrate community's condition. Two analytical approaches can be used for Level 3 assessments: *multimetric* analysis, or *multivariate* analysis. To make accurate assessments between sites, using either multimetric or multivariate analysis techniques, *the same level of identification must be used for each taxonomic group for all sites being compared.* Because levels of identification can vary between taxonomists or between sites due to maturity of specimens or preservation quality, each data set should be checked by a taxonomist for identification consistency.

Multimetric Analysis

This approach is the same as that used for Level 2, except more metrics are incorporated into the analysis. The metrics and associated scoring criteria for Level 3 metric assessments are listed below.

Taxa Richness
This is the total number of invertebrate taxa identified from the sample.

Mayfly Richness
This is the total number of mayfly taxa identified from the sample.

Stonefly Richness
This is the total number of stonefly taxa identified from the sample.

Caddisfly Richness
This is the total number of caddisfly taxa identified from the sample.

Sensitive Taxa
This is the number of taxa identified that are known to be very sensitive to stream disturbance. The list of taxa that qualify as "sensitive" are listed in Appendix F.

Sediment Sensitive Taxa
Some taxa are known to be very sensitive to inputs of fine sediment. The presence of one or more of these taxa indicate that fine sediments are probably not a major concern.

Modified HBI
"HBI" stands for *Hilsenhof Biotic Index*. This is an index of a taxa's sensitivity to organic enrichment that typically occurs as a result of excessive nutrient inputs. Index values for individual taxa range from 1 to 10. Low scores indicate high sensitivity (found only in waters with low organic enrichment). High scores indicate low sensitivity (tolerant of waters with high organic enrichment). HBI index values for each taxa are listed in the taxa list for Oregon streams in Appendix F.

% Tolerant Taxa
This is the percent of the invertebrate community made up of taxa tolerant to disturbance. Taxa counted as "tolerant" taxa are listed in Appendix F. Divide the abundance of tolerant taxa by the total number of organisms sorted from the sample, and multiply by 100.

% Sediment Tolerant Taxa
This is the percent of the invertebrate community made up of taxa tolerant to fine sediments (see Appendix F). Divide the abundance of sediment tolerant taxa by the total number of organisms sorted from the sample, and multiply by 100.

% Dominant (single taxa)
This is the total abundance of the single most abundant taxon in the sample divided by the total number of organisms sorted from the sample, multiplied by 100. A high percent of a single taxon indicates some disturbance has likely occurred to the invertebrate community. After calculating each individual metric score add them together for the total score or biotic index. Stream condition levels are based on the ranges of total scores listed below.

Table 12-5. Genus/species level metrics and scoring criteria.

Metric	Raw Value	Scoring Criteria 5	3	1	Score (Circle)
Taxa Richness		>35	19-35	<19	5 3 1
Mayfly Richness		>8	4-8	<4	5 3 1
Stonefly Richness		>5	3-5	3	5 3 1
Caddisfly Richness		>8	4-8	<2	5 3 1
Sensitive Taxa		>4	2-4	<2	5 3 1
Sediment sens. Taxa		>2	1	0	5 3 1
Modified HBI		<4.0	4-5	>5.0	5 3 1
% Tolerant Taxa		<15	15-45	>45	5 3 1
% Sed Tol Taxa		<10	10-25	>25	5 3 1
% Dominant (single taxa)		<20	20-40	>40	5 3 1

Score Range	Stream Condition
>39	No Impairment: passes level 3 assessment. Indicates good diversity of invertebrates and stream conditions with little or no disturbance.
30-39	Slight Impairment: evidence of some impairment exists.
20-29	Moderate Impairment. clear evidence of disturbance exists.
<20	Severe Impairment. conditions indicate a high level of disturbance.

Multivariate Analysis

Level 3 assessments can also be analyzed using multivariate analysis techniques. In this approach, reference sites (high quality, least disturbed sites) are compared as a benchmark against the sites of interest (test sites). The method has two basic elements: the development of a relatively sophisticated predictive model based upon reference conditions, and direct comparisons of the stream taxa collected at a test site against model predictions.

Multivariate analysis requires the appropriate computer software and knowledge of multivariate statistical techniques. While this limits its current use by volunteer groups, multivariate analysis is a powerful technique that the Monitoring Team plans to make more accessible to groups in the future. Contact the mentors listed in this chapter for more information about multivariate analysis.

Quality Assurance

Overview

Quality assurance procedures (QA) assess the environmental variability, sampling procedures validity, repeatability of the sample methods, and identification quality. The quality assurance procedures involve a system of following standard methods and protocols, duplicate

sampling, and identification reviews. Please refer to Chapter 4 for a general discussion of data quality.

Field QA Sample

Ten percent of all stream sites sampled, or one sample per survey, whichever is greater, should have a duplicate set of field samples collected. The duplicate sample is from the same sample reach. This is called a field quality assurance sample (FQA). Field QA samples look at the natural variability within a riffle and insure that the field sampling method is repeatable. This sample is sorted and identified the same as any other sample.

Laboratory QA Samples

Ten percent of all composite samples collected or one sample per survey, whichever is greater, is re-sorted for an additional 300 specimen subsample from the original preserved composite sample. The result is a duplicate sample from the same composite. This is a laboratory quality assurance sample (LQA). Lab QA samples look at the variability inherent in the subsampling procedure and insure that the subsampling method is repeatable and within an acceptable range of variability.

Type Collection

It is useful to maintain a macroinvertebrate type collection for each major basin, watershed, or ecoregion studied. This collection has a representative of each taxon identified and serves as a basin record, and as a reference for checking identifications.

Identification Review

For Level 3 assessments, data should be reviewed by an experienced taxonomist for anomalous identifications. Randomly selected samples should also be identified by an experienced entomologist independently of the first identification. Finally, specimens entered into the type collection should be checked by an experienced entomologist for accurate identification.

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Chapter 13

Pesticides and Toxins Protocol

The following protocol describes methods to determine if pesticides or chemical toxins are present in surface waters of streams. Pesticides include herbicides, insecticides, rodenticides, and other chemicals used to control unwanted vegetation or pests. Chemical toxins can include a suite of materials often associated with urban or industrial discharges. Examples of chemical toxins include chlorinated phenols and polychlorinated biphenyls (PCBs). How each individual, group or agency works its way through the protocol will depend on their respective technical background, experience, and the goals of the monitoring project. By following the protocol recommendations given below for maintaining sample integrity, collection methods, and sample analysis options, it will be possible to develop regional data sets. These data sets will be extremely valuable to the OPSW effort to restore and protect salmonid habitat throughout Oregon.

Mentor Contact

As with any monitoring project, questions will come up that are not answered or covered sufficiently in this protocol. Therefore, a group of mentors that are agency experts in monitoring have been identified. These mentors may be contacted with specific questions about particular monitoring goals and efforts. Questions about pesticides and toxins monitoring should be directed to one of the following:

ODF Monitoring Coordinator
Liz Dent (503) 945-7493
E-mail : liz.f.dent@state.or.us
Oregon Department of Forestry
2600 State Street
Salem, Oregon 97310

or

Statewide DEQ Volunteer Monitoring Coordinator
Karen Williams (503) 229-5983
E-mail: williams.karen@deq.state.or.us

General information of pesticide and toxics is available from the following references:

Background Information On Pesticides and Toxins

This section and the following sections will focus on monitoring for pesticides associated with nonpoint source activities like agriculture or forestry. However, urban point sources can also be a source of pesticides and industrial/urban toxins. When samples are being collected for these materials in urban areas, the same guidelines to avoid contamination or degradation of the samples should be used.

Why Monitor?
High levels of pesticides or toxins in water may affect fish and other aquatic organisms' health and productivity. Toxicity is related to both the level of exposure (dose or concentration) and the duration of the exposure (acute or chronic). Some chemicals are highly mobile in water while others are not. In order to be detected, chemicals must first enter the water column through either a direct application to the water body, aerial drift, transport through ground water, or overland flow.

After entering the water column, chemicals differ in their potential effects. Some herbicides, depending on their concentration

and duration, may indirectly effect aquatic animals through effects on aquatic plants. Insecticides, rodenticides, or fungicides may have direct effects on aquatic animals depending on the chemical and concentration. Chemicals used in forest, agricultural, and urban settings differ. County extension offices or the Oregon Department of Environmental Quality (DEQ) are resources for associating land use and stream habitat setting with the kinds of chemicals that may be affecting streams.

Pesticide Use and Runoff Patterns in Oregon

A number of studies have been conducted or are underway to assess the introduction of pesticides into Oregon waters. The USGS recently published results of monitoring in their report entitled "Occurrence of Trace Elements and Organic Compounds and Their Relationship to Land Use in the Willamette River Basin, Oregon, 1992-1996" (Anderson, Rinella and, Stewart, 1996). They found that

> "…of the 25 most frequently detected pesticides, 3 were found primarily at urban sites, 6 were found primarily at agricultural sites, and 7 were found at all types of sites except for forested."

For some land-uses, chemicals can only be detected for a short time immediately after a spray operation or when a storm causes runoff (see the discussion under *Selecting and Sampling Sites* below). For example, Oregon Department of Forestry (ODF) monitoring has found herbicide concentrations at or below detection levels in most of their samples even when they were collected at the time of a herbicide application (ODF 1992). Watershed Councils need to carefully consider when and if to monitor for these chemicals.

Monitoring Plan Considerations

A monitoring plan provides a guide for how, when, and where to monitor surface water toxins. A more detailed discussion about developing a monitoring plan is provided in Chapter 2 (*Monitoring Strategy and Plan)* in this document.

Selecting and Sampling Sites

Most chemicals reside for a very short time within the water column of a flowing stream. Through time they either:

- metabolically break down into other compounds,
- remain active but settle into the stream sediments,
- or dilute into undetectable, biologically benign amounts.

As a result, sampling to detect pesticides in the water column often requires that sampling occur immediately after chemical application or following a storm event. Depending on chemical stability and longevity in the environment, run-off events after heavy rainfall may cause more contamination to streams then aerial drift during spray application.

Most pesticides will be collected as nonpoint source samples. This means that the potential pollution source originates from a large area of land and may enter the water column through a number of means and at a number of points. Point source pollution sampling may also apply in some agricultural and urban settings. Sampling for point source pollution is relatively simple. The effluent from a pipe, culvert, or other outlet originating from the polluting source (factory, sewage treatment plant, feedlot) is a direct connection to the stream and sampling can be done at the discharge site. Point source sampling should occur at the mouth of the outlet pipe from the pollution source where the pipe

links to the stream system. Samples may be collected directly from the outlet.

For nonpoint source sampling, the sample point should generally be located within 200 feet downstream from the lower edge of the chemical application or land use boundary. It is critical throughout the sampling process that neither the sampling equipment nor the personnel come into contact with the chemical. This contact could contaminate the water samples. Therefore, do not sample too close to the operation. However, a distance too far from the application may introduce confounding factors such as tributaries or incoming ground water which can dilute concentrations in the flow. The sample point should also have easy access, even at night, and the stream should be deep enough to adequately fill the collection jar. A uniform stream bottom also facilitates stream flow readings.

Sample Analysis and Equipment Options

Laboratory methods for pesticides and toxic chemicals tend to be exacting and expensive because these chemicals are usually present in the water in very small amounts. Also, they occur in an already chemically-complex water column. Quality assurance is important for both the collection and transport of samples to the laboratory and also in the analysis.

