QAPP for Status and Trends Monitoring of Small Streams in Puget Lowlands Ecoregion for Monitoring Conducted using Pooled RSMP Funds contributed by Western Washington Municipal Stormwater Permittees

Appendices A - O

Prepared by:
Washington State Department of Ecology

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Appendix A. Regional stormwater monitoring strategy

Background

The Puget Sound Stormwater Work Group (SWG) was assembled in 2008 at the request of Ecology and the Puget Sound Partnership to develop recommendations for a monitoring and assessment strategy to improve our understanding of the effects of stormwater in the Puget Sound region. In 2010, the SWG finalized the overall strategy for monitoring in the document *2010 Stormwater Monitoring and Assessment Strategy for the Puget Sound Region (SWAMPPS)* (SWG, 2010). These recommendations were submitted to the Washington State Department of Ecology (Ecology) and Puget Sound Partnership for consideration in the development of an integrated stormwater monitoring program focused on the Puget Sound region. The 2010 Strategy included "55 Key Recommendations" for a new stormwater assessment and monitoring program.

The 2010 Strategy describes four components of a robust program: status and trends monitoring of receiving waters impacted by stormwater runoff; effectiveness studies to evaluate best management practices and programmatic approaches to manage stormwater; source identification and diagnostic monitoring to improve pollution reduction efforts; and research to increase knowledge of stormwater effects on biota and treatment approaches to reduce effects.

The SWG followed the 2010 Strategy with 33 recommendations for municipal permit monitoring (http://www.ecy.wa.gov/programs/wq/stormwater/municipal/rsmp.html). These recommendations outlined a plan for implementing a core subset of the 2010 Strategy through municipal stormwater permits issued to local governments in Puget Sound.

Status and trends monitoring design

For receiving water monitoring, the SWG proposed a sampling strategy compatible with current statewide status and trends monitoring, based on the existing statewide monitoring program (Cusimano, et al., 2006). Stream health is assessed at each sampling site within the study areas (populations) by measuring biological condition using macroinvertebrate and periphyton indicators in a rotating sampling design. Biological condition is determined using multi-metric models (e.g., Karr, 1991) or multivariate models (e.g., Wright, 1995). Both types of empirical models classify stream health based on comparison to reference conditions. Stressors affecting biological conditions are estimated using concurrent habitat and chemical measurements.

The status of each stream population is determined by estimating the percentage of stream length with impaired biological conditions and their associated stressors. The trends within stream populations are estimated by comparing measurements between five-year periods. Estimates for status and trends monitoring are made using statistical evaluations of the data using protocols developed for the national Environmental Monitoring and Assessment Program (EMAP) (EPA, 2005). For the purposes of assessing stormwater impacts, the study design characteristics will take into account the desire for Puget Sound-scale estimates at a 90% confidence level and potential for stratification of samples into other categories (e.g., land uses).

The 90% confidence level is determined by the variance of the indicator variable and the sample size within populations, as per www.epa.gov/nheerl/arm/surdesignfaqs.htm. Several evaluations have been conducted by EPA and Ecology scientists to determine the physical, chemical, and biological variables and their *signal/noise* properties (e.g., Kaufmann, et al., 1999). These evaluations determine the magnitude of change that occurs in a single variable when some sort of human-induced influence is present. Variables can change over the short-term or the long-term depending on how much artificial change is necessary to measure a response.

Assumptions underlying the design

This monitoring program design is based on several assumptions. The SWG recommended selecting sampling sites based on stream order (Strahler, 1957). Only 1st, 2nd, and 3rd order streams within the study areas were considered because these smaller streams are assumed to be more impacted by stormwater than larger order streams. Zero order streams are assumed to be intermittent and therefore not appropriate for comparison of stream benthos populations.

The SWG also assumes that two assessment regions (Urban Growth Area (UGA) and non-UGA) are different. This assumption is based on the differences in stormwater management efforts required by the permits are targeted inside UGA boundaries, and the differences in overall land use. Streams in urban and urbanizing areas are assumed to be more (or differently) influenced than streams outside urban and urbanizing areas.

This monitoring design assumes that the sites will be useable over the long term. The site layout is designed for a long-term monitoring program rather than for a targeted study. While the data collection can proceed using the same protocols for either type of study, a targeted study may choose to use a habitat- specific approach, rather than reach-wide approach, thereby reducing the noise and amplifying the signal created by the conditions under investigation. This study design assumes that general trends in watershed and ecosystem health can be described with the parameters outlined here.

Regional stormwater monitoring objectives

This monitoring framework is designed to answer the following core broad-scale monitoring questions:

- What are the status and trends of instream water quality, biological, and habitat conditions for 1st, 2nd, and 3rd order (small) streams in Puget Lowlands?
- What are the status and trends of the water quality, biological, and habitat conditions for 1st, 2nd, and 3rd order (small) streams in Puget Lowlands, both inside and outside of City/UGAs?

In addition to core questions, numerous others queries can be made from individual indicators and corresponding metrics (e.g., biological metal tolerance index inside and outside UGAs). Also, site-specific evaluations of data can be useful for answering questions at local scale and will improve stormwater managers' understanding of stream condition and stressors.

Coordination

A programmatic objective of the 2010 Strategy is to efficiently allocate limited resources for monitoring activities. To the maximum extent possible, data collection efforts within the Puget Sound region should be identified and leveraged for purposes of reporting and interpreting results. For example:

- Ecology's status and trends monitoring effort for Watershed Health and Salmon Recovery collected base-level monitoring in the Puget Sound SRR in 2009 and is expected to return in 2013. Assessment data collected during these periods can be incorporated into data analysis and reporting efforts for this stormwater QAPP.
- In 2012 and 2013, the U.S. Geological Survey analyzed the existing streamflow gaging network in Puget Sound and identified opportunities to use data from current gages to inform other Puget Lowland monitoring sites, (USGS, 2012 and 2013).
- Many local governments have active, robust monitoring programs. Their capacity and findings should be leveraged as much as possible in implementing the regional monitoring program and interpreting the results.

Scale of regional monitoring

Monitoring for this QAPP is focused at three landscape scales:

- Wadeable streams in the Puget Lowlands.
- Wadeable streams within UGA boundaries in the Puget Lowlands.
- Wadeable streams outside UGAs (non-UGA) in the Puget Lowlands.

These areas are the focus of important stormwater management, resource conservation and protection efforts. Information generated for each of these regions can be useful for Ecology, local governments, and agencies managing aquatic resources that are impacted by stormwater.

Since management for water quality improvements usually occurs at a local scale, the framework design provides for answering habitat and water quality questions at smaller, watershed scales (e.g., sub-watershed). This would require local partners to contribute to funding and sampling at this scale. At smaller scales, estimates of stream condition are more closely related to the causes

of aquatic resource impairments. In addition, the focus on sub-watersheds is understood and readily used by local governments, who are likely to participate in data collection efforts and become users of data generated by the monitoring program.

Indicators

The SWG (SWG, 2010) recommended monitoring specific biota, habitat, and chemical indicators related to stormwater runoff and stormwater impacts. The basic list of parameters comes from existing state status and trends study designs. Additional chemicals of interest (PAHs, flame retardants, pesticides, pharmaceuticals and hormones) are pollutants in small stream waters and sediments, many of have been detected in streams and are known to impact biota. The selected variables are useful in indicating change and identifying patterns over the broad spatial scale (e.g., ecoregion or inside/outside UGA boundaries) and at the stream-reach level. Some variables integrate the characteristics of streams over the broad spatial scale while others are sensitive to small changes in a stream.

Appendix B. Data quality assurance

B-1. Data quality indicators for benthic macroinvertebrates and periphyton

The integrity of the data collected by this project is upheld by maintaining a high quality and addressing the five objectives below. The quality of the sampling protocol is checked by analyzing the degree of sampling and visit precision, attempting to maintain less than 20% variation among reference stream data for taxa richness in benthos and periphyton samples. The aim is to collect samples that are representative of community and ecological conditions for each stream. Data are collected with common protocols used by other regional biological monitoring programs. This improves data comparability and usefulness among colleagues in biomonitoring.

Visit precision

Visit precision measures variability in the sampling method and is related to the variability of collecting a composite sample in a reach. Visit precision is estimated by collecting duplicate composite samples of the invertebrate and periphyton communities within the same reach during the same day at 10% of the reaches sampled annually. Visit precision is calculated using the RSD from two replicate composite samples and should be < 20% in reference streams when using the taxa richness metric.

Bias

Sampling bias is the difference between the population mean and the true value. Bias usually describes a systematic difference reproducible over time and is characteristic of both the measurement system, and the analyte(s) being measured (Kammin, 2010). Bias may be caused by the same field investigator conducting the same task at each site. It may also occur due to consistent misinterpretation of protocols by a group of field investigators.

Representativeness

Representativeness measures the degree to which a sample reflects the population from which it came - a data quality indicator (USGS, 1998). For ambient monitoring, sites should be representative of minimally- or least-disturbed conditions in the sampled stream. For targeted monitoring, the sites should be representative of the range of conditions in the sample area. The sampling protocols in the appendices are designed to produce consistent and repeatable results in each stream reach. Physical variability within reaches is accounted for through reach-wide sampling of the various depths, substrates, and flow conditions throughout the stream.

Completeness

Completeness is defined as the amount of valid data obtained from a data collection project compared to the planned amount and is usually expressed as a percentage (EPA, 1997). Our target for completeness of data is 100%. Sample loss is minimized with sturdy sample storage vessels and adequate labeling of each vessel. Sample vessel type and labeling information are described under the sections "Sampling Stream Macroinvertebrates", and "Sampling Periphyton" in the Appendix D. Sample contamination occurs when containers are improperly sealed or

stored. Loss of material or desiccation diminishes the integrity of the sample. If the validity of the information from the sample is in question, the sample will be flagged and excluded from analysis.

Completeness is determined by four criteria:

- Number of samples collected compared to the sampling plan.
- Number of samples shipped and received in good condition by the laboratory and the taxonomy contractor.
- Laboratory's ability to produce usable results for each sampling event.
- Sample results accepted by the project manager.

Comparability

Comparability describes the degree to which different methods, data sets, and decisions agree or can be represented as similar (EPA, 1997). Comparable data sets allow for sharing data with other organizations that adhere to the same protocols, such as field sampling and analytical methods.

In the spring of the year prior to monitoring the project manager will organize a training session for the WHM field work. Field staff will be expected to follow the protocols presented during training, especially when they are updated over the SOPs listed in this QAPP. At this time, the staff will verify, by signature, that applicable SOPs and protocols were reviewed during the training. This will improve the comparability of data collected within the program.

Biological monitoring efforts within Ecology use the applicable protocols followed by Washington's Status and Trends Monitoring Program. These protocols are similar to those of others in the region, including the Oregon Department of Environmental Quality's bioassessment program, and EPA's Regional Environmental Monitoring and Assessment Program (R-EMAP). Following these commonly accepted protocols will result in data that is comparable to other regional programs.

B-2. Data quality indicators for water and sediment chemistry

Accuracy

Accuracy is the measure of agreement between a measurement's result and the true or known value. Analytical accuracy can be found by analyzing known reference materials or known standards (LCS, ms/msd, and/or surrogates). A common metric is the percent recovery of a spike. Factors that influence analytical accuracy include laboratory calibration procedures, sample (field and laboratory) preparation procedures, and laboratory equipment or deionized water contamination.

Sensitivity

Sensitivity refers to the concentration that the analytical method can positively identify and report analytical results. The laboratory's "detection" limit indicates their sensitivity for a given method. The reporting limits specified in Tables 20-21 indicate the target quantitation limit established from experience at Manchester Environmental Laboratory (MEL) and King County

Environmental Laboratory (KCEL). Other laboratories, that may test the samples with short holding times, may be able to achieve different reporting limits, and permittees and their laboratories may report on lower limits than expressed in the MQO tables.

Precision

Precision is the measure of nearness of repeated measurements to the same value over time. Precision of samples and data collected will be evaluated using field replicate and laboratory duplicate sample analyses. Poor precision of field replicates may be due to heterogeneity of the water or sediment sample. Field replicates should be evaluated at the targeted relative percent difference (RPD) or relative standard deviation (or RSD) as listed in Tables 17-18. Other reasons for poor precision may include contamination, problems with sampling, or poor sensitivity of the analytical methods. Bias and blanks will assist with determining a reason for poor precision. Analytical precision is measured using laboratory duplicate (split) and matrix spike/matrix spike duplicate samples. Matrix spikes and matrix spike duplicates (ms/msd) are prepared by adding a known concentration of a compound to the sample and determining the concentration of that spike in the sample matrix. The matrix spike and matrix spike duplicate are compared to provide an estimate of the precision of the laboratory method.

Precision of a duplicate pair is calculated as the relative percent difference (RPD), which is usually expressed as a percentage:

Equation 1: Relative Percent Difference

$$\text{RPD} = \frac{|C_1 - C_2|}{\bar{x}} \times 100\%$$

Where:

RPD = relative percent difference
C_1 = concentration of original sample
C_2 = concentration of duplicate
$\bar{x}$ = mean of samples

Precision of more than two sample replicates is calculated as the relative standard deviation (RSD), which is expressed as a percentage:

Equation 2: Relative Standard Deviation

$$\text{RSD} = \frac{\text{S}}{\bar{x}} \times 100\%$$

Where:

RSD = percent relative standard deviation
S = standard deviation
$\bar{x}$ = mean of samples

Precision in trend analysis is further described in Hallock and Ehinger (2003).

Bias

Bias represents systematic error and can be used to describe a tendency or preference in one direction. Bias in water quality or sediment quality samples can be assessed based on the analyses of method blanks, field blanks, trip blanks, matrix spikes, and laboratory control samples (LCS). In the field, bias from meters will be consistently evaluated by testing against known standards.

Field blank results greater than the reporting limit (RL) should be flagged (e.g., *B) as* blank contamination. Depending on the type of blank collected (trip, transfer, or equipment), it may be possible to re-run the blank and/or re-clean the equipment that may have contaminated the field blank. Typically, associated project samples within five times the blank concentration are qualified as an estimate (*J*).

Status and trends monitoring programs are ongoing, long-term projects. Typically, we assume that any batch-specific bias in the chemical analyses will be corrected so that long-term bias will not occur within a single laboratory method. Sampling bias will be minimized by strictly adhering to the protocols discussed and referenced here.

Laboratory bias

Method blanks and matrix spikes are used to identify potential laboratory or sample matrix biases affecting results. Laboratory method blanks should not exceed the reporting limit. If this occurs, the associated blank concentration may be defined as the new reporting limit. For conventional parameters (e.g., turbidity, nutrients) and metals and samples with identified contaminants, the sample concentration must be at least five times the method blank concentration for the result to be considered valid. Sample concentrations within this five times de-facto reporting limit will be flagged by the laboratory as *B*, and should be reviewed by permittees.

For all samples identified with organic compound (e.g., phthalates, PBDEs, PCBs) laboratory contaminants, the concentration must be within ten times the blank concentration, de-facto reporting limit. This result will be flagged as *B*, and should be reviewed by permittees. Permittees will determine how many samples are affected and if corrective actions are necessary.

The targeted range for percent recovery of matrix spikes and matrix spike duplicates (ms/msd) varies according to the parameter, as shown in Tables 20-21. Percent recovery for matrix spikes will be calculated using:

Equation 3: Percent Recovery for MS/MSD

$$\%R = \frac{(X_s - X_o)}{C_s} \times 100\%$$

Where:

$\%R$ = percent recovery
X_s = spike sample result
X_o = original sample amount
C_s = concentration of spike

The goals for percent recovery of LCS vary for each parameter. Percent recovery for LCS will be calculated using:

Equation 4: Percent Recovery for LCS

$$\%R = \frac{M}{T} \times 100\%$$

Where:

%R = percent recovery
M = measured value
T = true value

Representativeness

Representativeness is a qualitative measure of the degree to which sample data represent characteristic environmental conditions or, more specifically, site conditions. Representativeness for water samples will be ensured by collecting water and sediment samples with consistency to the described protocols. Water collection consists of a single, near surface water sample taken from bridges or, in a few cases from the riverbank, where the river appears to be well-mixed vertically and horizontally. Although vertical heterogeneity of sediment-associated chemical species does occur, especially in large rivers, homogeneity is assumed for our objectives of characterization and trend analysis. This assumption should not be made for some constituents if our data are to be used for loading analyses.

Likewise, temporally, we assume 12 monthly samples are representative for our purposes. However, those using the data for annual loading estimates should evaluate this assumption, particularly for constituents that tend to be "patchy," like sediment, or highly associated with “first flush” effects, like total phosphorus. The time of day when samples are collected is determined by the logistics of sampling all stations and delivering the samples to the lab for timely analysis. Attempts should be made to sample a particular station at the same time each trip during a given year, but the time of sample collection often changes. Results for constituents with large diurnal variations (temperature, pH, and dissolved oxygen) may need to be adjusted statistically to a common time prior to trend analysis, for example.

Completeness

Completeness is the percentage of measurements judged to be valid over the total number of measurements compared to the amount of data deemed necessary to meet monitoring objectives. Completeness goals in terms of number of samples are set to the number required by the permit and this QAPP. Laboratory analysis can improve completeness by processing samples within their holding times.

Comparability

Comparability is a qualitative measure designed to express the confidence with which one data set may be compared to another. Standard sampling procedures, analytical methods, units of measurement, reporting rules, and reporting limits will be applied to meet the goal of data comparability. Comparability is limited by other MQOs because data sets can be compared with confidence only when precision and accuracy are known.

Appendix C. Watershed health measurement procedures

C-1. Site verification and layout for small streams

Personnel responsibilities

This method is performed by 2 or more trained staff. Agencies conducting monitoring are responsible for gathering permissions for property access, if necessary.

Equipment, reagents, supplies

- GPS
- *GPS Positions Form*
- Measuring rod
- 50-m tape
- Flagging
- Permanent marker
- Laser rangefinder
- Soft-lead pencil
- *Site Verification Form*
- Wading gear
- No. 2 pencil
- Maps

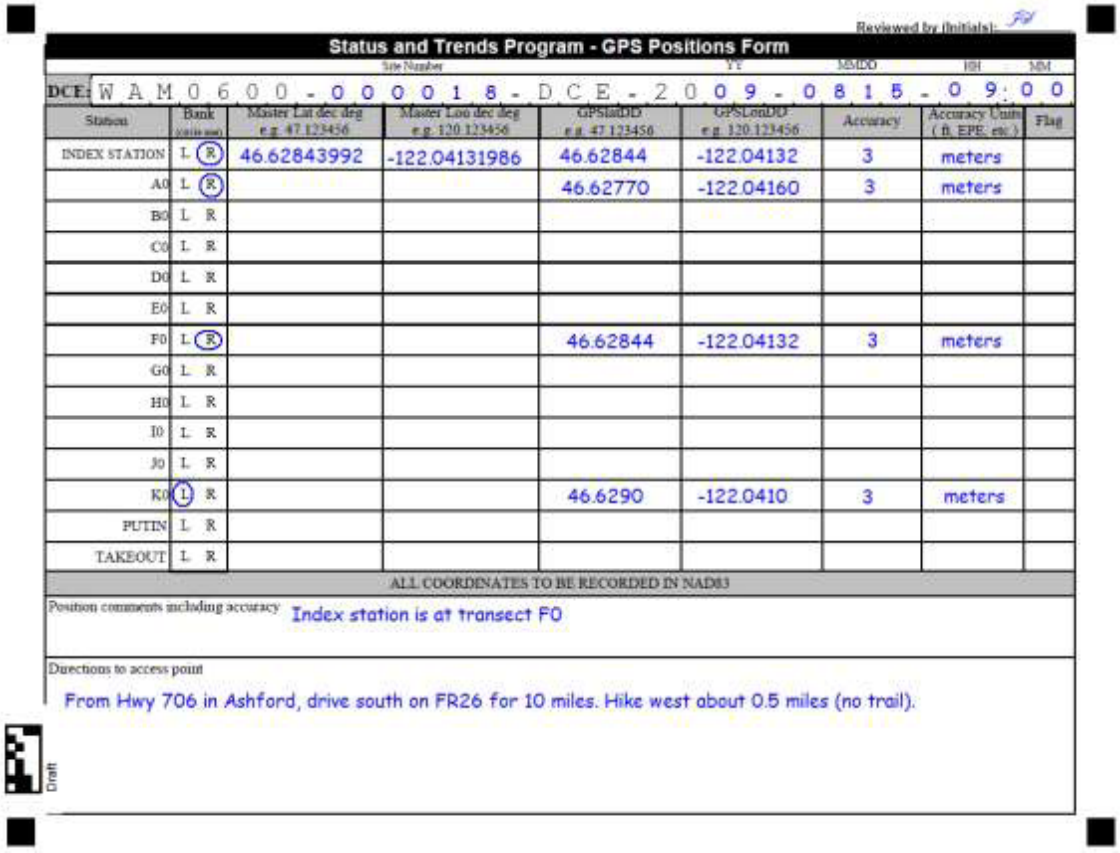
Summary of procedure

The crew first establishes the data collection event by:

(1) Navigating to the site using the Master Sample site coordinates provided on the RSMP website (www.ecy.wa.gov/programs/wq/stormwater/municipal/rsmp.html).
(2) Verifying that they are at the correct location and determining the site suitability for sampling.
(3) Defining the upper and lower boundaries and transects within the site.

Establish the data collection event

Before leaving the office, refer to the *GPS Positions Form* (Figure C-1.1). Enter the SITE_ID portion of the Data Collection Event (DCE) using a number 2 pencil. Enter the Latitude and Longitude as listed. Navigate to the site using the GPS receiver. Upon arrival, record the date (MMDD) and time (military) portion of the DCE. Record the GPS-measured coordinates for the Index Station. Identify the bank at which these coordinates were measured (left and right are interpreted when facing downstream). Also note the precision of the GPS measurement. Other notes on location can also be recorded. Record the turn-by-turn directions taken to reach the site's access point.



Reviewed by (Initials):

Status and Trends Program - GPS Positions Form

Site Number | YY | MMDD | HH | MM

DCE: WAM0600-000018-DCE-2009-0815-09:00

Station	Bank (circle one)	Master Lat dec deg e.g. 47.123456	Master Lon dec deg e.g. 120.123456	GPSLatDD e.g. 47.123456	GPSLonDD e.g. 120.123456	Accuracy	Accuracy Units (ft, EPE, etc.)	Flag
INDEX STATION	L (R)	46.62843992	-122.04131986	46.62844	-122.04132	3	meters	
A0	L (R)			46.62770	-122.04160	3	meters	
B0	L R							
C0	L R							
D0	L R							
E0	L R							
F0	L (R)			46.62844	-122.04132	3	meters	
G0	L R							
H0	L R							
I0	L R							
J0	L R							
K0	(L) R			46.6290	-122.0410	3	meters	
PUTIN	L R							
TAKEOUT	L R							

ALL COORDINATES TO BE RECORDED IN NAD83

Position comments including accuracy: Index station is at transect F0

Directions to access point:

From Hwy 706 in Ashford, drive south on FR26 for 10 miles. Hike west about 0.5 miles (no trail).

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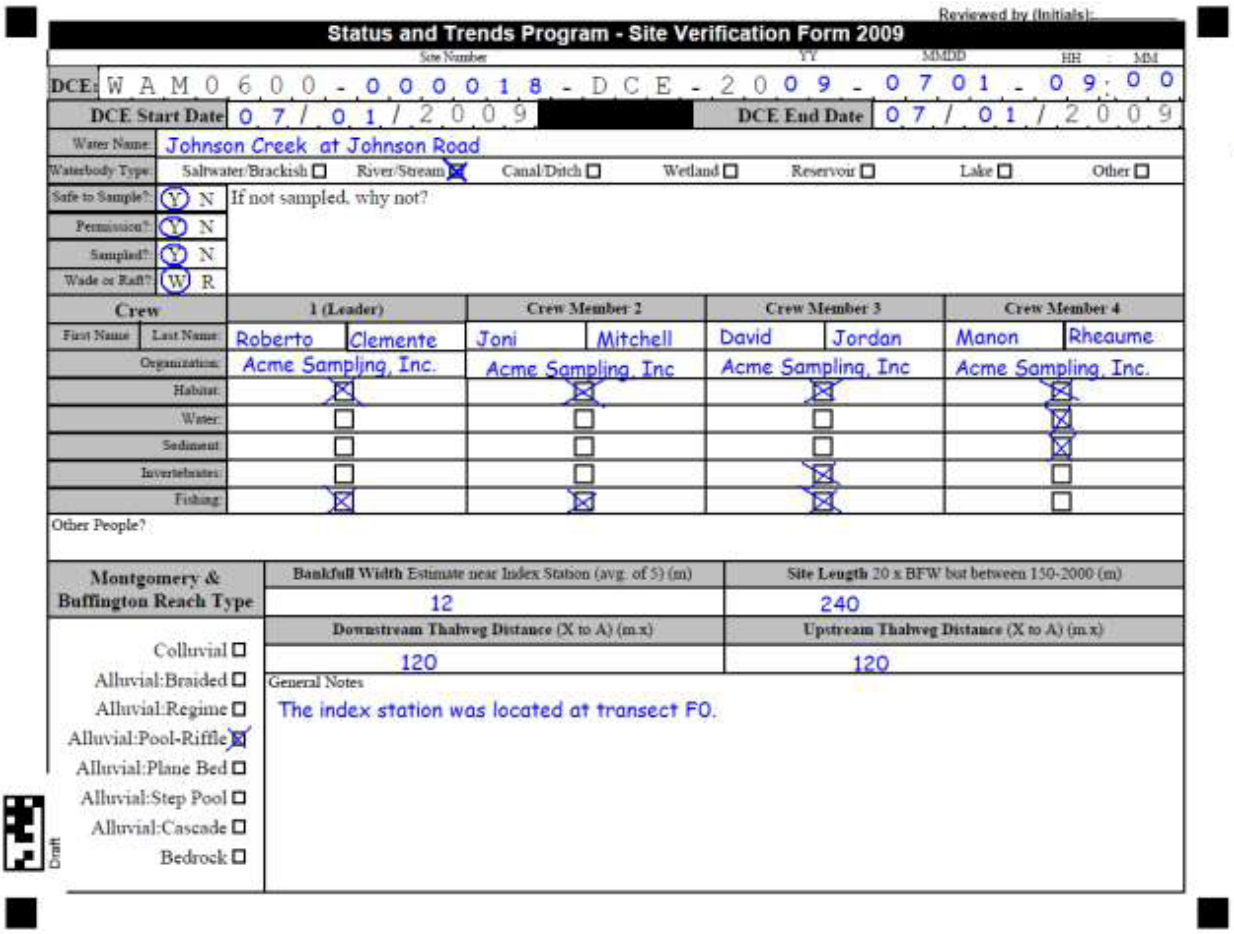
Figure C-1.1. The *GPS Positions Form* with example data. Note: Sometimes streams have re-routed after production of the map from which the Master coordinates were generated. In these cases navigate to the closest (most representative) point on the stream.

Determine site suitability

After arriving and recording the DCE, determine whether the site is suitable for sampling. Refer to the *Site Verification Form* (Figures C-1.2, and C-1.3).



Reviewed by (Initials):

Status and Trends Program - Site Verification Form 2009

	Site Number	YY	MMDD	HH	MM
DCE:	WAM0600-000018-DCE-	2009	-0701	-09:	00

DCE Start Date	07/01/2009	DCE End Date	07/01/2009

Water Name: Johnson Creek at Johnson Road

Waterbody Type: Saltwater/Brackish ☐ River/Stream ☒ Canal/Ditch ☐ Wetland ☐ Reservoir ☐ Lake ☐ Other ☐

Safe to Sample? (Y) N — If not sampled, why not?

Permission? (Y) N

Sampled? (Y) N

Wade or Raft? (W) R

Crew	1 (Leader)	Crew Member 2	Crew Member 3	Crew Member 4
First Name / Last Name	Roberto Clemente	Joni Mitchell	David Jordan	Manon Rheaume
Organization	Acme Sampling, Inc.	Acme Sampling, Inc	Acme Sampling, Inc	Acme Sampling, Inc.
Habitat	☒	☒	☒	☒
Water	☐	☐	☐	☒
Sediment	☐	☐	☐	☒
Invertebrates	☐	☐	☒	☐
Fishing	☒	☒	☒	☐

Other People?

Montgomery & Buffington Reach Type	Bankfull Width Estimate near Index Station (avg. of 5) (m)	Site Length 20 x BFW but between 150-2000 (m)
	12	240
	Downstream Thalweg Distance (X to A) (m.x)	**Upstream Thalweg Distance** (X to A) (m.x)
	120	120

Colluvial ☐
Alluvial:Braided ☐
Alluvial:Regime ☐
Alluvial:Pool-Riffle ☒
Alluvial:Plane Bed ☐
Alluvial:Step Pool ☐
Alluvial:Cascade ☐
Bedrock ☐

General Notes

The index station was located at transect F0.

Draft

Figure C-1.2. The front side of the Site Verification Form with example data.

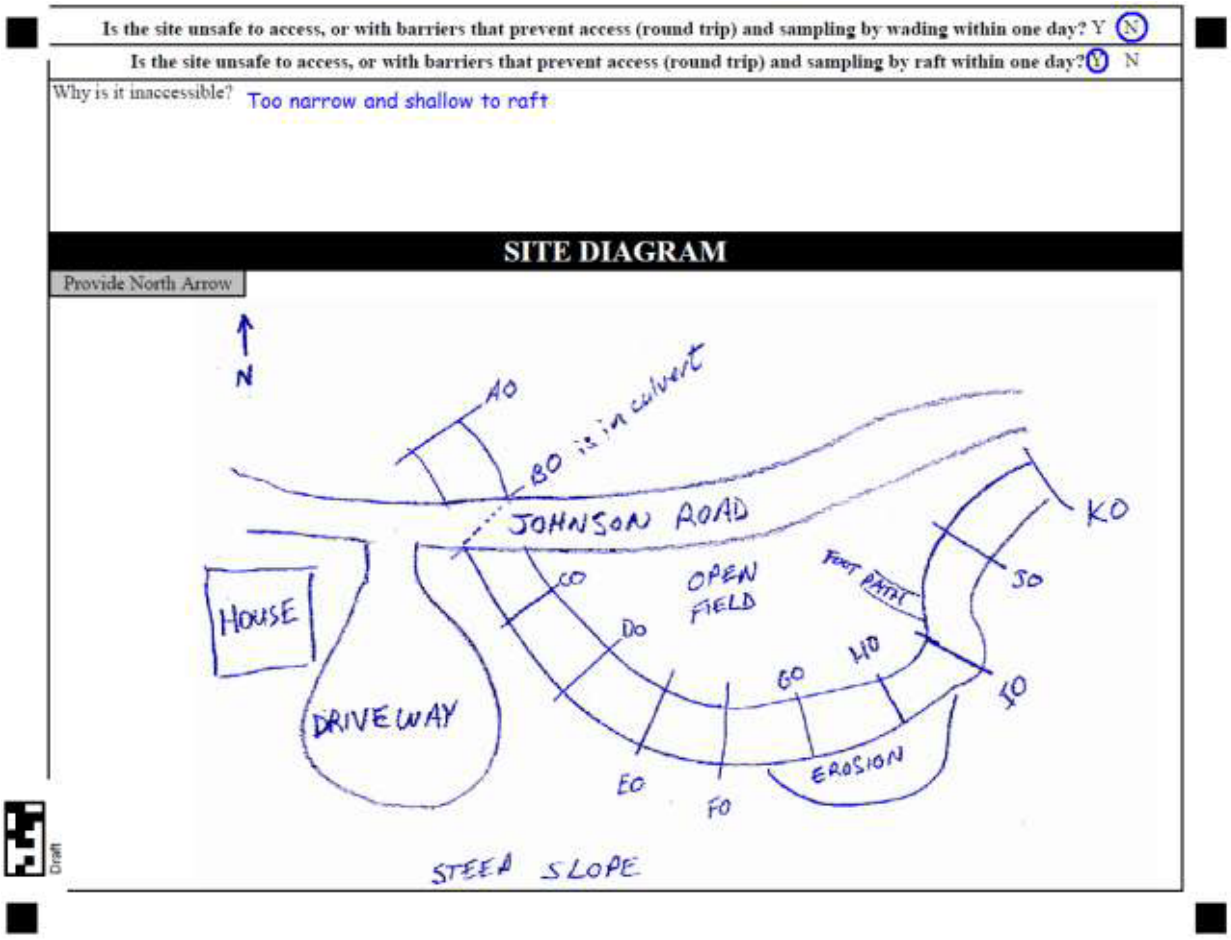


Figure C-1.3. The backside of the Site Verification Form, with example data.

Desktop evaluation of the site was performed earlier according to the method described elsewhere in this protocol. Verify that conditions at the site are truly suitable for sampling during the day of arrival. Complete the appropriate fields in the top third of the front side of the *Site Verification Form*, indicating whether the site is being sampled. The site should not be sampled if it is deemed:

- Unsafe to enter.
- To have permission denied by land owners.
- Not a stream or river (e.g., a wetland, lake).
- Not freshwater.
- Within an artificial channel (e.g., canal or ditch).
- Not perennial.
- Not with surface flow for more than 50% of the length.

Record event information

Next, on the *Site Verification Form* (Figure C-1.2), record the information below about the data collection event.

Crew

Record the names of those who are in the crew. Also note the organization that each staff represents. The crew lead will be recorded in column 1. Staff sampling roles can be recorded later, after the day is done, by using the check boxes provided on the form.

Site

BANKFULL STAGE

Near the Index Station (X), visually estimate the bankfull stage. This is best done after considerable training. There are good on-line sources of training materials for identifying bankfull stage identified on PNAMP monitoring methods website (https://www.monitoringmethods.org/Method/Details/3838), including:

1. www.pnamp.org/sites/default/files/BuffingtonPPT_v.2.ppt (Buffington, 2006)
2. www.dnr.wa.gov/Publications/fp_bfw_video_pt1.wmv www.dnr.wa.gov/Publications/fp_bfw_video_pt2.wmv (Grizzel, 2008)
3. www.stream.fs.fed.us/publications/bankfull_west.html (Leopold, et al., 1995)

Bankfull stage height is *not* a value that gets recorded on the *Site Verification Form*. The crew merely uses their visual estimate to help understand where to measure bankfull width.

BANKFULL WIDTH

Using the estimated bankfull level, measure the channel width at each of 5 transects near the Index Station:

1. The Index Station (X)
2. 1 bankfull width upstream from X
3. 2 bankfull widths upstream from X
4. 1 bankfull width downstream from X
5. 2 bankfull widths downstream from X

Record the average (nearest meter) of these 5 bankfull width measurements on the *Site Verification Form* (Figure C-1.2). Width measurements can be made using a 50-m tape, a measuring rod, or (if the channel is wide) a laser rangefinder.

SITE LENGTH

Multiply the average bankfull width times 20. This value (whole meters) is the site length for a path that follows the main flow of the river. However, for any site with bankfull width less than 8 meters, the site length will be extended to 150 m, the minimum length for a sampling reach. Record the site length on the *Site Verification Form* (Figure C-1.2). Sampling methods for waded streams are restricted to sites that are less than 25 meters wide (less than 500 m long).

RELATIVE POSITION OF THE INDEX STATION (X) WITHIN THE SITE

The index station (X) is normally located at the middle of the site (i.e. at major transect F). On the *Site Verification Form* (Figure C-1.2), record the distance (tenths of meters) from X to the bottom of the site (i.e., to major transect A) and the distance from X to the top of the site (i.e., to major transect K). This distance is measured along the thalweg channel. Unless there is a reason to adjust the position of X, the distance will be equal to half the site length, in each direction. The relative position of X can be adjusted for several reasons: to keep the top or bottom of the site in lands where permission has not been denied, to keep from changing Strahler (1957) stream order (at the 1:100,000 scale), or to account for barriers such as lakes. The location of the Index Station's coordinates can never be changed. These are pre-defined by the survey design. Although the site position can change relative to X (called "sliding" the site), the site must always contain X.

BED FORM

Assess the site for its predominant reach type according to Montgomery and Buffington (1993, 1997). Review the source materials hot-linked in the references to help understand the differences between bed forms. These references discuss details and provide images of examples. First decide whether the site is predominated by a reach that is colluvial, alluvial, or bedrock. Colluvial streams have a low chance of being sampled by this status and trends program, because we are limiting our sample to perennial streams. Bedrock streams are confined locations with little depositional material present. Most streams sampled will be alluvial. Next, if the site is predominantly alluvial, decide which one of the following sub-classifications can be used to describe the site.

- cascade
- step-pool
- plane-bed
- pool-riffle
- regime
- braided

Place an X in the appropriate box of the *Site Verification Form* (Figure C-1.2) to describe the predominant bed form within the site. Refer to the references (Montgomery and Buffington, 1993, 1997, 1998) for help. Figures C-1.4 and C-1.5 might help.

