Quality Assurance Project Plan for Status and Trends Monitoring of Small Streams in the Puget Lowlands Ecoregion

for Monitoring Conducted by City of Redmond, Washington to Comply with NPDES Stormwater Permit Special Condition S8.B Requirements

August 2014

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This template Quality Assurance Project Plan is available on the Department of Ecology's Regional Stormwater Monitoring Program (RSMP) website for National Pollutant Discharge Elimination System (NPDES) municipal stormwater permittees at http://www.ecy.wa.gov/programs/wq/stormwater/municipal/status.html.

Data for the RSMP will be available on Ecology's Environmental Information Management (EIM) website at www.ecy.wa.gov/eim/index.htm. Search Study ID, RSMP_PLES2015. Data from City of Redmond will be under Study ID RSMP_RD_PLES2015.

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For Monitoring Conducted by City of Redmond, Washington to Comply with NPDES Stormwater Permit Special Condition S8.B Requirements

August 15, 2014

Approved by:

Signature: _____ Date: _____
Tanya MacFarlane, City of Redmond Field Lead

Signature: _____ Date: _____
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Signature: _____ Date: _____
Brandi Lubliner, Ecology RSMP Coordinator

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Anne Dettelbach, Ecology Regional Permit Manager

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Introduction

Development of a Stormwater Monitoring Strategy for the Puget Sound Region

The Stormwater Work Group (SWG) is a coalition of federal, tribal, state, and local governments; business, environmental, and agricultural entities; and academic researchers. All SWG members have interests and a stake in the Puget Sound watershed health and recovery. The SWG was convened by the Puget Sound Partnership (PSP) and the Washington State Department of Ecology (Ecology) in October 2008 to develop a regional stormwater monitoring strategy and to recommend monitoring requirements in National Pollutant Discharge Elimination System (NPDES) Municipal stormwater permits issued by Ecology. In 2012, the SWG became the first "topical workgroup" included in the Puget Sound Ecosystem Monitoring Program (PSEMP), an organization designed to coordinate regional monitoring efforts to assist in providing information to support Puget Sound recovery efforts.

An overall strategy for stormwater monitoring and assessment for the Puget Sound region was developed by the SWG in 2010 (SWG, 2010a). This strategy, summarized in Appendix A, included recommendations for status and trends monitoring in small streams in the Puget Sound watershed and in the Puget Sound nearshore, with a focus on an integrated approach to quantify stormwater pollutant impacts in small streams and in Puget Sound, and providing information to inform adaptive management of stormwater to reduce harm to the greater Puget Sound watershed ecosystem.

The SWG also recommended a specific NPDES municipal permittee-funded plan for monitoring the effects of stormwater in small streams and nearshore, under the current conditions in the Puget Sound region (SWG, 2010b). The resulting program, a subset of the overall strategy, is called the Regional Stormwater Monitoring Program (RSMP). Specifically, the RSMP includes status and trends monitoring of water quality and "watershed health" (physical habitat, water quality, sediment chemistry, and biological communities) in small streams in the Puget Sound lowlands; and of sediment quality, and mussel contaminants in the marine nearshore of Puget Sound. All the RSMP status and trends monitoring follows a probabilistic design (SWG, 2010a) that is compatible with ongoing status and trends monitoring programs such as Ecology's statewide monitoring program entitled *Status and Trends Monitoring for Watershed Health and Salmon Recovery* (WHSR) (Cusimano et al., 2006). Additional information about the experimental design, the goals, and the objectives for status and trends and other monitoring in the RSMP can be found in Appendix A of this report, in SWG (2010a and 2010b), and at the RSMP website (http://www.ecy.wa.gov/programs/wq/stormwater/municipal/status.html).

This QAPP has been generated to document the City of Redmond's status and trends monitoring design, independent of the regional status and trends monitoring, as Redmond chose to collect data within Redmond instead of paying into the regional program. This QAPP is a standalone QAPP and documents how the City of Redmond will collect status and trends data that is comparable to the regional status and trends monitoring program.