Different types of laboratory and field methods can be used for analyzing pesticides and toxins. Some analytical methods can be relatively simple and inexpensive. Commonly, the most precise and accurate methods are costly and involve trace analytical techniques and equipment. Some chemicals are more difficult to analyze than others. The cost of the analysis is directly related to the complexity of the analysis (type of chemical being detected, matrix of chemicals in the water) and the minimum detection limit requirements.

Setting Appropriate Detection Limits

For litigation or situations that require quantifiable proof and testing to very low concentrations, laboratory methods can provide excellent answers with low margins of error. These tests are expensive, however, ranging from $200 to $300 per sample. Laboratory tests using a gas chromatograph, high performance liquid chromatograph (HPLC), or even mass spectrometer, can detect some chemicals down to ppb (parts per billion) and ppt (parts per trillion).

Often, however, analysis of such small amounts of chemicals in water samples are not needed. Rather, local groups may want to know if the chemical levels exceed an EPA, DEQ, or other water-quality standard. These “action” concentrations can be much greater than a few ppt or even ppb. The Watershed Council needs to consider carefully what detection limits are appropriate for which chemicals as part of its monitoring plan.

Immunoassay Alternatives

One emerging analytical alternative is the use of immunoassay techniques. These tests involve the use of an antibody to the chemical of interest. Sites on the antibody are tied up by the chemical. A colorometric reaction is often used which can indicate the concentration of the chemical in the water. Immunoassay techniques provide an inexpensive alternative where they provide sufficient detection limits, precision, and accuracy. Costs range from about $150 to $400 for up to twenty tests, depending on the chemical. Detection limits as low as 0.05 ppb are reported for some immunoassay methods but problems can occur with false readings attributable to other chemicals.

Immunoassay test kits are only available for a limited number of chemicals. Other chemicals still need to be analyzed in a lab. However, if the chemical of interest can be analyzed by

available immunoassay techniques, these tests provide quick and accurate results.

General Equipment Considerations

The type of analysis planned as well as the chemical(s) to be tested will dictate the types of equipment needed. Collecting samples and sending them to a certified laboratory for analysis will require one set of sampling equipment. Immunoassay kits come with the equipment required to collect the samples to perform the specific analyses.

To successfully collect data for the OPSW, care must be taken to collect clean, uncontaminated water grab samples. The type of collection jars used is part of that process. If the samples will be analyzed by immunoassays, the immunoassay kit will contain the appropriate collection jar. However, if samples will be sent to a lab, the lab must be contacted prior to ordering sample jars. Some chemical toxins react to the plastics used in non-glass jars while others are not affected. Other chemicals require frozen storage until analysis, which means plastic containers will be needed. Most labs require that samples be stored in jars with special lids which provide a barrier and seal against the introduction of outside contaminants after closing. Be sure to ask what jars are best for the chemical(s) that will be sampled.

Required Equipment for the Laboratory Method

- Appropriate sample jars and lids
- Labels
- Sterile gloves
- Ziplock" bags larger than the sample jars
- Ice/frozen water jugs/blue ice packs
- Cooler
- Stopwatch
- Permanent waterproof marker
- Lab forms and clipboard
- Flow meter
- Watch

The jars and gloves can be ordered from any scientific supply company. Other equipment is easy to find and purchase. Be certain to confirm the sample collection requirements with the laboratory where samples will be sent for analysis. The Oregon Department of Agriculture (ODA) Laboratory has defined specific container and storage temperature requirements for given chemicals. Some samples must be analyzed within 24 hours of collection or else they must be frozen. If samples will be mailed to the lab, the lab technicians may be able to recommend the most efficient and safest method to prevent sample degradation.

Contact a local ODA or DEQ office or a monitoring mentor to locate the nearest analytical laboratories.

Equipment Required for Immunoassay Methods

Field kits exist for some pesticides that can screen for those compounds in the field. The kits contain the following:

- Test tubes and rack
- Assay calibrators
- Control
- Appropriate enzymes
- Appropriate substrates
- Stop solutions

Other recommended equipment include the following:

- Sterile gloves

- Lab glasses
- Stopwatch
- Flow meter
- "Write-in-the-rain" data sheets
- Clipboard
- Watch

A variety of laboratory and environmental testing companies offer immunoassay kits. Inquire with the EPA, DEQ, or city/county water quality offices for assistance and recommendations on criteria to decide which kit will best fit the monitoring needs.

Laboratory Quality Assurance

In addition to delivering samples in good condition to the laboratory, utilizing a laboratory that can provide analysis of the samples at the desired level of sensitivity, accuracy, and precision is also important. Some of the components to high quality analysis include the use of *pre-treatment control samples*, *field* and *laboratory blanks*, *internal standards*, and *surrogate spikes*. The following discussion provides a short overview of the treatments used to insure quality laboratory analyses. If more information is desired about any one these quality assurance processes, contact the mentor listed in this section.

Pre-treatment control samples are collected before an operation begins. They provide a measure of existing chemical load or possible interferences with the analytical technique. Field and laboratory blanks are control samples that provide an indication of potential contamination in the field or from the sample collection or analytical equipment and extraction materials. Internal standards are either measured amount of the chemical of interest or distilled water added to the sample which react similarly to the chemical of interest (referred to as the "analyte"). "The ratio of the internal standard response to the analyte response is called the relative response factor, and it calculates analyte concentration" (Keith 1991). Internal standards are added at the end of the extraction process. Surrogate spikes are added at the beginning of an analysis. Like an internal standard, they are chemicals which are expected to behave similarly to the analyte. Surrogate spikes allow for determination of recovery efficiencies (percent recovery) as part of the sample clean-up and preparation (Keith 1991). Be prepared to ask questions and work with the laboratory to insure that data quality is maintained.

Field Protocol

The following field protocol is appropriate for pesticide sampling below a planned spray operation. Collecting water grab samples for pesticide analysis is a relatively easy process. Two important considerations must be remembered. The first is:

Do not contaminate the samples in the following ways.

- Entering the spray area
- Driving through the spray area
- Coming in contact with operators or their equipment
- Exposing the collection jars to potential contamination
- Coming in contact with the water column before it enters the collection jar (stand downstream, do not let clothing or skin contact the water upstream)

The second important consideration is: *Remember to collect a control sample and then collect subsequent samples on the appropriate time schedule.*

Following these two important points will facilitate a successful sample collection.

Sample Location

Sample sites should have already been selected. Samples will be collected approximately 200 feet downstream of the edge of treatment unit. Access to the sampling site should be done without walking or driving through the treatment unit. The site should be protected from drift, have a uniform cross-section (no backwater or eddies), and have adequate flow to facilitate sample collection.

Sample Timing

A control sample will be collected within 24 hours prior to the start of the application. Follow the Laboratory Sample procedure below or the methods indicated with the immunoassay kit.

After collecting the control sample, measure the velocity of the stream (ft/sec) using a velocity or flow meter (contact the mentors for information regarding measuring flow in the field). Estimates of velocity can also be obtained using the informal "chip" method. Using a brightly colored, buoyant object (light-colored wood chip, cherry, tennis ball, etc.), record the time it takes the object to travel a pre-measured distance. This provides a "feet traveled per second" reading. Three or more readings should be taken and averaged. Record the stream velocity.

Five more water quality samples will be collected based on the travel time of the water moving through the treatment unit. Samples will be collected approximately 15 minutes, and 2, 4, 8, and 24 hours after the first swath has been sprayed near the buffer strip. The actual time of collection is calculated as follows:

Equation: $$\frac{L/v}{60 \text{ seconds}} + 15 \text{ minutes} = 15 \text{ minute sample time}$$

where, L = length of stream between top of treatment area and sample point plus length of stream between bottom of treatment unit and sample point divided by 2 (ft).v = average velocity of stream (ft / sec)

Runoff Sampling

Runoff sampling is appropriate at all sites where a runoff event occurs within the first 72 hours of the chemical application. Samples can be collected within the first 12 hours after the first runoff event. Only one runoff sample need be collected as long as the sample captures the runoff event. However, depending on the monitoring objectives, samples may be collected at the beginning, middle, and end of the event.

Operator Questionnaire

It may be worthwhile to gather data about the chemical application from the operator. A form template is provided below for operators or landowners to fill out. The information requested deals with how the chemicals were applied. If this monitoring site's results will be compared to others, this kind of information is invaluable (Figure 13-1).

Landowner:_____
Person's name completing questionnaire: _____
Unit Name:_____
Date of Application: _____

Weather Conditions:
Please fill in measurements of:

	Time	Time	Time	Time	Time	Time	Time	Time
Wind Speed:	_____	_____	_____	_____	_____	_____	_____	_____
Wind Direction	_____	_____	_____	_____	_____	_____	_____	_____
Relative Humidity	_____	_____	_____	_____	_____	_____	_____	_____
Temperature	_____	_____	_____	_____	_____	_____	_____	_____

Chemical Application
Start time _____

End time _____Was chemical directly applied within 60 ft of the stream? _____
Target vegetation/pest:_
Active ingredient pesticide: _____ lbs/acre applied_____
Additional pesticide used: _____ lbs/acre applied_____
Surfactant added: _____ amount/acre _____
Other additives:_____ amount/acre _____
Application rate for final spray mixture_____ amount/acre _____
Carriers used: _____
EPA Registration number _____ Trade Name _____

Operation
Helicopter/plan/tractor model: _____
Flight altitude: _____
Air/ground speed: _____
Boom length:_____ Boom Pressure _____
Flight centerline offset from edge of buffer:_____
Half Boom used ____ Yes ____ No
Nozzle type, size, angle, orientation:_____
Number of nozzles: _____

Figure 13-1. Operator questionnaire.

Procedures

Laboratory Sample Procedure. Arrive at the sampling site without physical contact with vehicles or personnel from the spray operation. Comply with the following procedure:

1. All equipment will be clean and free of chemical residues.
2. For each sample, put on a new pair of surgical-type sanitary gloves and pick up container.

3. Fill out two labels identifying the sample, the date, the location, and the time. Place one on the bottle and one on the lid. When using a plastic container, the sample number should be written directly on the bottle as well as on the label.

4. Stand downstream of the sample location. Do not let clothing make contact with the water.

5. Triple-rinse sample container in the stream water (unless a preservative is used) at the sample site. Empty rinse water downstream.

6. At the sample time, face upstream and slowly sink container into the mainflow of the water column until the lip of the jar is just below the surface. Fill container.

7. Fill out Water Quality Sampling form (Figure 13-2).

Draw a schematic map of the unit, streams, buffers, and application patterns.

Notification number: _____

Applied pesticide: _____

Stream name: _____

Monitor's name(s): _____

Spray start time: _____

Average stream velocity: _____(ft/sec) _____

Sampling start time:_____Date:_____

SAMPLE DESCRIPTION	SAMPLE COLLECTION DATE TIME	SAMPLE ID NUMBER
Control Sample		
15 minute		
2 hour		
4 hour		
8 hour		
24 hour		
Runoff Sample #1 (optional)		
Runoff Sample #2 (optional)		
Runoff Sample #3 (optional)		

Figure 13-2. Water quality chemical sampling form.

Upon submission of the water samples to the lab, a *Universal Sample Collection and Laboratory Report* form and a *chain of custody form* usually need to be completed and turned in. Copies of these can be obtained from the lab.