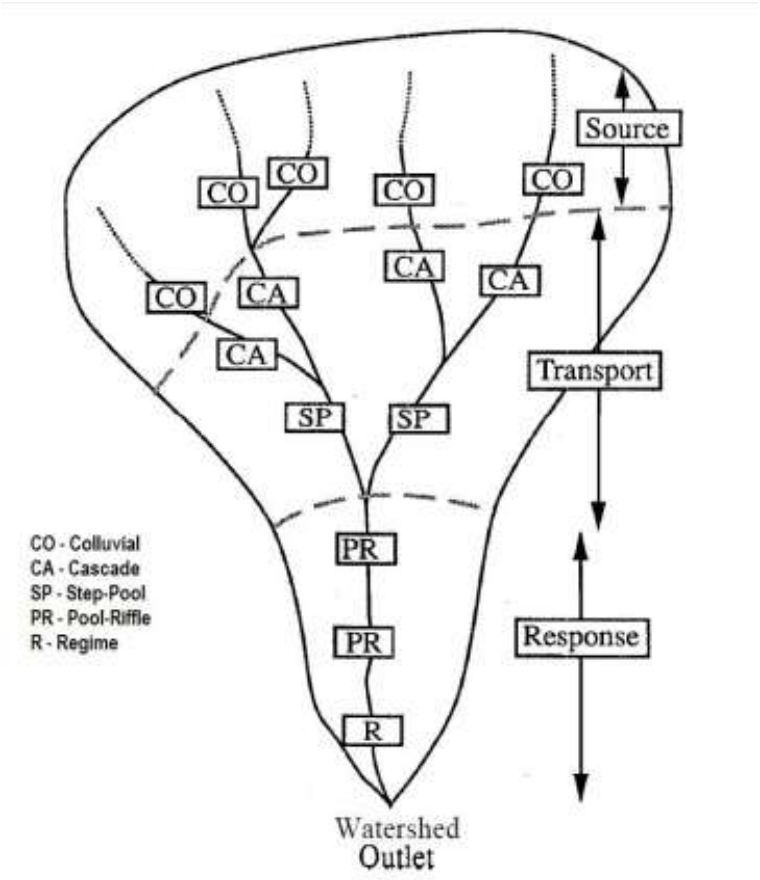


Figure C-1.4. Idealized positions (aerial view) of bed form types within a watershed.

Modified from Figure 22 of Montgomery and Buffington (1993).

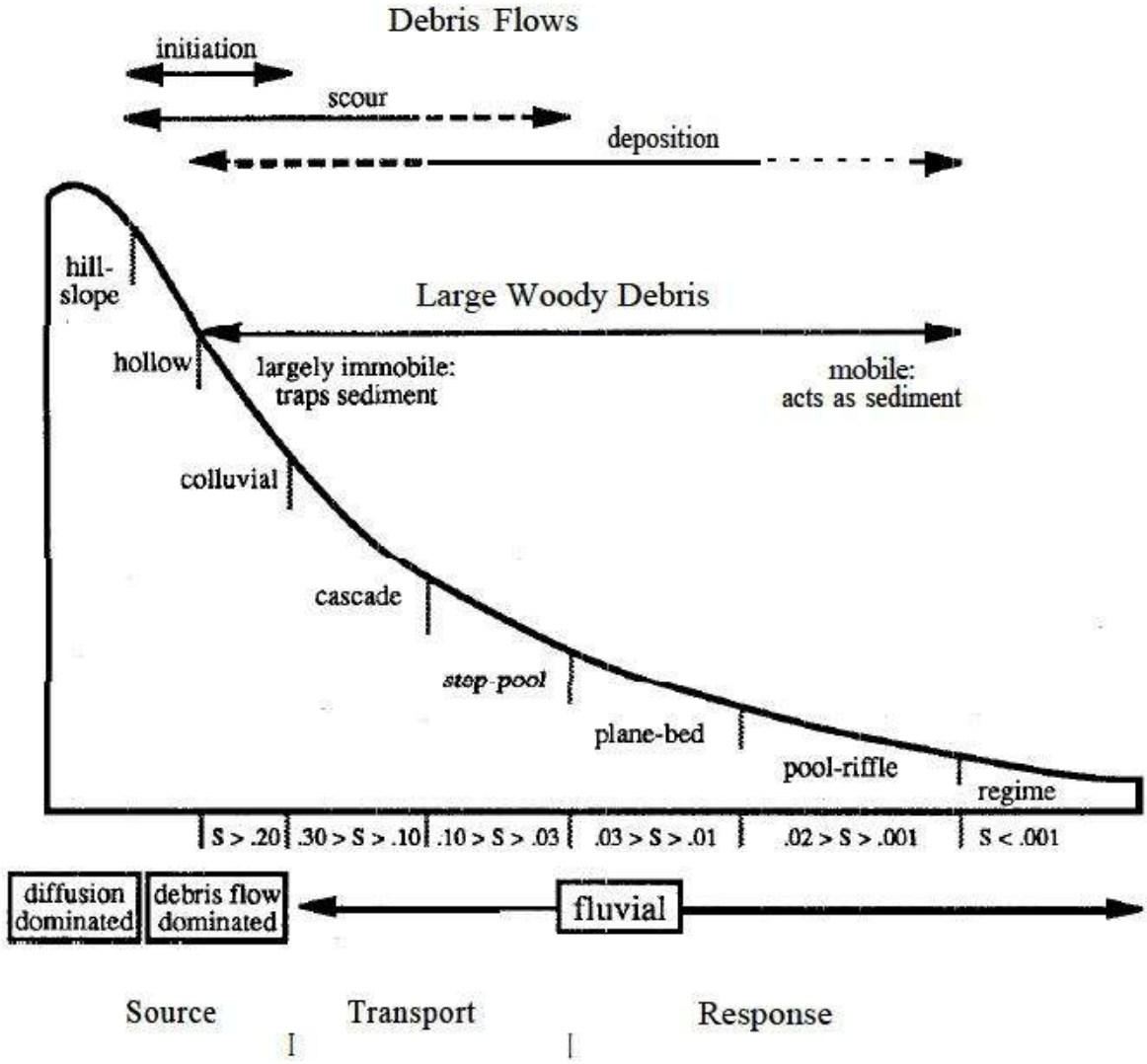


Figure C-1.5. Idealized positions (plan view) of bed form types within a watershed.

From Figure 16 of Montgomery and Buffington (1993).

Lay out the reach

There are 3 types of transects that define the stream site (Table C-1.1): thalweg transects, major transects, and minor transects.

Thalweg transects

Conceptually divide the stream site length using 101 transects which are perpendicular to the thalweg. These are called Thalweg Transects. They occur at regular intervals (0.2 bankfull widths). Thalweg transects, except for those that are also major transects (see below), do not need to be marked. Thalweg transects are useful in concept for describing relative positions within the site.

Major transects

Use orange flagging and a permanent marker to mark each of the 11 equidistant major transects. The lowest is *transect A0*, the highest is *transect K0*. Measure the distance between transects using either a 50-m tape or a measuring rod, by following the thalweg of the stream. The distance between flags should be 1/10th of the site length or (or 2 times the estimated bankfull width at the index station).

Minor transects

Ten minor transects occur midway between the 11 major transects (Table C-1.1). The distance between major and minor transects is $1/5^{th}$ of the site length (or 1 bankfull width). Minor transects don't need to be marked.

Table C-1.1. The relative position of all transects on a stream site.

Station	Thalweg Transect	Major Transect	Minor Transect	Distance from Bottom (Bankfull Widths*)
A0	Yes	Yes		0
A1	Yes			0.2
A2	Yes			0.4
A3	Yes			0.6
A4	Yes			0.8
A5	Yes		Yes	1
A6	Yes			1.2
A7	Yes			1.4
A8	Yes			1.6
A9	Yes			1.8
B0	Yes	Yes		2
B1	Yes			2.2
B2	Yes			2.4
B3	Yes			2.6
B4	Yes			2.8
B5	Yes		Yes	3
B6	Yes			3.2
B7	Yes			3.4
B8	Yes			3.6
B9	Yes			3.8
C0	Yes	Yes		4
C1	Yes			4.2
C2	Yes			4.4
C3	Yes			4.6
C4	Yes			4.8
C5	Yes		Yes	5
C6	Yes			5.2
C7	Yes			5.4
C8	Yes			5.6
C9	Yes			5.8
D0	Yes	Yes		6
D1	Yes			6.2
D2	Yes			6.4
D3	Yes			6.6

Station	Thalweg Transect	Major Transect	Minor Transect	Distance from Bottom (Bankfull Widths*)
D4	Yes			6.8
D5	Yes		Yes	7
D6	Yes			7.2
D7	Yes			7.4
D8	Yes			7.6
D9	Yes			7.8
E0	Yes	Yes		8
E1	Yes			8.2
E2	Yes			8.4
E3	Yes			8.6
E4	Yes			8.8
E5	Yes		Yes	9
E6	Yes			9.2
E7	Yes			9.4
E8	Yes			9.6
E9	Yes			9.8
F0	Yes	Yes		10
F1	Yes			10.2
F2	Yes			10.4
F3	Yes			10.6
F4	Yes			10.8
F5	Yes		Yes	11
F6	Yes			11.2
F7	Yes			11.4
F8	Yes			11.6
F9	Yes			11.8
G0	Yes	Yes		12
G1	Yes			12.2
G2	Yes			12.4
G3	Yes			12.6
G4	Yes			12.8
G5	Yes		Yes	13
G6	Yes			13.2
G7	Yes			13.4
G8	Yes			13.6
G9	Yes			13.8
H0	Yes	Yes		14
H1	Yes			14.2
H2	Yes			14.4
H3	Yes			14.6
H4	Yes			14.8
H5	Yes		Yes	15
H6	Yes			15.2
H7	Yes			15.4
H8	Yes			15.6
H9	Yes			15.8
I0	Yes	Yes		16
I1	Yes			16.2
I2	Yes			16.4
I3	Yes			16.6

Station	Thalweg Transect	Major Transect	Minor Transect	Distance from Bottom (Bankfull Widths*)
I4	Yes			16.8
I5	Yes		Yes	17
I6	Yes			17.2
I7	Yes			17.4
I8	Yes			17.6
I9	Yes			17.8
J0	Yes	Yes		18
J1	Yes			18.2
J2	Yes			18.4
J3	Yes			18.6
J4	Yes			18.8
J5	Yes		Yes	19
J6	Yes			19.2
J7	Yes			19.4
J8	Yes			19.6
J9	Yes			19.8
K0	Yes	Yes		20

*For very small (with length of 150 m), the transect spacing is 1/100th of the site length, and might not be 0.2 bankfull widths.

Record coordinates

Record the GPS-measured coordinates at the bottom of the site (transect A0), and at the top of the site (transect K0). Note the bank at which the GPS was used and the accuracy of the measurements. You might also record coordinates for other major transects too, but this is not required for the waded streams.

C-2. In-situ measurements in small streams

Purpose and scope

This explains the methods to collect in-situ measures of temperature, dissolved oxygen, pH, and conductivity at small streams using a multi-probe, based on SOP EAP033. Grab sample collection methods are described in SOP EAP034.

Ecology's ambient water quality program collects dissolved oxygen data using the Winkler titration method in the field; however, this is not necessary for permittees conducting monitoring. Therefore the sections of SOP EAP034 referencing DO samples should be disregarded and the protocol for use of LDO meters EAP033 used instead. The calibration techniques are discussed in EAP033 and in the manufacturer's websites. Relevant information from Adams (2010b) is retained here.

Personnel responsibilities

This method is performed by 1 or more persons. This method is applied at every DCE, at the start of the sampling event. Staff performing this method must have been trained.

Equipment, reagents, supplies

- No. 2 pencil.
- *Chemistry and Sampling Form*.
- Calibration Form.
- Hydrolab (or equivalent), components, maintenance kit (Swanson, 2007).
- Hydrolab (or equivalent) Manuals (Hach 1999; 2006a; 2006b).

Summary of procedure

Calibrate the instrument before sampling. Measure the stream twice.

Verify quality control

Prior to sampling

Ensure that the calibrations and that QC checks have been performed according to EAP033. Record calibration information on *Meter Calibration form* (Table C-2.1). Circle "Yes" on the top section of the *Chemistry and Sampling Form* (Figure C-2.1) for each sensor that checked out. Proceed with measurements using sensors that are within criteria.

Table C-2.1. An example Meter Calibration Form, with examples data records.

Recorder:

Project Name: _____ **Date:** _____

Sonde #: _____ **Time** _____

Temp: _____ **Barometric Pressure:** _____

PRE Field Run CALIBRATION

	Meter Reading	**Buffer Value**	**Buffer Temp**	**Comments**
Conductivity-100 µS/cm	*94*	*100*		*post-calibration reading was unstable in buffer; ranging between 95 - 105*
Conductivity-1000 µS/cm				
pH - 7				
pH - 10				
pH - 4				*linearity check; not calibrated to buffer value*
DO % Saturation				
Temperature check with NIST thermometer				

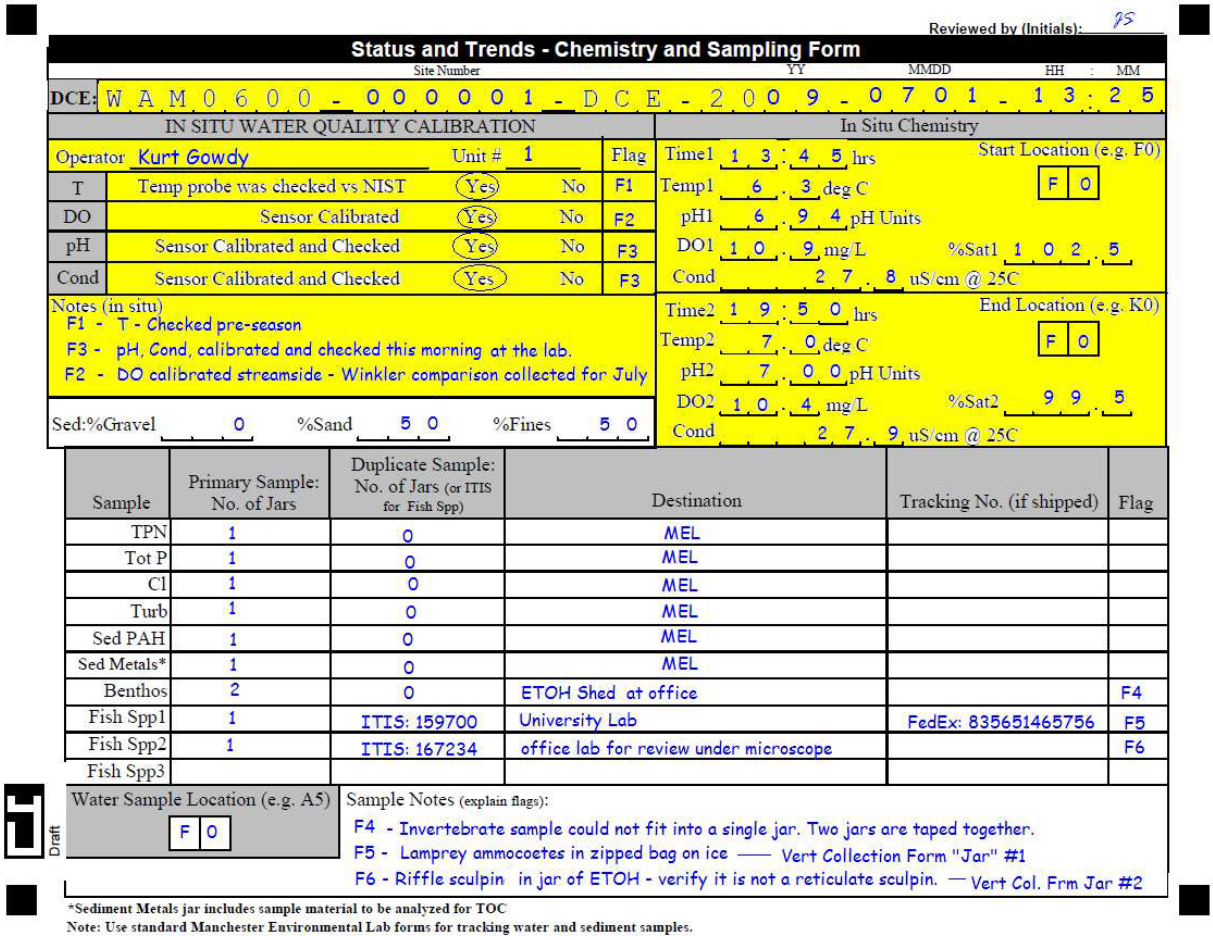
POST Field Run Check

	Meter	**Buffer**	**Comments**
Conductivity-100 µS/cm	*98*	*100*	
Conductivity-1000 µS/cm			
pH - 7			
pH - 10			
pH - 4			
DO % Saturation			

Measure

Measure pH, water temperature, dissolved oxygen, oxygen percent saturation, and specific conductivity during a DCE. Record time (military) and location (thalweg transect). Both sets of in-situ measurements should usually be made near the middle elevation of the site, on the main channel. Measurements should *always* be taken within the boundaries of the site (between transects A0 and K0).

Place the probes into the stream and let them thermally equilibrate to the stream temperature. This might take 3-5 minutes. Then hold the sensors so that they are just below the surface of the water, and completely immersed. Avoid any turbulence. Make sure that readings are stable. On the *Chemistry and Sampling Form* (Figure C-2.1), record temperature (˚ C, nearest tenth), pH (pH unit, nearest hundredth), specific conductivity (uS/cm at 25˚ C, nearest tenth), dissolved oxygen (mg/L, nearest tenth), and oxygen percent saturation (nearest tenth).



Reviewed by (Initials): JS

Status and Trends - Chemistry and Sampling Form

DCE: WAM0600 - 0000001 - DCE - 2009 - 0701 - 13:25 (Site Number; YY; MMDD; HH : MM)

IN SITU WATER QUALITY CALIBRATION

Operator: Kurt Gowdy — Unit #: 1

				Flag
T	Temp probe was checked vs NIST	(Yes)	No	F1
DO	Sensor Calibrated	(Yes)	No	F2
pH	Sensor Calibrated and Checked	(Yes)	No	F3
Cond	Sensor Calibrated and Checked	(Yes)	No	F3

Notes (in situ)
F1 - T - Checked pre-season
F3 - pH, Cond, calibrated and checked this morning at the lab.
F2 - DO calibrated streamside - Winkler comparison collected for July

Sed:%Gravel 0 %Sand 50 %Fines 50

In Situ Chemistry

Start Location (e.g. F0): F0

Time1 13:45 hrs
Temp1 6.3 deg C
pH1 6.94 pH Units
DO1 10.9 mg/L %Sat1 102.5
Cond 27.8 uS/cm @ 25C

End Location (e.g. K0): F0

Time2 19:50 hrs
Temp2 7.0 deg C
pH2 7.00 pH Units
DO2 10.4 mg/L %Sat2 99.5
Cond 27.9 uS/cm @ 25C

Sample	Primary Sample: No. of Jars	Duplicate Sample: No. of Jars (or ITIS for Fish Spp)	Destination	Tracking No. (if shipped)	Flag
TPN	1	0	MEL		
Tot P	1	0	MEL		
Cl	1	0	MEL		
Turb	1	0	MEL		
Sed PAH	1	0	MEL		
Sed Metals*	1	0	MEL		
Benthos	2	0	ETOH Shed at office		F4
Fish Spp1	1	ITIS: 159700	University Lab	FedEx: 835651465756	F5
Fish Spp2	1	ITIS: 167234	office lab for review under microscope		F6
Fish Spp3					

Water Sample Location (e.g. A5): F0

Sample Notes (explain flags):
F4 - Invertebrate sample could not fit into a single jar. Two jars are taped together.
F5 - Lamprey ammocoetes in zipped bag on ice — Vert Collection Form "Jar" #1
F6 - Riffle sculpin in jar of ETOH - verify it is not a reticulate sculpin. — Vert Col. Frm Jar #2

Draft

*Sediment Metals jar includes sample material to be analyzed for TOC
Note: Use standard Manchester Environmental Lab forms for tracking water and sediment samples.

Figure C-2.1. The *Chemistry and Sampling Form*, with examples of in-situ data records.

C-3. Estimating discharge in small streams

(This protocol was moved from Appendix C –Watershed health measurement procedures section C-3 to Appendix E – Water quality sampling procedures section E-5. Discharge and stage measurements will be monitored monthly under the RSMP. For measuring flow during the WHM visit, refer to the protocols in E-5).

C-4. Sediment chemistry sampling

This section draws on sediment sampling protocols for sampling and sieving composite sediment samples in streams from USGS National Field Manual (USGS, 2005) and NAWQA protocols (USGS, 1994).

This method explains how to collect and process bed-sediment samples for Watershed Health monitoring. A composite sediment sample will be composed of sub-samples taken from 5 different shallow-water stations in the site. The composite sample will be processed (sieved) in the field to make two unique samples. The first sample will be sieved to less than 2.0 mm and analyzed for multiple organic compounds (PAHs, pesticides, phthalates, PBDEs, PCBs, PPCPs, and H/S) percent solids, total-organic carbon (TOC) and grain size. The second sample will be sieved to less than 63 µm and analyzed for metals (arsenic, cadmium, chromium, copper, lead, silver and zinc).

Personnel requirements

This sampling can be performed by one person in the field, but would likely be done more efficiently by a two-person team during a day-long Watershed Health Sampling event. Pre-sampling cleaning activities should be performed by staff familiar with MSDS and safety procedures. Staff collecting sediments should not use sunscreen and mosquito repellent until finished collecting the samples.

Equipment, reagents, supplies

Equipment and supplies for collecting and processing stream bed-sediment samples for analyses of trace elements and organic contaminants are listed in Table C-4.1. The use of each is explained in the following discussions of preparation for sampling, sampling procedures, and sample processing. The number of supplies depends on the total number of sites permittees are responsible for.

Summary of procedure

These procedures are derived from methods described in Johnson (1997), Blakley (2008a), and Manchester Environmental Laboratory (2008), with the additional sieving procedures described by Radke (2005) and Shelton and Capel (1994). The sample-collection strategy focuses on obtaining samples of fine-grained surficial sediments from natural depositional zones during low-flow conditions and on compositing samples from several depositional zones within a stream reach.

Table B-4.1. Equipment and supplies for collecting and processing bed sediment samples. Use uncolored or white non-metallic sieve and utensils to process bottom material for samples that will be analyzed for metals. Use a stainless steel sieve and polyfluorocarbon (Teflon) utensils to process bottom material for samples that will be analyzed for organic compounds. Brass is acceptable but not recommended.

Sampling and Processing
Bowl, glass, flat bottom, approximately 5 L, 12-in diameter
Sieve, stainless steel, 2.0 mm, 3" diameter (for organics sample)
Sieve Frame, Nylon, 8" diameter (for metals sample)
Nylon sieve cloth, 63 micron (for metals sample)
Funnel, polyethylene, 8" diameter
Policeman, Teflon (to aid sieving)
Spatula, scoop, and spoon, all Teflon
Syringe, plastic, 50ml
Wash bottle (labeled) with Liquinox or Alconox
Wash bottle (labeled) with acetone (pesticide grade)
Wash bottle (labeled) with 10% nitric acid
Wash bottle, plastic 500-ml
Wash bottle, Teflon 500-ml
Deionized water
Personal protective gear as specified by the MSDS
Sample containers (analytical laboratory will supply) – see Table 12 in QAPP
Miscellaneous
MSDS
Gloves - Non-powdered nitrile
Cooler and Ice
Polyethylene bags
Foam sleeves for shipping
Ice
5-gallon plastic bags
Sample Tags/bottle labels (with laboratory-assigned sample numbers)
Aluminum foil

Pre-sampling preparation

Sample Numbers, Jars, and Tags

Prior to sampling staff will obtain sample numbers, sample jars, and labels from laboratories conducting the analysis.

Cleaning

Prior to sampling, the field crew will clean necessary sampling tools (including spares). These are the cleaning steps for each reusable piece of sampling equipment that comes in contact with the sediment sample:

1. Washing in non-phosphate detergent and hot tap water
2. Rinsing with hot tap water
3. Rinsing with 10% nitric acid
4. Rinsing with deionized water three times
5. Air drying in clean area free of contaminants

6. Rinsing with pesticide-grade acetone
7. Air drying in clean area free of contaminants

After drying, equipment will be wrapped in aluminum foil (shiny side out) and stored in polyethylene bags until used in the field. Sampling equipment will be dedicated to the station and will only be used at subsequent stations following cleaning in accordance with the above procedures, which are based on EPA guidelines (EPA, 1990).

Sampling

Use clean equipment at each site. Collect the composite sample by sampling quiescent sediment from each of three suitable locations at each of three to five stations at the site. A suitable location will have these characteristics:

- Surface sediment is dominated by particles < 2 mm diameter (coarse sand or smaller),
- Water depth above the sediment is < than 30 cm,
- The station is always under water throughout the day.
- Anywhere within 10 bankfull widths (upstream or downstream) of the index station.
- Upstream from where staff have entered the stream channel.

Using a Teflon spoon, scoop, or spatula, carefully remove the top 2 cm of sediment and place it into a glass mixing bowl. The spatula can remove thin layers of surficial sediments, and the scoop or spoon can remove the bed material from between rocks and debris. Sieving is easier if the sandy material is avoided. Care must be taken to prevent the fine sediments from being washed away by the stream when bringing the sample to the surface. Collect a total of about 1.5 L of wet sediment.

Sample processing

Sediment samples for several types of analyses will be processed from the single composite sample from a site. One sample will be sieved to less than 63 µm and analyzed for metals. A second sample will be sieved to less than 2.0 mm and analyzed for multiple organic compounds and total-organic carbon. The third sample also will be sieved to less than 2.0 mm and analyzed for percent particle-size distribution less than 63 µm (sand/silt).

Prepare for sample processing:

1. Park the field vehicle as far away from any nearby road(s) as possible and turn off motor (road dust and vehicle emissions can contaminate samples) in order to isolate the sample-processing area from potential contaminants.
2. Set up field-processing area. Preferable areas would be in a van or a building located near the sampling site. If not available, a foldable table can be used onsite.
    a. Spread a large, uncolored or white plastic (non-metallic) sheet over the area where inorganic sample processing is taking place.
    b. Use heavy-duty aluminum sheeting over the area where organic sample processing is taking place.
    c. Keep sample-processing equipment covered (when not processing sample), and keep all sample containers covered or capped.

3. Field rinse processing equipment with native stream water to ensure that all cleaning solution residues are removed, and to equilibrate equipment with sampling environment.
4. Wear powderless, disposable gloves while processing sample. Avoid contact with any potential source(s) of contamination. For example, keep gloved hands off any reactive (metal or plastic) objects when processing samples.

Sieving

Two different sieves are required to process a sample for trace elements and organic contaminants. A 63-µm mesh nylon-sieve cloth held in a plastic frame is used for sieving sediment samples for trace-element analyses, and a 2.0-mm stainless-steel sieve is used for processing samples for organic-contaminants analyses. Wear nitrile gloves and thoroughly mix (homogenize) the composite sample in the glass bowl using the Teflon spatula until a uniform color and texture is achieved. Decant excess water from sample into an appropriate, nonreactive wash bottle, being careful not to lose fine material.

Metals samples

- Stretch the 63-µm mesh nylon-sieve cloth over the plastic-sieve frame and attach retaining ring. Assemble in series the 63-µm mesh nylon cloth sieve and the plastic funnel over a 500-mL plastic sample container.
- Place a small amount of composite sample onto the 63-µm mesh nylon sieve with the spatula. "Pressure sieve" the sample using native water that has been collected directly from the stream into the 500-mL plastic-wash bottle. The fine sediments pass through the sieve with the stream of water delivered by the wash bottle.
- Work small amounts of bed material through the sieve at a time, discarding the material remaining on the sieve. It is not necessary to sieve all the material that is less than 63 µm in each aliquot.

  NOTE: Shaking the sieve aggressively will help separate the fines.

- If additional wash water is needed, allow the sieved sediment/native water to settle several minutes and decant only the native water back into the wash bottle for reuse. Continue to reuse the native water until the necessary amount of sediment sample is obtained (a depth of approximately 1 cm in the sample container). The specific analytical laboratory can tell you how much sample material is needed for the analyses of inorganic constituents; typically that will be about 10 g (dry weight) of sieved sediment.

Organics samples

Place the 2.0-mm stainless-steel sieve over a 500-1,000-mL glass sample container. Gently work an aliquot of the sample through the sieve with a teflon policeman or spatula. Do not use water. The bottom of the sieve may require periodic removal of the material that adheres to it. Fill the sample container approximately half full or until an adequate amount of sample material has been collected; about 500 mL of wet sediment is typically needed for analyses of organic contaminants and TOC.

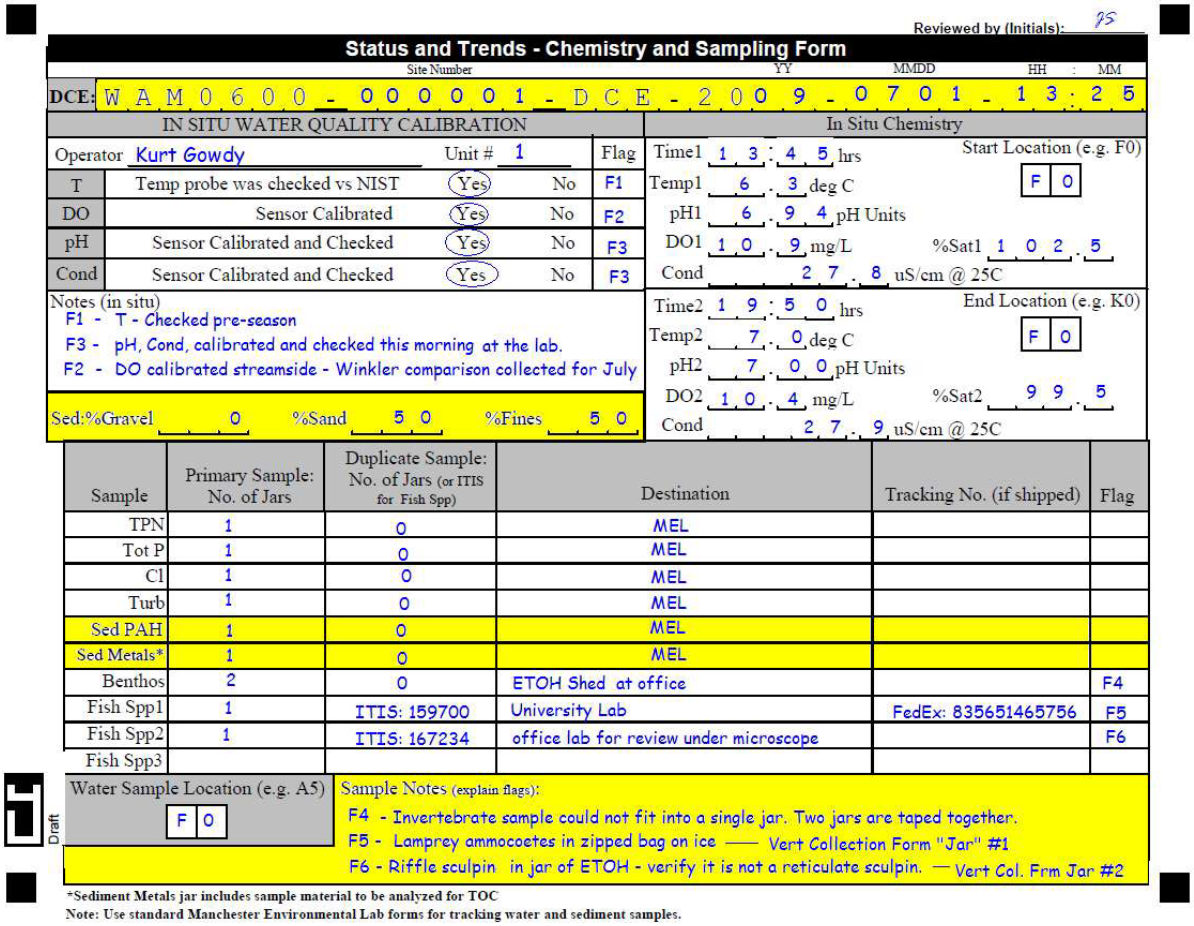
Particle size samples

Using the same 2.0-mm sieve described above, continue to sieve until approximately 2 cm of wet sediment accumulates into a 500-1,000-mL plastic sample container.

Reserve a scoop of the homogenized sample for conducting an estimate on the physical composition of the sediment. Gravel should never be a dominant component of the sample. Sand is gritty to the touch. Fines are not. Record percent gravel, percent sand, and percent fines on the field form. Field-determined grain size estimation is categorized as follows: gravel (>2 mm), sand (2-16 mm), and fines (silt/clay/muck).

Labeling, storage, and shipping

For all samples, label each jar, place into polyethylene bags, and store in a small portable cooler of ice. Record sample information, including number of jars representing each sample on a field form. Figure C-4.1 provides an example form for *Chemistry and Sampling*. Use the appropriate column depending upon whether documenting the primary sample or a duplicate for the date.



Reviewed by (Initials): JS

Status and Trends - Chemistry and Sampling Form

Site Number | YY | MMDD | HH : MM

DCE: WAM0600_0000001_DCE_2009_0701_13:25

IN SITU WATER QUALITY CALIBRATION

Operator: Kurt Gowdy | Unit #: 1

				Flag
T	Temp probe was checked vs NIST	(Yes)	No	F1
DO	Sensor Calibrated	(Yes)	No	F2
pH	Sensor Calibrated and Checked	(Yes)	No	F3
Cond	Sensor Calibrated and Checked	(Yes)	No	F3

Notes (in situ)
F1 - T - Checked pre-season
F3 - pH, Cond, calibrated and checked this morning at the lab.
F2 - DO calibrated streamside - Winkler comparison collected for July

Sed:%Gravel: 0 | %Sand: 50 | %Fines: 50

In Situ Chemistry

Time1: 13:45 hrs | Start Location (e.g. F0): F0
Temp1: 6.3 deg C
pH1: 6.94 pH Units
DO1: 10.9 mg/L | %Sat1: 102.5
Cond: 27.8 uS/cm @ 25C

Time2: 19:50 hrs | End Location (e.g. K0): F0
Temp2: 7.0 deg C
pH2: 7.00 pH Units
DO2: 10.4 mg/L | %Sat2: 99.5
Cond: 27.9 uS/cm @ 25C

Sample	Primary Sample: No. of Jars	Duplicate Sample: No. of Jars (or ITIS for Fish Spp)	Destination	Tracking No. (if shipped)	Flag
TPN	1	0	MEL		
Tot P	1	0	MEL		
Cl	1	0	MEL		
Turb	1	0	MEL		
Sed PAH	1	0	MEL		
Sed Metals*	1	0	MEL		
Benthos	2	0	ETOH Shed at office		F4
Fish Spp1	1	ITIS: 159700	University Lab	FedEx: 835651465756	F5
Fish Spp2	1	ITIS: 167234	office lab for review under microscope		F6
Fish Spp3					

Water Sample Location (e.g. A5): F0

Sample Notes (explain flags):
F4 - Invertebrate sample could not fit into a single jar. Two jars are taped together.
F5 - Lamprey ammocoetes in zipped bag on ice — Vert Collection Form "Jar" #1
F6 - Riffle sculpin in jar of ETOH - verify it is not a reticulate sculpin. — Vert Col. Frm Jar #2

Draft

*Sediment Metals jar includes sample material to be analyzed for TOC
Note: Use standard Manchester Environmental Lab forms for tracking water and sediment samples.

Figure C-4.1. The *Chemistry and Sampling Form*, with fields for sediment chemistry data highlighted.

If you are sampling close to your vehicle, immediately place samples in a cooler of ice. Otherwise bring a small cooler for samples to the field sampling location. Place samples into a cooler of ice as soon as possible.

Check the tag to ensure that the SITE_ID number is recorded. Also record in waterproof ink or pencil:

- Project name
- Data and time that appears in the DCE
- Field sampler names
- Laboratory number
- Parameters for analysis
- Other information needed

Label information will meet the contract laboratory needs.

Sample crews will complete laboratory analysis forms and chain-of-custody forms (if separate) and submit samples to a courier or directly to a laboratory.

C-5. Bank measurements at major transects in waded streams

Purpose and scope

This method explains how to collect measurements for Watershed Health monitoring at each of 11 equidistant transects at each site. Measurements in this procedure will be restricted to one main channel. Instruments included on the procedure include distance measuring devices (e.g., measuring rod, laser rangefinder, 50-m measuring tape), and hand-levels.

Personnel responsibilities

This method is performed by two people. This method is applied at every DCE, at each major transect. Staff performing this method must have been trained.

Equipment, reagents, supplies

- No. 2 pencil
- Measuring rod
- 50-m tape
- Laser rangefinder
- Hand level
- Clinometers
- Calculator

Summary of procedure

Refer to the *Major Transect Form* (Figures C-5.1 and C-5.2). At each of the major Transects (A0- K0), assess the main channel. Measure these channel characters: bankfull width, wetted width, bar width, bankfull height, and bank instability. Describe flags.