Scope of this Quality Assurance Project Plan

Ecology issued NPDES municipal stormwater permits for Phase I and Phase II communities (Ecology, 2012a, b) effective August 2013 through July 2018. All permittees located in Puget Sound were given two options to comply with the permits' Special Condition S8.B for status and trends monitoring requirements.

Option 1: Pay a prescribed amount into a pooled fund to support RSMP Status and Trends monitoring. These permittees' role is limited to providing permit-defined amounts of funding for coordinated implementation of monitoring at sites throughout the Puget Sound region.

Or

Option 2: Conduct their own status and trends monitoring at specific, assigned sites inside their jurisdictional boundaries, following the same protocols as those used for the RSMP.

In fall 2013, the City of Redmond officially selected the Option 2. This Quality Assurance Project Plan (QAPP) defines the permit-required small streams status and trends monitoring that will be conducted by Redmond. This QAPP serves as the Ecology-approved "RSMP QAPP" referenced in the permits. This QAPP defines the site confirmation and sampling protocols that the permittee will follow while conducting the monitoring as well as the data and reports that the city will produce to document the monitoring results. This QAPP was developed in accordance with Ecology's QAPP guidelines (Lombard and Kirchmer, 2004).

City of Redmond Roles and Responsibilities

The City of Redmond will conduct monitoring in assigned small streams sites in their jurisdictions. Redmond must submit the completed version of this QAPP to Anne Dettelbach, Ecology permit manager by July 15, 2014 for approval prior to sampling. The key completion dates for Redmond's required monitoring activities including site confirmation, field and laboratory work, data entry into Ecology's Environmental Information Management (EIM) database, and submission of monitoring summary reports, are summarized in Table 1.

Table 1. Key completion dates for permittees' QAPPs, monitoring activities, and reports for status and trends monitoring in small streams.

Due	Item	Description
June 15, 2014	Site selection and verification	Permittees have confirmed all sites to be monitored, including sufficient additional sites to sample if sampling attempted at any of the original sites is unsuccessful.
July 15, 2014	QAPP revisions due	Revised QAPP completed and submitted to Ecology.
August 15, 2014	Ecology QAPP review complete	QAPP reviewed within 30 days; RSMP Coordinator approves or provides comments via Permit Manager. Permittees are informed whether an inter-laboratory comparison will be done.
September 15, 2014	Deadline for final QAPPs	Final QAPP submitted to RSMP Coordinator with all comments addressed.
October 2014 - September 2015	Water quality sampling	Permittees conduct water quality sampling at the required number of lowland stream sites.
June - October 2015	Watershed health sampling	Permittees conduct watershed health (physical habitat, sediment chemistry, and biological) sampling at the required number of lowland stream sites.
December 31, 2016	Electronic data submittal due	All QA/QC'ed data submitted to Environmental Information Management (EIM) database.
March 31, 2016 and March 31, 2017	Stream monitoring reports due	Summary reports submitted to permit manager with Annual Report.

Table 2. City of Redmond project staff roles and responsibilities.

Phase I Permittees	Implementation of Stormwater Permit Monitoring	
Name/Contact	**Role**	**Responsibility**
Jerallyn Roetemeyer 425-556-2824	NPDES Stormwater Monitoring Project Manager	Manage overall compliance activities; verify whether QAPP is followed and monitoring data are of known and acceptable quality; ensure adequate training of staff, complies with corrective action requirements
Tanya MacFarlane 425-556-2764	Field Lead	Manage and oversee monitoring activities and sampling decisions; coordinate laboratory selection and delivery, and equipment maintenance; manage internal and external field teams.
Joe Capis Don Swayne Other TBD	Field Assistant(s)	Assist in collecting and processing field samples.