Sample Storage and Delivery to Laboratory. The lab should be notified ahead of time about delivery. Samples should be put immediately in watertight cold storage with a leak-proof cooling device (blue-ice, frozen water jugs, double-bagged ice cubes) and remain so until delivered to the lab. Samples should be transported to the laboratory as soon as possible. At no time should any sample be in contact with personnel directly involved with the chemical operation.

Sample Analysis and Evaluation. The samples may be analyzed individually to determine concentrations of the chemical throughout time. This is highly accurate but the most expensive option because each of the six samples will be billed. A 24-hour average can be approximated from these results with the following formula. This formula applies a time-proportionate weighting factor to each grab result.

Equation:

$$24\text{-hr average concentration} = 15\text{-min}(0.02) + 2\text{-hr}(0.08) + 4\text{-hr}(0.10) + 8\text{-hr}(0.30) + 24\text{-hr}(0.50)$$

(Note: 15-min, 2-hr, 4-hr, etc., refers to the pesticide concentration collected at those time intervals, and not the time the sample was collected.)

Samples can also be combined into one sample for analysis. Composites will usually be formed by the lab so the samples should be delivered in individual containers. Analyzing composites results in losing the ability to detect a 24-hour maximum concentration. The decision to analyze composites or not is a budgetary one.

Immunoassay Procedure. The procedures for the immunoassay tests will be detailed in the test instructions within the kit. All sample integrity and contamination concerns from the laboratory methods apply to the immunoassay tests as well. Six samples through time, including a control, should be taken. Sample vials should be labeled and kept separate from other samples. New sterile gloves and clean equipment should be used with each sample. A stable and sheltered area in the field either near the stream or near the vehicle should be established to complete the tests because they may take up to two hours to complete. Refer to the directions and technical assistance offered by the test manufacturer for more questions.

Analyzing Data

The level of analysis will depend on the initial objectives of the monitoring project. If the goal is to determine if pesticide or introduced toxin levels are above a state or federal standard, then the laboratory or immunoassay results will answer the question affirmatively or negatively. If the goal is to determine if post-application levels exceed control levels, then lab or test results will indicate that as well.

If the monitoring project goal involves comparing different site responses or application rates or techniques, then a more complicated analysis will be required. The sample size (number of individual spray operations) will need to be larger. Multiple sites or spray applications may be compared as long as the environmental conditions that differ between sites are thoroughly and completely

measured and documented. Graphical comparisons of the condition of interest will be required. Contact the OPSW surface water toxins monitoring mentor for assistance on collecting reliable and pertinent environmental data and analysis options.

Individual sites may be compared through time as well. Changes in flow and other factors that directly affect potential toxin levels must be documented and changes in buffers or management techniques that may indirectly affect toxin levels should also be recorded to maximize the value of the data collection effort.

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Appendix A

Oregon Salmon Plan Monitoring Framework

The OPSW monitoring team has developed the following monitoring framework to guide and coordinate monitoring efforts. The components of this framework are described below and in Table A-1. Most questions related to monitoring will address one or more of these components.

For more information on the OPSW monitoring team, please contact Kelly Moore with the Governor's Natural Resource Office (541-757-4263 ext. 226).

I. Condition Assessment

What are the historical, current and future desired conditions in the watershed that restoration activities and changes caused by restoration activities can be measured against?

Historical, current and desired future condition are monitored in this component.

II. Ecological and Cultural Trends

What are the trends in the productive capacity and resilience of Oregon's aquatic ecosystems and salmonid populations?

Monitoring the trends of salmon populations and aquatic ecosystems over time and space and inferring how much of these trends are due to indirect management influences is reported in this component.

I. Management Actions

Are resource-management activities being implemented in accordance with the Oregon Plan?

Implementation of the plan and individual land-use practices are monitored and reported in this component.

II. Management Effects

Are resource-management activities effective at meeting their specific objectives and supporting the mission and goals of the Oregon Plan?

Consistency of various management activities with the goals of the Oregon Plan and the effectiveness of those activities in meeting the plan's goals are monitored and reported in this component.

III. Research

What are the cause and effect and/or mechanistic relationships between salmon, salmon habitat and resource management? And, what are some improved technology and methods that can be applied to answer the questions?

Issues better addressed with research are reported in this component as well as references to guide monitoring activities and interpretation of results.

Table A-1. Revised conceptual framework and example of how the sediment issue could be addressed with this framework.

	Condition Assessment	**Ecological & Cultural Trends**	**Management Actions**	**Management Effects**	**Research Topics**
Monitoring Indicators	Historical sediment sources and sinks	Turbidity/ percent fines in substrate	Road hazard survey/road maintenance & reconstruction	Reduce delivery of sediment to channels from roads.	Suspended sediment in road drainage ditches and structures
Monitoring & Research Questions	What have been the sources of sediment over the period of record (air photos)?	What is the annual trend and range in turbidity? What is the range of turbidity levels during storm events?	Are landowners implementing the road hazard survey? Are road maintenance & reconstruction activities being implemented properly?	Are the road improvement practices reducing sediment delivery to stream channels?	Do road-improvement practices reduce suspended sediment loads in drainage ditches that deliver to channels?
Coordination and Oversight Standards					
Agency Responsibility					

Appendix B

Monitoring Types

The appropriate monitoring design depends on the purpose for monitoring and the resources available to monitor. It is impossible to monitor everything, everywhere, all the time, so experience and judgment must be used to select the appropriate type and intensity of monitoring. Six general monitoring types are useful for monitoring activities associated with the OPSW: ***baseline***, ***trend***, ***implementation***, ***effectiveness***, ***compliance***, and ***validation*** (Ice et al. 1996) (Figure B-1). Because the purpose of each of these monitoring types is different, their requirements are also different. The six monitoring types are described below.

Baseline

Baseline monitoring is designed to characterize existing or undisturbed conditions for comparison with other monitoring activities. This type of monitoring can be useful as a starting point for other monitoring efforts (especially trend monitoring, project monitoring, and effectiveness monitoring). Sites for baseline monitoring must be carefully selected to insure they are representative of the conditions with which they will be compared. Upstream monitoring is often used to set the baseline for temperature changes observed downstream. However, because many factors influence temperature through a reach, before and after monitoring, or *temporal* baseline monitoring, can greatly strengthen interpretation of results.

Trend

This monitoring type requires development of a record over time (usually five years or more). Sites must be established which are "stable" and not impacted by ancillary factors. For example, if the purpose for monitoring is to determine the long term trend in stream temperature with recovery of riparian shade following a wildfire, then monitoring sites would need to be located downstream of the wildfire site. But monitoring sites would also need to be positioned where changing influences, like a new upstream reservoir (which can control temperature by regulating flows and the temperature of water releases), can be avoided or accounted for in the monitoring plan. Measurement methods must also be "repeatable" over the monitoring period.

Implementation

This type of monitoring assesses whether activities were carried out as planned. The most common example of this monitoring is an assessment of Best Management Practice (BMP) or forest practice rule implementation. Implementation monitoring of stream temperature response might focus on determining whether the forest practice rules for shade retention are being met.

Effectiveness

Effectiveness monitoring is used to determine whether properly implemented control practices work. An example of the effectiveness monitoring is the stream temperature monitoring conducted as part of the Alsea Watershed Study to determine the effectiveness of forest buffers in minimizing increases in stream temperature following logging (Brown 1970). The ODF (1994) protocols are specifically designed to develop information to assess the effectiveness of the forest practice rules for riparian areas to meet temperature goals. ***Project monitoring*** looks at the effectiveness of a particular project and the combination of

measures used to protect water quality. Effectiveness monitoring requires that the conditions influencing performance be assessed and that control measures be properly implemented.

Compliance

Compliance monitoring is a special type of effectiveness monitoring to determine whether specific performance standards are met. For stream temperature, compliance monitoring would be designed to determine whether the stream temperature increase follows upstream management approaches or exceeds water-quality standards. The location, frequency, and method of measurement may be specified as part of the standard.

Validation

This type of monitoring is used to assess the performance of a model or standard. A validation study might be designed to monitor fish populations and stream temperature simultaneously for a variety of conditions to determine whether the current water quality standards provide appropriate protection and whether assumed relationships between fish and temperature are valid.

Clearly stating the purpose of a monitoring effort and developing a sampling plan is important in answering questions about where to locate the monitoring, what frequency and how to monitor, and how many monitoring sites are appropriate. The project coordinator is directed also to the EPA *Volunteer Monitor's Guide to Quality Assurance Project Plans* (EPA 1996)

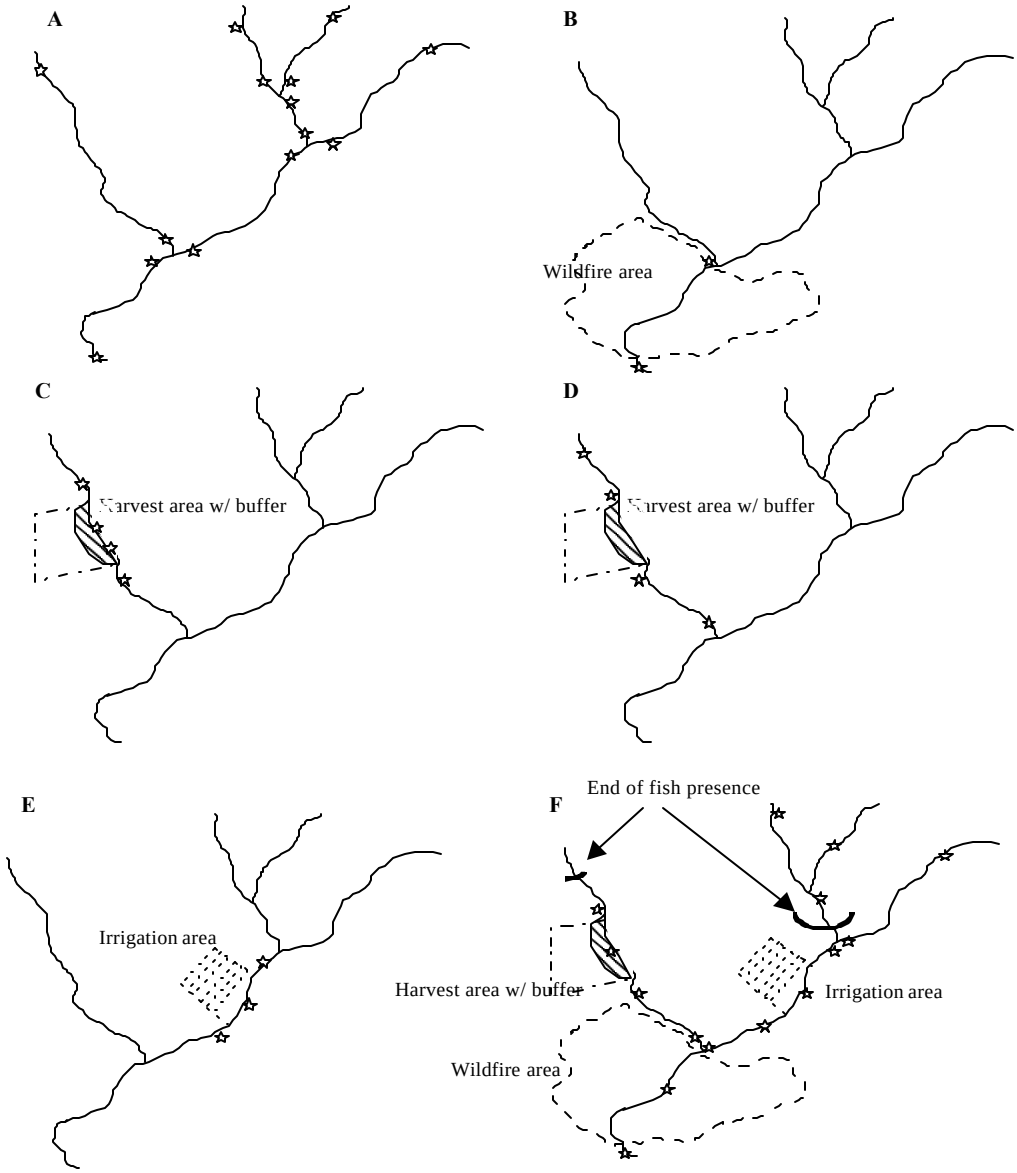


Figure B-1. Schematic examples of monitoring types applied within a sub-basin. (✰) indicate locations of stream temperature monitoring. Spatial scale is an important consideration in determining which monitoring type will best suit a monitoring objective. This diagram is intended to be as "scale-less" as possible so as to illustrate the concepts behind the monitoring types. Careful consideration of monitoring objectives is critical before directly applying figure locations to a field situation. A = baseline monitoring for basin characteristics. B = trend monitoring of recovery from wildfire at two stations over 8 years. C = implementation monitoring to assess shade retention on a recent harvest site. D = effectiveness monitoring to determine if a streamside buffer is effective in protecting stream temperature. E = compliance monitoring to determine if field irrigation withdrawal and return flow are increasing stream temperature above the state standard. F = validation monitoring to test the response of fish to stream temperature changes. Stream temperature and fish presence is measured at each site in the basin.