BANK		
		Flag
Wetted Width XXX.X m	3.2	
Bar Width XX.X m	0	
Bankfull Width XXX.X m	5.4	
R Bankfull Height cm	35	
L BankfullHeight cm	32	
LB Instability %	50	F1
RB Instability %	0	

Figure C-5.1. A portion of the Major Transect Form, with example data for this method.

Flag	Comments
F1	slumping bank with cow prints

Figure C-5.2. A portion of the Major Transect Form, with an example flag qualifier.

Channel dimensions

Bankfull stage

At the transect, visually estimate the bankfull stage. This is best done after considerable training, see Section C-2.Use this visual estimate to help understand where to measure bankfull width and bankfull height.

Bankfull width

After locating the bankfull stage at each bank, measure the bankfull width (Figure C-5.3) to the nearest tenth of a meter. Record this value on the *Major Transect Data Form* (Figure C-5.1). Width measurements can be made using either a 50-m tape, a measuring rod, or (if the channel is wide) with a laser rangefinder.

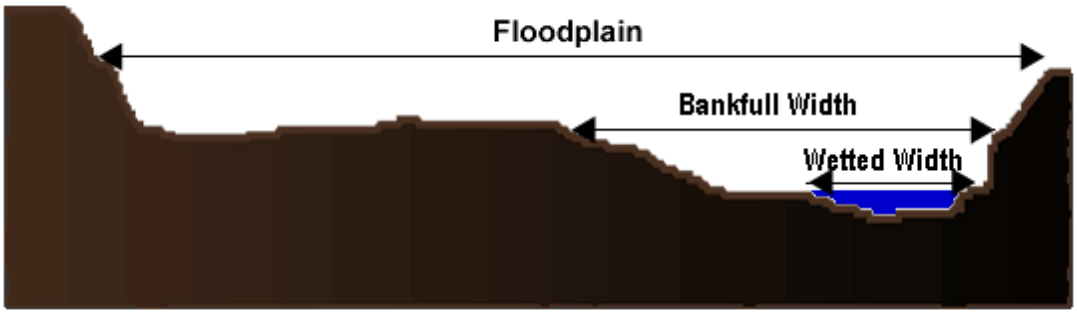


Figure C-5.3. Diagram of widths at the transect (Modified from Endreny 2009).

Wetted width

Observe the wetted margins of the channel. On the *Major Transect Data Form* (Figure C-5.1), record the wetted width (or horizontal distance between these margins) to the nearest tenth of a meter. Do *not* subtract for bars.

Bar width

Using the measuring rod, measure the width of each bar within the wetted channel. Record the sum (nearest tenth of a meter) for bar width.

Bankfull height

Bankfull height is measured using a surveyor's rod with hand level or clinometer. On the *Major Transect Form* (Figure C-5.1), record bankfull height data in whole centimeters. Record the right bankfull height and left bankfull height (Figure C-5.4).

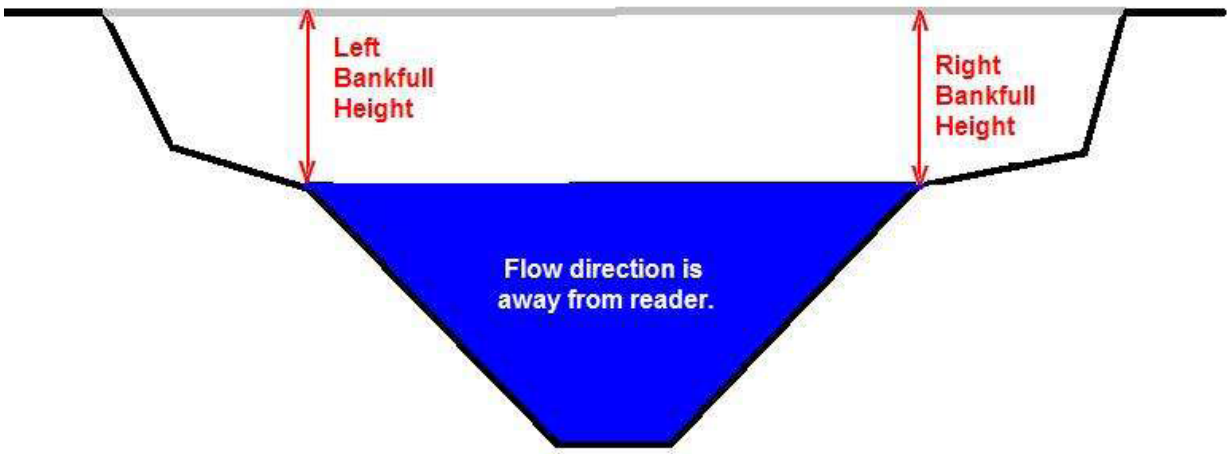


Figure C-5.4. Diagram of the left and right bankfull height measurements.

Bank instability

For waded streams, evaluate how much of a 10-m length of each bank (centered on the primary transect) is unstable. Limit your observations of bank stability to the portion of the bank at and below the bankfull stage. A bank is unstable if it has eroding or collapsing banks. It may have the following characteristics:

- Sparse vegetation on a steep surface
- Tension cracks
- Sloughing

On the *Major Transect Form* (Figure C-5.1), record right bank instability (%) and left bank instability (%).

C-6. Substrate and depth measurements at major transects in waded streams

Purpose and scope

This method explains how to measure substrate characteristics for Watershed Health monitoring at each of 11 equidistant transects at each site. Measurements in this procedure will be restricted to one main channel. This method must be preceded by the Major Transects Method. Instruments included on the procedure include distance measuring devices (e.g., measuring rod, or 50-m measuring tape, caliper), leveling device (hand level or clinometer) and a10-cm PVC ring.

Personnel responsibilities

This method is performed by two people. This method is applied at every DCE, at each major transect. Staff performing this method must have been trained.

Equipment, reagents, supplies

- No. 2 pencil
- measuring rod
- 50-m tape
- PVC ring
- hand-level
- Clinometers
- Calculator

Summary of procedure

Refer to the *Major Transect Data Form* (Figure C-6.1). At each of the major Transects (A0-K0), assess the main channel (channel number 0). Record these characters at each of 11 equidistant stations across the bankfull width:

- Wetted depth
- Bankfull depth
- Substrate type code
- Embeddedness

Station location

Identify the position along the transect. Example stations along a transect would be:

1. **left bank** – at the left bankfull stage.
2. **.1** – 10% distance across the channel.
3. **.2** – 20% distance across the channel.
4. **.3** – 30% distance across the channel.
5. **.4** – 40% distance across the channel.
6. **.5** – halfway across the channel.
7. **.6** – 60% distance across the channel.
8. **.7** – 70% distance across the channel.
9. **.8** – 80% distance across the channel.
10. **.9** – 90% distance across the channel.
11. **right bank** – at the right bankfull stage.

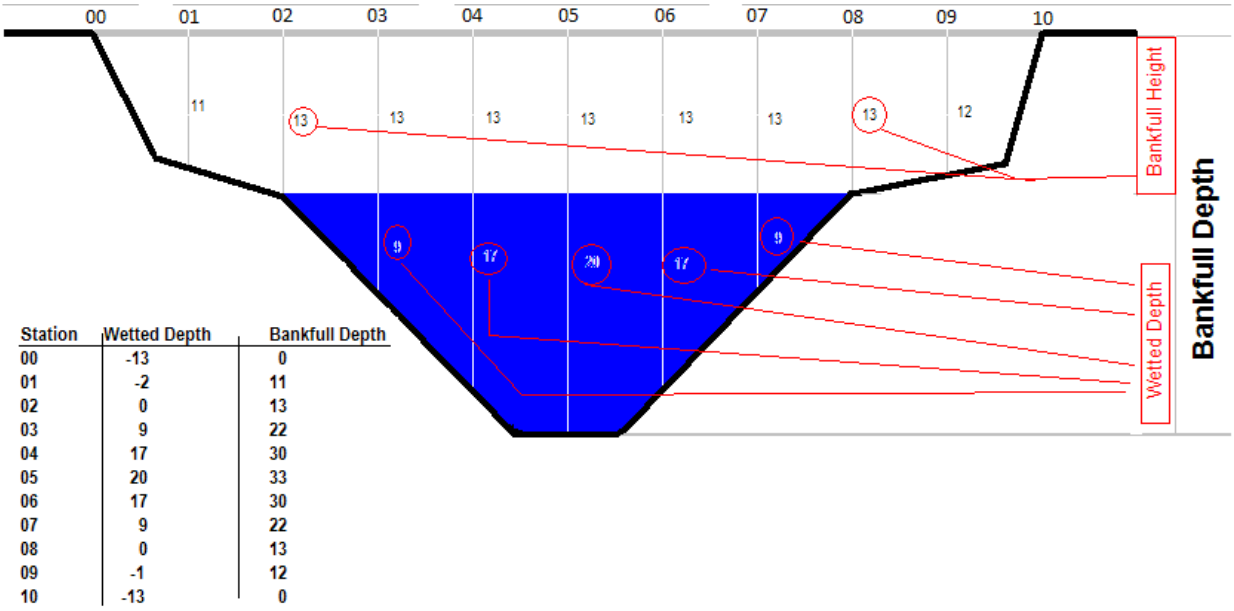
On the Major Transect Form (Figure C-6.1), insert data for depths, substrate type and embeddedness next to each station code. Describe flags (Figure C-6.2). Examples of data can be found in Figures C-6.1, C-6.2, and C-6.3.

SUBSTRATE					
	Wet Depth	BF Depth XXX CM	Size Class	Embd. 0-100%	Flag
left bank	-13	0	SA	100	
.1	-2	11	GF	90	
.2	0	13	GC	50	
.3	9	22	CB	25	
.4	17	30	SB	5	
.5	20	33	CB	25	
.6	17	30	CB	10	
.7	9	22	GC	10	
.8	0	13	WD	90	F1
.9	-1	12	FN	100	
right bank	-13	0	SA	100	

Figure C-6.1. Part of the Major Transect Form with example data for this method.

Flag	Comments
F1	WD = partially buried Douglas fir log, about 60 cm diameter

Figure C-6.2. Part of the Major Transect Form with example flag descriptions.



Station	Wetted Depth	Bankfull Depth
00	-13	0
01	-2	11
02	0	13
03	9	22
04	17	30
05	20	33
06	17	30
07	9	22
08	0	13
09	-1	12
10	-13	0

Figure C-6.3. Transect diagram showing example data for wetted depth, bankfull depth, and bankfull height. The bankfull depth equals the wetted depth plus average bankfull height.

Station depth

For each station, record depth in whole centimeters. This should be the easiest to measure of either wetted depth or bankfull depth. The bankfull depth equals the wetted depth plus average bankfull height. Therefore, if you know one type of depth and the mean bankfull height, you also know the other type of depth.

Substrate type

After recording depth, estimate the substrate particle type at the front of the measuring rod, where it rests on the surface of the streambed. Estimate the size class of that particle based on the intermediate axis length. Record the substrate type code. The choices are listed in Table C-6.1. For fine gravel, coarse gravel, and cobble, use calipers to measure the intermediate axis length of the particle and confirm your estimate of size. For larger sizes, use the measuring rod to confirm your estimate.

Particles smaller than 100 mm are evaluated using a 10-cm ring surrounding the sample point. All particles within the ring are evaluated for size and embeddedness, not just the point. Record the estimated average for surface substrate within the ring.

Table C-6.1. Substrate codes, types, and sizes.

Code	Type	Range Size	Size Gauge
RS	Bedrock (smooth)	>4 m	larger than a car
RR	Bedrock (rough)	>4 m	larger than a car
RC	Concrete/Asphalt	>4 m	larger than a car
XB	Large boulder	1-4 m	meter stick to car
SB	Small boulder	>250 mm–1 m	basketball to meter stick
CB	Cobble	>64 mm–250 mm	tennis ball to basketball
GC	Gravel, coarse	>16 mm to 64 mm	marble to tennis ball
GF	Gravel, fine	>2 mm to 16 mm	ladybug to marble
SA	Sand(2-16 mm)	>0.06 mm to 2 mm	gritty to ladybug
FN	Fines(silt/clay/muck)	<0.06 mm	non gritty
HP	Hardpan- hardened fines	any size	
WD	Wood	any size	
OT	Other (doesn't fit choices above)	any size	

Embeddedness

At each station, touch the nearest particle to foot of the measuring rod then look at it. Estimate embeddedness (%). This is the fraction of a particle's surface that is surrounded by (embedded in) sand or finer sediments (≤ 2 mm). By default, sand or fines are 100% embedded. By default, bedrock is 0% embedded.

Particles smaller than 100 mm are evaluated using a 10-cm ring surrounding the sample point. All particles within the ring are evaluated for size and embeddedness, not just the point. Record the estimated average for surface substrate within the ring.

C-7. Shade measurements at major transects in waded streams

Purpose and scope

This method explains how to measure shade for Watershed Health monitoring at each of 11 equidistant transects at each site. Measurements in this procedure will be restricted to one main channel. This method must be preceded by the Major Transects Method. Instruments included on the procedure include a distance measuring device (e.g., measuring rod) and a convex densiometer (modified according to Mulvey, et al., 1992).

Personnel responsibilities

This method is performed by one person. This method is applied at every DCE, at each major transect. Staff performing this method must have been trained.

Equipment, reagents, supplies

- No. 2 pencil
- *Major Transect Form*
- Measuring rod or 50-m tape
- Modified convex densitometer

Summary of procedure

Refer to the *Major Transect Form* (Figure C-7.1). At each of the major Transects (A0-K0), assess the main channel (channel number 0). Use a convex densiometer (Lemmon, 1957) that has been modified according to Mulvey, et al. (1992). It has 17 intersections. See Figure C-7.2.

DENSIOMETER MEASUREMENTS

(0-17Max)

		Flag			Flag
CenUp	5		CenR	9	
CenL	0		Left	0	
CenDwn	4		Right	17	

Figure C-7.1. Densiometer portion of The Major Transects Form, with example data.

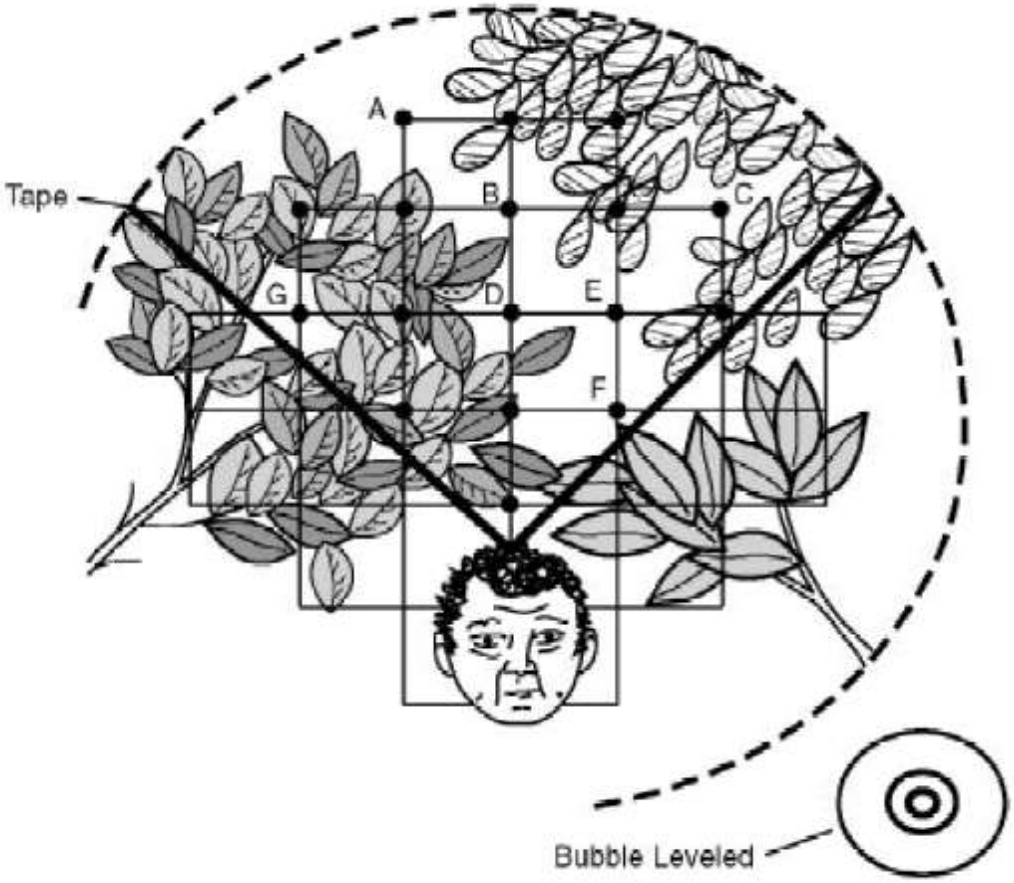


Figure C-7.2. An example reading from a modified convex densiometer. It shows 10 of 17 intersections with shade (a score of "10").

Note the proper positions of the bubble and head reflection (from Mulvey, et al., 1992).

Record how many of the 17 cross-hairs have shade over them. Record for each of six directions on the major transect (Figure C-7.3):

- Facing the left bankfull stage.
- Facing the right bankfull stage.
- Bankfull channel center, facing upstream.
- Bankfull channel center, facing right.
- Bankfull channel center, facing downstream.
- Bankfull channel center, facing left.

At each wetted station, hold the densiometer 30 cm above the water. At each dry station, hold the densiometer 30 cm above the ground. Bank readings should be able to detect shade from riparian understory vegetation such as ferns.

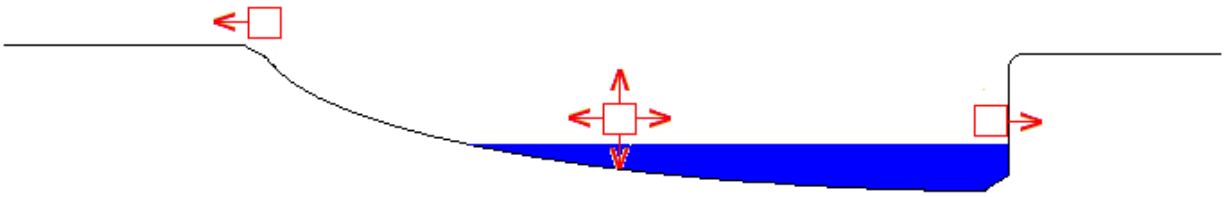


Figure C-7.3. Stations for densiometer measurement on each major transect. The densiometer is held level, and 30 cm above water for wet stations and 30 cm above ground for dry stations.

C-8. Estimating fish cover at major transects in waded streams

Purpose and scope

This method explains how to estimate fish cover for Watershed Health monitoring at each of 11 equidistant transects at each site. Measurements in this procedure will be restricted to one main channel. This method must be preceded by the Major Transects Method. Instruments included on the procedure include a distance-measuring device (e.g., measuring rod).

Personnel responsibilities

This method is performed by one person. This method is applied at every DCE, at each major transect. Staff performing this method must have been trained.

Equipment, reagents, supplies

- No. 2 pencil
- *Major Transect Form*
- Measuring rod or 50-m tape

Summary of procedure

This method is derived from that of Peck, et al. (2006). Within the main channel, evaluate 11 plots (Figure C-8.1) with these characteristics:

- Centered at each major transect.
- Extends 5 meters upstream of each transect.
- Extends 5 meters downstream of each transect.
- Beneath the wetted surface.
- Visually assess the percentage of the water surface that has fish cover provided by each of 10 cover types.

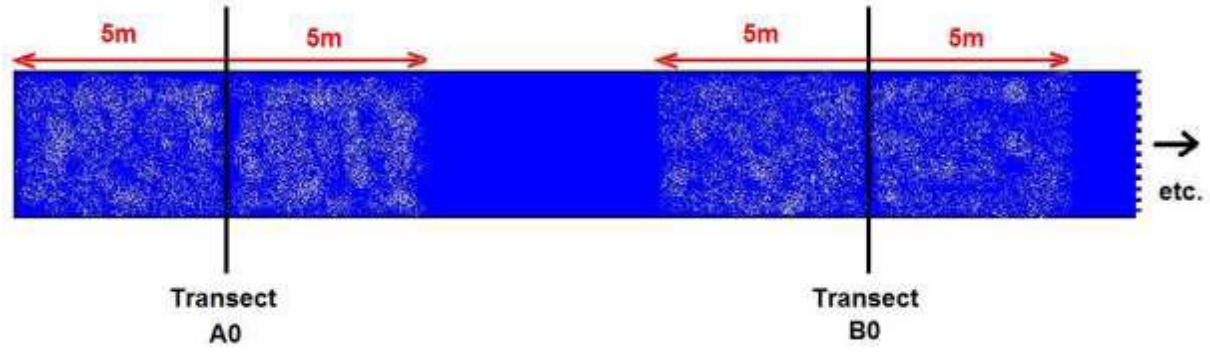


Figure C-8.1. Diagram of fish cover plots at each major transect of the main channel.

Refer to the *Major Transect Form* (Figure C-8.2). Circle the cover code that best characterizes each cover type.

FISH COVER	0 = Absent (0%) 1 = Sparse (<10%) 2 = Moderate (10-40%) 3 = Heavy (40-75%) 4 = Very Heavy (>75%) (circle one)	
	Cover in Channel	Flag
Filamentous Algae	**(0)** 1 2 3 4	
Macrophytes	**(0)** 1 2 3 4	
Woody Debris	0 **(1)** 2 3 4	
Brush	0 1 **(2)** 3 4	
Live Trees or Roots	**(0)** 1 2 3 4	
Overhanging Veg. =<1 m of Surface	0 1 2 **(3)** 4	
Undercut Banks	0 **(1)** 2 3 4	
Boulders	**(0)** 1 2 3 4	
Artificial Structures	**(0)** 1 2 3 4	
Bryophytes	**(0)** 1 2 3 4	

Figure C-8.2. Fish Cover portion of The Major Transects Form, with example records.

C-9. Human influence at major transects in waded streams

Purpose and scope

This method explains how to collect measurements for Watershed Health monitoring at each of 11 equidistant transects at each site. Measurements in this procedure will be restricted to one main channel. This method must follow the method for establishing major transects.

Personnel responsibilities

This method is performed by one person. This method is applied at every DCE, at each major transect. Observations are made at each bank of the main channel. Staff performing this method must have been trained.

Equipment, reagents, supplies

- No. 2 pencil
- *Major Transect Data Form*
- Measuring device (rod, tape, rangefinder)

Summary of procedure

This procedure is derived from Peck, et al. (2006) and Moberg (2007). Refer to the *Major Transect Data Form* (Figures C-9.1 and C-9.2). At each of the major Transects (A0-K0), assess the main channel. Record the appropriate *influence proximity code* for each of 13 human *influence types* (Figure C-9.1) relative to riparian plots (Figure C-9.3) on each bank of the transect. Influence proximity codes are:

- 0 = absent.
- 1 = beyond the plot, but within 30 meters of the bankfull margin.
- 2 = within the 10 meter by 10 m riparian plot.
- 3 = at least partially within the bankfull channel.

HUMAN INFLUENCE	0=not present, 1= 10-30m, 2= 0-10m, 3= on bank		
	Left Bank	Right Bank	Flag
Wall/Dike/Revetment/ Riprap/Dam	(0) 1 2 3	(0) 1 2 3	
Buildings	(0) 1 2 3	0 1 (2) 3	F1
Unpaved Motor Trail	0 (1) 2 3	(0) 1 2 3	
Clearing or Lot	(0) 1 2 3	(0) 1 2 3	
Human Foot Path	(0) 1 2 3	(0) 1 2 3	
PavedRoad/Railroad	0 (1) 2 3	(0) 1 2 3	
Pipes (Inlet/Outlet)	(0) 1 2 3	0 1 2 (3)	F2
Landfill/Trash	0 (1) 2 3	0 1 2 (3)	F3
Park/Lawn	0 1 (2) 3	(0) 1 2 3	
Row Crops	(0) 1 2 3	(0) 1 2 3	
Pasture/Range/Hay Field	(0) 1 2 3	(0) 1 2 3	
Logging Operations	(0) 1 2 3	(0) 1 2 3	
Mining Activity	(0) 1 2 3	(0) 1 2 3	

Figure C-9.1. A portion of the Major Transect Form, with example data.

Flag	Comments
F1	single-family home
F2	possible irrigation source
F3	beer cans

Figure C-9.2. A portion of the Major Transect Form with example comments for data flags.

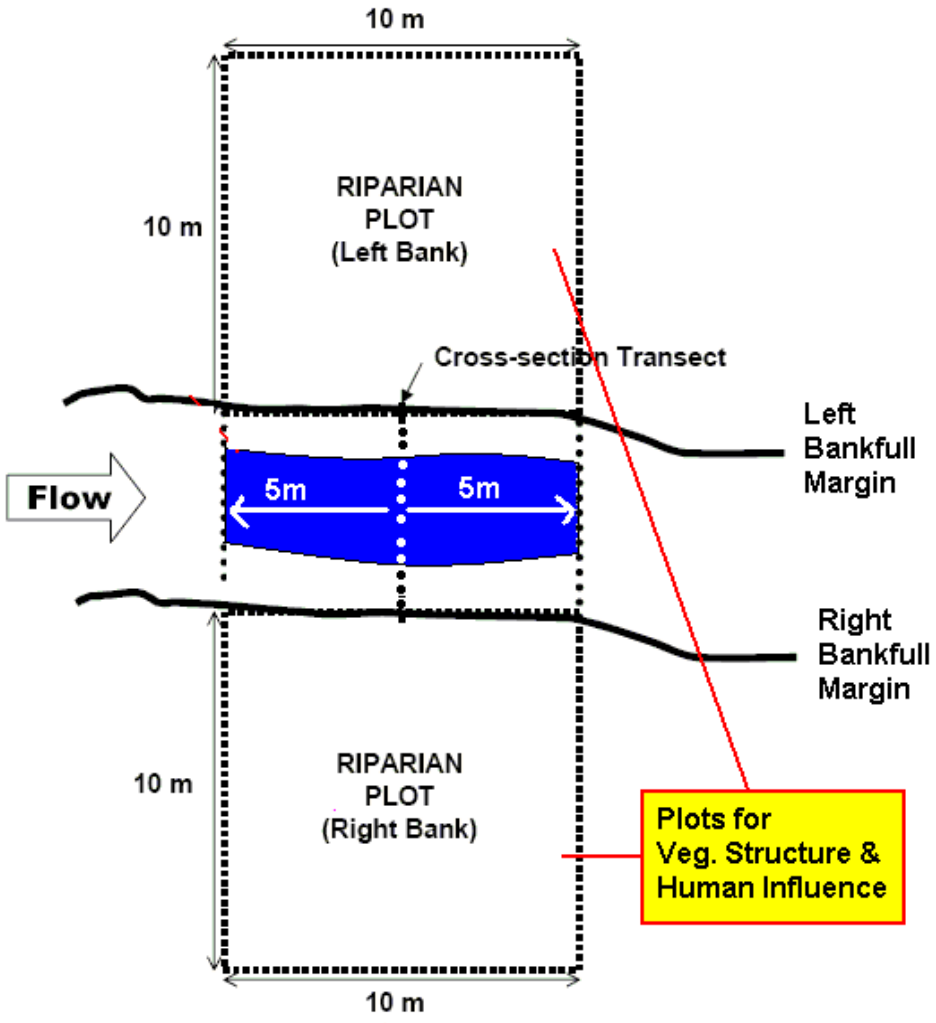


Figure C-9.3. Riparian plots.

C-10. Riparian vegetation structure at major transects in waded streams

Purpose and scope

This method explains how to collect measurements for Watershed Health monitoring at each of 11 equidistant transects at each site. Observations in this procedure will be restricted to one main channel. This method must follow the method for establishing major transects.

Personnel responsibilities

This method is performed by one person. This method is applied at every DCE, at each major transect. Observations are made at each bank of the main channel. Staff performing this method must have been trained.

Equipment, reagents, supplies

- No. 2 pencil
- *Major Transect Data Form*

Summary of procedure

This procedure is derived from Peck, et al. (2006) and Moberg (2007). Refer to the *Major Transect Data Form* (Figure C-10.1).

RIPARIAN

0 = Absent (0%)
1 = Sparse (<10%)
2 = Moderate (10-40%)
3 = Heavy (40-75%)
4 = Very Heavy (>75%)

D = Deciduous
C = Coniferous
E = Broadleaf Evergreen
M = Mixed
N = None

RIPARIAN VEGETATION COVER	Left Bank	Right Bank	Flag
Canopy (>5 m high)			
Woody Vegetation Type	D C E **(M)** N	D **(C)** E M N	
BIG Trees (Trunk >0.3 m DBH)	0 **(1)** 2 3 4	0 1 2 **(3)** 4	
SMALL Trees (Trunk <0.3 m DBH)	0 1 2 **(3)** 4	0 **(1)** 2 3 4	
Understory (0.5 to 5 m high)			
Woody Vegetation Type	D C E **(M)** N	D C E **(M)** N	
Woody Shrubs & Saplings	0 1 2 **(3)** 4	0 1 **(2)** 3 4	
Non-Woody Herbs, Grasses, & Forbs	0 **(1)** 2 3 4	0 **(1)** 2 3 4	
Ground Cover (<0.5 m high)			
Woody Shrubs & Saplings	0 1 2 **(3)** 4	0 1 **(2)** 3 4	
Non-Woody Herbs, Grasses and Forbs	0 1 **(2)** 3 4	0 1 **(2)** 3 4	
Barren, Bare Dirt or Duff	0 **(1)** 2 3 4	0 1 **(2)** 3 4	

Figure C-10.1. A portion of the Major Transect Data Form, with example data.

On each major transect of the main channel, assess a plot on each bank. Each plot extends 5 meters downstream, 5 meters upstream, and 10 meters back from the bankfull margin (Figure C-10.2). The riparian plot dimensions can be estimated rather than measured. On steeply sloping channel margins, plot boundaries are defined as if they were projected down from an aerial view.

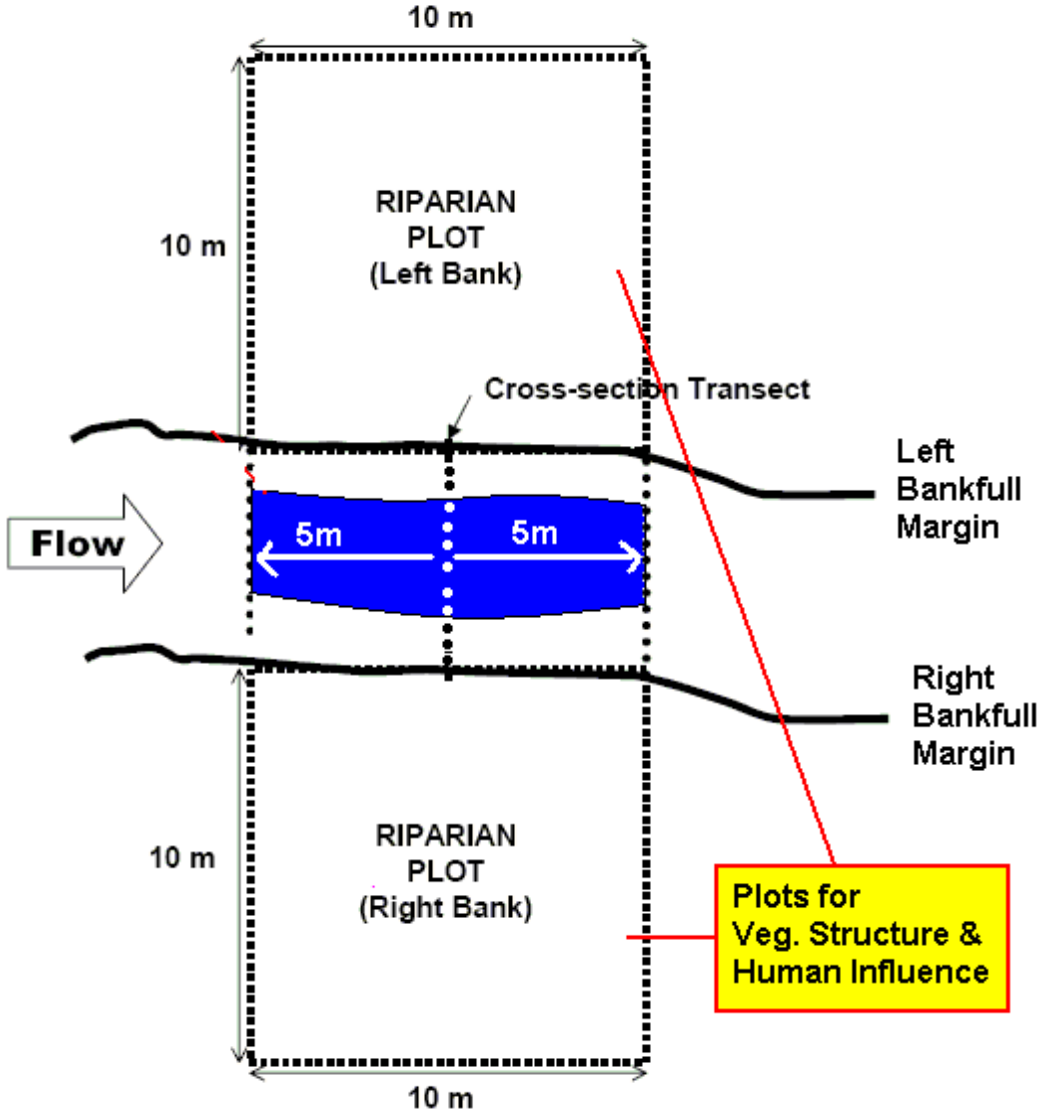


Figure C-10.2. Riparian plots.

Conceptually divide the riparian vegetation into three layers:

- Canopy (> 5 m high).
- Understory (0.5 to 5 m high).
- Ground Cover layer (< 0.5 m high).

Within each layer, consider the type of vegetation present and the amount of cover provided. Do this independently of what is contained in higher layers. Cover quantity is coded on the field form (Figure C-10.1) as follows:

- 0 - absent.
- sparse (< 10% cover).
- 2 - moderate (10-40% cover).
- 3 - heavy (40-75% cover).
- 4 - very heavy (> 75% cover).

The maximum cover in each layer is 100%, so the sum of the cover for the combined three layers could add up to 300%.

Canopy

On the *Major Transect Form* (Figure C-10.1), circle the appropriate vegetation type code (D, C, E, M, or N). Type codes are defined on the form. Then circle the appropriate cover quantity code (0, 1, 2, 3, or 4) for each of 2 classes:

- Big trees - trees having trunks larger than 0.3 m diameter (at breast height).
- Small trees - trees having trunks smaller than 0.3 m diameter (at breast height).

Understory

On the *Major Transect Form* (Figure C-10.1), circle the appropriate vegetation type code (D, C, E, M, or N) for any *woody* vegetation that might be present. Then circle the appropriate cover quantity code (0, 1, 2, 3, or 4) for each of 2 classes:

- Woody vegetation - such as shrubs or saplings.
- Non-woody vegetation - such as herbs, grasses, or forbs.

Ground cover

Circle the appropriate cover quantity code (0, 1, 2, 3, or 4) for each of 3 classes:

- Woody (living).
- Non-woody (living).
- Bare dirt (or decomposing debris).

The sum of cover quantity ranges for these 3 types of ground cover should include 100%.

C-11. Measuring thalweg depth in waded streams

Purpose and scope

This method explains how to collect incremental depth measurements for Watershed Health monitoring when traversing the length of the stream site. It also describes assessing the presence of bars and edge pools. Observations in this method will be restricted to the main channel.

Personnel responsibilities

This method is performed by two people: one person measures and another person records. This method is limited to the main channel. It must be preceded by the method for verification and site layout. Staff performing this method must have been trained.

Equipment, reagents, supplies

- No. 2 pencil
- *Thalweg Data Form*
- Measuring rod

Summary of procedure

This procedure is derived from Peck, et al. (2006) and Moberg (2007). Refer to the *Thalweg Data Form* (Figure C-11.1).

Transect A	Thalweg Depth (cm)	Bar? (circle)	Edge Pool? (circle)
.0	69	Y (N)	(Y) N
.1	70	Y (N)	Y N
.2	75	Y (N)	Y N
.3	87	Y (N)	Y N
.4	70	Y (N)	Y N
.5	75	Y (N)	Y N
.6	33	Y (N)	Y N
.7	34	Y (N)	Y N
.8	32	(Y) N	Y N
.9	33	Y (N)	(Y) N

Figure C-11.1. A portion of the Thalweg Data Form, with example data.

While walking up the main channel, measure thalweg depth (cm) at each of 101 thalweg transects. To reference location:

- Record the letter code for the lowest major transect referenced (e.g., A).
- Record depth and occurrence data into the appropriate thalweg transect row (e.g., .0).