Ecology's role

Ecology's RSMP Coordinator will either approve or comment on this completed QAPPs and transmit approval or comments to the permittee via the permittee's Ecology Regional Permit Manager by August 15, 2014. After the sampling is completed and the permittee has completed quality assurance (QA) and quality control (QC) review of the data and submitted it to EIM, Ecology staff will review and notify the permittees with data quality corrections and when the data is ready for final upload to EIM. The RSMP Coordinator will review monitoring reports. Ecology permit managers will review all submittals for permit compliance purposes. Ecology staff and their responsibilities are listed in Table 3.

Table 3. Ecology project staff and responsibilities.

Ecology Staff	Administration of Stormwater Permits	
Name, Program, Location	**Role**	**Responsibility**
Brandi Lubliner - WQP Lacey, WA	RSMP Coordinator	Ongoing implementation and administration of RSMP. Reviews and approves completed QAPPs and project deliverables from permittees' monitoring efforts.
Anne Dettelbach - WQP NWRO: Bellevue, WA	Permit Manager	Ecology's contact for stormwater permittees including Redmond. Reviews QAPP and monitoring reports for permit compliance.
William Kammin Lacey, WA	Ecology Quality Assurance Officer	Draft template QAPP review and approval.
WQP staff, Lacey, WA	EIM Coordinator	Reviews and QAs data submitted by permittees and RSMP contractors.
EAP staff, Lacey, WA	Watershed Health database coordinator	Reviews and QAs data submitted by permittees and RSMP contractors.

NWRO: Northwest Regional Office
SWRO: Southwest Regional Office
EIM: Environmental Information Management database
WQP: Water Quality Program
EAP: Environmental Assessment Program

Coordination and Training

The City of Redmond will contribute data collection information and results to the RSMP. The City of Redmond is required to participate in a one-day RSMP or Ecology sponsored field-based training (to be held in summer 2015) for sediment and habitat sampling prior to conducting that sampling.

Sampling Site Selection and Evaluation

The sampling site selection and evaluation process described in this section of the QAPP concerns selecting suitable sites for inclusion in the RSMP based largely on a field visit to candidate sites before the sampling begins. Additional site suitability details that are considered

on the day of sampling are described in the specific sections of this QAPP detailing the sampling methods.

Site Lists

Sampling sites within the City of Redmond will be selected from a list of candidate sites referred to as the RSMP Master Sample Site list. Redmond will evaluate and confirm the suitability of sites in numerical order from lowest to highest in the ORDER column. The required numbers of sites to be monitored are stated in permit condition(s) S.8.B.2., which is twelve (12) sites for Redmond. Redmond will select from the list the required number of candidate sites located within their jurisdictional boundaries.

The initial 100 RSMP candidate sites (50 for each assessment region) are shown in Figure 1. The remaining candidate sampling sites will serve as replacement sites within Redmond. In the event that any of the candidate sites are found to be unsuitable for sampling, and/or in the event that an insufficient number of candidate sites are located in the permittee's jurisdiction, the permittee will sample sites from the remaining master sampling site list. If, from the remaining master sampling list, there still is not enough sites suitable for sampling, the permittee will sample only the suitable sites from the list.