Appendix C

Watershed Data for Interpretation of Temperature Information

Information about watershed and site conditions may be needed to interpret the information collected. The ODF Temperature guidelines (ODF 1994) provide recommendations for documenting stream and stand conditions. Guidelines include diagramming the site and stream segment; acquiring aerial photos; photographing stream segments; measuring typical depths and wetted widths; estimating substrate composition; measuring the percent of the stream exposed to sunlight with densiometer[12] measurements of canopy cover, and making a general description of each stream (type of shade, tributaries, management history, etc.). Confounding factors discussed in **Monitoring Site Selection** such as beaver ponds or presence of springs need to be documented as they may influence interpretation of results. Collecting information about the property ownership, a contact person, and any management information such as cropping, grazing, irrigation, timber harvesting, and site preparation will also be useful.

Detailed information about physical riparian and watershed measurements can be obtained from the ***Physical Habitat Team Report***. At a minimum, data on the shade characteristics at the site and immediately upstream and downstream of the site should be collected. The recommendation of the Water-Quality Monitoring Protocol team, based on personal experience, is that the vegetation immediately at the monitoring site and at least 1,000 feet upstream from the site should be characterized by taking eleven measurements at 100-foot intervals. These measures may then be averaged to obtain a general numeric description of the stream segment influencing stream temperature at the thermometer location.

[12] A densiometer is a convex mirror engraved with a cross-shaped grid of 24 quarter-inch squares. The mirror reflects trees and other objects above a stream or in a forest stand and is used to quantify shade or canopy closure. (Available from Forestry Suppliers ~$100).

Appendix D

Road Hazard Inventory

Background

The most common sources of sediment in rural and forested areas are from unsurfaced roads. Monitoring source areas of sediment can identify inputs of sediment to the stream system that may need to be mitigated. Ideally this should be done on a watershed scale, because other sources of sediment are also present in the watershed. This protocol only addresses road-related sources of sediment.

Erosion associated with roads and ditches typically includes both surface erosion and landslides. Road construction disturbs and compacts soils and prevents revegetation. Therefore, in the forested landscape, roads are the greatest potential source of sediment outside the stream channels. This can occur in the form of surface erosion or landsliding.

Past monitoring indicates three major areas of concern for road-related erosion. One concern is excess spacing of cross drainage on steep gradient roads. Another is a side ditch routed over long distances with direct discharge into channels. Finally, road-related landslides are typically associated with steep sidecast material. The three major elements (Table D-1) of the road hazard inventory address these road concerns.

Table D-1. Elements of road hazard inventory

Inventory Elements	Area of concern
Stream crossing structures	Washouts of crossings and fish passage through culverts
Sidecast fill on steep slopes	Sidecast-related landslides entering channels
Road surface drainage systems	Muddy drainage waters delivered to streams

In order to use this protocol, several terms need to be understood by monitoring participants:

Road prism
Cross section of roadway from the top of the excavated area (cut) to the toe of the fill.

Cutslope
Slope created by excavation into the natural hillslope. The cutslope is steeper than the natural slope.

Sidecast
Unconsolidated excavated material pushed to the slope below the road. Sidecasts are generally not used as part of the road and are steeper than the natural slope.

Fillslope
Excavated material placed below the road and intended to serve as part of the road.

Inslope
Road surface that is sloped so that all water drains toward the ditch or cutslope.

Outslope
Road surface that is sloped so that all water drains toward the fillslope or sidecast.

Berm
A continuous pile of fill and/or aggregate, usually on the outside edge of a road which prevents surface water from leaving the road.

Cross drain culvert
A culvert installed under and across a road to carry ditch water to the downslope side of a road..

Stream crossing culvert
A culvert installed in a stream channel intended to carry stream flow under the road.

Bridge
A structure intended to carry vehicles over a stream or other feature, usually consisting of a span and abutments.

Log puncheon
A drainage structure made of logs (often cedar) and no longer in common use.

Ford
A stream crossing where stream flow covers the crossing for all or part of the year.

Waterbar
A constructed ditch and berm designed to direct water across the road.

Dips
A cross drainage structure where a low spot is excavated along the profile of the road and where surface water of stream flow is directed across the road.

Grade break
Location where the road grade reverses (typically on a saddle or ridge) and surface water automatically drains away from the road surface in question.

Ditch
Trench constructed at the toe of a cutslope and intended to keep water off the road surface. Ditch water is drained down slope along the road to some point of relief or cross drain.

Landing
An area constructed for logging equipment and log handling operations. Landings may be at the end of roads, or constructed as wide spots in the road. They are typically wider than the rest of the logging road.

Ridge Road
Ridge roads are located on or near the ridgeline (most or all of the road on the top one-third of the slope).

Midslope Road
A road located between a ridge and stream channel

Valley Road
Any road which generally parallels a stream in places, usually in the former riparian area of the stream.

Equipment Needs

In order to successfully and efficiently collect road data, the following equipment is needed:

- Vehicle—a vehicle (pick-up or utility rig) is preferred for road access, although a mountain bike can also be used where access is poor.
- Two person crew—a single person can collect the necessary data, although a crew of two can be more effective. The inventory person or crew can also be used to mark culverts and to flag locations needing immediate maintenance attention.
- Distance Measuring Instrument (DMI) and Hip Chain (String Box)—a DMI or other device that records vehicle travel distance in feet is recommended to accurately record distances while traveling along roads. Impassable roads are measured with a hip chain (string box).

- Clinometer—a clinometer is used to determine average road gradient and hillslope steepness. More accurate measurement tools (engineer's level) are required for any actual repair activity.
- Scaled rod or staff and a measuring (loggers) tape—lengths of culverts and bridges will be measured with these tools.
- ODF stream classification maps—on USGS 7.5 minute quad maps and/or other maps showing roads and streams are also needed.
- Global Positioning System (GPS)—GPS may be used to map road features. However, use of GPS to date has significantly slowed data collection, and is not an essential component of this protocol. GPS efficiency is poor in areas of narrow canyons or when the canopy is wet.
- Data Logger—direct data entry into a field data-logger as it is being collected can be very efficient.
- Computer System and Software—inventory information should be entered into relational databases. Relational databases are probably the most effective tool for making sense of large amounts of information. Commonly available software can be used to query the database to find high erosion hazards or barriers to fish migration.
- Geographic Information System (GIS)—data can be entered into a GIS system without GPS data using *dynamic segmentation*. If GPS has been used, the locations of features can be directly input to a GIS system.

Site Selection

The road hazard inventory is designed to assess all roads under a given ownership or within a given watershed. The protocol provides information to help landowners identify roads of concern and prioritize repair activities. It does not provide all the information necessary to implement those repairs. Timely inspection and subsequent maintenance or repair activity on forest roads will benefit fish and fish habitat. Therefore, inventories should eventually be conducted on all road miles that potentially affect fish habitat.

Prioritizing site selection depends on the monitoring question being asked. However, in general, road inventories should first be conducted in areas where roads pose higher risk to anadromous fish and their habitats. This can be determined from:

- Landowner knowledge
- Topographic maps showing:
  - stream crossings of fish bearing streams,
  - midslope roads on steep slopes, and/or
  - steep, long road grades leading to channel crossing

Landowners are encouraged to use this protocol for road management purposes other than erosion hazard reduction. Possible uses include routine maintenance and surfacing decisions.

Road Hazard Field Methods

<u>Overall Methodology</u>

Begin at a road junction or other landmark. Take measurements described in the **Surface Drainage Section** below. As you travel along the road, measure the distance (DMI or other device starting at 0), until encountering a drainage feature and or stream crossing. This is referred to as road stationing. Record distance traveled, repeat surface drainage measurements and take **Culvert/Bridge and/or Stream Crossing Details** (described below), whichever are applicable. Record observations

of general road characteristics (described in next section) for the entire road.

General Road Characteristics

Each road should be identified by name or number, according to the system normally used by the landowner. General characteristics are normally collected only once for each road. The following observations are used to classify each road and can be documented on a form such as in Table D-2:

Road identification by name, numbering system or other means.

Road use by management activity.

> active roads: roads used for timber haul in the past year
>
> inactive roads: include all other roads used for management since 1972; and
>
> orphaned roads: overgrown roads or railroad grades not used since 1972.

Surfacing material is described as asphalt, clean rock (new quarry rock); old rock (more common); or dirt.

Road location is described as ridge, midslope, or valley as the location of most of the road.

Width of the entire road is estimated (from the outside edge to the base of the cutslope).

For ownerships where *georegion, geology or soils* are variable and have a great influence on erosion, these classifications should also be documented.

Record whether the road is *outsloped* or has a *ditch*.

Record the *location* of the road with respect to a landmark. This may be done with the GPS unit or on a map.

Surface Drainage

Between drainage features, information is collected on the erosion potential and sediment delivery potential of the roadway. The typical road conditions between each drainage feature are categorized to identify erosion problems. The following observations and measurements are made to identify symptoms of high erosion on road segments that best describe the condition of the entire segment:

Road Grade
Road grade (slope) is measured in percent, with an estimated average when the slope changes. Slope is recorded as positive if the direction is up from the measuring point or feature, and negative when the direction is down from the feature. A positive slope drains toward the feature, a negative slope drains away from the feature.

Road Surface Condition
Road surface condition is described as good, rutted, bermed, or eroded (gullied).

Ditch
Ditch is described by function as good (capable of holding runoff without serious erosion), cutting, diverted, or full.