These thalweg stations are located 0.2 bankfull widths apart from each other; bankfull width is based on an estimate made during the site layout. While measuring thalweg depth, also evaluate whether each of these features is present at each thalweg transect:

- Bar.
- Edge pool.
- Circle “Y” for “yes” and “N” for “no”.

C-12. Large woody debris tally for waded streams of western Washington

Purpose and scope

This method explains how to count pieces of large woody debris in waded streams for Watershed Health monitoring when traversing the length of the stream site. Observations are limited to the main channel. This method applies to streams of western Washington (west of the Cascade ridge), where natural conditions are expected to include larger sizes of wood.

Personnel responsibilities

This method is performed by one person. This method is applied at every DCE. Observations are made while walking upstream in the main channel. Staff performing this method must have been trained.

Equipment, reagents, supplies

- No. 2 pencil
- *Thalweg Data Form*
- Measuring rod
- Calipers

Summary of procedure

This procedure is derived from Peck, et al. (2006) and Moberg (2007). One person, while walking upstream, counts the number of pieces of large woody debris (LWD), that are (at least partially) within the bankfull channel of each stream segment (e.g., A0 to B0) in the main channel. Pieces are tallied according to size classes (Table C-12.1), which differ by region (Table C-12.2).

Table C-12.1. Size classes for large woody debris.

Large Woody Debris measured in each thalweg			
Length (meters)			
West	>2-5 m	>5-15 m	>15 m
10-30 dia (cm)			
30-60 dia (cm)			
60-80 dia (cm)			
>80 dia (cm)			
Central & East	>1-3 m	>3-6 m	>6 m
10-15 dia (cm)			
15-30 dia (cm)			
30-60 dia (cm)			
>60 dia (cm)			

Table C-12.2. Washington State regions used to determine which size class of woody debris to use during bioassessment.

Region	LWD Size Class Used
Puget Sound	West
Coastal	West
Lower Columbia	West
Mid Columbia	East
Upper Columbia	East
Snake	East
Northeast Washington	East
Unlisted Washington	East

LWD: Large, woody debris

Considering taper

Wood pieces have a taper. Considerations for taper are illustrated in Figure C-12.1. The diameter of a log is based on the thickest end. The length of a log only counts the portion that has a diameter of more than 10 cm.

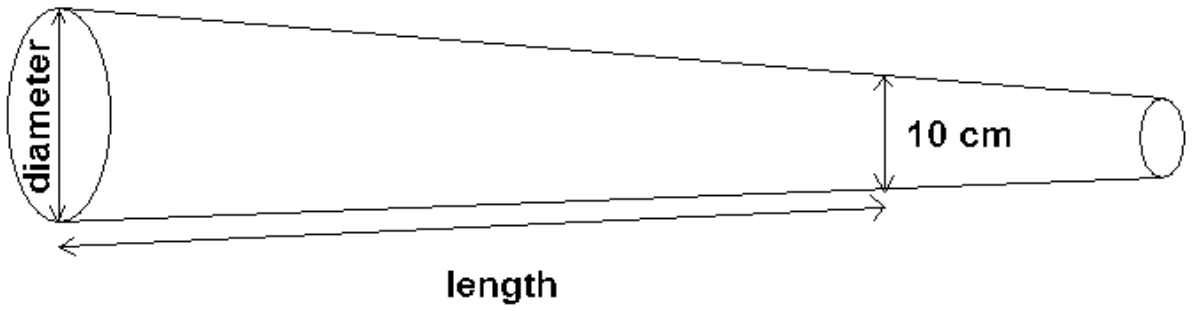
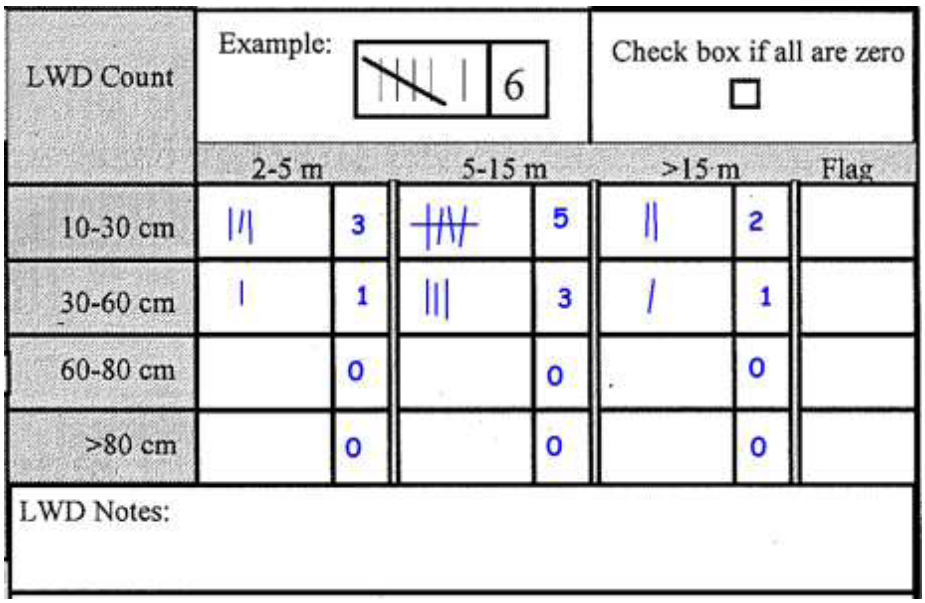


Figure C-12.1. Diagram of how to estimate the dimensions of a log.

Record

Refer to the *Thalweg Data Form* (Figure C-12.2). Identify and tally large, woody debris (LWD) pieces that lie in the bankfull channel. After tallying, sum the marks separately for each size class and enter the number into the corresponding box for each class.



LWD Count	Example: 6		Check box if all are zero ☐			
	2-5 m		5-15 m		>15 m	Flag
10-30 cm	𝍸	3	𝍸	5	‖	2	
30-60 cm	|	1	‖|	3	/	1	
60-80 cm		0		0		0	
>80 cm		0		0		0	
LWD Notes:							

Figure C-12.2. A portion of the Thalweg Data Form, with example data.

C-13. Habitat unit descriptions along the main channel thalweg

Purpose and scope

This method explains how to identify and count habitat units for Watershed Health monitoring when traversing the length of the stream site. The habitat unit descriptions are based on the Hawkins, et al. (1993) classification system (Figure C-13.1). Observations in this method will be restricted to the main channel.

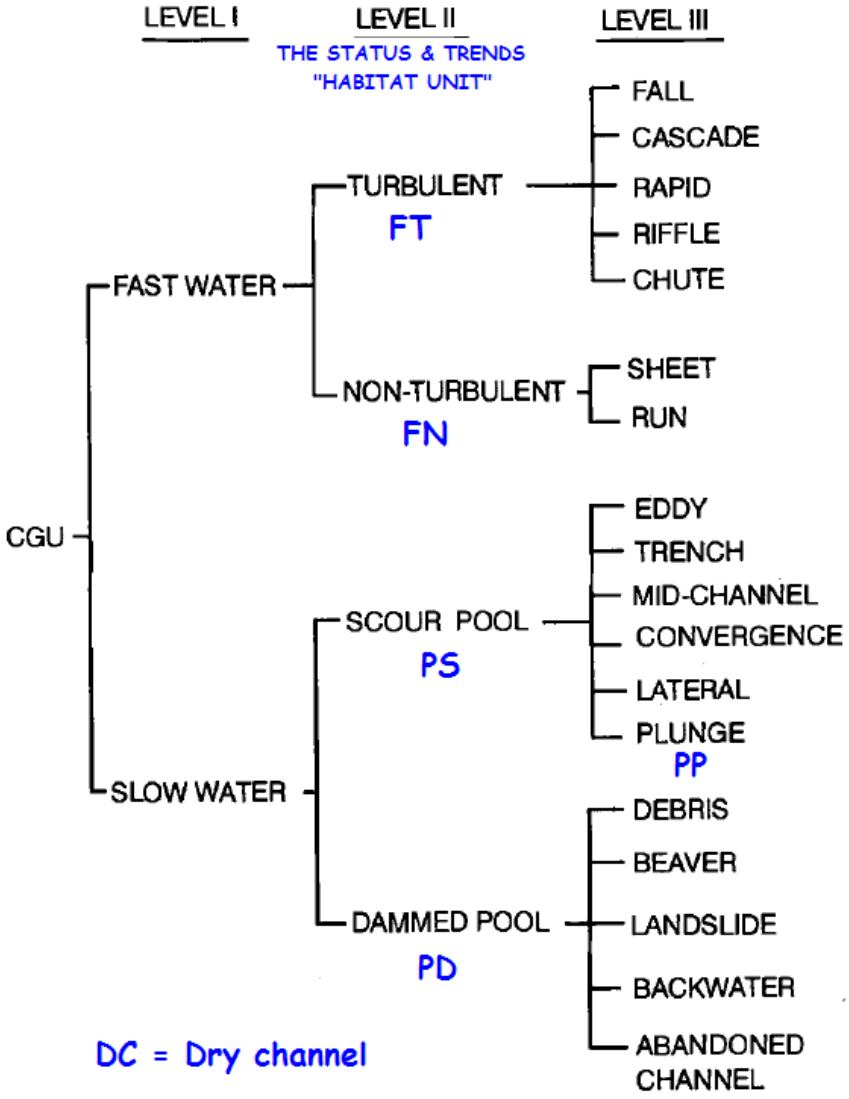


Figure C-13.1. Categories of channel geomorphic units (CGU) described by Hawkins, et al. (1993) and their three levels of resolution.

This figure is modified from Hawkins, et al. (1993), with status and trends habitat unit codes displayed in blue text.

Personnel responsibilities

This method is performed by one person and who dictates data to a second person who records. This method is applied at every DCE. Observations are made while walking upstream in the thalweg of the main channel. Staff performing this method must have been trained.

Equipment, reagents, supplies

- No. 2 pencil
- *Thalweg Data Form*
- Measuring rod
- 50-m tape or laser rangefinder

Summary of procedure

This procedure is derived from Moberg (2007). Refer to the *Thalweg Data Form* (Figures C-13.2 and C-13.3). Identify and code habitat units consecutively during the walk upstream. A separate Thalweg Data Form is recorded for sets of observations that span between major transects. Data will include:

- Type code
- Unit identity (number)
- Pool forming code
- Depths (for pools)

Habitat Unit Number	Habitat Unit Type FT, FN, PS, PD, PP,DC	Pool Forming Code (N, W, R, B, F)		HU Width (m.x)	Max Pool Depth (cm)	Crest Pool Depth (cm)	Channel Unit Notes:
		Code 1	Code 2				
1	PD	W	B	3.5	90	30	Pool formed by both boulder & wood
2	PP	W		4.2	75	15	
3	FN	N		4.8			

Figure C-13.2. A portion of the Thalweg Data Form, with example data for habitat unit type, pool forming code, habitat unit width, and pool depths.

Transect A	Thalweg Depth (cm)	Bar? (circle)	Edge Pool? (circle)	Habitat Unit Number
.0		Y N	Y N	1
.1		Y N	Y N	1
.2		Y N	Y N	1
.3		Y N	Y N	1
.4		Y N	Y N	2
.5		Y N	Y N	2
.6		Y N	Y N	3
.7		Y N	Y N	3
.8		Y N	Y N	3
.9		Y N	Y N	3

Figure C-13.3 A portion of the Thalweg Data Form, with example data for habitat unit locations relative to thalweg transects.

Type code

With each step up the thalweg, evaluate the wetted channel for conformity to the Hawkins, et al. (1993) classification system (Figure C-13.1). We are focusing on Level II designations. The main division is between slow water (pools) and fast water (e.g., cascades, riffles, or runs). All habitat units (except plunge pools or dry channels) must be at least as long as half the wetted width. All pools have specific depth criteria: the maximum depth must be at least 1.5 times the depth at the pool crest. Record the unit type code (Table C-13.1) on the *Thalweg Data Form*.

Table C-13.1. Habitat unit type codes.

Unit	Type	Description
FT	Fast Turbulent	(riffle, cascade, waterfall)
FN	Fast Non-Turbulent	(sheet, run)
PS	Scour	pool
PD	Dammed	pool
PP	Plunge	pool
DC	Dry	channel

Unit number

After you designate the habitat unit type (Table C-13.1), assign a habitat unit number. These are consecutive number counts for the whole stream site. For each form, record data for any new habitat units that appear since the last encountered major transect. For example, if habitat units numbered 1, 2, and 3 were recorded between major transects A and B, then new units encountered between B and C would begin with habitat unit number 4.

Pool forming code

On the *Thalweg Data Form* (Figure C-13.2), record the pool forming code (Table C-13.2) to describe the obstruction that led to pool formation. Assign "N" for habitat units other than pools. If pool formation could be associated with two types (e.g., boulder *and* large wood), use both columns on the form, with one code per column.

Table C-13.2. Pool forming codes.

Pool Forming Code	Description
N	Not a pool
W	Large Woody Debris
R	Root wad
B	Boulder/Bedrock
F	Fluvial (non-specific stream process)

Habitat unit width

Estimate the average wetted width (nearest tenth of a meter) of the habitat unit for the full course of its length. Record this value on the *Thalweg Data Form* (Figure C-13.2) A measurement is not required. Just consider the relative width compared to the width measurements performed at nearby major transects and minor transects.

Pool depths

With a measuring rod, measure water depth (cm) in each of two locations in the thalweg of pools:

- at the crest.
- at maximum depth.

Crest depth is measured differently, depending upon the pool type. For scour pools and plunge pools, the crest depth is measured where water exits the pool. For dammed pools, the crest depth is measured where water enters the pool. Record crest depth and maximum depth on the *Thalweg Data Form* (Figure C-13.2). No data needs to be recorded for non-pool habitat units.

Position

After identifying and describing habitat units (Figure C-13.2), record the position of each habitat unit relative to thalweg stations (Figure C-13.3).

C-14. Side-channel descriptions

Purpose and scope

This method explains how to identify and count side-channels of waded streams for Watershed Health monitoring when traversing the length of the stream site. Observations are limited to portions of side channels that occur next to the sampled part of the main channel (above Transect A0 and below Transect K0).

Personnel responsibilities

This method is performed by one person who dictates to another. This method is applied at every DCE. Observations are made while walking upstream to measure thalweg depths of the main channel. Staff performing this method must have been trained.

Equipment, reagents, supplies

- No. 2 pencil
- *Thalweg Data Form*
- Measuring rod
- Field notebook

Summary of procedure

This procedure is derived from Moberg (2007). Refer to the *Thalweg Data Form* (Figures C-14.1 and C-14.2). Identify and count side channels occurring within the length of the sample site. Estimate their widths.

Identify and count

Identify and code side channels consecutively for the entire streams site. Number them as encountered while walking upstream. Note their presence for each of the 101 Thalweg Transects of the stream site. This will require 11 *Thalweg Data Forms* to complete (A-K).

Transect A	Thalweg Depth (cm)	Bar? (circle)	Edge Pool? (circle)	Habitat Unit Number	Side Channel Numbers				
.0		Y N	Y N						
.1		Y N	Y N		1				
.2		Y N	Y N		1				
.3		Y N	Y N		1				
.4		Y N	Y N		1	2			
.5		Y N	Y N		1	2			
.6		Y N	Y N		1	2			
.7		Y N	Y N		1	2			
.8		Y N	Y N		1	2	3		
.9		Y N	Y N		1	2	3		

Figure C-14.1. A portion of the Thalweg Data Form, with example data showing the presence or absence of side-channels at each Thalweg Transect.

Estimate width

For each channel, estimate wetted width (nearest tenth of a meter). Make at least one representative measurement (in a notebook) between each major transect then visually estimate an average value for the length of the side-channel. Record this channel average on the *Thalweg Data Form* (Figure C-14.1). In your width estimate, do *not* include portions of the channel that occur below transect A0 or above transect K0.

Side Channel Number	Width (m.x)	Side Channel Notes:
1	1.0	left side of main channel
2	2.3	diverts from channel 1, not from main channel
3	3.7	Right side of main channel

Figure C-14.2. A portion of the Thalweg Data Form, with example data for channel width.

C-15. Width and substrate measurements at minor transects in waded streams

Purpose and scope

This method explains how to measure width and substrate characteristics for Watershed Health monitoring at each of 10 equidistant transects at each site. Measurements in this procedure will be restricted to one main channel. This method is performed in conjunction with the method for measuring thalweg depth. Instruments included on the procedure include distance-measuring devices (e.g., measuring rod, or 50-m measuring tape, caliper) and a10-cm ring.

Personnel responsibilities

This method is performed by two people: an observer and a recorder. This method is applied at each minor transect. It is performed in conjunction with the method for measuring thalweg depth. Staff performing this method must have been trained.

Equipment, reagents, supplies

- No. 2 pencil
- Measuring rod
- 50-m tape
- Calculator
- 10-cm ring

Summary of procedure

Measure the channel width and then make observations about substrate size at 11 equidistant stations across the minor transect.

Widths

At each minor transect, measure distance (tenth of meters) for:

- Bankfull width.
- Wetted width.
- Total bar width (sum for all bars).

Record these widths on the *Thalweg Data Form* (Figure C-15.1).

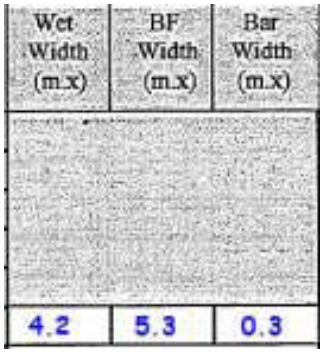


Figure C-15.1. Part of the Thalweg Data Form, with example data for widths at the minor transect.

Station location

Identify the ***Transect Station LeftRight***. Example stations for minor transect A5 would be:

12. **A500** – at the left bankfull stage.
13. **A501** – 10% distance across the channel.
14. **A502** – 20% distance across the channel.
15. **A503** – 30% distance across the channel.
16. **A504** – 40% distance across the channel.
17. **A505** – half way across the channel.
18. **A506** – 60% distance across the channel.
19. **A507** – 70% distance across the channel.
20. **A508** – 80% distance across the channel.
21. **A509** – 90% distance across the channel.
22. **A510** – at the right bankfull stage.

Substrate type

Hold the measuring rod vertically and rest it on the substrate at each station. Estimate the substrate particle type at the front of the measuring rod, where it rests on the surface of the streambed. Estimate the size class of that particle based on the intermediate axis length. Record the *substrate type code* (Table C-15.1) on the *Thalweg Data Form* (Figure C-15.2) for each station. For coarse gravel and cobble, use calipers to measure the intermediate axis length of the particle and confirm your estimate of size. For larger sizes, use the measuring rod to confirm your estimate. Particles smaller than 100 mm are evaluated using a 10-cm ring surrounding the sample point. All particles within the ring are evaluated for size and embeddedness, not just the point. Record the estimated average for surface substrate within the ring.

Substrates at 5	LB	01	02	03	04	05	06	07	08	09	RB	Substrate Notes:
	SA	GF	GC	CB	CB	XB	XB	XB	XB	XB	FN	Stations 5-9 are one boulder

Figure C-15.2. Part of the Thalweg Data Form, with example data for substrate types along the minor transect.

Table C-15.1. Substrate codes, types, and sizes.

Code	Type	Range Size	Size Gauge
RS	Bedrock (smooth)	>4 m	larger than a car
RR	Bedrock (rough)	>4 m	larger than a car
RC	Concrete/Asphalt	>4 m	larger than a car
XB	Large Boulder	1-4 m	meter stick to car
SB	Small boulder	>250 mm–1 m	basketball to meter stick
CB	Cobble	>64 mm–250 mm	tennis ball to basketball
GC	Gravel, coarse	>16 mm to 64 mm	marble to tennis ball
GF	Gravel, fine	>2 mm to 16 mm	ladybug to marble
SA	Sand(2-16 mm)	>0.06 mm to 2 mm	gritty to ladybug
FN	Fines(silt/clay/muck)	<0.06 mm	non gritty
HP	Hardpan- hardened fines	any size	
WD	Wood	any size	
OT	Other (doesn't fit choices above)	any size	

C-16. Measuring slope and bearing in small streams

Purpose and scope

This method describes how to measure slope and bearing of the main channel at each site during a data collection event (DCE) for Watershed Health monitoring. It applies to waded streams. This method requires use of a hand level, measuring rod, and a compass to make incremental measurements across each of at least 20 segments of the stream site.

Personnel responsibilities

Two persons perform this activity: one *rodder* who holds a measuring rod in a vertical position and a *sighter* who sights on the rodder with a hand level and compass to record data. Crew members must be trained prior to performing this method.

Equipment, reagents, supplies

- Hand level (5x magnification)
- Monopod for hand level
- Measuring rod (telescoping)
- Compass (handheld, magnetic)
- Range finder
- 50-meter tape
- *Slope and Bearing Form*
- Pencil

Summary of procedure

A two person-crew performs this procedure incrementally, once for each of at least 20 segments of the main channel for the entire site. Segments evaluated are normally between major and minor transects (e.g., A5-A0), but intermediate measurements may be used if necessary (e.g., due to thick vegetation or sharp bends in the channel). There should be no space between segments and no overlap of segments. The crew can either work moving up the stream or down, depending on efficiency of overall work flow. We will describe the technique for working from the top of the stream, downward. This method is based on modifications of Peck, et al. (2006) and Moberg (2007).

Slope

The sighter stands at the water's edge of a transect at a higher elevation (Figure C-16.1). This person will sight downstream toward a measuring rod at a lower transect. Use a monopod to rest the hand level at a fixed eye height. The rodder holds the measuring rod vertically, with its base at the surface of the water. The rodder can assist by pointing to the numbers on rod and adjusting up or down as directed by the sighter. Record these things on the *Slope and Bearing Form* (Figure C-16.2):

- Identity of transect where the sighter stands
- Identity of transect where the rodder stands
- Eye height (cm)
- Level height (cm)

Note: Sometimes it is easier to sight in the wetted channel rather than at the edge, to avoid vegetation. If the monopod or measuring rod rests below the surface of the water, subtract that depth from the eye height or level height.

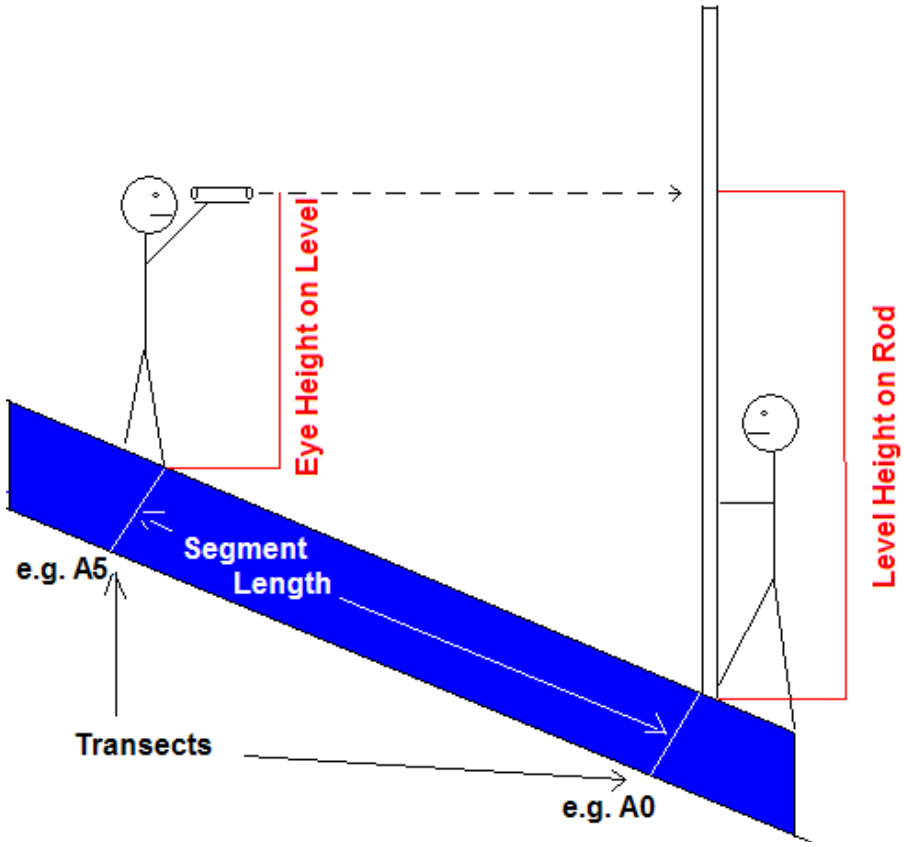
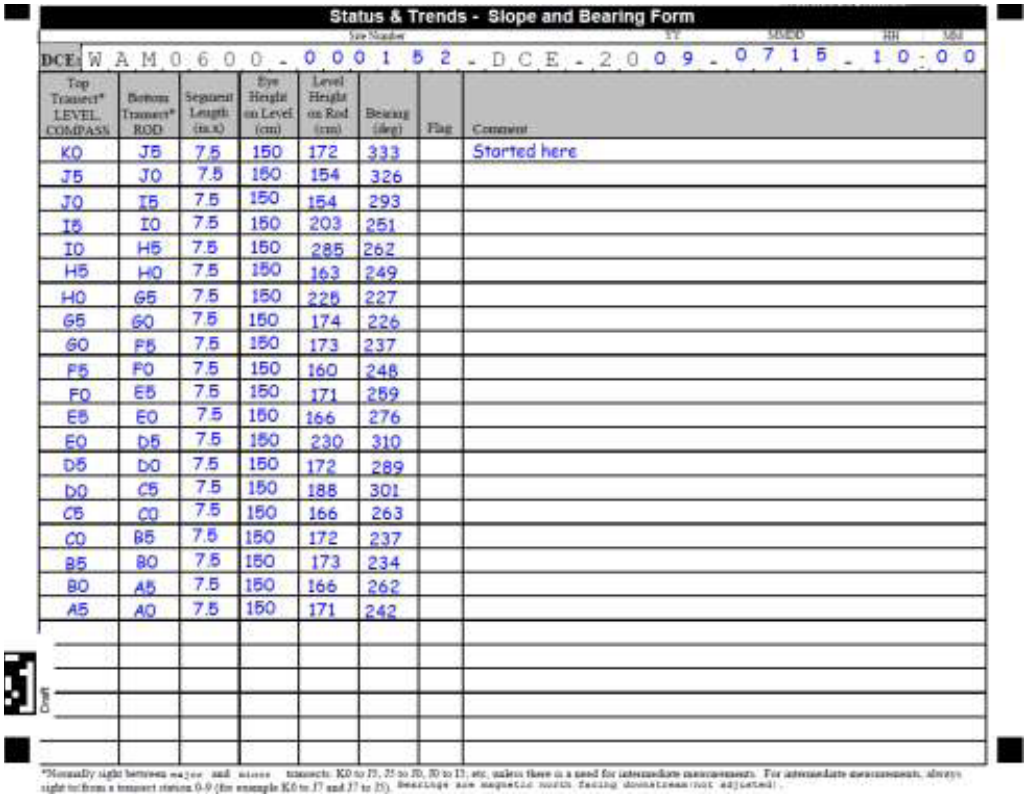


Figure C-16.1. Crew positions when measuring the slope and bearing.



Status & Trends - Slope and Bearing Form

Site Number: DCE: WAM0600-000152-DCE-2009-0715-10:00 (YY MMDD HH MM)

Top Transect* LEVEL COMPASS	Bottom Transect* ROD	Segment Length (m.x)	Eye Height on Level (cm)	Level Height on Rod (cm)	Bearing (deg)	Flag	Comment
K0	J5	7.5	150	172	333		Started here
J5	J0	7.5	150	154	326		
J0	I5	7.5	150	154	293		
I5	I0	7.5	150	203	251		
I0	H5	7.5	150	285	262		
H5	H0	7.5	150	163	249		
H0	G5	7.5	150	225	227		
G5	G0	7.5	150	174	226		
G0	F5	7.5	150	173	237		
F5	F0	7.5	150	160	248		
F0	E5	7.5	150	171	259		
E5	E0	7.5	150	166	276		
E0	D5	7.5	150	230	310		
D5	D0	7.5	150	172	289		
D0	C5	7.5	150	188	301		
C5	C0	7.5	150	166	263		
C0	B5	7.5	150	172	237		
B5	B0	7.5	150	173	234		
B0	A5	7.5	150	166	262		
A5	A0	7.5	150	171	242		

*Normally right between major and minor transects: K0 to J5, J5 to J0, J0 to I5, etc, unless there is a need for intermediate measurements. For intermediate measurements, always right to/from a transect minus 0.5 (for example K0 to J7 and J7 to J5). Bearings are magnetic north facing downstream (not adjusted).

Figure C-16.2. The *Slope and Bearing Form*, with example data.

Bearing

The sighter stands at a transect at a higher elevation (Figure C-16.1). This person will sight downstream toward the rodder at a lower transect. The sighter will then point the compass toward the rodder and parallel to the thalweg. On the *Slope and Bearing Form*, record the bearing (magnetic north) of the thalweg between the top and bottom of the segment.
Note: If sighting from bottom to top, record the bearing south.

Appendix D. Field and laboratory sampling procedures for benthos and periphyton

D-1. Field sampling benthos in small streams

Purpose and scope

This method describes how to collect benthic macroinvertebrate samples for conducting community-level assessments for Washington's Watershed Health Monitoring Program. Data will be used to describe biological integrity and ecological quality (or taxonomic loss). It applies to waded streams. This method requires measurement of the associated physical and chemical environmental variables described in other methods within this protocol.

Personnel responsibilities

One person or more performs this activity. Staff performing this method must have been trained.

Equipment, reagents, supplies

- Wide-mouth polyethylene jar (128 oz or 3.8 L).
- D-Frame kick net with these characteristics.
    - Frame mouth that is 1 ft (30.5 cm) wide by 1 ft tall.
    - 500-μm mesh net.
- 95% Ethanol (add 3 parts by volume for each part sample).
- Label (waterproof) for jar exterior.
- Label (waterproof) for jar interior.
- Soft-lead pencil.
- Clear tape.
- Electrical tape.
- Pocket knife.
- Wading gear.

Summary of procedure

Invertebrate sampling is one of the first methods to be performed on-site, after site verification and layout. It starts concurrently with water sampling, with initial components of the benthos sample collected downstream of the water sample. One kick sample is collected at each of 8 transects and added to the composite sample for the site. This method is taken from Hayslip (2007) with some details provided by Peck, et al. (2006).

Choose transects

Randomly choose 8 transect stations out of these 11:

- A0
- B0
- C0
- D0
- E0
- F0
- G0
- H0
- I0
- J0
- K0

Identify kick stations

Start at the lowest transect and work upstream. At each transect, visually estimate the distance from left to right where the stream bottom will be sampled (Table D-1.1). Half the stations are in mid-channel. Half are in margins. If the water is too deep to sample at any station, collect the sample from the nearest feasible location. The kick net normally allows sampling up to about 50 cm depths.

Table D-1.1. Components of the macroinvertebrate composite sample.

Kick Station	Distance across wetted channel (left to right)
1st	25%
2nd	50%
3rd	75%
4th	50%
5th	25%
6th	50%
7th	75%
8th	50%

Collect each kick

A different procedure is needed depending upon whether the station sits within flowing water or slack water. Flowing water is where the stream current can sweep organisms into the net. Slack water is where water is so slow that active net movement is required to collect organisms.

Flowing water stations

Once the kick station is determined, place the net opening into the face of flow. Position the net quickly and securely on the stream bottom to eliminate gaps under the frame. Collect benthic macroinvertebrates from a 1ft² (0.9 m²) quadrant located directly in front of the frame mouth. Work from the upstream edge of the quadrant backward and carefully pick up and rub stones directly in front of the net to remove attached animals. Quickly inspect each stone to make sure you have dislodged everything and then set it aside. If a rock is lodged in the stream bottom, rub it a few times, concentrating on any cracks or indentations.

After removing all large stones, keeping the sampler securely in position, starting at the upstream end of the quadrant, kick the top 4 to 5 cm of the remaining finer substrate within the quadrant for 30 seconds. Pull the net up out of the water. Immerse the net in the stream several times or splash the outside of the net with stream water to remove fine sediments and to concentrate organisms at the end of the net. After completing the sample, hold the net vertically and rinse material to the bottom of the net.

After taking a sample, examine the contents of the net. Pick out coarse rocks and sticks. Closely examine them for clinging organisms; pick these animals off of the debris and place them into the sample jar. Discard the debris and empty the net's remaining contents into the sample jar. Add enough ethanol to the sample jar so that the resulting solution consists of 1/3 sample and 2/3 ethanol (by volume).

Slack water stations

Visually define a rectangular quadrant with an area of 1 ft^2 (0.09 m^2). Inspect the stream bottom within the quadrant for any heavy organisms, such as mussels and snails. Remove these organisms by hand and place them into the sample jar. Pick up any loose rocks or other larger substrate particles within the quadrant and hold them in front of the net. Use your hands to rub any clinging organisms off of rocks or other pieces of larger substrate (especially those covered with algae or other debris) into the net. After scrubbing, place the larger substrate particles outside of the quadrant.

Vigorously kick the remaining finer substrate within the quadrant with your feet while dragging the net repeatedly through the disturbed area just above the bottom. Keep moving the net all the time so that the organisms trapped in the net will not escape. Continue kicking the substrate and moving the net for 30 seconds.

After 30 seconds, remove the net from the water with a quick upstream motion to wash the organisms to the bottom of the net. After taking a sample, examine the contents of the net. Pick out coarse rocks and sticks. Closely examine them for clinging organisms; pick these animals off of the debris and place them into the sample jar. Discard the debris and empty the net's remaining contents into the sample jar. Add enough ethanol to the sample jar so that the resulting solution consists of 1/3 sample and 2/3 ethanol (by volume).

Special circumstances

For samples located within dense beds of long, filamentous aquatic vegetation, kicking may not be effective. Use a knife to sample only the vegetation that lies within the quadrant. Don't include parts of the strands that extend beyond the quadrant.

Label and seal the composite sample

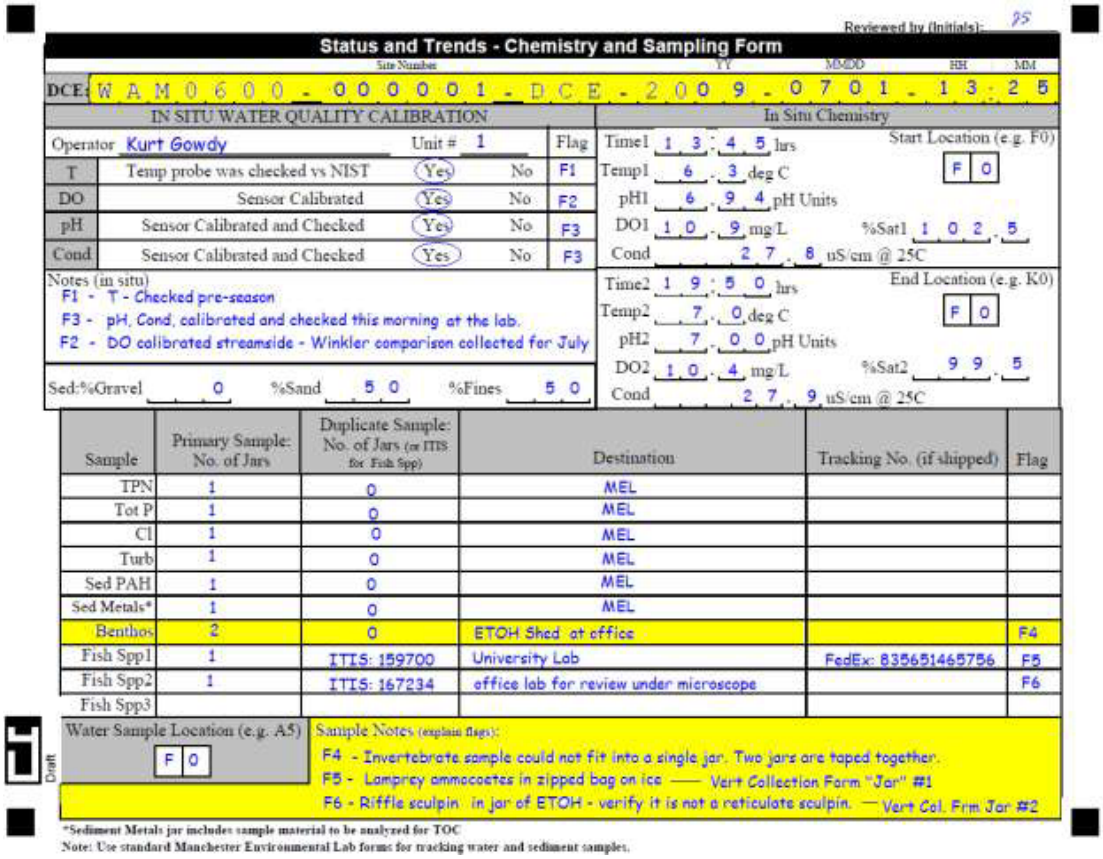
Using a number 2 pencil, complete two benthos jar labels (Figure D-1.1). Place one into the sample. Screw on the lid and seal it closed using electrical tape. Attach the other benthos label to the outside of the jar using clear tape. Record the DCE, which includes the Site ID, and site arrival time (year, month, day, hour, and minute). It should match the DCE recorded on the Site Verification Form. Be sure to note which transects were sampled, and which of these were sampled using the slack water technique.