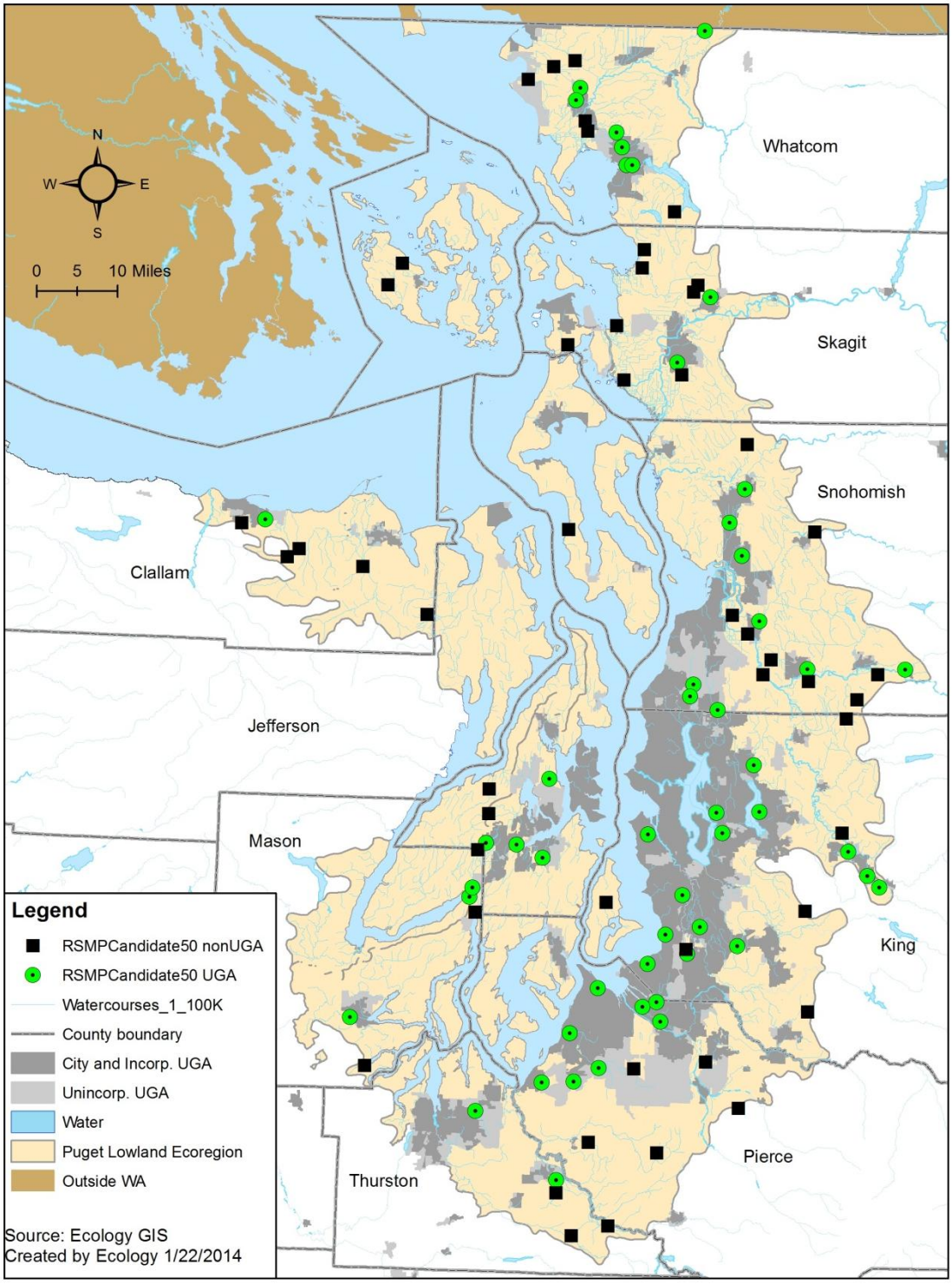

Figure 1 Initial 100 candidate site locations for the UGA and non-UGA assessment areas in the Puget Lowlands Ecoregion for the RSMP.

Candidate Sites

Candidate and alternative small stream sites for Redmond "Option 2" permittee are listed in Table 4 and shown in Figure 2. RSMP site lists are also available on Ecology's RSMP website at www.ecy.wa.gov/programs/wq/stormwater/municipal/status.html.