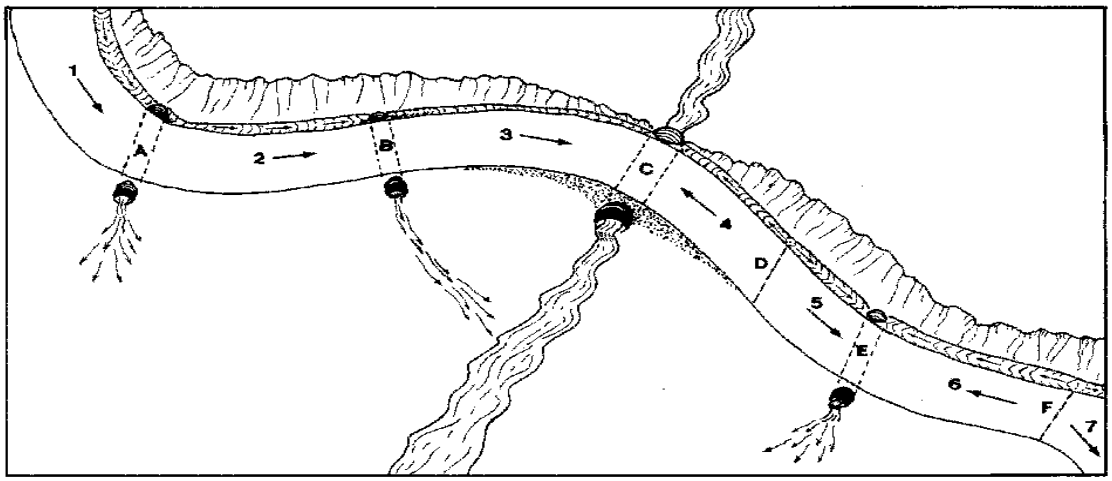
Cutslope
Cutslope is described as good (stable), ravel problems, or slides into the road.

Delivery
Delivery of sediment to streams from that length of road is described as “yes,” “possible,” “no,” or “bypassed” (water flows past the drainage feature and not off of the road).

Road length draining to drainage.
The length of road draining to each drainage feature can be calculated by use of several commonly available database or spreadsheet programs. For properly functioning outsloped roads there are no cross drainage features, only stream crossing features.

Drainage and Stream Crossings

Drainage data is collected at each drainage feature where collected drainage water is directed away from or under the roadway, and also at drainage divides. Drainage features include: stream crossing culverts, bridges, log puncheons, fords, cross-drain culverts, waterbars, dips, other relief, landings, and grade breaks. For each drainage feature, record the distance from road stationing and the type of feature so that drainage spacing can be determined. Landowners may also choose to locate features such as gates and water pump chances. A typical length of road with drainage patterns and features is shown in Figure D-1.



A. Cross-drain culvert, sediment filtered and not delivered to stream.
B. Cross-drain culvert with sediment delivery from segment 2 to stream.
C. Stream-crossing culvert, sediment from road segments 3 and 4 delivered to stream.
D. Drainage divide.
E. Cross-drain culvert, possible sediment delivery to stream.

Figure D-1. Typical road surface drainage and drainage features.

Culvert (and bridge) Detail

The following information is collected for all culverts (stream crossing and cross drain) and bridges:

Diameter/Span
Diameter/span of the culvert (diameter for round, rise and span for arch) or span length (for bridge) is measured in inches (for culverts) and feet (for bridges).

Condition
Condition of the culvert is described as good; mechanical damage, sediment blockage, rusted, bottom out, collapse, animal (beavers), wood blockage, natural bottom (gravel) [more than one description may be appropriate in this category].

Inlet Opening
Inlet opening is estimated as a percent or original (design) opening.

Stream Crossing Detail

Stream crossings are an extremely important part of the road system. Improperly functioning stream crossings can result in loss of the roadway through washouts and channel diversions. Stream crossings can also be barriers to fish movement. At each crossing structure, information should be collected by getting out of the vehicle and taking

measurements at the inlet end and near the outlet end of the structure. In addition to the culvert detail, the following information should be collected at each stream-crossing culvert (Figure D-2).

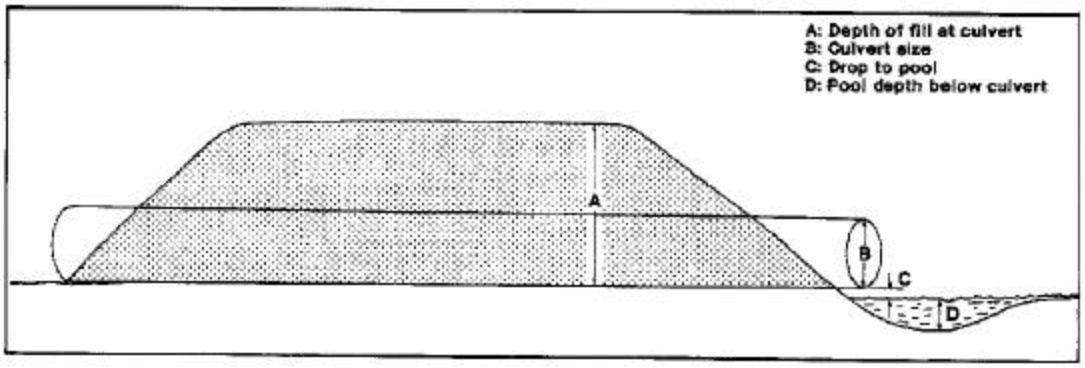


Figure D-2. Stream-crossing culvert with key dimensions.

Fish Presence
Fish presence (species, if known, from ODF classification maps or other sources). Categories are: "fish use"; "unknown fish use"; "no fish use"; or "anadromous fish use".

Diversion Potential
Diversion potential (for streamflow diverted onto the road surface and eroding the roadway or fill) is categorized as "high," "medium" or "low."

Culvert Slope
Culvert slope is measured for "fish use," or "unknown fish use" streams only.

Fill Height
Fill height is estimated from the channel bottom to the road surface at the downstream end.

Outlet Drop
Outlet drop is the distance from the bottom of the pipe to the elevation of the pool, in feet (measure countersunk outlets as negative drops). This can vary with stream discharge, so measurement should generally be taken during summer flow.

Resting Pool
Resting Pool below the pipe is categorized for fish use, or possible fish use streams only as "good" (at least two feet deep and six feet long); "fair" (at least one foot deep and four feet long); or "absent."

Sediment Filtering
Sediment filtering opportunities around the crossing are noted as "utilized, " "not utilized," or "not available."

Sidecast Detail

Sidecast-related landslides are reasonably expected along particularly steep sections of road (Table D-3) . Depending on georegion, geology, soil, and drainage, the natural slopes (below the road) for a steep section can be as gentle as 50% (in wet areas with weak sidecast and drainage problems). In areas with well-drained materials with uniform slopes and no or very limited signs of old slides, the appropriate slope may be 65 or 70%. Sections of road which have experienced past sidecast-related landslides should also be inventoried.

The beginning and ending points used to characterize sidecast stability will be different than those used to characterize drainage. Therefore, a separate database is used (Table D-3).

Begin characterizing sidecast stability at the point in the road where steepness indicates a slope failure hazard exists. This may be, and usually is, at some distance between drainage features. Record this station distance from the road junction or landmark using the same stationing methods as recording from drainage features. Also record the ending point in the same manner. The following features are then used to describe typical conditions over the steep sections:

Average Natural Slope Steepness
Average natural slope steepness under the sidecast (if present).

Indicators of Movement
Indicators of movement described as "none," "cracks," a "drop in the outside of the prism," or "signs of old sidecast slides."

Vegetation
Vegetation on the sidecast is described as "none," "cover (grass or brush)," "reproduction (plantation)," or "forest."

Fill Condition
Fill condition is described as "at least 15% steeper than the natural slope," "logs exposed," or "good."

Fill Depth
Fill depth at the outside edge of the road is estimated to the nearest foot as a vertical measurement.

Downslope Risk
Downslope risk to streams is described by a qualitative rating of the slope to the nearest stream channel: "low," "moderate," or "high" based on the presence and size of benches or landings between the site and the nearest channel.

Forms

Example data sheets suitable for relational databases are shown in Tables F-2 and F-3. One data sheet has been designed for surface drainage and stream crossings (Table D-2), and another data sheet for sidecast (Table D-3), since the beginning and ending points of areas of sidecast rarely coincide with drainage location. Codes for the data sheet are explained on the pages following the data sheets. The codes have been designed with one or two digits (underlined) to reduce the size of the code sheets.

Table D-2. Field data sheet for surface drainage and stream crossing details and examples of collected data. In this example attention is required on the last entry because the culvert is partially blocked.

Road name/number_____ Road Use: Active, Inactive, Orphaned Surfacing: Dirt, Rock, Clean rock

Road Location: Ridge, Midslope, Valley _____ Avg. Width _____ Ditch: yes/No Outslope: Yes/no

Inventoried by:_____ Date_____ Geology/Soils _____

			Surface Drainage					Culvert Detail					Stream Crossing Detail						
Station	Fea-ture	Attn.	Grade	Ditch	Cut-slope	Sur-face	Deli-very	Diam/ span	Mat	Cond-ition	% open	Outlet	Fish	Diver	Fill	Filter	Slope	Drop	Pool
0	JN	N	12	G	G	G	Y												
352	GB	N	-1	G	G	R	Y												
537	DP	N	6	G	G	G	Y												
681	BR	N	11	G	G	G	Y	56											
1686	DP	N	10	G	G	G	Y												
2016	SC	N	14	G	G	G	Y	24	S	G	100	6	N	H	9	N	6	6	N
2026	SC	Y	16	G	G	G	Y	12	S	M	20	G	N	H	7	N	8	5	N

Feature codes:

SC = stream crossing culvert CC = cross drain culvert BR = bridge JN = road junction GB = grade break PN = log puncheon

DR = any other ditch relief I = features requiring immediate attention WB = waterbar DP = dip LD = landing

PC = pump chance G = gate

Table D-3. Field data sheets for sidecast details. Example included.

Road name/Number:_____ Date_____

Inventoried by_____

Sidecast Detail								
Station Start (ft)	Station end (ft)	% slope Below	Movement Indicators.	Vegetation	Fill condition	Fill depth	Downslope Risk	Remarks
3413	3814	70	S	F	C	2	H	Stream has washed out road.

Codes:	Codes:	Codes	Codes:
Cracks	None	Steep 15	Low
Drop	Veg.	Logs	Moderate
Slide Activity	Reprod.	Good	High
None	Forested		

Road Data Analysis

Road data should be analyzed to determine which roads, drainage systems, and/or stream crossings:

- are not functioning properly,
- may be delivering sediment to fish-bearing streams,
- do not pass fish (calculated from the data collected, refer to ODF&W fish passage protocol),
- and/or pose a risk to fish bearing streams (road-related landslides).

A number of indicators for potential sediment problems may exist. Examples include:

- Average distance to first cross drain is over 500 feet and road grade is greater than 6%;
- Culverts that are more than 50% blocked;
- Logs in fills;
- Steep sidecast with high downslope risk;
- Fish bearing streams with culverts that have a >0 foot outlet drop, gradient over 1% and are not retaining sediment or do not have baffles.

Calculations of the road data can be done with a spreadsheet or database to address these road maintenance, sediment, and fish-passage related concerns.

Road-related results can be combined with turbidity and channel information to understand erosion and sediment processes in your watershed. It is important to recognize that a correlation between the three measurements may not reflect cause-and-effect relationships. In general such relationships can only be achieved with a properly designed and controlled study. However, over time the data will be useful for understanding environmental trends.