500 µ D-frame kick	**Benthos Jar Label** Jar ___ of ___
Project	Date____ Name_____
Stream	
Who collected? (full name)	
8 1-ft$_2$ Transects **(circle all sampled)**	A B C D E F G H I J K Transects sampled using slack-water technique: _____
Collectors Notes	
DCE	WAM06600-__ __ __ __ __ __-dce-2015__ __ __ __-__ __ __ __ m m d d h h m m

Figure D-1.1. The benthos jar label.

Enter data to the C*hemistry and Sampling Form*

The sample jars will be stored by field crews and delivered *en mass* to the analytical laboratory at the end of the field season. The *Chemistry and Sampling Form* (Figure D-1.2) will be used to keep track of sample jar information. Note the Sample ID and number of jars per Sample ID. If there is more than one jar for a Sample ID, then ensure that the jars are located together. Taping the jars together with clear tape may be helpful. For destination, note the immediate place to where the sample will be stored, shipped, or delivered.



Reviewed by (Initials): PS

Status and Trends - Chemistry and Sampling Form

DCE: WAM0600 - 0000001 - DCE - 2009 - 0701 - 1325
(Site Number — YY — MMDD — HH — MM)

IN SITU WATER QUALITY CALIBRATION

Operator: Kurt Gowdy Unit #: 1

				Flag
T	Temp probe was checked vs NIST	Yes	No	F1
DO	Sensor Calibrated	Yes	No	F2
pH	Sensor Calibrated and Checked	Yes	No	F3
Cond	Sensor Calibrated and Checked	Yes	No	F3

Notes (in situ)
F1 - T - Checked pre-season
F3 - pH, Cond, calibrated and checked this morning at the lab.
F2 - DO calibrated streamside - Winkler comparison collected for July

Sed:%Gravel 0 %Sand 50 %Fines 50

In Situ Chemistry

Time1 13:45 hrs Start Location (e.g. F0): F0
Temp1 6.3 deg C
pH1 6.94 pH Units
DO1 10.9 mg/L %Sat1 102.5
Cond 27.8 uS/cm @ 25C

Time2 19:50 hrs End Location (e.g. K0): F0
Temp2 7.0 deg C
pH2 7.00 pH Units
DO2 10.4 mg/L %Sat2 99.5
Cond 27.9 uS/cm @ 25C

Sample	Primary Sample: No. of Jars	Duplicate Sample: No. of Jars (or ITIS for Fish Spp)	Destination	Tracking No. (if shipped)	Flag
TPN	1	0	MEL		
Tot P	1	0	MEL		
Cl	1	0	MEL		
Turb	1	0	MEL		
Sed PAH	1	0	MEL		
Sed Metals*	1	0	MEL		
Benthos	2	0	ETOH Shed at office		F4
Fish Spp1	1	ITIS: 159700	University Lab	FedEx: 835651465756	F5
Fish Spp2	1	ITIS: 167234	office lab for review under microscope		F6
Fish Spp3					

Water Sample Location (e.g. A5): F0

Sample Notes (explain flags):
F4 - Invertebrate sample could not fit into a single jar. Two jars are taped together.
F5 - Lamprey ammocoetes in zipped bag on ice — Vert Collection Form "Jar" #1
F6 - Riffle sculpin in jar of ETOH - verify it is not a reticulate sculpin. — Vert Col. Frm Jar #2

Draft

*Sediment Metals jar includes sample material to be analyzed for TOC
Note: Use standard Manchester Environmental Lab forms for tracking water and sediment samples.

Figure D-1.2. The *Chemistry and Sampling Form*, with fields that are relevant to benthos sampling highlighted.

D-2. Taxonomic lab sampling benthos from small streams

Purpose and scope

Taxonomic identification is conducted by a lab that employs taxonomists certified by the Society for Freshwater Science at the genus level. The taxonomist should have experience with the freshwater macroinvertebrates of the Pacific Northwest. All major orders of freshwater macroinvertebrates are identified to at least the genus level (see Appendix I), including the Chironomidae (See Appendix J) and Simuliidae, and to species where existing taxonomic keys are available. Taxon groups normally identified to coarser taxonomic levels include: Lumbriculidae, Naididae, Oligochaeta, select families of the Coleoptera, Planariidae, and Acari. If the taxonomist has a compelling reason (Appendix K) that a specimen cannot be identified to the genus level, they may decide to aggregate individuals in the next highest taxonomic level.

Personnel responsibilities

One person or more performs this activity. Staff performing this method must have been trained.

Summary of procedure

Sample preparation

Samples are sub-sampled using a 500-organism count. According to Ecology protocols (Plotnikoff and Wiseman, 2001), macroinvertebrates are removed from a minimum of two randomly chosen squares from a 30 square sub-sampling grid. The dimension of each square is 6 cm x 6 cm and the grids overall dimensions are 30 cm x 36 cm. The sample material is thoroughly mixed and spread evenly across the grid. All organisms are removed from randomly chosen squares until a minimum of 500 macroinvertebrates are removed from the sample and placed in alcohol for subsequent identification under the dissecting scope. If a grid square is dominated by a single taxon, additional grids are selected and sorted, and notes are made in the report. In some cases, there may be less than 500 organisms in the whole sample. When the target count of organisms has been reached or the specified amount of material has been sorted, a special large and rare protocol may be followed, with these organisms placed in an additional labeled vial.

Large and rare specimen identification

The remainder of the sample material in the tray will be searched for any large or rare taxa that may have been missed in the sub-sampled fraction. These specimens will be identified and placed together in a vial labeled “Large and Rare Taxa” for the voucher collection. This scan will include any adult aquatic invertebrates, which will be archived separately (not to be identified and included in data set) for anyone interested in looking at the material in the future.

Reporting requirements

The contractor will complete all analyses by the dates mentioned above and submit all final data and QA results via the Puget Sound Stream Benthos website, including:

- Sample ID.
- ITIS number.
- Taxon name.
- Taxon count.
- Density estimate per sample of each species in square meters.
- Percent Sample Sorted – website will ask for number or squares counted out of total number of squares.
- Number of individuals per species of organisms in a life stage other than juvenile (i.e. eggs, adults, larval) in each sample.
- Number of individuals that were damaged to the point that identification to the required resolution was not possible.

D-3. Field sampling periphyton in small streams

(This protocol is being updated by Chad Larson (Ecology) and this update represents a near final version. The published update is expected in Fall 2014 and any changes are expected to be minor. All updates will be captured in the Spring 2015 training.)

Purpose and scope

Periphyton are important primary producers and chemical modulators in stream ecosystems. As such, periphyton can be more sensitive to certain stressors such as nutrients, salts, sediment, and temperature compared to other aquatic organisms. Measures of periphyton community structure, diversity, and density are useful in the assessment of biological condition for surface waters. For more information on periphyton and their use in bioassessment, refer to Barbour, et al. (1999) and Stevenson, et al. (1996).

Personnel responsibilities

One person or more performs this activity. Staff performing this method must have been trained.

Equipment, reagents, supplies

- Wide-mouth polyethylene jar (1 L).
- Firm bristled toothbrush.
- luminum foil.
- Wash bottle.
- Plastic funnel.
- Digital caliper
- Volumetric pipette
- 5-10% Lugol's solution (potassium iodide, iodine crystals, glacial acetic acid, DI water)
- One gallon Ziploc bag.
- Large plastic bowl or tray to catch rinsate.
- Filtered native stream water using Grade 4 filter paper 20-25 µm (e.g., Whatman 9.0 cm, supplier No. 1004-090) + Buchner funnel (e.g., Scienceware® 9.0 cm, supplier No. 146030000).
- Label (waterproof) for jar exterior.
- Soft-lead pencil.
- Clear tape.
- Electrical tape.
- Pocket knife.
- Wading gear.

Summary of procedure for multi-habitat sampling

Periphyton sampling can be performed on-site with macroinvertebrate sampling, after site verification and layout. One sample is collected at each of 8 randomly selected major transects and are added to a single composite sample jar for the site. Samples are to be collected in close proximity to (but not within) the randomly selected macroinvertebrate sample locations. See *Benthos Sampling* (Adams 2010) for description of selecting random sample locations. The

objective is to collect a composite sample that is representative of the periphyton assemblage present in the reach. Therefore, sample all substrates roughly in proportion to their areal coverage in the reach (see list and descriptions of sampling methods below), which can be approximated when marking transects for the reach. For example, if approximately half the transects in a reach are composed largely of epilithic (coarse substrate) and epipelic (silt) habitats respectively, then a composite sample should contain material from four transects each of epilithic and epipelic habitats. Additionally, if present, pick specimens of macroalgae (i.e. filamentous forms) by hand in proportion to their relative abundance in the reach. Combine all samples into a common container.

Habitat	**Substrate type**	**Approximate surface area (1 transect)**	**Approximate surface area (8 transects)**
Epilithic	Coarse substrate	44.2 cm^2	353.43 cm^2
Epidendric	Woody snags	Variable, see method description	Variable, see method description
Episammic	Pea gravel/sand	17.4 cm^2	139 cm^2
Epipelic	Silt	17.4 cm^2	139 cm^2
Epiphytic	Emergent vegetation	Variable, see method description	Variable, see method description
Epiphytic	Submerged vegetation	Variable, see method description	Variable, see method description

Filtering Native Stream Water for Rinsate

Rinsate used while collecting periphyton should preferentially come from filtered water obtained directly from the stream being sampled. Prior to collection of periphyton, stream water should be filtered through grade 3 filter paper (particle retention 6 µm) using a Buchner funnel and collected into wash bottle.

Sampling Method for Epilithic (Coarse Substrate) Habitats

Epilithic or course substrate is likely to be the most frequently encountered habitat sampled for periphyton.

1. Carefully remove a rock from each randomly selected sample locations, retaining the rock's orientation as it occurred in the stream to avoid loss of periphyton. Rocks should be relatively flat and range in size from about 4 cm (coarse gravel) to 10 cm (small cobble) in diameter. Collect one rock per randomly selected sample location if the diameter of the rock selected is equal to or exceeds 7.5 cm, otherwise select a second rock. If possible, select rocks that are similar with respect to size, depth, and exposure to sunlight. Gently place the rock(s) in a plastic tray; do not stack them upon one another. Transport the tray to a convenient sample-processing area. Where possible, process the sample out of direct sunlight to minimize degradation of chlorophyll.
2. Scrub only the upper surface of each rock with a firm-bristled toothbrush using a circular motion. When rocks are greater than 10 cm (medium to large cobbles), firmly brush only a portion of the upper rock surface around 7.5 cm in diameter. Do not brush the sides or bottom of rocks. If needed, remove any filamentous algae and mosses by scraping with a

knife and place in a separate plastic tray. Use a knife or scissors to cut algal filaments or moss into roughly 2 to 3 mm segments. Gently brush other larger plant material that may be attached to the rocks but do not collect the plants. Rinse the sampled rock surface, attached plants and toothbrush bristles with a rinse bottle containing filtered stream water. Use rinse water sparingly, but be thorough. Collect rinsate in the plastic tray containing any filamentous algae or mosses and repeat for the remaining rocks. The volumes of water used to rinse separate collections must be carefully monitored to ensure that the total sample volume after compositing does not exceed 475 ml so as to allow enough empty volume to add Lugol's and not exceed a 500-ml sample-volume limitation. After sample processing is complete, measure and record the total rinsate volume (now considered the composite sample volume) on the datasheet and sample label and pour the rinsate through a funnel into a 1 L Nalgene® sample bottle.

3. For each rock processed, cover the surface with a sheet of aluminum foil. Either trim the foil with a knife or fold the foil to match the area sampled. Place the trimmed/folded foil templates into a labeled collection envelope and attach to the field data sheets.
4. Process the composite sample following steps described in Subsample Processing Procedures to extract subsamples for chlorophyll *a* analysis and taxonomic identification.

Sampling method for epidendric (woody snag) habitats

Collecting quantitative periphyton samples from epidendric habitats presents a challenge because they generally have an irregular surface and are difficult to remove without loss of periphyton biomass. Use the following method to address these difficulties when sampling epidendric habitats:

1. Select one piece of woody snag material from each transect where epidendric habitat is the dominant habitat. Select a piece of wood that is greater than 1 cm in diameter that has likely been submerged for most of the year to allow for sufficient periphyton colonization but is not smothered by bottom sediments.
2. Carefully remove an approximately 10 to 20 cm long section of each woody snag with pruning shears or a hand saw and place in a plastic tray. Transport the tray to a convenient sample-processing area. Where possible, process the sample out of direct sunlight to minimize degradation of chlorophyll.
3. Scrub the entire surface of the woody section with a firm-bristled toothbrush. If needed, remove any filamentous algae and mosses by scraping with a knife and place in the plastic tray. Use a knife or scissors to cut algal filaments or moss into roughly 2 to 3 mm segments. Rinse the toothbrush and the section of wood with a rinse bottle containing filtered stream water. Use rinse water sparingly, but be thorough. Collect rinsate in the plastic tray containing any filamentous algae or mosses. Set the section of wood aside. Repeat for the remaining woody sections. Keep the total sample volume less than 500 mL (i.e. sample plus Lugol's preservative). After sample processing is complete, measure and record the total rinsate volume (now considered the composite sample volume) on the datasheet and sample bottle and pour the rinsate through a funnel into a 1 L Nalgene® sample bottle.
4. Measure the length and diameter (take an average of three diameter measurements) of each cleaned woody section and calculate the total sampled surface area by using the following formula (assumes a cylinder):

   Total Sampled Area (cm^2) = $\sum_{i=1}^{n}(\pi)(d)(l)$
   Where,
   n = number of discrete collections
   π = 3.1416
   d = mean diameter of each woody section, in centimeters
   l = length of each woody section, in centimeters

   Alternatively, a foil template can be used (see Epilithic habitat method) for irregularly shaped woody sections. Record the sampled surface areas on the datasheet.
5. Process the composite sample following the steps described in Subsample Processing Procedures to extract subsamples for chlorophyll *a* analysis and taxonomic identification.

Sampling method for episammic (pea gravel/sand) and epipelic (silt) habitats

Quantitative microalgal periphyton samples are collected from the upper 5 to 7 mm layer of episammic (pea gravel _ 5 mm/sand) and epipelic (silt) habitat in organically-rich depositional areas of the reach. Use the following method to sample episammic or epipelic habitats:

1. At each transect where episammic habitat is the dominant habitat, select a location in a shallow, organically-rich depositional zone that consist of either pea gravel, sand, or silt substrate.
2. At each sampling location, hold the lid of a plastic Petri dish (47 mm diameter) upside down in the water; gently stir/shake the lid to remove air bubbles without disturbing the substrate.
3. With the lid still submerged, turn the inside of the lid toward the substrate that will be sampled without disturbing the substrate.
4. Carefully and slowly, press (in cookie cutter fashion) the lid into the substrate.
5. Slide the lid onto a spatula to enclose the discrete collection. Holding the Petri dish firm against the spatula, carefully wash extraneous sediment from the spatula and lift out of the water.
6. Transport the Petri Dish and spatula to a convenient sample-processing area. Where possible, process the sample out of direct sunlight to minimize degradation of chlorophyll.
7. Invert the lid and remove the spatula. Be careful not to lose any of the discrete sample still adhering to the spatula.
8. Rinse the substrate from the lid and spatula with a rinse bottle containing filtered stream water into a 1 L Nalgene® sample bottle. Use rinse water sparingly, but be thorough, being mindful that total composite sample should not exceed 500 mL. After sample processing is complete, measure and record the total rinsate volume (now considered the composite sample volume) on the datasheet.
9. The total sample surface area for each discrete sample collected with a 47 mm Petri dish is approximately 17.4 cm^2. Record the sampled surface area on the datasheet.
10. Measure water depth and velocity at the point where each of the eight discrete collections were removed using a top-setting rod and velocity meter and record on the datasheet. Assuming the sun is directly overhead, determine the relative degree of riparian shading (e.g., shaded, partial, or full sun) at each sample location and record on the datasheet.
11. Process the composite sample following the steps described in Subsample Processing Procedures to extract subsamples for chlorophyll *a* analysis and taxonomic identification.

Sampling method for epiphytic (emergent vegetation) habitats

Sampling emergent macrophytes is appropriate only when it is not possible to sample other preferred targeted habitats (e.g., epilithic, epidendric, episammic, and epipelic) due to their absence or rare occurrence in the reach. Collecting quantitative microalgal periphyton samples from emergent vegetation presents a challenge because of varying sizes and shapes in vegetation and the care needed to remove the vegetation without loss of periphyton biomass. Use the following method to address these difficulties when sampling epiphytic (emergent vegetation):

1. All samples should be collected from live specimens of the same emergent species or group of closely-related emergent species that are common in the reach to facilitate sampling of the same species/group in the future. This will allow for representative comparisons of periphyton data over time. Groups of closely-related emergent species such as Carex (sedges), Juncus (rushes), Polygonum (smartweed) and Typha (cattails) are all suitable.
2. Record the species or group of closely-related species that will be sampled on the field datasheet.
3. Select a total of three sections of emergent vegetation from each transect where emergent vegetation is the dominant habitat. Each section represents a stem or leaf (no roots); however, all three sections must be of the same type. NOTE: Do not cause unneeded disturbance to the emergent vegetation or excessive wave-action when wading; epiphytic periphyton are often loosely attached and easily dislodged.
4. Prior to sampling a section, the un-submerged portion of the selected emergent vegetation should be removed with pruning shears or scissors at water level and discarded. Select a large diameter/width section of stem or leaf from the submerged portion of the emergent vegetation just below water level. Select sections that have likely been submerged for most of the growing season to allow for sufficient periphyton colonization but which are not smothered by bottom sediments.
5. Carefully remove an approximately 10 to 20 cm long section of each stem or leaf with pruning shears or scissors and place in a plastic tray. Transport the tray to a convenient sample-processing area. Where possible, process the sample out of direct sunlight to minimize degradation of chlorophyll.
6. Measure water depth and velocity at the point where each section of emergent vegetation was removed using a top-setting rod and velocity meter and record on the datasheet.
7. Gently brush the entire surface of the stem or both sides of a leaf section with a soft-bristled toothbrush. If needed, remove any filamentous algae and mosses by brushing and place in a separate plastic tray. Use a knife or scissors to cut algal filaments or moss into roughly 2 to 3 mm segments. Rinse the toothbrush and the stem or leaf section with a rinse bottle containing filtered stream water. Use rinse water sparingly, but be thorough. Collect rinsate in the plastic tray containing any filamentous algae and mosses. Set the stem or leaf section aside. Repeat for the remaining stem or leaf sections. Keep the sample volume less than 500 mL. After sample processing is complete, measure and record the total rinsate volume (now considered the composite sample volume) on the datasheet and pour the rinsate through a funnel into a 1 L Nalgene® sample bottle.

8. For cylindrical-shaped stem or leaf samples, use a digital caliper to measure the length and diameter (take an average of three diameter measurements) of each cleaned stem/leaf section and calculate the total sampled surface area by using the following formula (assumes a cylinder):

   Total Sampled Area (cm^2) = $\sum_{i=1}^{n}(\pi)(d)(l)$

   Where,
   n = number of discrete collections
   π = 3.1416
   d = mean diameter of each cylindrical stem section, in centimeters
   l = length of each cylindrical stem section, in centimeters

9. For triangular stem samples, use a digital caliper to measure the width of all three sides (measure the width of each side and take an average) and length for each cleaned stem section. Calculate the total sampled surface area by using the following formula (assumes an equilateral triangle):

   Total Sampled Area (cm^2) = $\sum_{i=1}^{n} 3(w)(l)$

   Where,
   n = number of discrete collections
   w = mean width of each triangular stem section, in centimeters
   l = length of each triangular stem section, in centimeters

10. For non-cylindrical leaf samples, place each cleaned leaf section on a sheet of aluminum foil. With a permanent marker or pen, trace the shape of the leaf section to match the area sampled. For large leaf sections, either trim the foil with a knife or fold the foil to match the area sampled. Mark “X 2” on each leaf template to note that both sides of the leaf section were sampled. Place the marked or trimmed/folded foil templates into a labeled collection envelope and attach to the field data sheets.

11. Process the composite sample following steps described in Subsample Processing Procedures to extract subsamples for chlorophyll *a* analysis and taxonomic identification.

Sampling method for epiphytic (submerged vegetation) habitats

Sampling submerged macrophytes is appropriate only when it is not possible to sample other preferred targeted habitats (e.g., epilithic, epidendric, episammic, epipelic) due to their absence or rare occurrence in the reach. Collecting quantitative periphyton samples from submerged macrophytes can be a challenge because several submerged macrophytes have small or finely dissected leaves, which present difficulties for accurately calculating the surface area of periphyton colonization. Furthermore, care is needed to remove the vegetation without the loss of periphyton biomass. Use the following method to address these difficulties when sampling epiphytic (submerged vegetation) habitats:

1. All samples should be collected from live specimens of the same submerged species or group of closely-related submerged species that are common in the reach to facilitate sampling of the same species/group in the future. This will allow for representative comparisons of periphyton data over time. Groups of closely-related submerged species such as Ceratophyllum (coontail), Myriophyllum (milfoil), Najas (water-nymph), and Potamogeton (pondweed) are all suitable.
2. Record the species or group of closely-related species that will be sampled on the field datasheet.
3. Select a total of three samples of submerged vegetation from each transect throughout the reach where epiphytic habitat is dominant. Each sample should consist of a single stem plus associated branches of the plant from the lowest healthy leaves to the tip. Submerged leafless stems should not be included. Select samples that have likely been submerged for most of the growing season to allow for sufficient periphyton colonization but which are not smothered by bottom sediments. NOTE: Do not cause unneeded disturbance to the emergent vegetation or excessive wave-action when wading; epiphytic periphyton are often loosely attached and easily dislodged.
4. Carefully remove the sample with pruning shears or scissors and gently place in a plastic tray. Transport the tray to a convenient sample-processing area. Where possible, process the sample out of direct sunlight to minimize degradation of chlorophyll.
5. Measure water depth and velocity at the point where each of the eight samples of submerged vegetation were removed using a top-setting rod and velocity meter and record on the datasheet.
6. Use a knife or scissors to cut the sample's stem and branches into 10 to 20 cm segments, preferably at the plant nodes. Do not cut through the leaves. NOTE: For samples with small or finely dissected leaves, the sampler has the option of discarding the leaves and only processing the stems and branches. If leaves are discarded, sampler must note this on the datasheet. Use a knife or scissors to cut any algal filaments or moss into roughly 2 to 3 mm segments and place in a separate plastic tray. Repeat for remaining samples. Within the plastic tray, separate the sample segments from the algal/moss segments.
7. Fill a 1 L Nalgene® bottle with approximately 200 mL of filtered stream water. Place sample segments in the bottle and cap the bottle. Shake the bottle vigorously for 30 seconds to dislodge attached periphyton. Open the bottle, and poor algal slurry into the separate composite sample bottle using a funnel and a small kitchen strainer placed above the strainer to capture individual macrophyte segments. Rinse macrophyte segments with

filtered stream water. Use rinse water sparingly, but be thorough; rinsate should flow through the strainer and into the composite bottle. Once all sample segments have been processed, place algal/moss segments in the 1 L Nalgene® bottle. Any dislodged periphyton remaining in the plastic tray should be inserted through a funnel into the 1 L bottle. Keep the sample volume less than 500 mL. After sample processing is complete, measure and record the total rinsate volume (now considered the composite sample volume) on the datasheet.

8. For cylindrical-shaped stems, branches and leaf samples, use a digital caliper to measure the length and diameter of each cleaned stem, branch or leaf segment[1]. Calculate the total sampled surface area by using the following formula (assumes a cylinder):

   Total Sampled Area (cm2) = $\sum_{i=1}^{n}(\pi)(d)(l)$

   Where,
   n = number of discrete collections
   π = 3.1416
   d = mean diameter of each cylindrical stem section, in centimeters
   l = length of each cylindrical stem section, in centimeters

   [1]For samples with ≥ 10 cylindrical-shaped leaves, take an average of the length and width measured from each of the randomly-selected leaf segments. Use these mean values and the above equation to obtain a surface area and then multiply by the total number of leaf segments in the sample. This alternative method may also be used to calculate the area for cylindrical-shaped branches when the sample contains ≥ 10 branches. In all cases, the stem must be measured in its entirety.

9. For non-cylindrical or broad-shaped leaf samples, place each cleaned leaf segment on a sheet of aluminum foil[2]. With a permanent marker or pen, trace the shape of the leaf segment to match the area sampled. For large leaf segments, either trim the foil with a knife or fold the foil to match the area sampled. Mark "X 2" on each leaf template to note that both sides of the leaf segment were sampled. Place the marked or trimmed/folded foil templates into a labeled collection envelope and attach to the field data sheets.

   [2]For samples with ≥ 10 non-cylindrical or broad-shaped leaves, randomly select three leaf segments and follow the above foil template procedure. Record the total number of leaves on the datasheet. Take the average surface area from the three measured leaf segments and multiply by the total number of leaf segments in the sample. Record the total area on the datasheet.

10. Process the composite sample following steps described in Subsample Processing Procedures to extract subsamples for chlorophyll *a* analysis and taxonomic identification.

Subsample processing procedures

Each composite sample processed in the field is used to extract subsamples for chlorophyll *a* analysis and taxonomic identification. Successful execution of subsample processing procedures described here is dependent on measuring and tracking the various volumes as the composite sample is processed. One subsample is extracted from each composite sample for the purpose of determining chlorophyll *a* in the laboratory. The remaining volume of the composite sample is

considered the ID subsample and is preserved for taxonomic identification. Subsampling processing procedures for periphyton composite samples are as follows:

1. In an area out of direct sunlight, assemble the filtration apparatus by attaching the filter base with rubber stopper to the filtration flask. Join the flask and a hand-operated vacuum pump (with pressure gage) using a section of tubing.
2. Place a 47 mm 0.7 micron glass microfiber filter (for example, Whatman® GF/F) on the filter base and wet with deionized, distilled, or filtered stream water. NOTE: Wetting the filter will help it adhere to the base in windy conditions. Attach the filter funnel to the filter base.
3. Prior to subsample, composite sample volume should be brought to 485 ml. Prior to extraction, homogenize the composite sample by vigorously shaking or using a battery-powered stirrer for 30 seconds.
4. Extract one 10 mL aliquot of homogenized composite sample using a volumetric pipette and dispense onto the middle of the wetted glass microfiber filter.
5. Filter the 10 mL aliquot for chlorophyll *a* with the vacuum pump using 7 to 10 psi.
    a. Examine the filter. An adequate amount of periphytic biomass for analysis is indicated by the green or brown color of material retained on the filter. If needed, extract additional 5 mL aliquots and filter until a green or brown color on the filter is apparent. *NOTE: For composite samples with abundant organic material and/or fine sediment, filtration of a 10 mL aliquot may not be possible. In these circumstances, filter one 5 mL aliquot. If no difficulties were apparent when filtering the first 5 mL aliquot, proceed with filtering a second 5 mL aliquot.*
    b. The filtered aliquot(s) represent the chlorophyll *a* subsample. Determine the number of aliquots filtered and record the chlorophyll *a* subsample volume on the datasheet. For example, 2 aliquots x 5 mL/aliquot = 10 mL subsample volume.
    c. Rinse the sides of the filter funnel with deionized, distilled, or filtered stream water, allow the water to be vacuumed completely before releasing the vacuum from the filtering apparatus.
    d. Using forceps, fold the filter into quarters with the filtered biomass inside. Place the folded filter into the 500mL poly vial already containing acetone. Label the vial and wrap in aluminum foil to exclude light exposure.
    e. The label will contain the following information:
        i. Site name.
        ii. Sample ID.
        iii. Collection date (mm-dd-yyyy).
        iv. Collection Time (24 hr.).
        v. Composite sample volume (mL).
        vi. Subsample volume (mL).
    f. Repeat the aliquot extraction and filtration processes if necessary for quality control duplicates.

g. Place chlorophyll a poly vials in a resealable plastic bag and place in a cooler containing ice. NOTE: The stream itself can be used to cool the samples if getting a cooler to the site is not that feasible. The re-sealable bag can be partially submersed in the stream, being careful to not introduce stream water to the bag, prior to transfer to the cooler with ice.

h. Coolers should be shipped within a few days after the subsamples have been prepared because of a 25-day holding time limit. Subsamples can be temporarily stored in a freezer (at -20°C) at the field office over weekends.

6. For the periphyton taxonomic sample, measure the volume of the remaining composite sample (which represents the ID subsample volume) and record on the datasheet.

7. Preserve the ID subsample with 5% to 10% Lugol's solution (see Sample Preservative-Lugol's Solution for preparation). Five percent should be sufficient for most samples, although up to 10% can be used for samples rich in organic matter. Record the preservative volume on the datasheet. The quantities of Lugol's solution required for selected sample volumes are:

    475 mL ID subsample, add 25 mL Lugol's solution.

8. Label the ID subsample with the following required information:
    a. Site name.
    b. Sample ID.
    c. Collection date (mm-dd-yyyy).
    d. Collection time (24 hr.).
    e. ID subsample volume (mL) [ID subsample + preservative].

Sample preservative-Lugol's solution

Prepare Lugol's solution by dissolving 20 g potassium iodide (KI) and 10 g iodine crystals in 200 mL distilled water containing 20 mL glacial acetic acid. Store Lugol's solution in an opaque plastic bottle.

Quality control

At least 10% of all collected composite samples must consist of duplicate composite samples (this may result in only one replicate for permittees conducting their own monitoring). Duplicate composite sampling consists of two samplers each with the same equipment, collecting simultaneously alongside (1) randomly selected locations for Epilithic samples, (2) woody snag locations for Epidendric samples, (3) shallow depositional locations for Episammic/Epipelic samples, or (4) locations of emergent or submerged vegetation for Epiphytic samples.

Following the processes described under Subsample Processing Procedures, the sampler that collected the duplicate composite sample, extracts two chlorophyll *a* subsamples from the duplicate composite sample. The remaining duplicate composite sample volume will be used for the duplicate ID subsample. Duplicate composite samples are collected to check the variability between field samplers while the two duplicate chlorophyll *a* subsamples provide an indication of precision and the quality of the duplicate composite sample homogenization. An illustration of the duplicate composite sample/subsample processes is provided below.

For each rock processed, cover the surface with a sheet of aluminum foil. Either trim the foil with a knife or fold the foil to match the area sampled. Place the trimmed/folded foil templates into a labeled collection envelope and attach to the field datasheets.

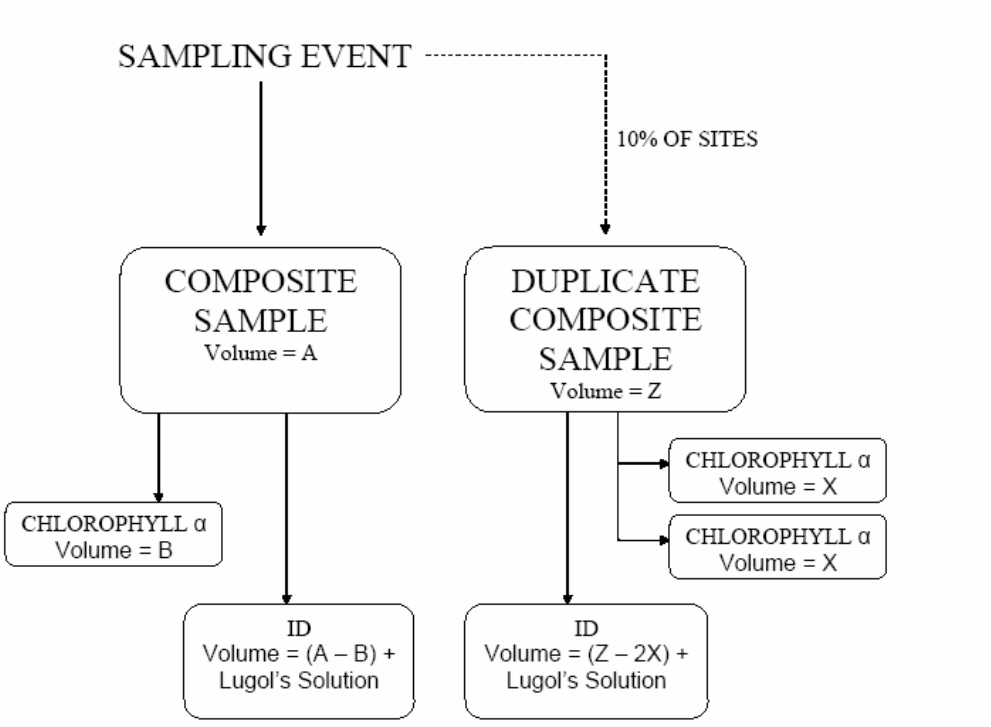
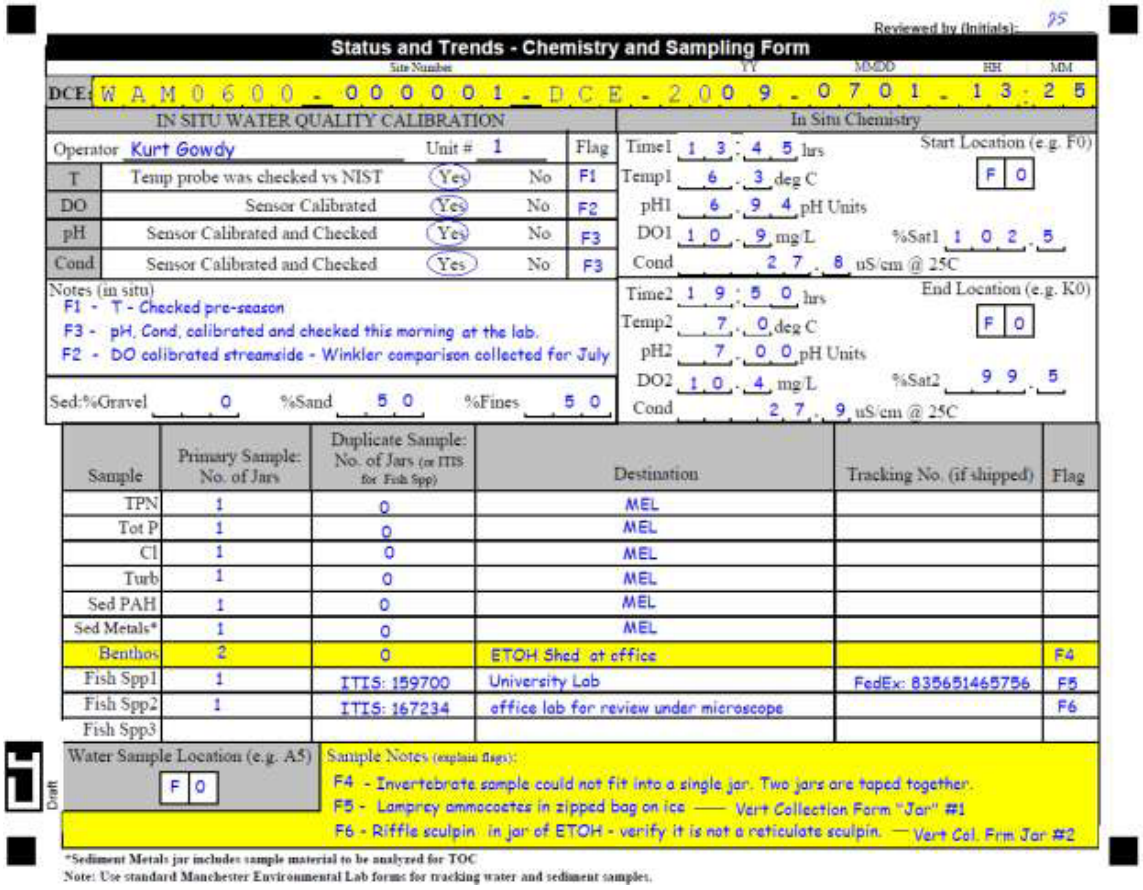


Figure D-3.1. Flow chart for the duplicate composite sample/subsample process.

Periphyton Jar Label Jar ___ of ___	
Project	20__ Monitoring in the _____ STR
Stream	
Who collected? (full name)	
8 samples/ 1 per transect (circle all sampled)	A B C D E F G H I J K
Collectors Notes	Total final volume_____mL
DCE	WAM06600-__ __ __ __ __ __-dce-2009__ __ __ __-__ __ __ __ m m d d h h m m

Figure D-3.2. Example periphyton jar label.