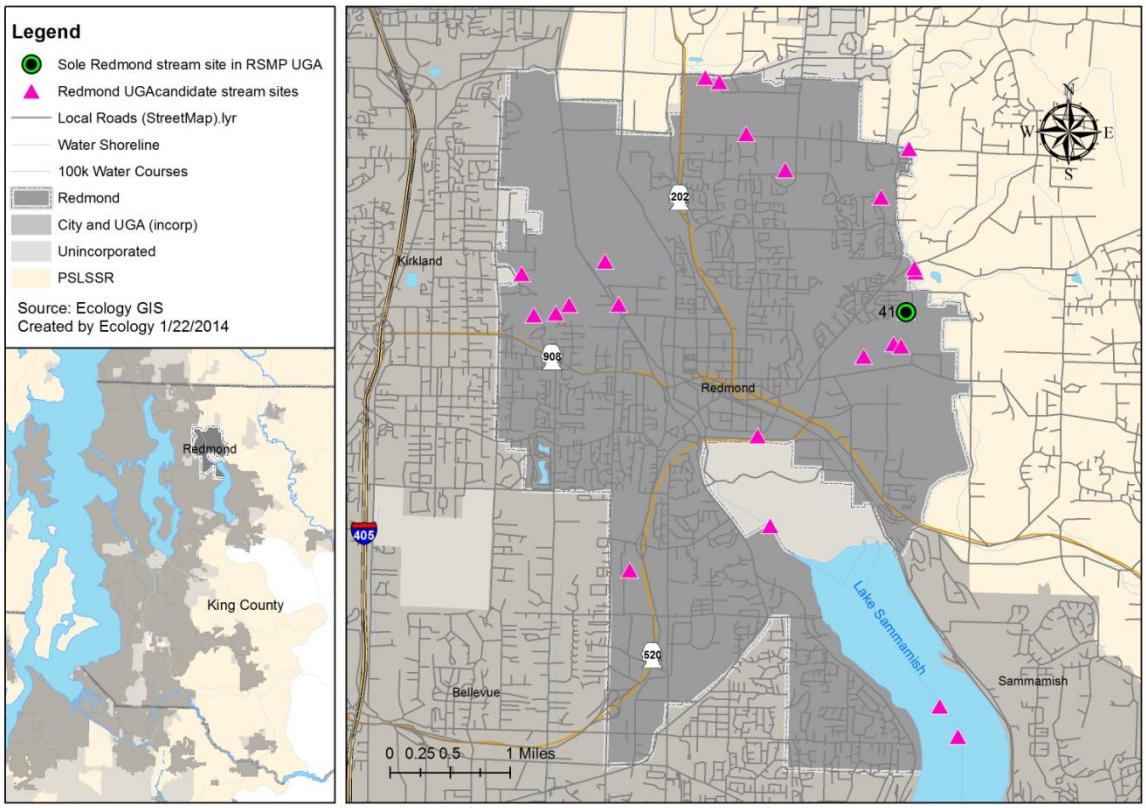

Figure 2. City of Redmond small stream candidate UGA sites.

Site Evaluation

Site evaluations, which must include a field visit to a candidate site, will determine the suitability of each site for monitoring to meet the RSMP goals. Site suitability is determined by selection criteria related to accessibility, hydrologic and geomorphic characteristics (flow, physical features, and salinity), and location relative to a candidate sites' original coordinates.

Candidate sites for evaluation are from the Master Sample Site list that was generated for Puget Lowland Ecoregion streams that drain to Puget Sound. Within that area, candidate sites are specified within each of the assessment regions: inside the Urban Growth Area (UGA) boundaries, and outside the UGA boundaries. Site evaluations will begin with the priority list of the initial 100 RSMP candidate sites, with 50 for each assessment region shown in the following "Site Lists" section. If any of those initial candidate sites are deemed unsuitable for monitoring, additional candidate sites for the relevant assessment region will be evaluated in the numerical order listed in the Master Sample Site list (from lowest to highest in the ORDER column).

The site evaluation procedure will start well in advance of the sampling season, ideally in March through June 2014, and will continue through the sampling season as necessitated by potential changes in site conditions that affect suitability for sampling. Desktop evaluation of candidate sites will be performed in advance of the initial site evaluation visit, and will include comparing candidate site coordinates to existing information on such items as surficial geology, parcel/property ownership, NHD waterbody type, historical streamflow and /or water quality data, and aerial photographs.

Criteria for Selecting a Suitable Sampling Site

Selection criteria for determining the suitability of a candidate site for monitoring to meet the RSMP goals are described below.