Appendix E

Sediment Deposition

Background

Sediment deposition occurs when the stream power is insufficient to continue transporting sediment particles and sediment settles or falls out of suspension. Where and when sediment deposition occurs depends on the

- size of the particle,
- channel morphology and
- stream flow characteristics.

Deposition takes place at both the smaller site scale (behind a big rock, in a deep pool) and the larger reach scale (lower channel gradient, meandering stream, reservoir).

Streambed material, referred to as channel substrate, is composed of a range of different sized particles. For example, some stream reaches have substrate composed mostly of bedrock while others have a mix of bedrock, cobble, sands, and gravels. In general, smaller particles are carried in suspension for the longest time.

Transportation and depositional reaches are ***not*** *ideal areas to monitor sediment deposition.* Past studies have found that changes in sediment deposition are difficult to detect in steep headwater streams and low-gradient rivers. Transitional reaches may be more responsive to changes in sediment and hydrologic regimes than headwater and valley-bottom streams. These intermediate size streams also provide important habitats for fish and aquatic invertebrates.

Describing the relative proportions of particles at a given site (particle size distribution) provides an index of the channel characteristics. If there are changes in the amount of sediment and the size of particles that are delivered to a stream reach, then the substrate characteristics may change. A change in channel morphology and hydraulics (for example placement of large woody debris enhancement projects) may result in a change in substrate even without a change in sediment delivery.

A stream system can be subdivided into *transportation* reaches, *transitional* reaches and *depositional* reaches. In general, transportation reaches consist of steeper headwater streams with large substrate (boulders and cobbles) that is stable during most flows. Fine sediments delivered to these reaches from the adjacent hillsides and streambanks are transported downstream during high flows. Conversely, depositional reaches consist of larger, lower-gradient, valley bottom stream channels that have depositional features (point-bars, floodplains, mid-channel point bars) that consist of fine sediments such as fine gravel, silts and clays.

Watershed management activities can affect watershed processes by altering sediment delivery to the stream network. Large inputs of fine sediment to the stream can degrade aquatic invertebrate and fish habitats and alter the structure and width of stream channels and adjacent riparian zones (MacDonald et al. 1991). Increased sediment input may elevate suspended sediment concentrations and turbidity. Fine sediments fill intergravel spaces used by aquatic insects and young fish. Pool

frequency and depth may be diminished and channel sinuosity and other channel characteristics can be appreciably changed.

This protocol can be used to develop some base-line data on substrate characteristics. However, there are limitations to what this protocol will reveal about a stream channel and potential impacts. For example, there can be significant aggradation (an increase in the elevation of the streambed due to sediment deposition) with no change in particle size distribution.

Terms

Particle size distribution
The relative proportions of a range of different-sized particles. For example a stream bed may be composed of 50% bedrock, 25% boulder, 10% cobble, 10% gravel and 5% sand at one sample point. This is the particle size distribution at that channel location. The particle size distribution can be described for a site, reach, or basin.

Substrate
Channel bed material (i.e., bedrock, sand, gravel) described in terms of its particle size distribution.

Habitat unit
Used to index fish habitat characteristics (see ODF&W fish habitat inventory methods). Typically describes characteristics of pools, riffles, and glides.

Equipment Needs

The following materials are needed to implement the field methods:

- Tape measure, 100 feet
- Wading rod or surveyor's rod
- 20"x20" screen with one inch grids
- Clear Plexiglas viewing tube
- Data forms
- Pencils
- Camera (optional)

Site Selection

Stream Reach
Watershed characteristics such as drainage area, landform, and stream gradient exert strong influences on the physical habitat of a stream. Thoughtful site selection and sample design acknowledge these relationships and may control some of the variability in the data and improve the sensitivity of the analysis.
Use the objectives and criteria described in Chapter 3 *Site Selection* to determine the location and number of stream reaches to be sampled. The number and location of sample sites will ultimately depend on the monitoring objectives. For example, if a particular management activity will be monitored, a transitional reach upstream and downstream of that activity might be monitored, both before and after the activity. If an instream restoration project is planned, monitoring the placement site before and after the placement for a few years will be necessary. If baseline or condition monitoring is the goal, then it may be necessary to randomly select multiple sections of the transitional reaches on a stream. If the focus is on spawning gravels or macroinvertebrates, then sample reaches should be located near these sites. Once sample reaches are designated, substrate measurements and observations are taken at eleven cross-sectional transects evenly spaced along the sample reach.

Selecting Sample Reaches
The pebble count and percent surface fine protocols presented here are most appropriate for *transitional* stream reaches (described in the background of this chapter). These transitional stream reaches often have moderate gradients of 2 to 6%, both erosion and

depositional features, and a mix of substrate sizes.

These methods are not appropriate for impoundments, wetlands, or large stream reaches that are too deep to safely wade. Each member of the sampling team must decide if the stream is safely wadeable by weighing factors such as depth, water velocity, and footing. A stream reach may be considered for sampling if more than 50% of the sample reach can be waded.

How many sites per stream?
The location and number of sites per stream depends on the objectives of the study, the type of impacts, and the resources available. It is important to sample enough sites to determine the inherent variability within and among different sites.

When are sites sampled?
Ideally, sampling will occur at or near low-flow conditions. Sampling should not occur during or soon after events such as a storm-related high-flows, because sampling may not be safe during this time and sampling results may be considerably different from samples collected during base flow conditions. During high flows finer sediment particles may be flushed from a coarse-bedded streambed and monitoring results may indicate an even coarser streambed surface than sampling results collected during lower flows (Adams and Beschta, 1980).

Field Methods

Overall Methodology
The pebble count method can be used to provide a representative estimate of the streambed particle size distribution (USEPA REMAP). Select the sample reach using the criteria discussed above. Establish eleven cross-sectional transects, evenly-spaced and perpendicular to the active stream channel. Substrate size will be measured at five sites on each of 11 transects using a modified pebble count method (Wollman 1954, Bain et al. 1985, and Plafkin et al. 1989).

Dimensions of the Sample Reach
Once the transitional sample reach has been established, the upstream and downstream boundaries of the sample reach must be determined. The length of a sample reach is 40 times the low-flow channel width or a minimum of 150 meters. Measure and record the wetted channel width of the stream at three locations that typify the stream channel. Do not include damp stream margins or isolated pools in these measurements. Average these three measurements to determine the average wetted width of the channel and multiply this average by 40 to determine the length of the channel to include within the sample reach.

Transects
Divide the sample reach into 10 equal segments. Beginning with the downstream end of the sample reach, establish the first of 11 transects (surveyor's flagging tied to vegetation may be useful for this purpose provided it is removed once the sampling is concluded). Each transect is oriented across the channel and is perpendicular to the longitudinal axis of the stream. Continue upstream following the wetted edge of the stream and establish another transect a distance equal to 4 wetted widths. Continue until the 11th and final transect is placed at the upstream end of the sample reach. Stay out of the stream as much as possible during this time to minimize disturbance of the substrate. This is especially important if water quality or macroinvertebrate samples will be collected in the same reach.

Pebble Count Method

1. At the downstream cross-section station, lay the surveyor's rod across the channel perpendicular to the flow, with the "zero" end at the left bank (determined when facing downstream). If the channel is too

wide for the rod, stretch the tape in the same manner.

2. Document the width of the channel from wetted bank to wetted bank. Divide the width of the channel by 4. This corresponds with the distance increments between sampling points. The sediment sampling points will be at the left and right banks and at 1/4th, 1/2, and 3/4 positions along the rod or tape. The result is a total of 5 measurements at each transect. For example, if the stream is 30 feet wide, a sediment measurement should be taken every 7.5 feet and at each bank. The bank measurements are taken just at the water's edge.

3. Place the measuring stick upright at the first sampling point at the end of the tape, being careful to stand downstream of the sample point. Read and record the depth. Pick up the substrate particle directly at the base of the stick (unless it is too big or too small), and visually estimate its diameter according to the following coded scale provided below. To minimize bias in this method, it is important to concentrate on correct placement of the measuring stick along the rod or tape. Place the center of the stick perpendicular and adjacent to the measurement increment on the outstretched tape or rod. Select the particle right at the bottom of the stick (not, for example, a more noticeable large particle that is just to the side of the stick). *There is a tendency to allow the rod to slip down the face of a rounded rock to a flat surface. If the rod lands on the side of a rounded rock, that is the particle to measure.* Record the particle as one of the following codes:

   BS = Bedrock (Smooth): >4000 mm; Smooth surface rock or hardpan (bigger than a car)

   BR = Bedrock Rough: >4000 mm; (bigger than a car)

   BL = Boulders: >250 to 4000 mm; (basketball to car size)

   CB = Cobbles: 64 to 250 mm; (tennis ball to basketball)

   GC = Gravel (Coarse): 16 to 64 mm; (marble to tennis ball)

   GF = Gravel (Fine): 2 to 16 mm; (ladybug to marble)

   SA = Sand: 0.06 to 2 mm; (<ladybug size, but visible as particles; gritty between fingers)

   FN = Fines: <0.06 mm; Silt Clay Muck; not gritty between fingers

   WD = Wood: Regardless of size

   OT = Other: Metal, tires, car bodies, etc. regardless of size (put in comments if "others").

4. Move successively to each of the remaining four positions along the rod or tape, repeating steps 3 and 4. Repeat the entire procedure at each new transect.

Table E-1. Field form.

Transect Number (5 per each transect)	Habitat Unit Type (pool, riffle, or glide)	Channel Width (ft)	Channel Max Depth (ft)	Channel Gradient (%)	Particle (Code)	Depth at sample site (ft)
1.1	Pool	25	1.3	2	BS	0.1
1.2	Pool	25	1.3	2	CB	0.5
1.3	Pool	25	1.3	2	CB	1.0
1.4	Pool	25	1.3	2	SA	0.8
1.5	Pool	25	1.3	2	BS	0.1
2.1	Glide	27	1.0	2.5	CB	0.1
2.2	Glide	27	1.0	2.5	BS	0.1
2.3	Glide	27	1.0	2.5	BS	1.0
2.4	Glide	27	1.0	2.5	SA	0.5
2.5	Glide	27	1.0	2.5	BS	0.1

If a mid-channel bar splits the wetted channel, the five sampling points shall be established as described above regardless of the bar. Consequently, sediment particles selected in some transects may be "high and dry." For dry channels, make cross section measurements across the unvegetated portion of the channel and within the scoured banks. Table E-1 is an example of a field data format to use when recording data.

Ancillary Data

When analyzing data it will be useful to have the following information:

- Channel width, maximum depth, and gradient at each transect
- Habitat unit type at each transect
- Tributary junctions within the sample reach
- Culverts that drain to the sample reach
- Stream-side management activities (roads, harvesting, pasture, trail, etc.)

Data Analysis

A number of techniques are available to characterize data findings. For in-depth relational analyses please contact one of the mentors listed in this manual.

Create a table to sum the number of particles within each size class for the entire reach (Table E-2). Calculate the percent of pebbles within each size class and the cumulative percent within each size class. Graph each of these statistics on the same chart (Figure E-1) using a log/normal scale (x-axis is log and y-axis is normal).

<u>Cumulative Frequency Distribution</u>
By graphing cumulative percent, monitors can determine what the dominant substrate is of a stream cross-section or reach. Values often reported are the D_{30}, D_{50}, and D_{75}. The D_{50} represents the median particle diameter. For example in Figure E-1 the D_{50} is 6 millimeters (mm). This means that 50% of the

particles are less than 6 mm and 50% are greater than 6 mm.