Reviewed by (Initials): JS

Status and Trends - Chemistry and Sampling Form

Site Number: DCE: WAM0600-000001-DCE-2009-0701-1325 (TY MMDD HH MM)

IN SITU WATER QUALITY CALIBRATION

Operator: Kurt Gowdy Unit #: 1

				Flag
T	Temp probe was checked vs NIST	(Yes)	No	F1
DO	Sensor Calibrated	(Yes)	No	F2
pH	Sensor Calibrated and Checked	(Yes)	No	F3
Cond	Sensor Calibrated and Checked	(Yes)	No	F3

Notes (in situ)
F1 - T - Checked pre-season
F3 - pH, Cond, calibrated and checked this morning at the lab.
F2 - DO calibrated streamside - Winkler comparison collected for July

Sed:%Gravel 0 %Sand 50 %Fines 50

In Situ Chemistry

Start Location (e.g. F0): F0

Time1 13:45 hrs
Temp1 6.3 deg C
pH1 6.94 pH Units
DO1 10.9 mg/L %Sat1 102.5
Cond 27.8 uS/cm @ 25C

End Location (e.g. K0): F0

Time2 19:50 hrs
Temp2 7.0 deg C
pH2 7.00 pH Units
DO2 10.4 mg/L %Sat2 99.5
Cond 27.9 uS/cm @ 25C

Sample	Primary Sample: No. of Jars	Duplicate Sample: No. of Jars (or ITIS for Fish Spp)	Destination	Tracking No. (if shipped)	Flag
TPN	1	0	MEL		
Tot P	1	0	MEL		
Cl	1	0	MEL		
Turb	1	0	MEL		
Sed PAH	1	0	MEL		
Sed Metals*	1	0	MEL		
Benthos	2	0	ETOH Shed at office		F4
Fish Spp1	1	ITIS: 159700	University Lab	FedEx: 835651465756	F5
Fish Spp2	1	ITIS: 167234	office lab for review under microscope		F6
Fish Spp3					

Water Sample Location (e.g. A5): F0

Sample Notes (explain flags):
F4 - Invertebrate sample could not fit into a single jar. Two jars are taped together.
F5 - Lamprey ammocoetes in zipped bag on ice — Vert Collection Form "Jar" #1
F6 - Riffle sculpin in jar of ETOH - verify it is not a reticulate sculpin. — Vert Col. Frm Jar #2

Draft

*Sediment Metals jar includes sample material to be analyzed for TOC
Note: Use standard Manchester Environmental Lab forms for tracking water and sediment samples.

Figure D-3.3. The *Chemistry and Sampling Form*, with fields that are relevant to periphyton sampling placed in the highlighted benthos fields. Use the flag to indicate volume in bottle.

D-4. Taxonomic lab sampling periphyton from small stream

Purpose and scope

Taxonomic identification is conducted by a lab that employs taxonomists with degrees in a related field. The taxonomist should have experience with the freshwater periphyton of the Pacific Northwest. A standardized level of identification is required to allow for valid comparison of periphyton data sets between sampling locations. The standard level of identification required by Ecology is that diatoms be identified to species (or species variety, if possible), and non-diatoms (soft-bodied algae) be identified to genus.

Personnel responsibilities

One person or more performs this activity. Staff performing this method must have been trained.

Summary of procedure

Sample processing and sub-sampling

Samples are homogenized in a blender to break up large colonies and filaments and to evenly distribute individual cells. A sub-sample is transferred to a Palmer-Maloney counting chamber, and 300 live algae cells are counted at 400X. The sub-sample is diluted or concentrated to achieve an optimum concentration of 10-20 cells per field. For small filamentous cyanobacteria (e.g., *Phormidium*) and for coenocytic algae that lack cell walls (e.g., *Vaucheria*), the counting unit will be a 10-micron length of filament. Diatoms are identified only as diatoms; soft (non-diatom) algae are identified to genus, and to species if possible. The number of dead (empty) diatom frustules is also recorded on the bench sheet during the count of 300 live cells.

After the identification of soft algae, the homogenized sample is re-agitated and a portion of the raw sample is extracted to make duplicate diatom slides. The diatom sub-sample is cleaned of organic matter using sulfuric acid, potassium dichromate, and hydrogen peroxide. Following several dilutions with distilled water, two permanent diatom slides are prepared from cleaned material using Naphrax, a high refractive index mounting medium. At least 300 diatom cells (600 valves) are counted at random and identified to species, and variety if possible. Diatom naming conventions should follow those adopted by the Academy of Natural Sciences (Philadelphia) for USGS NAWQA samples (Morales and Potapova, 2000) as updated in 2005 (Morales and Charles, 2005) http://diatom.ansp.org/nawqa/Workshops.aspx. Separate bench sheets will be used for algae and diatom counts, and these will provided to Ecology when samples are completed each year.

Counts of soft algae and diatoms will be conducted with compound research microscopes using brightfield transmitted light or differential interference contrast. Proportional counts will be performed using 100X oil immersion objectives and 10X oculars. After enumeration, the "voucher" slides will be prepared for storage and sent to Ecology's RSMP coordinator. These voucher slides may be used for an additional validation step if so decided by the RSMP.

Remaining sample volume will be re-preserved and stored by the contract laboratory. Contractor storage of sample fractions may be as long as six months. Ecology may request that a subset of samples be returned for quality assurance purposes. Non-requested sample fractions will be appropriately discarded by the contractor.

Sample submittal and data turn-around

Samples will be collected during the Index period, between early June and late October, and will be shipped to the contractor laboratory no later than December 1 in the year sampling occurred. Contract language with the laboratory will outline responsibility for costs of shipping samples and forms to the contract laboratory. The processing of samples and reporting of data will be completed within 90 days of the receipt of the samples by the contract laboratory. Data reported by the laboratory will consist of:

1. The original (signed) chain-of-custody form.
2. A taxonomic matrix for each sample based on the required level of identification. (Excel or Access format).
3. Metric calculations (Excel or Access format).
4. Copies of bench sheets used by the laboratory.
5. Copies of all QA/QC documentation.

Reporting requirements

Instructions for submitting periphyton data to the Department of Ecology's Environmental Information Management (EIM) are outlined in Periphyton Identification and Numeration (Counts) EIM Data Entry. Briefly, a taxa list for each sample should be reported with the associated count number for each taxon. The following should accompany this list:

- Sample ID.
- Surface area sampled (cm^2).
- volume of sample sent to laboratory, including preservative (mL).
- volume of sample counted by laboratory (mL).
- Taxon name.

The contract laboratory will analyze the taxonomy matrix for each sample and summarize with a suite of periphyton metrics. In most cases, diatoms and non-diatoms will be analyzed separately with a few exceptions (combined metrics) outlined below. This list represents the minimum metric reporting requirements. The contractor is encouraged to provide additional metrics if available and relevant.

Combined metrics

1. Total number of algal divisions.
2. Total number of algal genera.
3. Total number of algal species.

Diatom metrics

1. Total number of diatom genera.
2. Total number of diatom species.
3. Shannon Diversity Index.
4. Pollution Index (Lange-Bertalot, 1979; Bahls, 1993; Barbour, et al., 1999).
5. Siltation Index (% *Navicula* + *Nitzschia* + *Surirella*) (Bahls, 1993; Barbour, et al., 1999).
6. Percent live diatoms (Hill, 1997).
7. Valves counted.
8. Cells counted.

9. Percent dominant species.
10. Pollution tolerance (% by category).
11. Disturbance index (Bahls, 1993; Barbour, et al., 1999).
12. Percent rhopalodiales.
13. Percent aerophiles.
14. Percent centrics.
15. Motility index.

Van Dam diatom metrics (% in each category)(Van Dam, et al., 1994)

1. pH.
2. Salinity.
3. Nitrogen uptake.
4. Oxygen demand.
5. Saprobity.
6. Trophic state.
7. Moisture.

Non-diatom metrics

1. Dominant phylum.
2. Percent 5 dominant genera.
3. Percent 10 dominant genera.
4. Total number of genera (within each non-diatom algal division).
5. Total number of non-diatom genera.

D-5. Instructions for estimating periphyton cell densities from samples collected using Ecology protocols

Information needed to estimate periphyton density (cells · cm^{-2}):

1. # cells counted in 'wet count' – this information is obtained from the processing lab and includes counts of 'soft algae' (i.e., green algae and cyanobacteria) and diatoms (number of diatoms counted only and not identified to genus/species).
2. Volume of sample collected (mL) – this is the volume of algal slurry collected and sent to processing lab. This is total volume of sample with preservative.
3. Volume of sample counted (mL)* – this information is obtained from the processing lab and represents the volume of sample counted in a counting chamber. This volume depends on the counting chamber used and number of fields of view counted.
4. Surface area scraped (cm^2) – this is estimated by person(s) collecting the sample as outlined above (C-3).

Wet count:
Typically, a minimum of 300 cells are enumerated using a 'wet mount' using a Palmer-Maloney counting chamber at 400× magnification. Soft algae (green algae and cyanobacteria) and diatoms (number of diatoms counted only and not identified to genus/species) are counted until a minimum of 300 cells/units has been reached. For filamentous green algae and cyanobacteria, one unit is each 10 µm length of that filament in the field of view. Additionally, the number of empty diatom cells may also be counted by the processing lab, but are not used to estimate totaldensity.

Estimating density requires that the volume of sample counted is given by the processing lab (example below).

*Equation used to determine mLs counted using Palmer-Maloney counting chamber:

((FeildsOfView]*0.307*0.4)*0.001)

where: 0.307 = area of field of view
0.4 = depth of counting cell
0.001 – to bring mm^3 up to mL

Diatom count:
Typically, a minimum of 600 diatom valves are identified to species and enumerated by the processing lab. After a portion of the algal slurry from a particular site/reach has been processed to remove diatom chloroplasts (this process also removes organic matter including soft algae), a drop of solution containing cleaned/empty diatom frustules is placed on a coverslip, where they are heat fixed and mounted on a slide with Naphrax™. Cells are enumerated with an oil objective at 1000× magnification. Information from this count of diatom valves will be used to calculate the relative proportion of diatoms attributed to each species (e.g., if total valves counted = 600 and valves of *Achnanthidium minutissimum* = 300, then relative proportion of *A. minutissimum* in the sample = 300/600, or 0.5).

Relative proportion for each diatom species is then used to estimate the number of diatom cells attributed to each species in the 'wet count'. For example, if 100 of 324 total cells counted in a Palmer-Maloney cell were diatoms, then 50 of them would be *A. minutissimum* using the value in the previous paragraph, and the remaining 50 diatoms cells would be split by the relative proportions of the remaining diatom species enumerated in the 'diatom count'.

EPA method for calculation of cell densities:
Cell densities (cells · cm^2) are determined by dividing the numbers of cells counted by the proportion of sample counted and the area from which samples were collected.

Example:

Total # cells/units counted in 'wet count' = 324
mls of sample sent to processing lab = 35
mls counted = 0.0007368
surface area scraped (cm^2) = 110.25

Total density (cells · cm^2) = (324/(0.0007368/35))/(110.25) = 139,599.8 cells · cm^{-2}

Morales, E. A., & Potapova, M. (2000). Third NAWQA Diatom Taxonomy Harmonization Workshop. *Patrick Center for Environmental Research. The Academy of Natural Sciences of Philadelphia. Report No. 00-8.*

Morales, E. A., & Potapova, M. (2005). Fifteenth NAWQA Workshop on Harmonization of Algal Taxonomy. *Patrick Center for Environmental Research. The Academy of Natural Sciences of Philadelphia. Report No. 06-07.*

Appendix E. Water quality sampling procedures

E-1. Day of sample collection

Samples and measurements should be collected from well-mixed and representative locations within the reach. The methods are summarized below:

- Dissolved oxygen, temperature, conductivity, and pH are measured in-situ with field meters. For the RSMP, dissolved oxygen will be measured by an LDO field meter, and not titrated by the Winkler method as described in SOP EAP034.
- Turbidity, total suspended solids, and nutrient samples are measured in a laboratory in samples collected using either a bucket collection or hand-dipped bottles.
- Fecal coliform samples are measured in a laboratory in samples collected with the flow-orienting bacteria sampler or hand-dipped using an autoclaved bottle.

Field processing of samples fulfills three essential purposes: (1) preserve (fix) samples, (2) prepare samples for shipment to the lab, and (3) conduct the first quality control checks (e.g., completeness of sampling).

Field meters

Multiprobe meters may be used to make in-situ field measurements. Methods for use of these multi-meters are described in Appendix D; use of meters will follow the manufacturer's website instructions for the most up-to-date guidelines.

On the day of sampling, field staff will calibrate the meters/probes as follows:

- For pH, using a two-point calibration with NIST-certified standards. Most small streams west of the Cascades or in moderate to high elevations will need to be calibrated with pH 7 and pH 4 standards. A 10 standard may be used as a linearity check. r.
- For conductivity, using a one-point calibration with NIST-certified 100 uS/cm conductivity standards. A zero conductivity check will also be performed.
- For dissolved oxygen, a water saturated air calibration method is suitable, following recommendations by the manufacturer.
- For temperature, probes must be factory calibrated. Instead of calibration, probes will be checked against a NIST-certified thermometer prior to the start of the project and at the end of the project.
- Recording the barometric pressure within an hour of sampling, recording results on the field data form, and noting the location (and elevation) of the barometric meter.

E-2. Water quality sample containers

For all samples, pre-cleaned sample containers will be used. For many of the sediment organic contaminants samples, the homogenized sample will be placed in the appropriate glass jars, which are supplied pre-cleaned by the laboratory to EPA QA/QC specifications (EPA, 1990) and that carry a certification of cleanliness from the suppliers. The sample container shipment documentation will record batch numbers for the containers.

E-3. Water quality sample processing and preservation

Some of the parameters to be analyzed (ammonia, nitrate-nitrite, total nitrogen, and total phosphorus) require chemical preservation to maintain the integrity of the samples and prevent them from degrading prior to laboratory analysis. Other parameters require filtration in the field.

Field filtered parameters

Orthophosphate

Filtration is required for orthophosphate within 15 minutes after sample collection. Samples for orthophosphate will be filtered using a disposable syringe and filter. Prior to filtering the sample, an aliquot of field sample will be passed through the syringe and filter as a rinse. After rinsing, the filtered sample will be collected and distributed into the laboratory sample bottles. Disposable filter set-ups may be used for each sample. A field filtration blank is prepared by bringing 250 mL of deionized water from the lab and filling the syringe in the same manner as a field sample.

Dissolved Metals

Filtration for dissolved metals will be done using disposable filtration units that are operated by a separate vacuum pump (peristaltic or hand pump). The water to be filtered will typically be collected as a grab sample using a separate 500 mL metals sample container (that is later discarded). The filtration units are pre-cleaned before use thus rinsing with extra field sample is not needed. Once filtered, the collection vessel of the filtration unit is removed, or sample is transferred to appropriate container, and labeled. A field filtration blank is prepared by bringing 500 mL of deionized water from the lab and filling the filtration unit in the same manner as a field sample.

Dissolved Organic Carbon

Samples for dissolved organic carbon will be filtered using a disposable syringe and filter. Prior to filtering the sample, an aliquot of field sample will be passed through the filter to rinse the filter and syringe. After rinsing, the filtered sample will be collected and distributed into the pre-preserved dissolved organic carbon laboratory sample bottles. Disposable filter set-ups may be used for each sample. A field filtration blank is prepared by bringing 250 mL of deionized water from the lab and filling the syringe in the same manner as a field sample.

For all filtered samples, filtering should occur within 15 minutes after sample collection. If filtering occurs after 15 minutes and before 24 hours, the sample will be J qualified. If field filtering occurs after 24 hours for orthophosphate, the sample will be rejected and labeled with an *R* on the field forms. Field sampling efforts, including time of collection and time of filtration and other activities, will be documented on a field sampling form.

Preservation

Sample cooling to 4º to 6ºC or less, but not freezing, is necessary for preservation of most of the parameters to be analyzed. Collected samples must be transferred from the field station to the lab in an ice-filled or blue-ice-filled cooler to maintain temperature requirements.

E-4. Sample collection for the Inter-laboratory Comparison Study

Water samples selected nutrients, turbidity and dissolved metals will be collected at 6 stations during 2 separate sampling events (one winter and one spring or summer season). Split samples for PAH analysis will be collected at 6 stations during a winter event only.

The sample for Ammonia, Nitrate/Nitrite and Turbidity will be collected as a grab sample using a 1 Liter polypropylene bottle that has been properly cleaned. This sample will mixed multiple times by inverting the bottle then used to quickly fill duplicate sample containers then preserved as appropriate. Dissolved metals will be sampled by splitting the filtrate from the collection unit into separate 500 mL clean metals bottles. No acid preservation will be done until the samples arrive at the lab, when equivalent portions of nitric acid is added to each bottle.

The PAH samples can’t be collected in a common container and then split since the parameters will stick to this container and potentially bias the final results. These samples must therefore be collected as individual grab samples into separate bottles. The 2 containers should be dipped simultaneously and as close together as possible.

E-5. Stage Height and Stream Discharge Measurement

Purpose and scope

This method describes how to collect stage height and flow data necessary for estimating instantaneous discharge (in cubic feet per second) during each monthly visit.

Personnel responsibilities

One person performs this activity. Staff performing this method must have been trained.

Equipment, reagents, supplies

- Soft-lead pencil.
- Distance measuring device (50-m tape or measuring rod).
- Flow Meter.
- Wading rod (top setting).
- Orange or other neutrally buoyant object (if needed).
- 5-gallon bucket.
- Stop watch.
- Field notebook.
- Calculator.

Preferred approach for measuring and estimating stage and stream flow for the RSMP sites

This section provides guidance for the RSMP contractors to follow when measuring stage (water-surface elevation) and stream flow during the monthly water quality sampling trips. Some detail on how to take a discharge measurement is provided below; however each agency is assumed to be capable of measuring discharge in a wadeable stream.

Stage

Every month a stage height measurement will be made. During a site visit, choose a suitable location where you can measure the stage every time to visit the site. A measurement of the stage is best done by installing and reading a staff gage at the site. However, any stable measurement point you can either install (rebar, T-post, staff gage, etc.) or is already there (bridge deck or railing, vertical armored wall, large rock, etc.) will work. The important thing is to measure the stage relative to the same point every time. Note that stream depth is generally not a reliable measure of stage since the stream bed can change over time. However, if there is a stable in-channel feature that acts as a control (bedrock or a cement weir, or culvert for example) where you can measure the depth in the same place every time, then that works.

A pressure transducer may also be installed at the site to measure stream depths. A manual stage measurement point is still needed so data are available to confirm/correct the pressure transducer data.

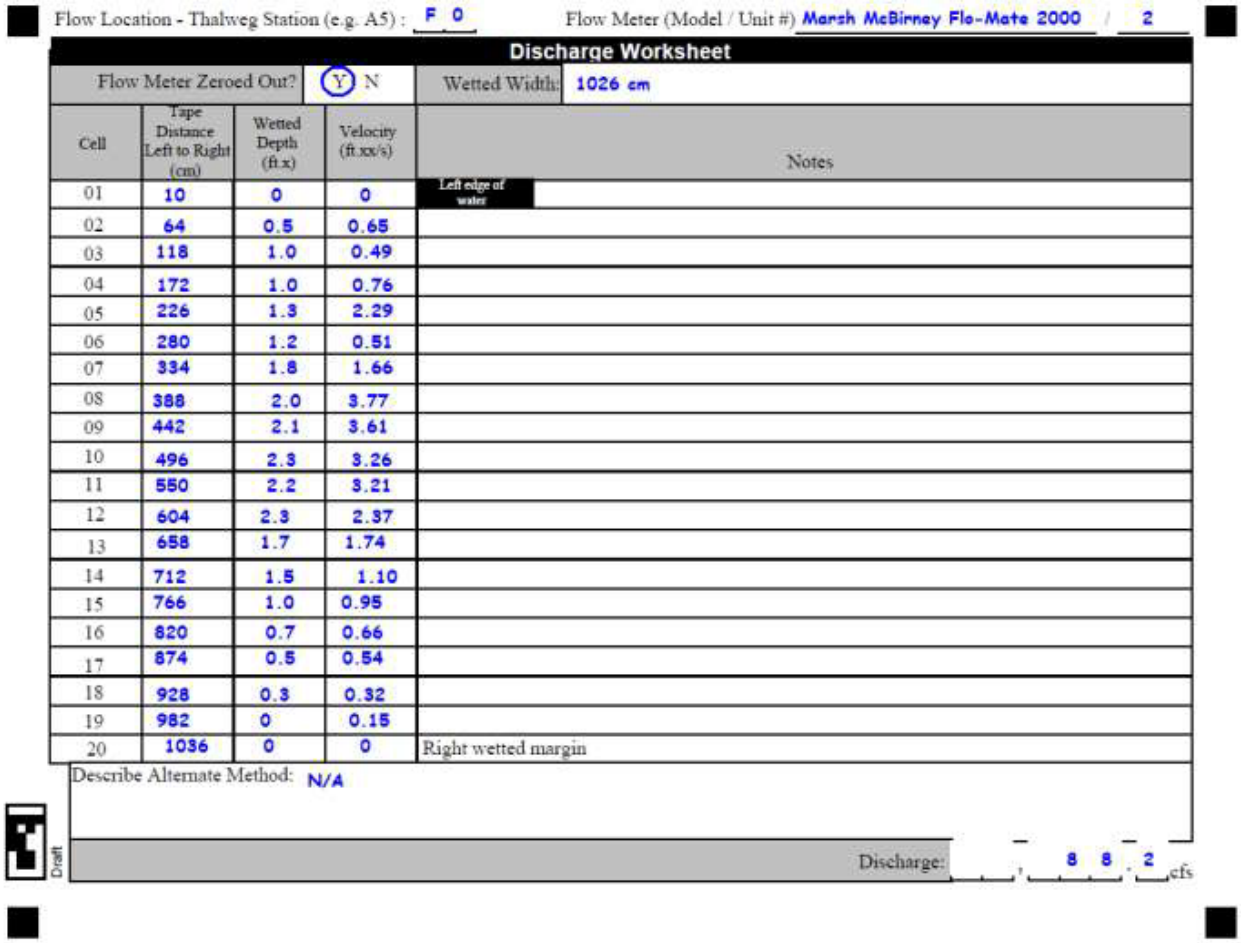
Stage data will be used to develop a rudimentary stage-discharge curve for each site to estimate flow for the visits when only stage was measured. These discharge estimates will be highly

uncertain because the nature of the stage-discharge relationship at high stages is difficult to predict. The relationship tends to have the most error at the higher end, which is when you will likely have only stage, but is the better than no estimate.

Velocity- Area Discharge Measurement

For most months of the year the sites should be wadeable and stream flow measured. Discharge is normally measured near the index station ("X") where there is uniform (non-turbulent) flow, but can be done anywhere in the reach. For method references see SOP EAP024 (Kardouni, 2013) and Kaufmann (2006). For operation of the flow meter, refer to the manufacturer's manual.

Use the Discharge Worksheet (Figure C-3.1), located on the back of the *Chemistry and Sampling Form*. Discharge can be calculated by converting widths to units of feet (nearest tenth) and applying the QWIN program (Larsen, 2005) as provided by PSNS&IMF (2006).



Flow Location - Thalweg Station (e.g. A5) : F 0 Flow Meter (Model / Unit #) Marsh McBirney Flo-Mate 2000 / 2

Discharge Worksheet

Flow Meter Zeroed Out? (Y) N Wetted Width: 1026 cm

Cell	Tape Distance Left to Right (cm)	Wetted Depth (ft.x)	Velocity (ft.xx/s)	Notes
01	10	0	0	Left edge of water
02	64	0.5	0.65	
03	118	1.0	0.49	
04	172	1.0	0.76	
05	226	1.3	2.29	
06	280	1.2	0.51	
07	334	1.8	1.66	
08	388	2.0	3.77	
09	442	2.1	3.61	
10	496	2.3	3.26	
11	550	2.2	3.21	
12	604	2.3	2.37	
13	658	1.7	1.74	
14	712	1.5	1.10	
15	766	1.0	0.95	
16	820	0.7	0.66	
17	874	0.5	0.54	
18	928	0.3	0.32	
19	982	0	0.15	
20	1036	0	0	Right wetted margin

Describe Alternate Method: N/A

Draft

Discharge: 8 8 . 2 cfs

Figure E-4.1. The Discharge Worksheet with example field data in blue.

Establish the cross-section

The velocity-area method is used at a transect location within the site that has the most of these conditions (based on Rantz, et al., 1982).

- The stream is straight.
- Depths are mostly greater than 0.5 ft (15 cm).
- Velocities are mostly greater than 50 ft/s (0.15 ms/s).
- Local habitat is not a pool.
- The channel is "U-shaped".
- The streambed is uniform and free of objects that cause turbulence.

Preference should be given to locations that are close to "X". Record the name of the nearest *Thalweg Transect*. Pull a measuring tape taught, perpendicular to the stream, and parallel to the stream surface (a measuring rod can be used for small streams). Record the tape value (cm) at the left wetted margin and at the right wetted margin. Subtract the left value from right value to determine the transect's wetted width. Record wetted width (cm) on the worksheet (Figure C-3.1).

Measure distance, depth, and velocity

Define about 15-20 equally spaced stations across the stream (possibly fewer for very small streams). To determine spacing between stations, divide the width by 20 and round up to a convenient number. Stations should not be closer than 10 cm to each other, even if this results in less than 15 stations. The first station is located at the left wetted margin, and the last station is located at the right wetted margin.

Use a calibrated flow meter equipped with a top-setting wading rod that has depth increments in tenths of feet. At each station, record the tape distance (cm) from left to right. Record the water depth (nearest 0.1 ft). Place the sensor 60% of the distance down from the surface (Figure C-3.2). Measure and record water velocity (nearest 0.01 f/s).

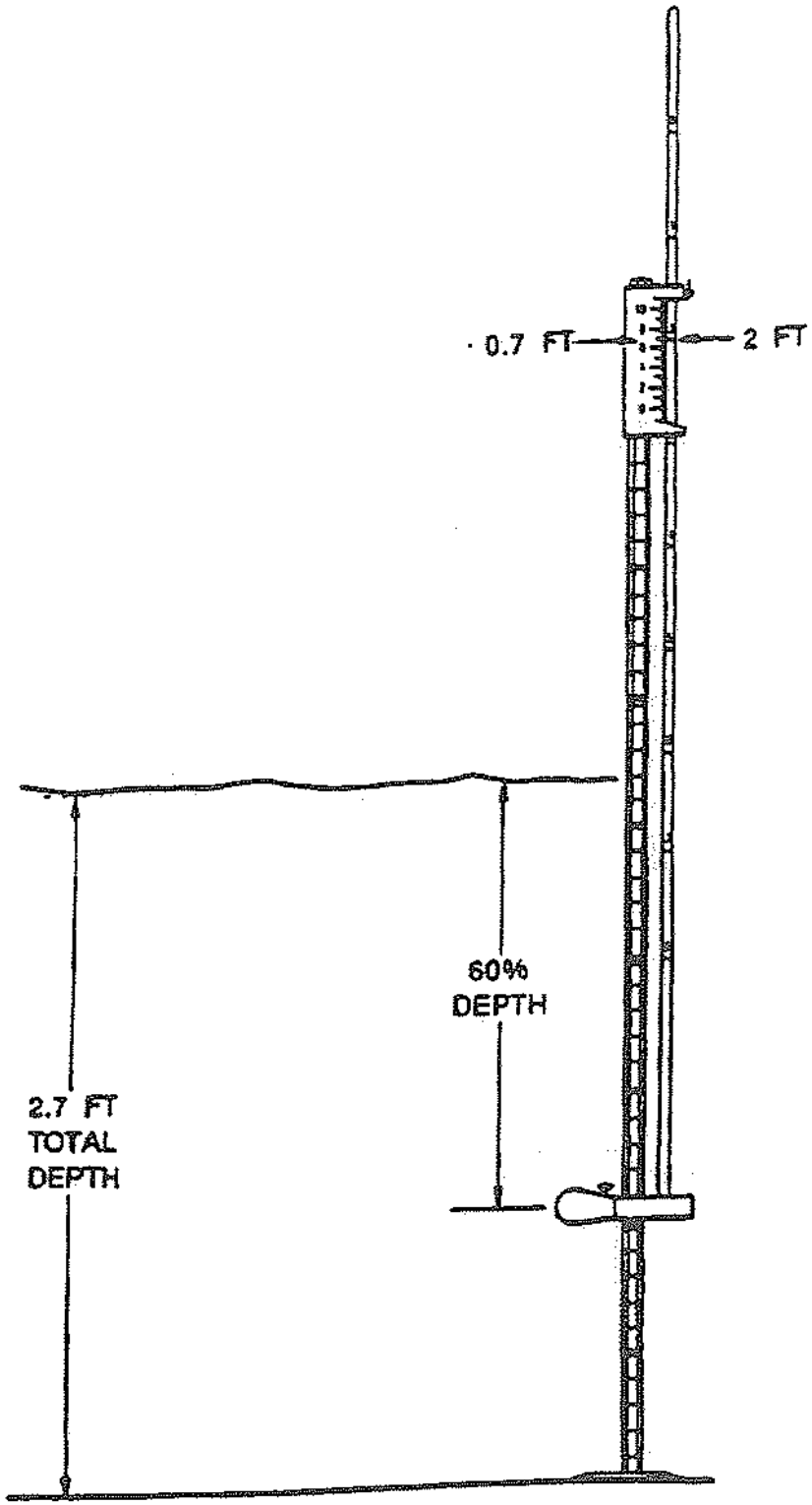


Figure E-4.2. Setting the wading rod at 60% depth when at a station that is 2.7 meters deep.

There may be some months when stream flow is too high or too low to measure directly. The following alternate methods are provided for those occasions.

- High flow – At a minimum, measure the stage as described above. If possible, estimate the mid-stream velocity using floating debris.
- Low flow – Measure the stage. If the flow is too low to measure discharge with your meter, try to estimate the flow volumetrically using a bucket and stopwatch or similar. Locate a place on the reach where flow is focused and can be collected into a bucket (e.g., hanging culvert). You might need to move some rocks to focus flow. You can also try to attempt to add a temporary weir for the collection, but it is not advised you leave this in place in order to not disturb the stream too much, especially around the habitat sampling.

Alternate methods

Mid-stream velocity using timed float

In the absence of a current meter, you can time the transport of a neutrally-buoyant object (e.g., oranges, plastic golf balls, sticks).to estimate velocity. This method is similar to the Velocity-Area method because discharge is calculated as the product of water velocity and the stream cross-sectional area. Requirements are:

- The object must float, but very low in the water.
- The object must be small enough to *not* drag bottom.
- The segment must be somewhat strait, uniform, and non-turbulent.
- The segment must be long enough that it takes 10 to 30 seconds for the float to pass.

Velocity

Compute water column velocity in a field notebook. Determine the average time (seconds) for the float to travel the segment. Repeat twice more, each time releasing at a different position across the width of the stream. Compute an average for the three times. Measure the length of the segment (ft). Divide the segment length by the average time of travel(s) to estimate surface velocity (ft/s). Multiply this surface velocity by 0.85 to estimate water column velocity.

Cross-sectional area

Compute cross-sectional area (ft^2) in a field notebook. This can be done by summing the area for at least two trapezoids to approximate the cross section of the stream (Figure C-3.3). These should be centered on the thalweg.

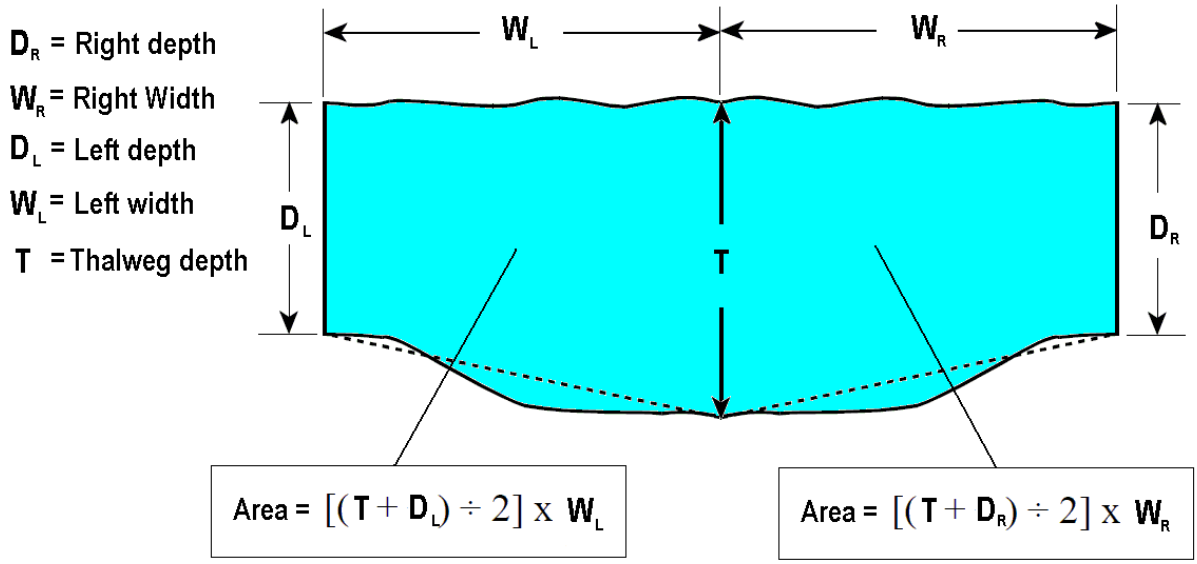


Figure E-4.3. The cross-sectional area of a stream segment as estimated by calculating the area of component trapezoids, centered on the thalweg.

Measure area for one or more cross-sections and average them. Only one cross-section is adequate if the channel is relatively uniform through the segment. Otherwise measure at these cross-sections:

- Near the top of the segment.
- Near the middle of the segment.
- Near the bottom of the segment.

If there is little change in channel width or depth, obtain measurements from a single "typical" cross-section within the segment.

Discharge

Convert cross-sectional area calculations to square feet (1 m^2 = 10.76391 ft^2). Then multiply water column velocity (ft/s) times the cross-sectional area (ft^2) to determine stream discharge for the site. Record this discharge (cfs) on the bottom of the discharge worksheet (Figure C-3.1). Also record "timed float" next to "Describe alternate method".

Timed bucket-filling

Place a bucket or other container with known volume below the discharge. Time how long it takes to fill the container. Repeat at least three times. Calculate discharge as the volume of the container divided by the average time to fill it. Use Table C-3.1 to translate from gallons or milliliters to cubic feet. Record discharge (cfs) at the bottom of the Discharge Worksheet (Figure C-3.1). Also record that the alternate method was by use of a timed bucket-filling.

Table E-4.1. Conversions for gallons or milliliters to cubic feet.

Gallons	Milliliters	Cubic Feet
0.1321	500	0.0176573
0.2642	1,000	0.0353147
1	3,785	0.1336806
5	18,927	0.6684028
7.480519	28,317	1

Existing gage data

If a nearby USGS, Ecology, or County gage is active, record discharge (cfs) at the bottom of the Discharge Worksheet (Figure C-3.1) and note the data source, next to "Describe alternate method".

For these sites located on the same stream as an existing gage, calculate the corresponding unit area discharge at the gaged location and adjust proportionally to estimate discharge at the ungagged site. Uncertainty due to changes in flow due to time-of-travel between the two gages will likely be very small relative to uncertainties in the proportional adjustment of discharge.

Appendix F. Quality control procedures

F-1. Quality control for in-situ meters

Purpose and scope

This method explains how to verify that in-situ meters used for the water quality monitoring are working properly. This section was written for certain meters used by Ecology and is provided as an example of a QC program for meters for permittees conducting monitoring. Permittees are not expected to conduct Winkler titrations for verification of dissolved oxygen measurements as is done by Ecology (Adams, 2010a and SOP EAP034).

Instruments included in the procedure include probes for measuring temperature, pH, conductivity, and dissolved oxygen (Minisonde Multiprobes). It also includes the instrument used for measuring water velocity (Marsh-McBirney FloMate-2000).

Personnel responsibilities

This method is performed by 1 or more persons. This method is applied at every DCE, before sampling. Staff performing this method must have been trained.

Equipment, reagents, supplies

- No. 2 pencil.
- Calibration Form.
- Flow Meter.
- Flow Meter batteries.
- Wading rod.
- Flow Meter Manual (e.g., Marsh-McBirney, 1990).
- Five-gallon bucket (for flow meter zero-adjust).
- Hydrolab, YSI (or equivalent), components, maintenance kit (Swanson, 2007).
- Multimeter Manuals.
- pH 7 buffer (7.00) – e.g., VWR - 23197-996.
- pH 4 buffer (4.01) – e.g., VWR - 23197-998.
- pH 10 buffer (10.01) – e.g., VWR - 23197-994.
- Conductivity Standard (100 uS) – e.g., VWR 23226-589.
- Conductivity Standard (1,000 uS) – e.g., VWR 23226-603.
- Conductivity Standard (alternate as available).
- Deionized water (DI).
- Tap Water.
- Lab tissues (e.g., KimWipes®).
- Barometer.