Accessibility Criteria

These criteria concern whether access to a site is permitted by the land owners, and if the site can be safely accessed and sampled throughout the year. A site may also be deemed unsuitable, or impracticable, for sampling certain if more than one hour is required to access the site from the nearest parking location.

Permission

If a candidate site is not obviously accessible through public property, property owners and/or tenants whose property will need to be accessed will, if feasible, be contacted prior to site evaluation. Parcel information gained from the desktop evaluation will be researched and a good faith effort to contact owners or tenants will be made. A site will be deemed unsuitable for sampling if permission has been denied by all land owners, tenants, or resource managers along the entire hydrologic reach (see *Location Criteria*, below). The Washington State Department of Natural Resources (WDNR, 2010) describes how to discern public and state-owned waters.

Safety

Overall safety conditions for access and sampling will be assessed prior to sampling, based on state and federal law and organizational policy. But it is ultimately the responsibility of the field crew at each time of arrival to decide if it is safe to enter the stream to conduct the sampling. Appropriate reasons for disqualifying a site from sampling may include:

- flow is too swift or too deep;
- route of entry is unstable;
- hostile people or animals are present.

Flow, Physical, and Salinity Criteria

These criteria concern the conditions of the stream and streambed with regard to the specific types of data desired for the RSMP. To be considered a suitable sampling site, the waterbody at the candidate site coordinates must be on a stream or small wadeable river, and not on a lake, pond, wetland, or estuary. Specifically, the waterbody must have:

- a net flow of water that is unidirectional;
- defined left and right banks readily discernible from mid-stream;
- uninterrupted surface-water flow for more than half the length of approximately 20 bankfull widths or a minimum of 150 meters surrounding the candidate site coordinates;
- perennial flow (as best as can be determined at the time of the site visit);
- flow in a natural channel that might have been highly modified, but was not constructed (such as canals, ditches, or pipelines);
- natural substrate on the channel bottom;
- freshwater, as defined by a water column with more than 95 percent of its depth with less than 1 part per thousand salinity at any time during the year. Multiple lines of evidence may be used to make this estimation (*e.g.*, vegetation, proximity to a known estuary, or salinity measurement).

Location Criteria

The following location rules apply such that the site reflects the intended probabilistic stream characteristics. During the site evaluation field visit, the field crew will attempt to access the site at the given coordinates or as nearby as possible, with recognition of the challenges of sampling in urban areas, particularly in gaining access to discretely defined locations. Ideally, a suitable sampling location will be located within 250 meters of the given candidate site coordinates.

However, if access, flow, physical, and chemical criteria are not met within this distance, the field crew may continue to investigate locations upstream and downstream of the initial reach with the objective finding a suitable site that maintains the original candidate site characteristics. More specifically, suitable sampling sites upstream and downstream of the candidate site coordinates must fall within these constraints:

- the final site is the same size class of the original candidate site; and
- there are no continuous surface-water inflows in excess of approximately 25 percent of the flow already in the reach[1]; and

[1] During the site confirmation process, questions about a how specific reach is defined should be directed to RSMP Coordinator Brandi Lubliner at 360-407-7140.

- either :
    - there is no substantial, abrupt change in adjacent land use such as from residential to industrial, or from native vegetation to developed conditions; or
    - the final site is less than 500m from the original candidate site coordinates.

The RSMP will determine how to interpret (*i.e.*, statistically weight) the data from all of the sites sampled by permittees who have chosen to conduct their own monitoring.

Documentation of Site Evaluations

Redmond must verify all sites given the suitability criteria above. Documentation of observations from both the desktop and field visits will be recorded and provided to the RSMP upon request. Permittees will provide a table listing the decisions and reasons for site selection or disqualification resulting from the site evaluations to the RSMP Coordinator by June 15, 2014.