The D_{30} is important in terms of effects on fish and macroinvertebrates. If 30% of the substrate is less than 2 mm in diameter, there may be adverse impacts to fish and macroinvertebrates. In Figure E-1 the D_{30} is 1 mm. The D_{75} represents the dominant substrate. In Figure E-1 the D_{75} is approximately 230 mm.

Table E-2. Example of spreadsheet organization for Figure E-1 calculations. Data represent 11 transects of pebble count data.

Particle Diameter (mm)	Total number for 11 x-sec.	Percent of Total (%)	Cumulative Percent (%)
<.06	5	9	9
<2	14	25	35
<16	17	31	65
<64	4	7	73
<250	3	5	78
<4000	12	22	100
	Total = 55	Total=100	

Percent of Total

The distribution of particle sizes throughout a transect or reach can be seen by plotting percent of total. This is valuable for determining if there is a "bi-modal" distribution (two peaks in the curve). This is illustrated in Figure E-1. In this example, although the D_{70} was 230 mm, most of the stream substrate is characterized between 2 and 12 mm.

Physical and Management Relationships

Once the sediment characteristics of a reach or watershed have been analyzed, relationships between management practices, restoration activities, channel gradient, channel width, channel depth, habitat unit type, and potential source areas can be examined. Assessments of whether the problem reaches have sediment sources that can be mitigated may also be done.

Remember that without proper study design, cause-and-effect relationships cannot be established. However, from a monitoring perspective, valuable information to help guide management decisions can be created.

For example, if instream fish restoration projects are planned, this information will provide good pre-treatment data. The substrate characteristics prior to restoration can be compared to substrate characteristics after the restoration activity. The data can also help determine where to place the instream structure.

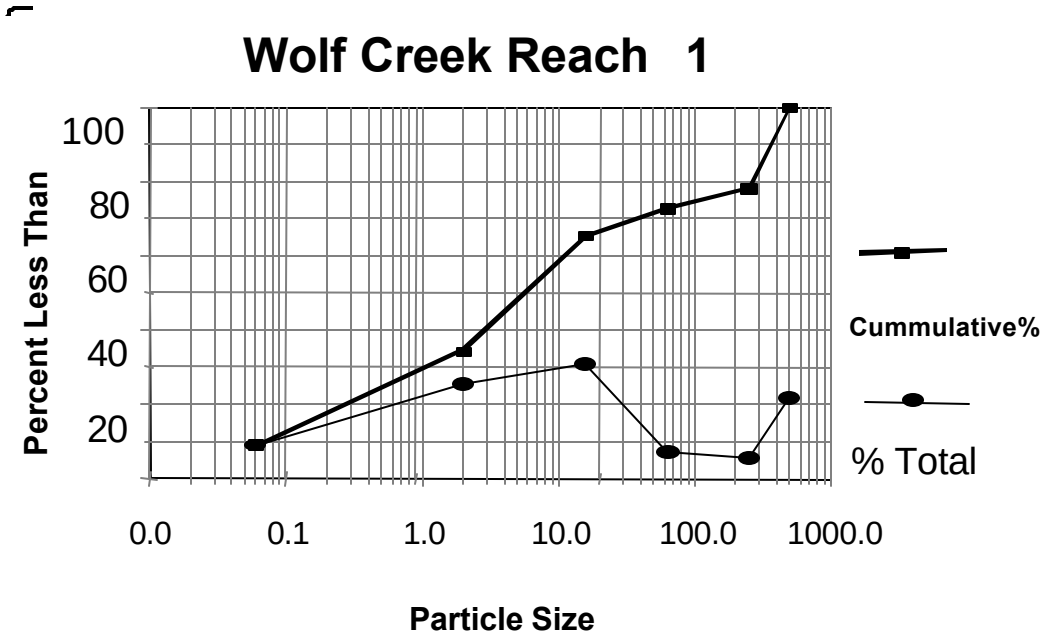


Figure E-1. Example of graphical display of data for a reach of stream.

Percent-Fines Grid Method

Once some of the preliminary analyses have been completed, problem areas or specific questions may be identified. For example, a stream cross-section may have identified where the substrate is predominantly fine material. Monitors can revisit these sites and implement the grid method described below to get more detailed information.

Percent Surface Fines: Grid Method

The grid method can be used to address very specific objectives or to focus on a particular site question where more detailed data on percent fines is needed. If sampling for macroinvertebrates, use the grid method at the site where the macroinvertebrates samples are collected. It is more time-consuming than the pebble-count methodology but reduces the potential for bias. The grid method can be used

to document all particle sizes as in the above procedure. However, in the following description we focus strictly on the percent of substrate made up of sands and fines.

Site selection

The grid method can be implemented at the same sample points or a subset of sample points used in the pebble count. If a subset of sites it used, be sure to spread them evenly throughout the 11 transects and alternate among the 5 site locations per transect.

Sampling Technique

1. Place the 20”x20” grid flat on the streambed surface and count the number of grid intersections that are directly above sand or fine sediment particles (for these purposes fine sediments are <2mm in size, or smaller than a ladybug or pea). Use the Plexiglas viewing tube or other device such as a scuba mask to improve viewing of the substrate by reducing distortion and glare from the surface of turbulent water.
2. Record the total number of grid intersections that are above sand and fine sediment particles in the appropriate location. A total of 400 intersections exist on the grid. Note if the site appeared to be a depositional area such as a pool or an erosional area such as a riffle.
3. Divide the total number of intersections overlying sand and fine sediment by the total number of intersections surveyed. This is an estimate of the percent of the streambed substrate that is occupied or covered by fine sediments.

Appendix F

Macroinvertebrate Taxa List

TAXON	HBI	SDTOL	SDINTOL	TOL	SENS
ANNELIDA					
HIRUDINEA	8			Yes	
OLIGOCHAETA	6	Yes		Yes	
Lumbriculidae	6	Yes			
Tubificidae	6	Yes		Yes	
POLYCHAETA	6				
ARTHROPODA					
ARACHNOIDEA "Hydracarina"	5				
CRUSTACEA					
Cladocera	8				
Copepoda	8				
Amphipoda	4				
Anisogammarus	4				
Eogammarus	6				
Gammarus	6			Yes	
Hyalella azteca	8				
Decapoda	6				
Pacifastacus	6				
Isopoda	8			Yes	
Ostracoda/Podocopa	8				
INSECTA					
Coleoptera					
Amphizoidae	1				
Chrysomelidae	6				
Dryopidae	5				
Dytiscidae	5			Yes	
Elmidae	4				
Ampumixis	4				
Atractelmis	4				
Cleptelmis	4			Yes	
Dubiraphia	6			Yes	
Heterlimnius	4				
Lara	4				
Microcylloepus	4			Yes	
Narpus	4				
Optioservus	4			Yes	
Ordobrevia	4				
Rhizelmis	2				
Stenelmis	5			Yes	
Zaitzevia	4			Yes	
Gyrinidae	5				
Haliplidae	5			Yes	
Brychius	5			Yes	
Haliplus	5			Yes	
Peltodytes	8			Yes	
Hydraenidae	4				
Hydraena	4				

TAXON	HBI	SDTOL	SDINTOL	TOL	SENS
Ochthebius	4				
INSECTA					
Coleoptera (cont.)					
Hydrophilidae	5				
Ametor	5				
Berosus	5			Yes	
Enochrus	5				
Helophorus	5				
Laccobius	5				
Paracymus	5				
Tropisternus	5				
Noteridae	4				
Pronoteus	4				
Psephenidae	4				
Acneus	4				
Dicranopselaphus	4			Yes	
Eubrianax	4			Yes	
Psephenus	4			Yes	
Ptilodactylidae	5				
Scirtidae	4				
Staphylinidae	5				
Diptera					
Brachycera					
Athericidae	4			Yes	
Atherix	4			Yes	
Dolichopodidae	4			Yes	
Empididae	6				
Chelifera	6				
Clinocera	6				
Hemerodromia	6				
Oreogeton	6				Yes
Wiedemannia	6				
Ephydridae	6				
Muscidae	6			Yes	
Limnophora	6			Yes	
Pelecorhynchidae	3				Yes
Glutops	3				Yes
Sciomyzidae	6				
Stratiomyidae	8			Yes	
Caloparyphus	8			Yes	
Euparyphus	8				
Syrphidae	6			Yes	
Tabanidae	8			Yes	
Mercomyia	6			Yes	
Silvius	6			Yes	
Tabanus	6			Yes	
Nematocera					
Blephariceridae	0				Yes
Agathon	0				Yes
Bibiocephala	0				Yes
Blepharicera	0				Yes
Dioptopsis	0				Yes
Philorus	0				Yes
Ceratopogonidae	6				
Ceratopogoninae	6				
INSECTA					
Diptera					
Forcipomyiinae	6				
Chironomidae	6				
Chironominae	6				

TAXON	HBI	SDTOL	SDINTOL	TOL	SENS
	6				
Nematocera					
Chironomidae					
Chironominae (cont.)					
Pseudochironomini	5				
Tanytarsini	6				
Diamesinae	2				
Orthocladiinae	5				
Podonominae	6				
Prodiamesinae	6				
Tanypodinae	7				
Pentaneurini	6				
Culicidae	8			Yes	
Deuterophlebiidae	0				Yes
Deuterophlebia	0				Yes
Dixidae	2				
Dixa	2				
Dixella	2				
Meringodixa	2				
Psychodidae	10				
Maruina	2				
Pericoma	4				
Ptychopteridae	7				
Bittacomorpha	7				
Ptychoptera	7				
Simuliidae	6				
Prosimulium	3				
Simulium	6				
Twinnia	6				
Tanyderidae	1				Yes
Thaumaleidae	8				
Thaumalea	8				
Tipulidae	3	Yes			
Molophilus	2	Yes			
Antocha	3	Yes			
Cryptolabis	3				
Dicranota	3	Yes			
Hesperoconopa	1				Yes
Hexatoma	2	Yes			
Limnophila	2	Yes			
Limonia	6	Yes		Yes	
Ormosia	3	Yes			
Pedicia	6	Yes			
Rhabdomastix	3				Yes
Tipula	4	Yes			
Ephemeroptera					
Ameletidae					
Ameletus	0				
Baetidae	4				
Acentrella	4				
Baetis	5				
Baetis bicaudatus	4				Yes
Baetis tricaudatus	6				
Callibaetis	9			Yes	
INSECTA					
Ephemeroptera					
Baetidae (cont.)					
Centroptilum	2			Yes	
Dactylobaetis	6				
Diphetor hageni	5				