Summary of procedure

Calibrate (conductivity, pH, dissolved oxygen, and velocity)

Calibration of multiprobe meters like Hydrolab will be conducted based on EAP033 (Swanson, 2007). Use the *Calibration Form* (Table F-1.1) to record calibrations and quality control checks. Each day, calibrate conductivity (COND), pH, dissolved oxygen (DO) and velocity. The DO should be calibrated on-site or near the site, to match local barometric pressure of calibration and sampling. The pH and conductivity calibration standards should be chosen to bracket expected values. For example, most small streams west of the Cascades or in moderate to high elevations will need to be calibrated with pH 7 and pH 4 standards; they will need to be calibrated with 0 and 100 uS conductivity standard. Many larger streams and rivers will need to be calibrated with pH 7 and pH 10 standards. The order of calibration is normally:

1. COND (Hydrolab)
2. pH (Hydrolab)
3. DO (Hydrolab)
4. Velocity (zero-adjust the flow meter)

Before calibrating, make sure that a post-sampling QC check measurement has been made to verify the quality of sampling at the previously sampled site. QC checking is discussed in detail later in this document.

Clean the Hydrolab Sonde and the Flow Meter Sensor
Refer to Swanson (2007) section 6.3 and to Marsh-McBirney 1990.

Rinse Hydrolab Sonde Between Each Operation
Rinse three times with tap water, three times with deionized water, then three times with the solution to be used for calibrating or testing.

Table F-1.1. An example Meter Calibration Form, with examples data records.

Recorder:

Project Name: _____ **Date:** _____

Sonde #: _____ **Time** _____

Temp: _____ **Barometric Pressure:** _____

PRE Field Run CALIBRATION

	Meter Reading	Buffer Value	Buffer Temp	Comments
Conductivity-100 µS/cm	*94*	*100*		*post-calibration reading was unstable in buffer; ranging between 95 - 105*
Conductivity-1000 µS/cm				
pH - 7				
pH - 10				
pH - 4				*linearity check; not calibrated to buffer value*
DO % Saturation				
Temperature check with NIST thermometer				

POST Field Run Check

	Meter	Buffer	Comments
Conductivity-100 µS/cm	*98*	*100*	
Conductivity-1000 µS/cm			
pH - 7			
pH - 10			
pH - 4			
DO % Saturation			

Calibrate conductivity to bracket expected field conductivity:

1. Dry the conductivity probe with a lab tissue (e.g., KimWipes®).
2. Using the Surveyor, enter 0 into SpCond, to dry calibrate to 0.
3. Fill the calibration cup to within a centimeter of the top of the calibration cup with dilute standard (either 100 uS or 1,000 uS) so that the probes are covered.
4. Make sure there are no bubbles in the cell; wait 2 minutes.
5. Using the Surveyor, enter the appropriate value for the standard into SpCond.

Table F-1.2. Theoretical pH values by temperature for each pH standard buffer.

Temp (˚C)	4[a]	7[b]	10[c]
4	4.00	7.09	10.26
5	4.00	7.08	10.25
6	4.00	7.08	10.23
7	4.00	7.07	10.22
8	4.00	7.07	10.21
9	4.00	7.06	10.2.0
10	4.00	7.06	10.18
11	4.00	7.05	10.17
12	4.00	7.05	10.16
13	4.00	7.04	10.14
14	4.00	7.04	10.13
15	4.00	7.03	10.12
16	4.00	7.03	10.11
17	4.00	7.02	10.10
18	4.00	7.02	10.09
19	4.00	7.02	10.08
20	4.00	7.01	10.06
21	4.01	7.01	10.05
22	4.01	7.01	10.04
23	4.01	7.00	10.03
24	4.01	7.00	10.02
25	4.01	7.00	10.01
26	4.01	6.99	10.00
27	4.01	6.99	9.99
28	4.01	6.99	9.98
29	4.01	6.99	9.98
30	4.02	6.98	9.97

Buffers: a Thermo 7.00, b Thermo 4.01, c Thermo 10.01.
From: www.thermo.com/com/cda/resources/resources_detail/1,2166,13217,00.html

Calibrate pH to bracket expected field pH

1. Pour the pH 7 buffer into the calibration cup to cover the sensor and reference electrode. Enter the theoretical pH value units into the Surveyor. Theoretical values are based on temperature of the standard and are listed in Table F-1.2.
2. Rinse and repeat step 1, using either the pH 10 buffer (when sampling in basic waters) or pH 4 buffer (when sampling in acidic waters).
3. On the calibration form, record the temperature and theoretical pH values that were used to calibrate. Also record adjustments that were needed to calibrate to these theoretical values.
4. On the calibration form, record the percent slope of the calibration (displayed on the Surveyor). Be sure this percent slope matches the criteria described on the form. Otherwise, recalibrate.

Calibrate DO using water-saturated air

1. Fill the calibration cup with about 1/2 inch of DI; it should be below the sensor cap.
2. Use KimWipes® to dry any droplets from the sensor cap.
3. Invert calibration cup's cap and gently rest it on the cup.
4. Wait 5 minutes, making sure that temperature stabilizes.
5. Determine local barometric pressure (mm Hg) and enter this value into the Surveyor.
6. Click "Calibrate". A "Calibrate Successful" will be displayed.

DO calibration notes:

1. To retain calibration accuracy between measurements, store with the sensor immersed in water or within a water-saturated air environment such as a sealed storage cup with at least 10 ml of water.
2. It is important to have the water-saturated air and the sensor at the same temperature. Therefore, store a jar of DI in the same environment as the sonde and calibrate in a similar air temperature as the water and sonde.
3. Stay out of direct sun or wind.
4. Refer to Table F-1.3 if necessary.

Table F-1.3. Unit conversions for pressure.

Atmospheres	Bars	Hg mm	inches Hg
1	1.01325	760	29.92126
0.9869233	1	750.0617	29.52999
0.001315789	0.001333224	1	0.03937008
0.03342105	0.03386388	25.4	1

Zero-adjust the flow meter

Zero the flow meter prior to each use. Refer to Marsh-McBirney 1990 (pages 8-9).

Quality control

Daily checks

Check pH and conductivity at the start and end of each DCE by measuring the quality control calibration standard (QCCS). Record the temperature of the QCCS too. *The pH should measure between 6.78 and 7.18 pH units.* Conductivity should measure between 65.3 and 85.3 uS/cm at 25 ˚C. Re-calibrate if the presampling check fails these criteria. Data from the DCE should be qualified if the post-sampling check fails these criteria. Record measurements on the *Calibration Form Monthly Checks.* Once monthly, check the accuracy of the DO sensor on the Hydrolab. Collect a Winkler sample at the same location and time as an in-situ DO reading. Winkler samples are collected and analyzed according to Mathieu (2007).

Twice-seasonal checks

Before and after the season, check the regular pH calibrations against dilute pH standards:

- pH 7 standard (6.97) – e.g., Thermo 700702.
- pH 4 standard (4.10) – e.g., Thermo 700402.
- pH 9 standard (9.15) – e.g., Thermo 700902.

Calibrate first with the regular buffers as for the daily calibrations (e.g., first 7 and 4), then check using the QCCS. Re-calibrate, this time using the dilute standards (e.g., 6.97 and 4.10). Measure the QCCS and compare the difference in QCCS measures between calibrations. Repeat for the high-pH calibrations (7 and 10; 6.97 and 9.15). Theoretical values by temperature for the dilute pH standards are found in Table F-1.4.

Table F-1.4. Theoretical values by temperature for the dilute pH standards.

Temp (˚C)	4[a]	7[b]	9[c]
10	4.10	7.01	9.27
11	4.10	7.01	9.26
12	4.10	7.00	9.25
13	4.10	7.00	9.25
14	4.10	7.00	9.24
15	4.10	7.00	9.23
16	4.10	6.99	9.22
17	4.10	6.99	9.21
18	4.10	6.99	9.21
19	4.10	6.98	9.20
20	4.10	6.98	9.19
21	4.10	6.98	9.18
22	4.10	6.97	9.18
23	4.11	6.97	9.17
24	4.11	6.97	9.16
25	4.11	6.97	9.16
26	4.11	6.96	9.15
27	4.11	6.96	9.14
28	4.12	6.96	9.13
29	4.12	6.95	9.13
30	4.12	6.95	9.12

a - Orion 700402,
b - Orion 700702,
c - Orion 700902

The Hydrolab's thermistor is factory-calibrated. Check the settings before and after the field season by comparing with an NIST-traceable thermometer. Verify that it measures to within 1˚ C of the thermometer reading. Do this in an ice water bath and in a warm water bath. Qualify the season's temperature data if the measures fall outside this range.

F-2. Quality control for laboratory analysis – benthos

Lab quality assurance samples

Macroinvertebrate sorting efficiency

Quality control procedures for initial sample processing and subsampling involves checking sorting efficiency. These checks are conducted on 100% of the samples by independent observers who microscopically re-examine 20% of sorted substrate from each sample. All organisms that were missed are counted. Sorting efficiency is evaluated by applying the following calculation:

$SE = n_1/n_2 \times 100$

where SE is the sorting efficiency, expressed as a percentage, n 1 is the total number of specimens in the first sort, and n 2 is the total number of specimens in the first and second sorts combined. Sorting efficiency is recorded on each benchsheet, and these data are entered into the Rhithron database. If 95% sorting efficiency is not achieved for a given sample, a failure is recorded on the benchsheet and in the database. The sorted portion of that sample is then completely re-sorted before the sorting efficiency test is repeated for that sample. Sorting efficiency statistics for each technician and for the entire laboratory are reviewed monthly. Sorting efficiency for each sample in a project is reported to the client in the technical summary document. Technicians who do not maintain the target sorting efficiency are given remedial training. Larger portions of the samples they process are examined for the sorting efficiency test until they are able to maintain the target sorting efficiency.

A second evaluation of the sub-sampling process is applied to a small proportion of samples processed in each month; typically one sample per week is subjected to the following test of precision of the sub-sampling process. The procedure is only applied to samples where the target number of organisms was achieved in less than half of the Caton grids. A sample is randomly selected, and a second sub-sample is re-sorted from the unprocessed sample remnant. A second technician performs this sort. The resulting sub-sample is identified, and Bray-Curtis similarity index is calculated for the results of both sub-samples. Results that are less than 90% similar would indicate the need for more thorough distribution of sample materials in the subsampling tray or more special attention given to easily missed taxa when sorting (i.e., increased magnification).

Taxonomic accuracy and precision

Taxonomic misidentification results in inadequate biological characterization of a stream. Errors in identification should be less than 5% of the total taxa in the sample. Re-identification of samples is conducted for 10% of the total number of samples in each year. Secondary identification is conducted by experienced taxonomists in order to maintain confidence in the data set. Difficult taxa should be sent to museum curators whose specialty includes members of the order in question. A voucher collection has been maintained by Ecology and is being transferred to the Orma J. Smith Museum of Natural History in Caldwell, Idaho for curation. A voucher collection should be prepared from the set of samples for the year and shipped to the address below:

The Orma J. Smith Museum of Natural History
College of Idaho
2112 Cleveland BLVD
Caldwell, ID 83605-4432

Documentation necessary for acceptance by the museum will be delivered to the successful bidder with the samples.

F-3. Quality control for laboratory analysis – periphyton

Taxonomic Accuracy and Precision: Taxonomic misidentification of diatom and algal taxa results in inadequate biological characterization of a stream. Errors in identification should be less than 5% of the total taxa in the sample. Re-identification of samples is conducted for 10% of the total number of samples in each year. Secondary identification is conducted by experienced taxonomists in order to maintain confidence in the data set. Difficult taxa should be sent to specialists whose area of expertise includes members of the order in question.

Chlorophyll *a* analysis on the filter with periphyton will be performed in accordance with the *Manchester Environmental Laboratory's Lab Users Manual* (MEL, 2008). Laboratory staff will consult the project manager if there are any changes in procedures over timeline or if other difficulties arise.

After the filtered subsamples are taken from the composite sample, the remaining sample volume will be transferred and preserved. The periphyton sample will be sent to the same contract laboratory as the benthos sample. Contractor storage of sample fractions may be as long as six months. Post-analysis, permittees may request that a subset of samples be returned for quality assurance purposes. Non-requested sample fractions will be appropriately discarded by the contractor.

Appendix G. Field forms for ambient biological monitoring in Washington

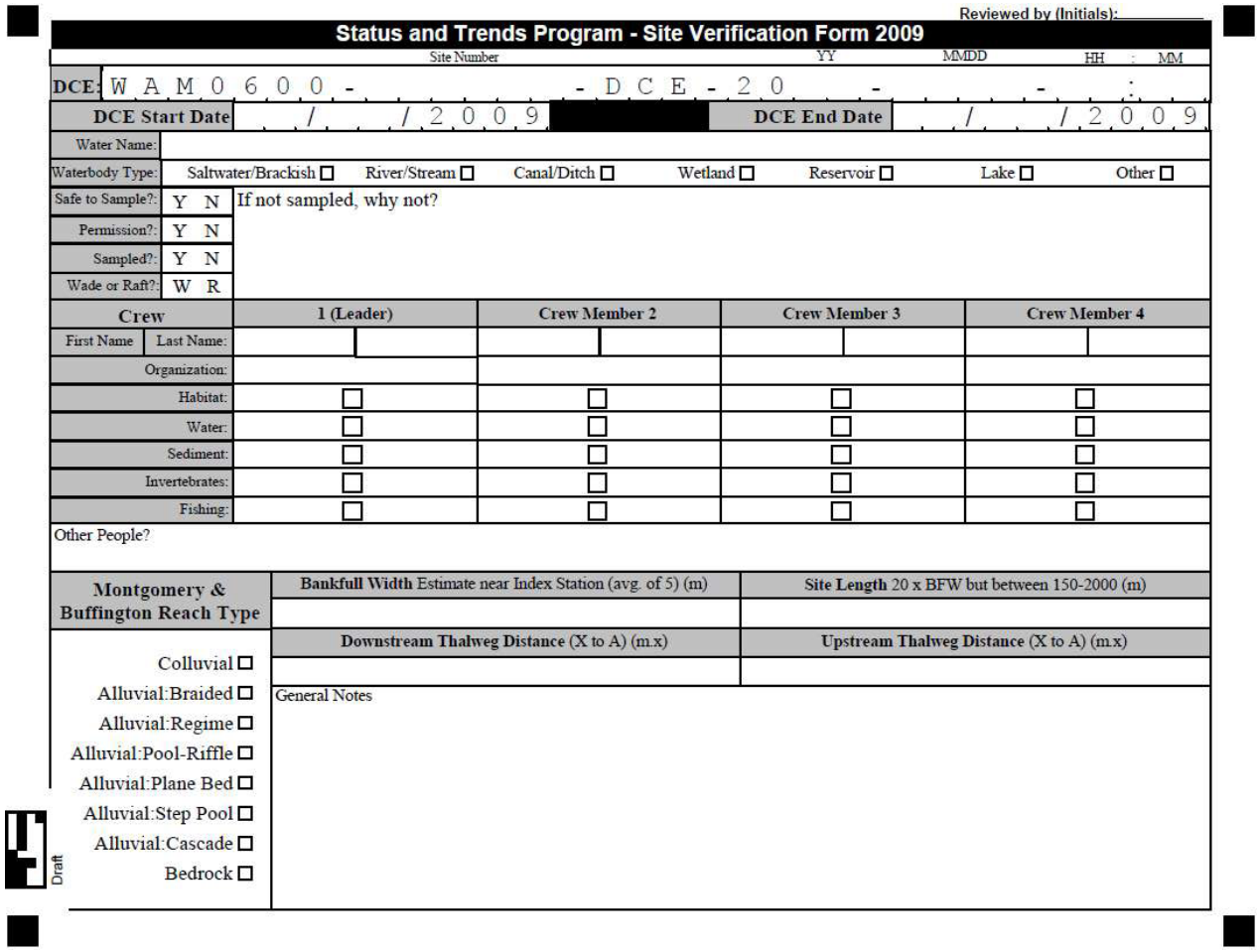
There are 8 data forms to record information in the field, for entry to Watershed Health Monitoring section of EIM. These are:

- GPS Positions Form
- Site Verification Form
- Site Diagram
- *Chemistry and Sampling Form*
- Discharge Worksheet
- Major Transect Form
- Thalweg Data Form
- Slope and Bearing Form

Table G-1. The juxtaposition of field forms and number of copies needed per data collection.

Front Side	Back side	Copies per DCE
Site Verification Form	Site Diagram	1
Chemistry and Sampling Form	Discharge Worksheet	1
Major Transect Form	Thalweg Data Form	11
Slope and Bearing Form	GPS Positions Form	1
Vertebrate Collection Form front	Vertebrate Collection Form back	1

See Figures G-1 to G-9 for each of the forms and logs.



Reviewed by (Initials): _____

Status and Trends Program - Site Verification Form 2009

Site Number | YY | MMDD | HH : MM

DCE: WAM0600 - - DCE - 20 - - :

DCE Start Date	/ / 2009	**DCE End Date**	/ / 2009

Water Name:

Waterbody Type: Saltwater/Brackish ☐ River/Stream ☐ Canal/Ditch ☐ Wetland ☐ Reservoir ☐ Lake ☐ Other ☐

Safe to Sample?: Y N — If not sampled, why not?

Permission?: Y N

Sampled?: Y N

Wade or Raft?: W R

Crew	1 (Leader)	Crew Member 2	Crew Member 3	Crew Member 4
First Name / Last Name:				
Organization:				
Habitat:	☐	☐	☐	☐
Water:	☐	☐	☐	☐
Sediment:	☐	☐	☐	☐
Invertebrates:	☐	☐	☐	☐
Fishing:	☐	☐	☐	☐

Other People?

Montgomery & Buffington Reach Type	**Bankfull Width** Estimate near Index Station (avg. of 5) (m)	**Site Length** 20 x BFW but between 150-2000 (m)
	Downstream Thalweg Distance (X to A) (m.x)	**Upstream Thalweg Distance** (X to A) (m.x)
Colluvial ☐		
Alluvial:Braided ☐	General Notes	
Alluvial:Regime ☐		
Alluvial:Pool-Riffle ☐		
Alluvial:Plane Bed ☐		
Alluvial:Step Pool ☐		
Alluvial:Cascade ☐		
Bedrock ☐		

Draft

Figure G-1. Site Verification Form.

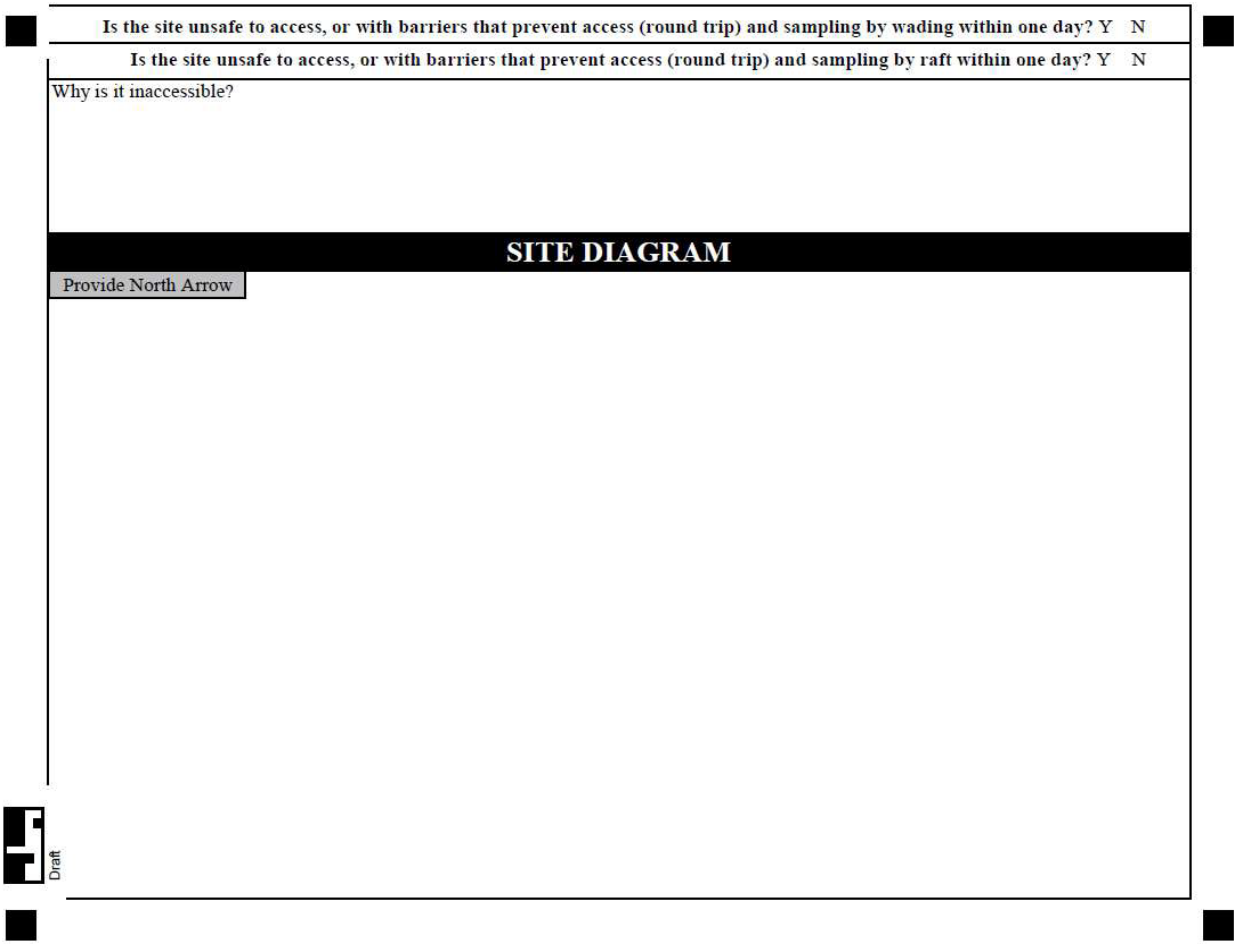
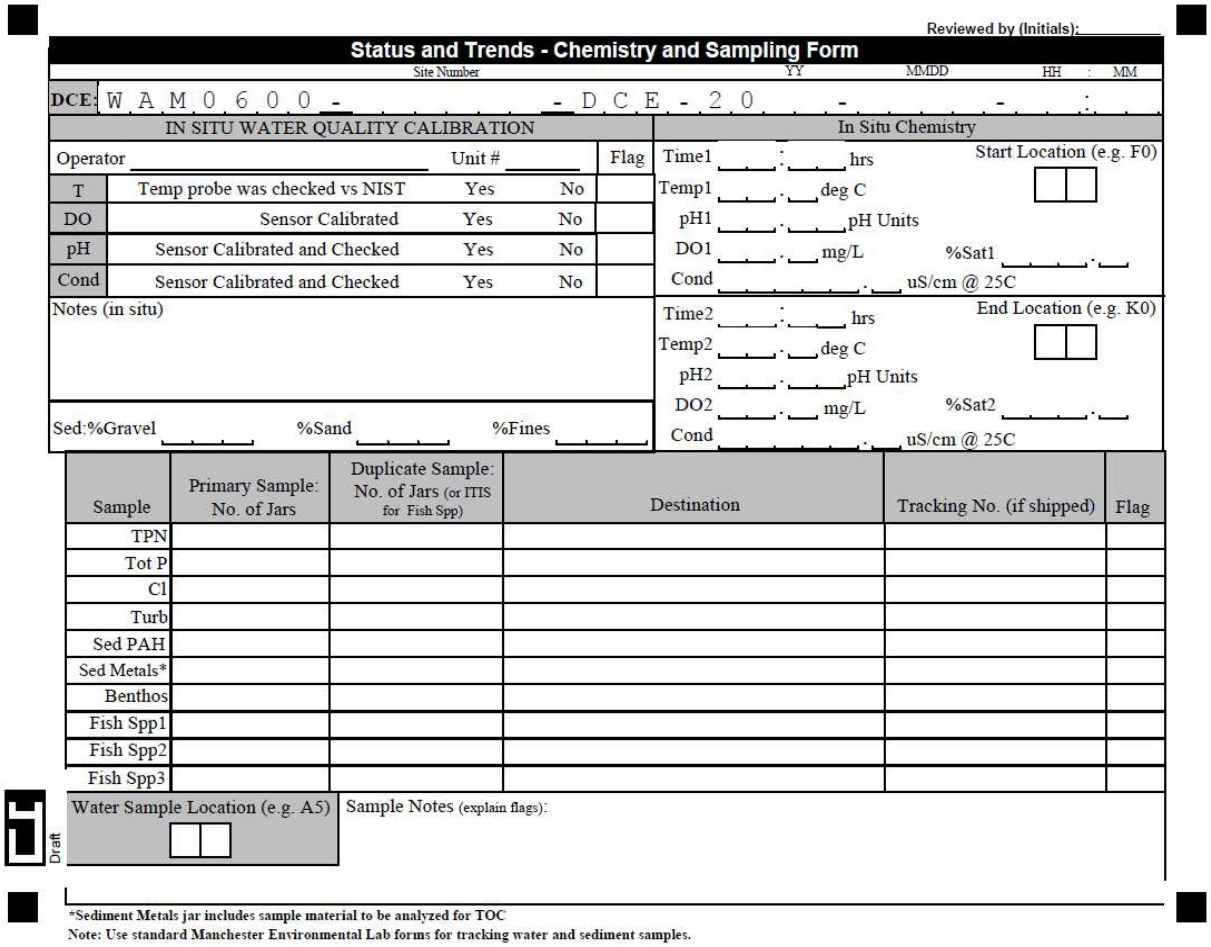


Figure G-2. Site Diagram Form.



Reviewed by (Initials):_____

Status and Trends - Chemistry and Sampling Form

Site Number | YY | MMDD | HH : MM

DCE: WAM0600 - _____ - DCE - 20____ - _____ - _____ : _____

IN SITU WATER QUALITY CALIBRATION

Operator _____ Unit # _____ Flag

T	Temp probe was checked vs NIST	Yes	No	
DO	Sensor Calibrated	Yes	No	
pH	Sensor Calibrated and Checked	Yes	No	
Cond	Sensor Calibrated and Checked	Yes	No	

Notes (in situ)

Sed:%Gravel _____ %Sand _____ %Fines _____

In Situ Chemistry

Start Location (e.g. F0) ☐☐

Time1 ____:____ hrs
Temp1 ____.____ deg C
pH1 ____.____ pH Units
DO1 ____.____ mg/L %Sat1 ____.____
Cond ____.____ uS/cm @ 25C

End Location (e.g. K0) ☐☐

Time2 ____:____ hrs
Temp2 ____.____ deg C
pH2 ____.____ pH Units
DO2 ____.____ mg/L %Sat2 ____.____
Cond ____.____ uS/cm @ 25C

Sample	Primary Sample: No. of Jars	Duplicate Sample: No. of Jars (or ITIS for Fish Spp)	Destination	Tracking No. (if shipped)	Flag
TPN					
Tot P					
Cl					
Turb					
Sed PAH					
Sed Metals*					
Benthos					
Fish Spp1					
Fish Spp2					
Fish Spp3					

Water Sample Location (e.g. A5) ☐☐

Sample Notes (explain flags):

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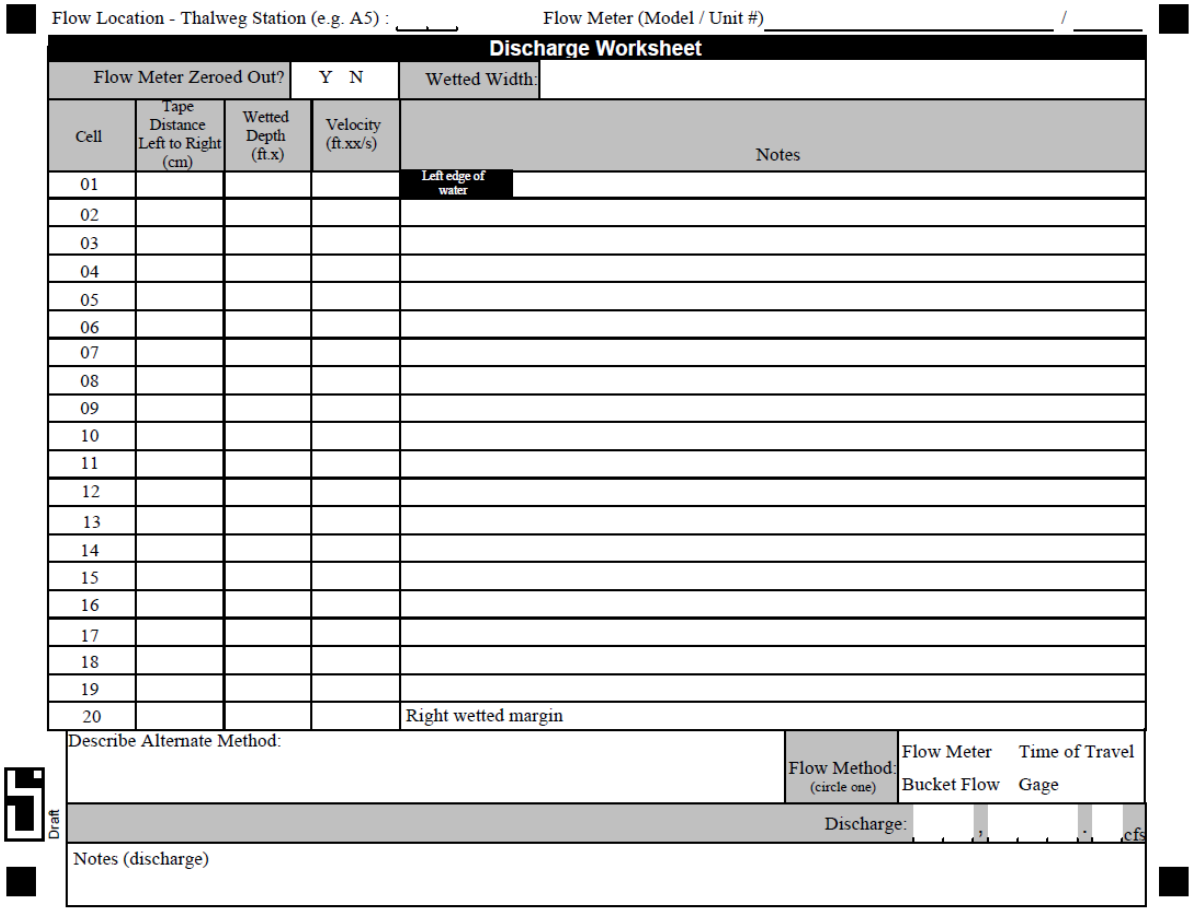
*Sediment Metals jar includes sample material to be analyzed for TOC
Note: Use standard Manchester Environmental Lab forms for tracking water and sediment samples.

Figure G-3. ***Chemistry and Sampling Form*****.**



Flow Location - Thalweg Station (e.g. A5) : ____ Flow Meter (Model / Unit #) _____ / _____

Discharge Worksheet

Flow Meter Zeroed Out?	Y N	Wetted Width:	

Cell	Tape Distance Left to Right (cm)	Wetted Depth (ft.x)	Velocity (ft.xx/s)	Notes
01				Left edge of water
02				
03				
04				
05				
06				
07				
08				
09				
10				
11				
12				
13				
14				
15				
16				
17				
18				
19				
20				Right wetted margin

Describe Alternate Method:

Flow Method: (circle one) Flow Meter Time of Travel Bucket Flow Gage

Discharge: ___,___.___cfs

Notes (discharge)

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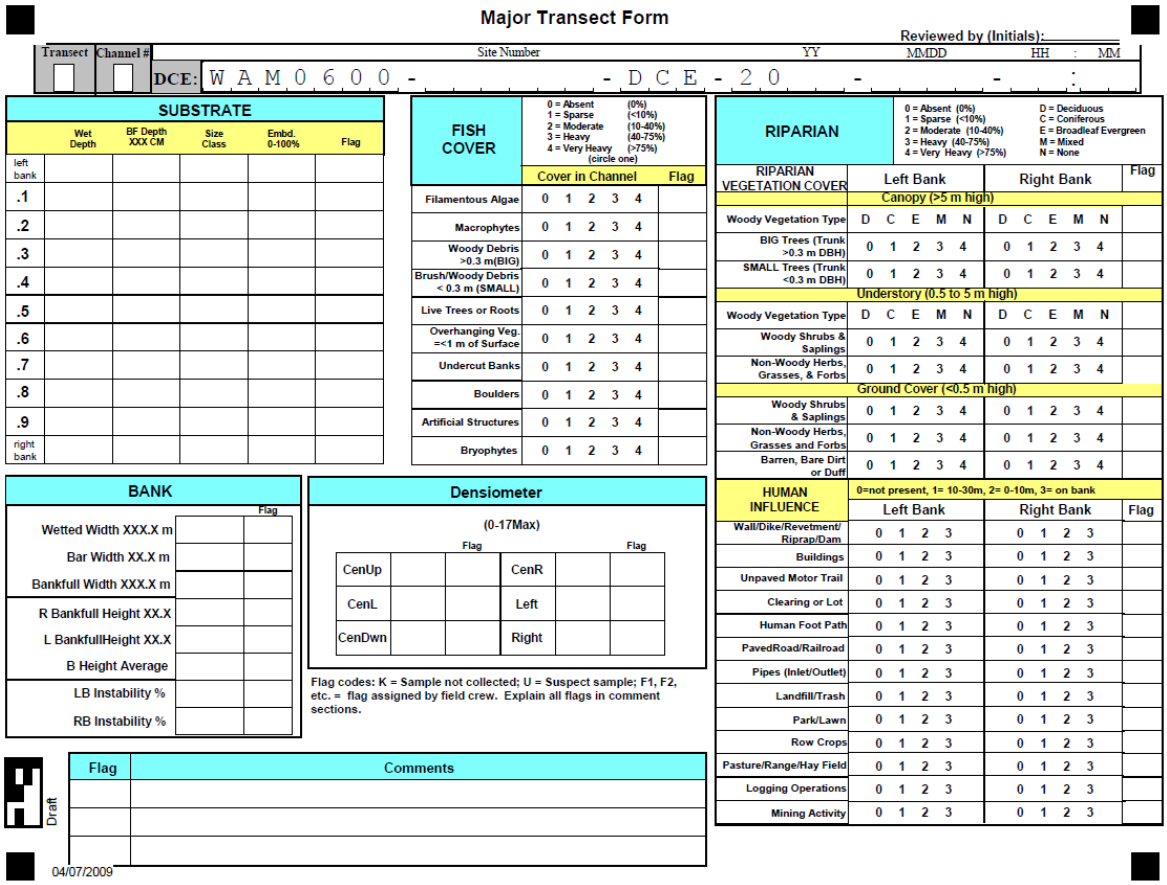
Figure G-4. Discharge Worksheet.



Major Transect Form

Reviewed by (Initials):_____

Transect | Channel # | Site Number: DCE: WAM0600- -DCE-20 - - | YY MMDD HH : MM

SUBSTRATE

	Wet Depth	BF Depth XXX CM	Size Class	Embd. 0-100%	Flag
left bank					
.1					
.2					
.3					
.4					
.5					
.6					
.7					
.8					
.9					
right bank					

FISH COVER

0 = Absent (0%)
1 = Sparse (<10%)
2 = Moderate (10-40%)
3 = Heavy (40-75%)
4 = Very Heavy (>75%)
(circle one)

	Cover in Channel	Flag
Filamentous Algae	0 1 2 3 4	
Macrophytes	0 1 2 3 4	
Woody Debris >0.3 m(BIG)	0 1 2 3 4	
Brush/Woody Debris < 0.3 m (SMALL)	0 1 2 3 4	
Live Trees or Roots	0 1 2 3 4	
Overhanging Veg. =<1 m of Surface	0 1 2 3 4	
Undercut Banks	0 1 2 3 4	
Boulders	0 1 2 3 4	
Artificial Structures	0 1 2 3 4	
Bryophytes	0 1 2 3 4	

RIPARIAN

0 = Absent (0%)
1 = Sparse (<10%)
2 = Moderate (10-40%)
3 = Heavy (40-75%)
4 = Very Heavy (>75%)

D = Deciduous
C = Coniferous
E = Broadleaf Evergreen
M = Mixed
N = None

RIPARIAN VEGETATION COVER	Left Bank	Right Bank	Flag
Canopy (>5 m high)			
Woody Vegetation Type	D C E M N	D C E M N	
BIG Trees (Trunk >0.3 m DBH)	0 1 2 3 4	0 1 2 3 4	
SMALL Trees (Trunk <0.3 m DBH)	0 1 2 3 4	0 1 2 3 4	
Understory (0.5 to 5 m high)			
Woody Vegetation Type	D C E M N	D C E M N	
Woody Shrubs & Saplings	0 1 2 3 4	0 1 2 3 4	
Non-Woody Herbs, Grasses, & Forbs	0 1 2 3 4	0 1 2 3 4	
Ground Cover (<0.5 m high)			
Woody Shrubs & Saplings	0 1 2 3 4	0 1 2 3 4	
Non-Woody Herbs, Grasses and Forbs	0 1 2 3 4	0 1 2 3 4	
Barren, Bare Dirt or Duff	0 1 2 3 4	0 1 2 3 4	

BANK

	Flag
Wetted Width XXX.X m	
Bar Width XX.X m	
Bankfull Width XXX.X m	
R Bankfull Height XX.X	
L BankfullHeight XX.X	
B Height Average	
LB Instability %	
RB Instability %	

Densiometer

(0-17Max)

	Flag		Flag
CenUp		CenR	
CenL		Left	
CenDwn		Right	

Flag codes: K = Sample not collected; U = Suspect sample; F1, F2, etc. = flag assigned by field crew. Explain all flags in comment sections.