Site Evaluation Results

Redmond followed the Criteria for Selection Suitable Sites defined by this QAPP. The results of evaluation has resulted in only seven probable sites, with one of the seven sites likely to have high flows all year around that will make it unsafe for sampling. As required, Table 4 below provides the full list of potential sites, identifies which sites meet the criteria in the sample yes/no column, and (if applicable) the reasoning for disqualifying a site is detailed in the notes column. Figure 3 is a map of sites that have been found suitable for Redmond's status and trends monitoring.

Mid-Study Changes

The City of Redmond will make a good faith effort to sample at the selected sites per the QAPP requirements. If a site becomes unsuitable for sampling during the course of the study, the RSMP Coordinator will be notified. Reasons a site may become unsuitable include, but are not limited to: a stream goes dry; the adjacent parcel(s) change ownership, and the new owner does not grant permission; or natural causes such as mudslides or animals make the site no longer safe to access.

If suitability conditions change prior to sampling a site for watershed health and sediment chemistry and the site is no longer suitable, then a new RSMP site needs to be identified from the list in order. If suitability conditions change after sampling the site for watershed health and sediment chemistry and the site is no longer suitable, sampling will simply discontinue for this round of RSMP sampling and conditions noted on the site lists.

Table 4. Redmond - Incorporated UGA Small Stream Sites-all sites located within King County.

ORDER	LON_DD	LAT_DD	Sample Yes/No	WRIA	Subwater-shed Name	NHD GNIS	Notes	Land Type/Owner	Redmond Location
41	-122.091069	47.68306	No	8	Bear Creek	Bear Creek	Not wadable, too deep, too swift	Friendly Village developed open space	Bear Creek at Friendly Village
58	-122.089474	47.687881	No	8	Bear Creek	Bear Creek	Not wadable, too deep, too swift, 50 m from site 784	Elm Court, developed open space	Bear Creek at Novelty Hill
158	~~-122.142431~~ -122.141	~~47.683244~~ 47.683	Yes	8	Bear Creek-Sammamish River	Peters Creek	Move site downstream 73m	Arena Sports (no access) Seneca (NGPE) City of Redmond, Wallace (NPGE), developed medium intensity	Peter's at Arena Sports, via Seneca, walk upstream
221	-122.093184	47.679188	No	8	Bear Creek	Bear Creek	Not wadable, too deep, too swift, bottom is muddy, access is challenging	WASHDOT CB Richard Ellis NGPE , herbaceous	Bear Creek at Evans Confluence
419	-122.091832	47.678924	No	8	Bear Creek	Evans Creek	Not wadable, too deep, too swift, channel incised	WASHDOT CB Richard Ellis NGPE, hay/ pasture	Evans upstream of Bear Confluence
459	-122.113141	47.6999	No	8	Bear Creek-Sammamish River		Channel braided and dry as of June 2014. Headwater channel-multiple joining channels	City of Redmond, developed low intensity	Headwaters of High School Creek
512	-122.117131	47.667758	Maybe	8	Bear Creek	Bear Creek	Not wadeable, too deep, too swift. Under Construction. Completion date approx. 9/14	City of Redmond, developed open space	Mouth of Bear Creek
626	-122.098486	47.677608	Yes	8	Bear Creek	Bear Creek	Not wadable in high flow. Bridge sampling/ for estimated flow use King County gage installed at Union Hill Road and Bear Creek. Bridge removal expected within 2 years	Swedish NGPE, shrub/scrub, replanted buffer in 2010, developed open space	Behind Swedish Medical Center
656	-122.125271	47.710295	Yes	8	Bear Creek-Sammamish River		Rechannelization completed in 2013	Greystone, NGPE plat not finaled developed open space	High School Creek upstream of NE 124th
657	-122.091099	47.702746	No	8	Bear Creek	Bear Creek	Not wadable, too deep, too swift, not safe, access issues	M&M Autobody not friendly, not working with us,	Up and downstream is out of Redmond City limits