TAXON	HBI	SDTOL	SDINTOL	TOL	SENS
Fallceon	5				
Procloeon	5				
Pseudocloeon	4				
Caenidae	7			Yes	
Caenis	7			Yes	
Ephemerellidae	1				
Attenella	2				
Caudatella	1				Yes
Drunella	0				
Drunella coloradensis	0				
Drunella coloradensis/flavilinea	0				
Drunella doddsi	0				Yes
Drunella flavilinea	0				
Drunella grandis	0				
Drunella pelosa	0				Yes
Drunella spinifera	0				Yes
Ephemerella	1				
Ephemerella aurivilli	1				
Ephemerella inermis/infrequens	1				
Ephemerella maculata	1				
Serratella	2				
Serratella teresa	2				
Serratella tibialis	2				
Timpanoga	7				
Ephemeridae	4				
Ephemera	4				
Ephemera simulans	7				
Hexagenia	6				
Hexagenia limbata	7				
Heptageniidae	4				
Cinygma	2				Yes
Cinygmula	4				
Epeorus	0				
Epeorus albertae	1				
Epeorus deceptivus	0				Yes
Epeorus grandis	0				Yes
Epeorus Ironopsis	0				
Epeorus longimanus	1				
Heptagenia	4				
Heptagenia/Nixe/Leucrocuta	4				
Ironodes	3				
Leucrocuta	1				
Nixe	4				
Rhithrogena	0				
Stenonema	5			Yes	
Isonychiidae	6				
Isonychia	2				
Leptophlebiidae	2				
Choroterpes	2			Yes	
Leptophlebia	4			Yes	
Paraleptophlebia	4				
Paraleptophlebia bicornuta	4				
Paraleptophlebia debilis	1				
Paraleptophlebia gregalis	1				
INSECTA					
Ephemeroptera (cont.)					
Paraleptophlebia temperalis	1				
Polymitarcydae	7				
Ephoron	7				
Siphlonuridae	7				
Parameletus	2				
Siphlonurus	7			Yes	

TAXON	HBI	SDTOL	SDINTOL	TOL	SENS
Tricorythidae	4			Yes	
Tricorythodes	4	Yes		Yes	
Hemiptera					
Belostomatidae	8				
Belostoma	8				
Lethocerus	8				
Corixidae	8				
Callicorixa	8				
Cenocorixa	8				
Corisella	8				
Hesperocorixa	8				
Sigara	8				
Trichocorixa	8				
Graptocorixa	8				
Gerridae	8				
Gerris	8				
Naucoridae	8				
Ambrysus	8				
Notonectidae	8				
Notonecta	8				
Saldidae	8				
Salda	8				
Saldula	8				
Veliidae	8				
Microvelia	8				
Rhagovelia	8				
Lepidoptera					
Pyralidae	5			Yes	
Petrophila	5			Yes	
Megaloptera					
Corydalidae	0		Yes		
Corydalus	0		Yes		
Dysmicohermes	0		Yes		
Neohermes	4		Yes		
Orohermes	0		Yes		
Sialidae	4			Yes	
Sialis	4			Yes	
Odonata					
Anisoptera					
Aeshnidae	5			Yes	
Aeshna	5			Yes	
Anax	8			Yes	
Cordulegastridae	3				
Cordulegaster	3				
Gomphidae	4				
Erpetogomphus	4				
Gomphus	4				
Octogomphus	4	Yes			
Ophiogomphus	4	Yes		Yes	
Libellulidae	9			Yes	
Sympetrum	10			Yes	
INSECTA					
Odonata (cont.)					
Zygoptera					
Coenagrionidae	9			Yes	
Amphiagrion	9			Yes	
Argia	7			Yes	
Coenagrion	8			Yes	
Coenagrion/Enallagma	8			Yes	
Enallagma/Ischnura	9			Yes	

TAXON	HBI	SDTOL	SDINTOL	TOL	SENS
Zoniagrion	9			Yes	
Lestidae	9			Yes	
Archilestes	9			Yes	
Plecoptera					
Capniidae	1				Yes
Capnia	1				Yes
Eucapnopsis	1				Yes
Paracapnia	1				Yes
Leuctridae	0				Yes
Despaxia	0				Yes
Leuctra	0				Yes
Moselia	0				Yes
Paraleuctra	0				Yes
Perlomyia	0				Yes
Megaleuctra	0				Yes
Nemouridae	2				
Amphinemura	2				
Malenka	2				
Nemoura	1				Yes
Ostrocerca	2				
Podmosta	2				
Prostoia	2				
Soyedina	2				
Visoka	0				Yes
Zapada	2				
Zapada cinctipes	2				
Zapada columbiana	2				Yes
Zapada frigida	2				Yes
Zapada Oregonensis Gr.	2				
Taeniopterygidae	2				Yes
Doddsia	2				Yes
Taenionema	2				Yes
Taeniopteryx	2				Yes
Chloroperlidae	1				
Alloperla	1				
Haploperla	0				Yes
Neaviperla	1				
Plumiperla	1				
Suwallia	1				
Sweltsa	1				
Kathroperla	0				Yes
Paraperla	0				Yes
Peltoperlidae	1				
Sierraperla	1				Yes
Soliperla	1				Yes
Yoraperla	1				Yes
Yoraperla brevis	1				Yes
Yoraperla mariana	1				Yes
INSECTA					
Plecoptera (cont.)					
Perlidae	1				
Calineuria	2				
Doroneuria	1				Yes
Hesperoperla	2				
Claassenia	3				
Perlodidae	2				
Cascadoperla	2				Yes
Isoperla	2				
Isoperla ebria	2				
Isoperla fulva	2				
Isoperla fusca	2				
Isoperla marmorata	2				
Isoperla mormona	2				

TAXON	HBI	SDTOL	SDINTOL	TOL	SENS
Isoperla petersoni	2				
Cultus	2				
Diura	2				
Frisonia	2				
Isogenoides	2				
Kogotus	2				
Megarcys	2				Yes
Oroperla	2				
Osobenus	2				
Perlinodes	2				
Pictetiella	2				
Rickera	2				
Setvena	2				Yes
Skwala	2				
Pteronarcyidae	0				
Pteronarcella	0				
Pteronarcys	0				
Pteronarcys californica	1				
Pteronarcys dorsata	0				
Pteronarcys princeps	0				Yes
Trichoptera					
Hydropsychidae	4				
Arctopsyche	1		Yes		
Parapsyche	1				
Parapsyche almota	2				
Parapsyche elsis	1		Yes		Yes
Cheumatopsyche	5			Yes	
Hydropsyche	4			Yes	
Philopotamidae	3				
Chimarra	4				
Dolophilodes	2		Yes		Yes
Wormaldia	3		Yes		
Polycentropodidae	6				
Neureclipsis	7				
Nyctiophylax	5				
Polycentropus	6				
Psychomiidae	2				
Psychomyia	2		Yes		
Tinodes	2				Yes
Apataniidae	1				Yes
Pedomoecus	0				Yes
Apatania	1				Yes
INSECTA					
Trichoptera (cont.)					
Brachycentridae	1				
Amiocentrus	3				
Brachycentrus	1				
Micrasema	1				
Oligoplectrum	2				
Calamoceratidae	1				
Heteroplectron	1				
Goeridae	1				
Goeracea	0				Yes
Helicopsychidae	3			Yes	
Helicopsyche	3			Yes	
Lepidostomatidae	1				
Lepidostoma	1				
Leptoceridae	4				
Ceraclea	3				

TAXON	HBI	SDTOL	SDINTOL	TOL	SENS
Mystacides	4				
Nectopsyche	3			Yes	
Oecetis	8			Yes	
Triaenodes	6			Yes	
Limnephilidae	4				
Allocosmoecus	0				Yes
Cryptochia	0				Yes
Dicosmoecus	1				
Dicosmoecus atripes	1				Yes
Dicosmoecus gilvipes	2				
Ecclisocosmoecus	0				Yes
Ecclisomyia	2				Yes
Onocosmoecus	1				
Asynarchus	4				
Chyranda	1				Yes
Clostoeca	4				
Desmona	0			Yes	
Grammotaulius	4				
Hesperophylax	3			Yes	
Homophylax	0				Yes
Hydatophylax	1				
Lenarchus	3				
Limnephilus	3			Yes	
Philocasca	0				Yes
Psychoglypha	0				
Pseudostenophylax	1				
Odontoceridae	2				
Nerophilus	2				
Phryganeidae	4				
Ptilostomis	3				
Sericostomatidae	3				
Gumaga	3				
Uenoidae	0				
Neophylax	3				
Neophylax occidentalis	1				
Neophylax rickeri	2				
Neophylax splendens	2				
Oligophlebodes	0				Yes
Farula	0				Yes
Neothremma	0				Yes
Sericostriata	0				
INSECTA					
Trichoptera (cont.)					
Glossosomatidae	0				
Agapetus	0				
Anagapetus	0		Yes		Yes
Glossosoma	1		Yes		
Protoptila	1				
Hydroptilidae	4				
Agraylea	8				
Alisotrichia	4				
Hydroptila	6			Yes	
Leucotrichia	6			Yes	
Neotrichia	4			Yes	
Ochrotrichia	4			Yes	
Oxyethira	3			Yes	
Stactobiella	4				
Palaegapetus	4				
Rhyacophilidae	1				

TAXON	HBI	SDTOL	SDINTOL	TOL	SENS
Himalopsyche	0				Yes
Rhyacophila	1				
Rhyacophila Alberta Gr.	0				Yes
Rhyacophila Angelita Gr.	0				
Rhyacophila arnaudi	0				
Rhyacophila Betteni Gr.	1				
Rhyacophila blarina	1				
Rhyacophila Brunnea Gr.	1				
Rhyacophila Coloradensis Gr.	2				
Rhyacophila grandis	1				Yes
Rhyacophila Grandis Gr.	1				Yes
Rhyacophila Hyalinata Gr.	1				
Rhyacophila Lieftincki Gr.	3				
Rhyacophila malkini	2				
Rhyacophila narvae	1				
Rhyacophila Nevadensis Gr.	2				
Rhyacophila oreta	0				Yes
Rhyacophila Oreta Gr.	0				Yes
Rhyacophila pellisa	1				
Rhyacophila Rotunda Gr.	0				Yes
Rhyacophila Sibirica Gr.	0				
Rhyacophila Vagrita Gr.	0				Yes
Rhyacophila valuma	1				
Rhyacophila Verrula Gr.	0				Yes
Rhyacophila Vofixa Gr.	0				Yes
MOLLUSCA					
Gastropoda					
Ancylidae	6			Yes	
Ferrissia	6	Yes		Yes	
Hydrobiidae	5				
Fluminicola	5			Yes	
Lymnaeidae	6	Yes		Yes	
Fossaria	6	Yes		Yes	
Lymnaea	6	Yes		Yes	
Stagnicola	6	Yes		Yes	
Physidae	8			Yes	
Physa	8			Yes	
Physella	8	Yes		Yes	
MOLLUSCA					
Gastropoda (cont.)					
Planorbidae	6	Yes		Yes	
Gyraulus	8	Yes		Yes	
Planorbella	7	Yes		Yes	
Promenetus	6	Yes		Yes	
Vorticifex	6	Yes		Yes	
Pleuroceridae	7				
Juga	7	Yes		Yes	
Valvatidae	8				
Valvata	8				
Pelecypoda					
Corbiculidae	9				
Corbicula	9	Yes		Yes	
Sphaeriidae	8				
Pisidium	8				
Unionidae	4				Yes
Gonidea	4				Yes
Margaritifera	4				Yes
COELENTERATA					
Hydridae	5			Yes	
Hydra	5			Yes	

TAXON		HBI	SDTOL	SDINTOL	TOL	SENS
NEMATODA		5				
NEMATOMORPHA		6				
PLATYHELMINTHES TURBELLARIA		4				
PORIFERA	Spongillidae	6				

BI = Biotic Index value. Used for calculating the HBI (Hilsenhof Biotic Index)

SD TOL = Sediment Tolerant Taxa

SDINTOL = Sediment Intolerant Taxa

TOL = Tolerant Taxa

SENS = Sensitive Taxa