HUMAN INFLUENCE

0=not present, 1= 10-30m, 2= 0-10m, 3= on bank

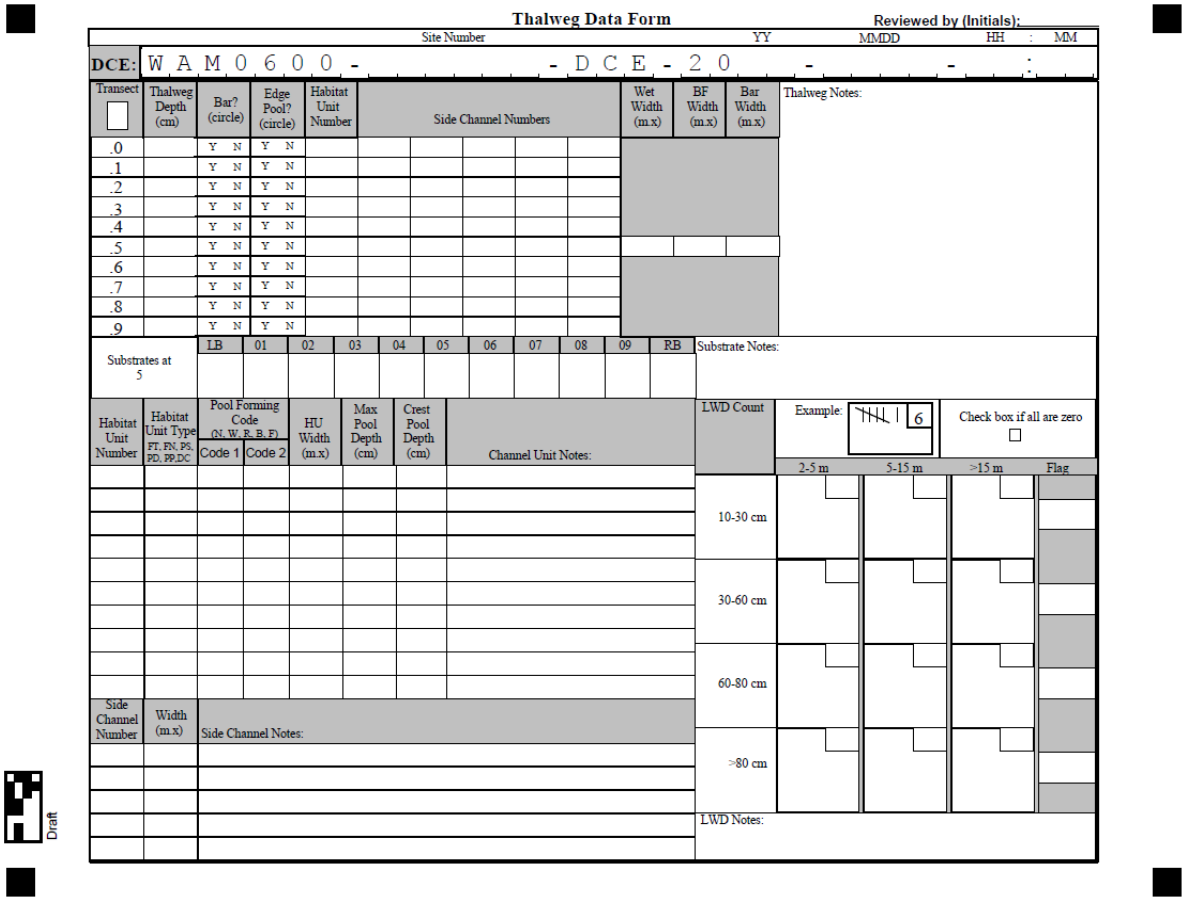
	Left Bank	Right Bank	Flag
Wall/Dike/Revetment/ Riprap/Dam	0 1 2 3	0 1 2 3	
Buildings	0 1 2 3	0 1 2 3	
Unpaved Motor Trail	0 1 2 3	0 1 2 3	
Clearing or Lot	0 1 2 3	0 1 2 3	
Human Foot Path	0 1 2 3	0 1 2 3	
PavedRoad/Railroad	0 1 2 3	0 1 2 3	
Pipes (Inlet/Outlet)	0 1 2 3	0 1 2 3	
Landfill/Trash	0 1 2 3	0 1 2 3	
Park/Lawn	0 1 2 3	0 1 2 3	
Row Crops	0 1 2 3	0 1 2 3	
Pasture/Range/Hay Field	0 1 2 3	0 1 2 3	
Logging Operations	0 1 2 3	0 1 2 3	
Mining Activity	0 1 2 3	0 1 2 3	

Flag	Comments

Draft

04/07/2009

Figure G-5. Major Transect Form.



Thalweg Data Form

Reviewed by (Initials):_____

Site Number | YY | MMDD | HH : MM

DCE: WAM0600 - - DCE - 20 - - :

Transect	Thalweg Depth (cm)	Bar? (circle)	Edge Pool? (circle)	Habitat Unit Number	Side Channel Numbers	Wet Width (m.x)	BF Width (m.x)	Bar Width (m.x)
.0		Y N	Y N					
.1		Y N	Y N					
.2		Y N	Y N					
.3		Y N	Y N					
.4		Y N	Y N					
.5		Y N	Y N					
.6		Y N	Y N					
.7		Y N	Y N					
.8		Y N	Y N					
.9		Y N	Y N					

Thalweg Notes:

Substrates at 5	LB	01	02	03	04	05	06	07	08	09	RB

Substrate Notes:

Habitat Unit Number	Habitat Unit Type (FT, FN, PS, PD, PP,DC)	Pool Forming Code (N, W, R, B, F) Code 1	Code 2	HU Width (m.x)	Max Pool Depth (cm)	Crest Pool Depth (cm)	Channel Unit Notes:

Side Channel Number	Width (m.x)	Side Channel Notes:

LWD Count — Example: 6 — Check box if all are zero ☐

	2-5 m	5-15 m	>15 m	Flag
10-30 cm				
30-60 cm				
60-80 cm				
>80 cm				

LWD Notes:

Draft

Figure G-6. Thalweg Data Form.

Reviewed by (Initials):___________

Status & Trends - Slope and Bearing Form

Site Number	YY	MMDD	HH : MM
DCE: W A M 0 6 0 0 - _ _ _ _ _ _ _ - D C E - 2 0	_ _ -	_ _ _ _ -	_ _ : _ _

Top Transect* LEVEL, COMPASS	Bottom Transect* ROD	Segment Length (m.x)	Eye Height on Level (cm)	Level Height on Rod (cm)	Bearing (deg)	Flag	Comment

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*Normally sight between major and minor transects: K0 to J5, J5 to J0, J0 to I5, etc, unless there is a need for intermediate measurements. For intermediate measurements, always sight to/from a transect station 0-9 (for example K0 to J7 and J7 to J5). Bearings are magnetic north facing downstream (not adjusted).

Figure G-7. Slope and Bearing Form.

Reviewed by (Initials):_____

Status and Trends Program - GPS Positions Form

Site Number YY MMDD HH : MM

DCE: WAM0600 - - DCE - 20 - - :

Station	Bank (circle one)	Master Lat dec deg e.g. 47.123456	Master Lon dec deg e.g. 120.123456	GPSlatDD e.g. 47.123456	GPSLonDD e.g. 120.123456	Accuracy	Accuracy Units (ft, EPE, etc.)	Flag
INDEX STATION	L R							
A0	L R							
B0	L R							
C0	L R							
D0	L R							
E0	L R							
F0	L R							
G0	L R							
H0	L R							
I0	L R							
J0	L R							
K0	L R							
PUTIN	L R							
TAKEOUT	L R							

ALL COORDINATES TO BE RECORDED IN NAD83

Position comments including accuracy

Directions to access point

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Figure G-8. GPS Positions Form.

Biomonitoring Program – Photo Log Form			
Photo Number	Date/time	Field Site Number	Photo Description

Figure G-9. Photo Log Form.

Appendix H. Field equipment checklist

Table H-1. Example checklist of equipment necessary to complete the procedures described.

Check Box (√)	Category	Item
	General	Pencils and pens
	General	Permanent markers
	General	Waders/Boots
	General	Camera
	General	Calculator
	General	Field notebook
	General	Calipers
	General	Scrub brush for boot cleaning
	General	GPS Unit/Maps
	Forms and labels	Site Verification Form x Site Diagram
	Forms and labels	Chemistry and Sampling Form x Discharge Worksheet
	Forms and labels	Major Transect Form x Thalweg Data Form
	Forms and labels	Slope and Bearing Form x GPS Positions
	Forms and labels	Calibration Form
	Forms and labels	Laboratory Analyses Required Form
	Forms and labels	Benthos Label (waterproof) for jar exterior
	Forms and labels	Benthos Label (waterproof) for jar interior
	Forms and labels	Periphyton Label (waterproof) for jar exterior
	Forms and labels	Periphyton Label (waterproof) for jar interior
	C.1 Site	50-m tape
	C.1 Site	Flagging
	C.1 Site Verification	Laser rangefinder
	C.2 In-situ stream meas	Multimeter for DO, pH, conductivity, and temperature (LDO type dissolved oxygen meter)
	C.2 In-situ stream meas	Multimeter Operation Manual
	C.2 In-situ stream meas	Multimeter Maintenance Kit
	C.3 Discharge	Flow Meter (Marsh-McBirney, SonTech FlowTracker, etc)
	C.3 Discharge	Wading rod (top setting)
	C.3 Discharge	Orange or other neutrally buoyant object
	C.3 Discharge	5-gallon bucket
	C.3 Discharge	Stop watch
	C.4 Sediment Chem	Bowl, glass, flat bottom, approximately 5 L, 12-in diameter
	C.4 Sediment Chem	Sieve Frame, Nylon, 8" diameter (for metals sample)
	C.4 Sediment Chem	Nylon sieve cloth, 63 micron (for metals sample)
	C.4 Sediment Chem	Funnel, polyethylene, 8" diameter
	C.4 Sediment Chem	Policeman, Teflon (to aid sieving)
	C.4 Sediment Chem	Spatula, scoop, and spoon, all Teflon
	C.4 Sediment Chem	Syringe, plastic, 50ml
	C.4 Sediment Chem	Wash bottle (labeled) with Liquinox or Alconox
	C.4 Sediment Chem	Wash bottle (labeled) with acetone (pesticide grade)

	C.4 Sediment Chem	Wash bottle (labeled) with 10% nitric acid
	C.4 Sediment Chem	Wash bottle, plastic 500-ml
	C.4 Sediment Chem	Wash bottle, Teflon 500-ml
	C.4 Sediment Chem	Deionized water
	C.4 Sediment Chem	Personal protective gear as specified by the MSDS
	C.4 Sediment Chem	Sample containers (analytical laboratory will supply)
	C.4 Sediment Chem	MSDS
	C.4 Sediment Chem	Gloves - Non-powdered nitrile
	C.4 Sediment Chem	Polyethylene bags
	C.4 Sediment Chem	Foam sleeves for shipping
	C.4 Sediment Chem	Aluminum foil
	C.5 Bank Measurements	Measuring rod
	C.5 Bank Measurements	Hand level
	C.5 Bank Measurements	Clinometers
	C.6 Substrate & depth	PVC ring
	C.7 Shade	Modified convex densitometer
	C.15 Width & Substrate	10-cm ring
	C.16 Slope & Bearing	Hand level (5x magnification)
	C.16 Slope & Bearing	Monopod for hand level
	C.16 Slope & Bearing	Measuring rod (telescoping)
	C.16 Slope & Bearing	Compass (handheld, magnetic)
	C.16 Slope & Bearing	Range finder
	D.1 Benthos	Wide-mouth polyethylene jar (128 oz or 3.8 L).
	D.1 Benthos	D-Frame kick net, (1 ft (30.5 cm) wide by 1 ft tall, 500-µm mesh D-frame kick net), or Surber sampler
	D.1 Benthos	Garden trowel or large spike to disturb substrate
	D.1 Benthos	Eye Protection
	D.1 Benthos	Eye Wash
	D.1 Benthos	95% Ethanol (add 3 parts by volume for each part sample)
	D.1 Benthos	Label (waterproof) for jar exterior
	D.1 Benthos	Clear tape
	D.1 Benthos	Electrical tape
	D.1 Benthos	Pocket knife
	D.3 Periphyton	Digital Caliper
	D.3 Periphyton	Small cooler (< 2 gal) for storing periphyton samples
	D.3 Periphyton	500mL poly vials
	D.3 Periphyton	47 mm 0.7 micron glass microfiber filters
	D.3 Periphyton	Velocity meter and top-setting rod
	D.3 Periphyton	Pruning shears, scissors, or small saw
	D.3 Periphyton	Nalgene® bottles (500 ml and 1000 ml)
	D.3 Periphyton	47 mm diameter Petri dishes
	D.3 Periphyton	Spatula (for inverting Petri dish)
	D.3 Periphyton	Collection envelopes
	D.3 Periphyton	Disposable serological volumetric glass pipette
	D.3 Periphyton	Wide-mouth polyethylene jars (1 L)
	D.3 Periphyton	Firm bristled toothbrush
	D.3 Periphyton	Aluminum foil

	D.3 Periphyton	Wash bottle (squirt type)
	D.3 Periphyton	Plastic funnel
	D.3 Periphyton	One gallon Ziploc bag
	D.3 Periphyton	Large plastic bowl or tray to catch rinsate
	D.3 Periphyton	95% Ethanol (add 3 parts by volume for each part sample)
	D.3 Periphyton	Deionized or distilled water
	D.3 Periphyton	Label (waterproof) for jar exterior
	F.1 In-situ QA/QC & Calibration	Disposable filter set-up
	F.1 In-situ QA/QC & Calibration	Filter set up with peristaltic or hand pump
	F.1 In-situ QA/QC & Calibration	Batteries for all equipment
	F.1 In-situ QA/QC & Calibration	Five-gallon bucket (for flow meter zero-adjust)
	F.1 In-situ QA/QC & Calibration	pH 7 buffer
	F.1 In-situ QA/QC & Calibration	pH 4 buffer
	F.1 In-situ QA/QC & Calibration	pH 10 buffer
	F.1 In-situ QA/QC & Calibration	Conductivity Standard (100 uS)
	F.1 In-situ QA/QC & Calibration	Conductivity Standard (1,000 uS)
	F.1 In-situ QA/QC & Calibration	Conductivity Standard (alternate as available).
	F.1 In-situ QA/QC & Calibration	Deionized water (DI)
	F.1 In-situ QA/QC & Calibration	Tap Water
	F.1 In-situ QA/QC & Calibration	Lab tissues (e.g., KimWipes®)
	F.1 In-situ QA/QC & Calibration	Barometric pressure-elevation chart
	F.1 In-situ QA/QC & Calibration	NIST certified thermometer

Appendix I. Standard taxonomic effort (except chironomidae)

Ephemeroptera

Genus, with exceptions noted:

Baetidae

- Acentrella—species.
- Acerpenna—species.
- Baetis—species.
- Baetodes—species.
- Cloeodes—species.
- Diphetor hageni—monotypic.
- Fallceon quilleri—distribution, genus for far SW US projects.
- Paracloeodes minutus—distribution.
- Psuedocloeon sp.—species.

Caenidae

- Amercaenis ridens—distribution.
- Caenis—species.

Ephemerellidae

- Attenella—species.
- Caudatella—species.
  - Note that C. cascadia has been synonymized with C. hystrix.
- Caurinella idahoensis—monotypic.
- Drunella—species.
  - Use D. coloradensis/flavinea for D. coloradensis and D. flavinea.
- Ephemerella—species.
  - Use E. inermis/infrequens for E. inermis and E. infrequens.
- Eurylophella—species.
- Serratella—species.
- Timpanoga hecuba—monotypic.

Ephemeridae

- Ephemera simulans—distribution.

Heptageniidae

- Epeorus—species for Rocky Mountain specimens, genus otherwise.
- Stenacron—species.
- McCaffertium—species.

Leptophlebiidae

- Neochoroterpes—species.
- Thraulodes—species.
- Traverella sp.—species.

Leptohyphidae

- Leptohyphes—species.
- Vacupernius packeri—monotypic.

Odonata
Species for mature specimens of most taxa (exception below), genus otherwise:
Coenagrionidae—genus.
Plecoptera
Genus for most taxa (exceptions below):
Capniidae—family except for late instar larvae.
Leuctridae—family except for late instar larvae.
Despaxia augusta—monotypic.
Moselia infuscata—monotypic.
Nemouridae
Visoka cataractae—monotypic.
Zapada—species.
Use Z. oregonensis gr. for members of that species group.
Perlodidae
Frisonia picticeps—monotypic.
Kogotus/Rickera—for indeterminate specimens.
Osbenus yakimae—monotypic.
Perlinodes aurea—monotypic.
Pictetiella expansa—monotypic.
Rickera sorpta—monotypic.
Perlidae
Acroneuria—species.
Calineuria californica—monotypic.
Claassenia sabulosa—distribution.
Hesperoperla—species.
Pteronarcyidae
Pteronarcys—species for mature specimens.
Hemiptera
Genus for most taxa (exceptions below):
Gerridae—ignore.
Veliidae—ignore.
Coleoptera
Genus for most taxa (exceptions below):
Elmidae
Ampumixis dispar—monotypic.
Atractelmis wawona—monotypic.
C. Barr undescribed sp.—used for that genus soon to be described by C. Barr.
Cleptelmis addenda—monotypic.
Macronychus glabratus—monotypic.
Ordobrevia nubifera—monotypic.
Rhizelmis nigra—monotypic.
Psephenidae
Eubrianax edwardsi—monotypic.
Megaloptera
Genus except for:
Orohermes crepusculus—monotypic.

Diptera

Larvae to genus with a few exceptions noted below and the following to family:

Thaumaleidae, Dolichopodidae, Syrphidae, Tabanidae, Ephydridae, Muscidae, Sciomyzidae

pupae to family except cased Simuliidae to genus, Antocha to genus.

Tipulidae

Rhabdomastix—larvae to species group.

Ceratopogonidae

Bezzia/Palpomyia—use for those two genera inseparable as larvae.

Chaoboridae

Eucorethra underwoodi—monotypic.

Psychodidae

Pericoma/Telmatoscopus—use for those two genera inseparable as larvae.

Stratiomyidae

Hedriodiscus/Odontomyia—use for inseparable larval specimens.

Trichoptera

Larvae generally to genus except monotypic species, other exceptions noted below:

Pupae to family.

Rhyacophilidae

Rhyacophila—most larvae to species group using Smith designations, with the following exceptions:

R. betteni gr.

R. malkini—distinctive.

R. leiftincki gr.

R. arnaudi—only N.A. species in group.

R. sibirica gr.

R. narvae—usually distinctive, leave at species group if unsure.

R. blarina—distinctive.

R. pellisa/valuma—use this for all R. atrata subgroup.

Hydropsychidae

Arctopsyche—larvae to species.

Parapsyche—larvae to species.

Potamyia flava—distribution.

Smicridea—to subgenus.

Polycentropodidae

Cyrnellus fraternus—distribution.

Psychomyiidae

Psychomyia—larvae to species.

Apataniidae

Pedomoecus sierra—monotypic.

Brachycentridae

Amiocentrus aspilus—monotypic.

Brachycentrus—larvae to species.

Calamoceratidae

Heteroplectron californicum—distribution.

Leptoceridae

Mystacides—larvae to species.
Oecetis—larvae of O. avara, O. disjuncta to species, all others to genus.
Limnephilidae
Allocosmoecus partitus—monotypic.
Amphicosmoecus canax—monotypic.
Chyranda centralis—monotypic.
Clostoeca disjuncta—monotypic.
Dicosmoecus—larvae to species.
Ecclicosmoecus scylla—distribution.
Hydatophylax hesperus—distribution.
Uenoidae
Neophylax—larvae to species.
Sericostriata surdickae—monotypic.
Lepidoptera
Larvae—Petrophila and Paraponyx to genus, most others to family, order if uncertain
pupae—order.
Cnidaria
Genus.
Nemertea
Genus.
Turbellaria
Phylum, except Polycelis to genus.
Nematoda
Phylum.
Nematomorpha
Phylum.
Gastropoda
Genus in most cases, with exceptions noted:
Valvatidae
Species for mature specimens, if immature leave at genus.
Hydrobiidae—family.
Lymnaeidae
Radix auricularia—monotypic.
Bivalvia
Genus for mature specimens.
Branchiobdella
Order (Branchiobdellida).
Polychaeta
Manayunkia speciosa—distinctive.
Hirudinea
Genus, with exceptions noted:
Erpobdellidae—family.
Glossiphoniidae
Glossiphonia complanata—distinctive.
Helobdella stagnalis—distinctive.
Piscicolidae
Piscicola—species for mature specimens.

Crustacea

Genus, with exceptions noted:
Astacidae—species.
Cambaridae—species for mature males.
Ostracoda—class.
Branchiopoda—ignore.
Copepoda—ignore.

Acarina

Genus for adults, use ‘Acari’ for indeterminate specimens, leave Oribatei at suborder (Oribatei).

Appendix J. Taxonomic effort for chironomidae

To maintain data consistency, Chironomidae identifications should be to the genus level when practical except for the following taxa:

Cardiocladius albiplumus

Cricotopus (Isocladius) Type I.

(EcoAnalysts in-house designation) presence of two "racing stripes" on the dorsum of the light colored (yellow) head, body with gray mottling, dark mentum with 15 teeth, first lateral teeth closely pressed to the median teeth, and the last two pairs of lateral teeth appear to be reduced and slightly separated from the other lateral teeth.

Cricotopus (Nostococladius) nostocicola

(previously referred to as *Cricotopus (Nostocladius)* sp. by EcoAnalysts, changed in 2002.)

Cricotopus bicinctus gr.

Cricotopus trifascia gr.

Heterotrissocladius

(identify to species group following Wiederholm, 1983 and 1986.)

Hyporhygma quadripunctatum (monotypic).

Lauterborniella agrayloides (monotypic).

Microtendipes

(identify to species group following Wiederholm, 1983 and 1986.)

Orthocladius Complex

(encompasses *Orthocladius* sp.; and *Cricotopus* sp. that are inseparable from *Orthocladius* sp.; and *Paratrichocladius* sp.)

Orthocladius (Symposiocladius) lignicola

Paralauterborniella nigrohalteralis (monotypic)

Paramerina/Zavrelimyia sp.

(This includes *Reomyia* sp.)

Paraphaenocladius "n. sp."

(EcoAnalysts in-house designation) single median tooth, "long" antenna, six antennal segments (sixth hairlike), second antennal segment with a "break."

Platysmittia

(identify to species following Epler, 2001 and Jacobsen, 1998.)

Potthastia

(identify to species group following Wiederholm, 1983 and 1986.)

Robackia

(identify to species following Wiederholm, 1983 and Epler, 2001.)

Saetheria tylus

(larval diagrams in Epler, 2001 and Merritt and Cummins, 1996.)
Tempisquitoneura merrillorum
Thienemannimyia gr. sp.
(consists of the genera *Arctopelopia*, *Conchapelopia*, *Hayesomyia*, *Helopelopia*, *Meropelopia*, *Rheopelopia*, *Telopelopia*, and *Thienemannimyia*.)

Tribelos jucundum

Tvetenia

(identify to species group following Bode, 1983 ie. discoloripes grp. and bavarica grp.)
Unniella multivirga (monotypic).
Xenochironomus xenolabis (monotypic).
Xylotopus par (monotypic).

Appendix K. Aggregation of taxa

1. When to aggregate

Unless the taxonomist has a compelling reason (listed below) to believe the unidentifiable individuals are unique, those individuals shall be aggregated.

An unidentifiable individual will be considered unique (distinct) if and only if:

A. Morphological characteristics preclude the unidentified individual from being any of the other taxa identified (in other words, the specimens cannot be the same thing). If they cannot be the same, then they are considered distinct.
B. Due to maturity an unidentifiable specimen is more than two major taxonomic levels removed from its nearest possible relative.

2. How to aggregate

Unidentifiable individuals shall be aggregated with the nearest related lower taxon or taxa using all available information (e.g., ecological, life history, developmental). In cases where no other clues are available, individuals shall be apportioned according to the relative abundance of each related taxon.

3. Notation

Each taxon that contains aggregated individuals will be marked using a simple note (e.g., A7, where "A" represents aggregate and "7" being the number of individuals aggregated).

Appendix L. Example Chain-of-Custody forms

Table L-1.1. Biological Chain-of-Custody form

PAGE ___ of ___ Biological Monitoring Project Chain-of-Custody Complete one form for each sample type	Project Lead:_____ Staff member:_____ submitting sample to lab Date samples transferred to Lab:_____ Date Project Lead downloads data:_____ Lab staff member receiving samples_____ Date samples received by Lab:_____ Date vouchers transferred to archive:_____ Date data available to Project Lead for download:_____ Archive staff member receiving vouchers:_____ Date vouchers received by archive:_____

Sample ID	Sample Type P=Periphyton M= Macroinvertebrate	Sample Location Latitude DMS	Sample Location Longitude DMS	# Jars/ sample
Total Samples Transferred				

PAGE ___ of ___
Biological Monitoring Project
Chain-of-Custody
Date Samples Transferred to Lab:

Sample ID	Sample Type P=Periphyton M= Macroinvertebrate	Sample Location Latitude DMS	Sample Location Longitude DMS	# Jars/ sample
Total Samples Transferred				

Table L-1.2. Water and Sediment Chain-of-Custody Form.

Laboratory Analyses Required

Page ____ of ____

Project Name: ______________________________

SIC: ___ ___ ___ ___ ___ **Program:** ______________________________

Send Results to: ____________________ **Mail Stop:** ______________

☐ Preliminary Investigation ☐ For HW Designation

☐ Monitoring ☐ For NPDES

Date Results needed by: ______________

Name/Reference # of QAPP for this project: ______________________

Sampling: Date (Year: ☐) Mo Day	Sampling: Time (Military) 0001 - 2400 Hr Mn	Field Station Identification	Manchester Lab Sample Number	Matrix Code	Source Code	No. of Containers	General Chemistry	Micro	Metals	Organic Chemistry
							Alkalinity; Conductivity; pH; Turbidity; ☐ Chloride ☐ Sulfate; Fluoride; Total Suspended Solids; Total Dissolved Solids; ☐ Total Solids ☐ % Sol ☐ % Vol Sol; TOC; DOC; BOD5; Oil & Grease (HEM); Ammonia; Nitrite/Nitrate; Total Phosphorus; Orthophosphate; TPN; ☐ Chlorophyll ☐ filters	☐ Fecal Coliform ☐ MF ☐ MPN; ☐ Total Coliform ☐ MF ☐ MPN; E. Coli ☐ MF ☐ MPN; % Klebsiella; Enterococcus	PP Mets; Mercury (Hg) ☐ low level; Hardness; Individual Elements (please list) ☐ Total ☐ Dissolved	VOA; BTEX; TPHG; TPHD; HCID Only (Hydrocarbon ID); PCB aroclors; ☐ Pest 1 ☐ w/PCB aroclors; ☐ Pest 2 ☐ w/PCB aroclors; ☐ Pest 3 ☐ w/PCB aroclors; Carbamates; PBBTMS; Herbicides; PBDE; BNA; PAH

Project Officer: ____________________

Phone Number: ____________________

Cell Number: ____________________

Samplers: ____________________

____

____

Chain of Custody Record

Relinquished By:	Received By:	Yr	Mo	Da	Hr	Mn	Seal I.D.	Condition of Seals	Comments (Temperature, Preserv., etc.)

Recorder: ____________________ Comments: ____

____

ECY 040-115 (Rev. 10/05)

Matrix Codes

Code	Description
10	Water
11	Field Filtered Water
12	Filter from Water
13	Water to be filtered upon receipt at lab
40	Soil/Sediment
41	Frozen Soil/Sediment (PSEP)
45	Semi-Solid/Sludge
70	Tissue
80	Oil/Solvent
90	Waste
00	Other (Use only if no other apply)

Source Codes

Code	Description
00	Unspecified Source
01	Unknown Liquid Media (Drum/Tank)
02	Unknown Liquid Media (Spill Area)
03	Unknown Liquid Media (Waste Pond)
10	**Water (General)**
12	Ambient Stream/River
13	Lake Reservoir
14	Estuary/Ocean
15	Spring/Seepage
16	Rain
17	Surface Runoff/Pond (general)
18	Irrigation Canal/Return Flow
20	**Well (General)**
21	Well (Industrial/Agricultural)
22	Well (Drinking Water Supply)
23	Well (Test/Observation)
24	Drinking Water Intake
25	Drinking Water (At Tap)
30	**Effluent Wastewater (General)**
31	Municipal Effluent
32	Municipal Inplant Waters
33	Industrial Surface Runoff/Leachate
34	Industrial Effluent
35	Industrial Inplant Waters
36	Industrial Surface Runoff/Pond
37	Industrial Waste Pond
38	Landfill Runoff/Pond/Leachate
40	**Sediment (General)**
42	Bottom Sediment or Deposit
44	Sludge (General)
45	Sludge (WastePond)
46	Sludge (Drum/Tank)
48	Soil (General)
49	Soil (Spill/Contaminated Area)

Code	Description
50	Bore Hole Material
60	**Air (General)**
61	Ambient Air
62	Source or Effluent Air
63	Industrial or Workroom Air
70	**Tissue (General)**
71	Fish Tissue
72	Shellfish Tissue
73	Bird Tissue
74	Mammal Tissue
75	Macroinvertebrate
76	Algae
77	Periphyton
78	Plant/Vegetation
80	**Oil/Solvent (General)**
81	Oil (Transformer/Capacitor)
82	Oil/Solvent (Drum Tank)
83	Oil/Solvent (Spill Area)
84	Oil/Solvent (Waste/Pond)
90	**Commercial Product Formulation**
95	Well Drill Water
96	Well Drill Mud
97	Well Sealing Material
98	Gravel Pack Material

Taken From MEL (2008).

Appendix M. Example Water Quality Field Data form

Field Data Report Form

Sampler(s): Weather: Date:

Station	Station Name	Time	Temp ºC	DO mg/L	DO #	Temp pH	True Meter	Cond uS/cm	Press in.Hg	ChkBr/ Corr.	Comments
23A160	*Chehalis R@ Dryad*	*9:50*	*15.8*	*9.5*	*15*	*16.1*	*7.03*	*53*	*29.05*	*0.27*	*Heavy rainfall during sample collection.*
						7.68	*7.03*				
QA-1	Quality Control										
	Sample										
QA-2	Quality Control										
	Sample										

Appendix N. Example WQI Monitoring Data submittal format

Table N-1.1. Example monitoring data format for Water Quality Index (WQI) calculations. The parameter order is important. If uncertain about beneficial uses (green cells), leave blank.

Station:	Swamp
Recreation Use:	Extraordinary
Aquatic Life (Temperature):	Core(16)
Aquatic Life (Oxygen):	Core
Supplemental Spawning:	None
Ecoregion:	2

	FC	Oxygen	pH	TP	TSS	Temp	TN	Turbidity
Date	col/100mL	mg/L	std. Units	mg/L	mg/L	C	mg/L	NTU
10/6/1997 10:30	63	8.6	6.95	0.00866	2	13.9	0.149	0.9
11/3/1997 9:30	70	8.7	6.82	0.014	3	10.6	0.31	3.7
12/1/1997 9:50	86	10.7	6.78	0.00924	5	6.8	0.438	3.5
1/7/1998 10:00	45	11.9	6.90	0.029	4	4.8	1.02	2.8
2/11/1998 10:20	85	12.9	7.29	0.029	7	4.8	1.02	1.3
3/7/1998 9:00	105	11.9	7.20	0.029	4	4.8	1.02	2.3
4/13/1998 11:15	53	10.9	7.22	0.0197	3	8.9	0.527	3.7
5/4/1998 13:40	4900	9.2	7.47	0.0219	42	17.4	0.338	3.5
6/10/1998 12:30	570	8.9	7.44	0.0257	20	15	0.328	2.8
7/7/1998 11:00	445	8.9	7.29	0.029	41	16.8	0.402	3.7
8/9/1998 09:45	345	9.9	7.39	0.039	4	17.8	0.202	3.5
9/9/1998 10:00	45	8.7	7.29	0.019	4	18.8	0.320	2.8

Appendix O. Glossary, acronyms, and abbreviations

Glossary

National Pollutant Discharge Elimination System (NPDES): National program for issuing, modifying, revoking and reissuing, terminating, monitoring, and enforcing permits, and imposing and enforcing pretreatment requirements under the Clean Water Act. The NPDES program regulates discharges from wastewater treatment plants, large factories, and other facilities that use, process, and discharge water back into lakes, streams, rivers, bays, and oceans.

Nonpoint source: Pollution that enters any waters of the state from any dispersed land-based or water-based activities. This includes, but is not limited to, atmospheric deposition, surface-water runoff from agricultural lands, urban areas, or forest lands, subsurface or underground sources, or discharges from boats or marine vessels not otherwise regulated under the NPDES program. Generally, any unconfined and diffuse source of contamination. Legally, any source of water pollution that does not meet the legal definition of “point source” in section 502(14) of the Clean Water Act.

Parameter: A physical, chemical, or biological property whose values determine environmental characteristics or behavior.

Point source: Sources of pollution that discharge at a specific location from pipes, outfalls, and conveyance channels to a surface water. Examples of point source discharges include municipal wastewater treatment plants, municipal stormwater systems, industrial waste treatment facilities, and construction sites that clear more than 5 acres of land.

Pollution: Contamination or other alteration of the physical, chemical, or biological properties of any waters of the state. This includes change in temperature, taste, color, turbidity, or odor of the waters. It also includes discharge of any liquid, gaseous, solid, radioactive, or other substance into any waters of the state. This definition assumes that these changes will, or are likely to, create a nuisance or render such waters harmful, detrimental, or injurious to (1) public health, safety, or welfare, or (2) domestic, commercial, industrial, agricultural, recreational, or other legitimate beneficial uses, or (3) livestock, wild animals, birds, fish, or other aquatic life.

Stormwater: The portion of precipitation that does not naturally percolate into the ground or evaporate but instead runs off roads, pavement, and roofs during rainfall or snow melt. Stormwater can also come from hard or saturated grass surfaces such as lawns, pastures, playfields, and from gravel roads and parking lots.

Watershed: A drainage area or basin in which all land and water areas drain or flow toward a central collector such as a stream, river, or lake at a lower elevation.

Acronyms and abbreviations

EAP	Environmental Assessment Program (at Ecology)
Ecology	Washington State Department of Ecology
e.g.	For example
EIM	Environmental Information Management database
EPA	U.S. Environmental Protection Agency
et al.	And others
GIS	Geographic Information System software
GPS	Global Positioning System
i.e.	In other words
MQO	Measurement quality objective
Non-UGA	Non-Urban Growth Area
NPDES	(See Glossary above)
PAH	Polycyclic aromatic hydrocarbons
PBDE	Polybrominated diphenyl ethers
PCB	Polychlorinated biphenyls
PLSSR	Puget Lowlands Salmon Recovery Region
PSAMP	Puget Sound Assessment and Monitoring Program
PSEMP	Puget Sound Ecosystem Monitoring Program (formerly PSAMP)
PSP	Puget Sound Partnership
QA	Quality assurance
QAPP	Quality Assurance Project Plan
QC	Quality control
RPD	Relative percent difference
RSD	Relative standard deviation
RSMP	Regional Stormwater Monitoring Program
SOP	Standard operating procedures
SWG	Stormwater Work Group
TOC	Total organic carbon
UGA	Urban Growth Area
WHSR	Status and Trends Monitoring for Watershed Health and Salmon Recovery
WQI	Water Quality Index

Units of measurement

°C	degrees centigrade
°F	degrees Fahrenheit
ft	feet
g	gram, a unit of mass
kg	kilograms, a unit of mass equal to 1,000 grams
km	kilometer, a unit of length equal to 1,000 meters
m	meter
mg	milligram
mg/Kg	milligrams per kilogram (parts per million)
mg/L	milligrams per liter (parts per million)
mL	milliliters
mm	millimeter
ng/g	nanograms per gram (parts per billion)
ng/Kg	nanograms per kilogram (parts per trillion)
ug/kg	micrograms per kilogram (parts per billion)