ORDER	LON_DD	LAT_DD	Sample Yes/No	WRIA	Subwater-shed Name	NHD GNIS	Notes	Land Type/Owner	Redmond Location
								developed medium intensity	
736	~~-122.1450~~ -122.142	~~47.688406~~ 47.688	Yes	8	Bear Creek-Sammamish River	Willows Creek	Move location to nearest stream 270 m to the southeast	PSE Easement, developed medium intensity	Willows Creek at Overlake Church
753	-122.151274	47.683061	No	8	Bear Creek-Sammamish River	Willows Creek	Not accessible, not unidirectional flow, no defined right and left bank, change of land use and greater than 25% increase in flows downstream	Emergent herbaceous wetlands PSE transmission lines, easement	Willows Creek at PSE property
784	-122.089709	47.688351	No	8	Bear Creek	Bear Creek	Not wadable, too deep, too swift, not safe, 50 m from site 58	Elm Court, developed open Space	Bear Creek and Novelty Hill Road
798	-122.114555	47.65697	No	8	Bear Creek-Sammamish River	Samma mish River	Not wadable, in Sammamish River	Park/Non Wilderness, developed open space	Sammamish River at Marymoor Park
814	~~-122.159812~~ -122.157	~~47.686714~~ 47.686	Yes	8	Bear Creek-Sammamish River		Move site downstream 270m, site is not located on a stream	PSE Transmission lines, easement, developed open space	Gun Club
874	-122.139456	47.651246	No	8	Bear Creek-Sammamish River		There is not a stream within 0.5 mile	Nintendo of America, developed high intensity	Nintendo Property, on the green roof
885	-122.157503	47.681774	Yes	8	Bear Creek-Sammamish River	Willows Creek	Channel is braided but have collected benthos in past years	Deciduous forest, Redmond City Park and Maple Brook Lane Homeowners Association	Willows Headwaters
886	-122.12775	47.710834	No	8	Bear Creek-Sammamish River		Site dry 4-6 months per year	City of Redmond	Kensington Estates Rehabilitation Project in 2010
900	-122.083631	47.635625	No	8	Lake Sammamish-Sammamish River		Not a qualifying stream. Site is in a lake.		Sammamish River
977	-122.080264	47.632008	No	8	Lake Sammamish-Sammamish River		Not a qualifying stream. Site is in a lake.		Sammamish River

Figure 3. Suitable Status and Trends Sites in Redmond

Measurement Quality Objectives

Measurement quality objectives for small stream monitoring described here are to obtain and analyze sufficient numbers of high quality samples to meet the goals and objectives of the RSMP status and trends program. Data quality indicators include precision, bias, sensitivity, representativeness, comparability, and completeness (Appendix B). The biological and habitat indicators adopted for this program come from Ecology's Watershed Health Monitoring Program for small streams (Adams, 2010a). The adopted water chemistry indicators come from Ecology's Ambient Water Quality monitoring program (Hallock, 2012), and the adopted sediment chemistry indicators come from multiple monitoring efforts within Ecology (Dutch et al., 2010; Johnson, A., 2010; and Meredith and Furl, 2008).

Field measurements

Measurements of water quality, sediment size estimation, and stream habitat variables are taken by field staff during a sample collection event. The City of Redmond must follow the collection methods, reporting requirements, and quality control (QC) procedures summarized in the *Field Operations* sections of this report. This approach will provide field measurement data that meet measurement quality objectives (MQOs) for status and trends monitoring for small streams under the RSMP, listed in Tables 19 and 20.

The City of Redmond will make a good faith effort to collect monitoring data described per QAPP requirements. If a water quality sample or measurement is missed on occasion, a second effort will be made to collect the sample within the same month. If a second attempt is also unsuccessful, then the RSMP Coordinator will be notified, and a third attempt is not required.

Reasons a sample or measurement may not be made include, but are not limited to: a stream goes dry; the stream site cannot be accessed due to high flow conditions, vandalism, extreme climatic conditions, or monitoring equipment has a sudden failure. Flow measurements may need to be estimated using high water mark, staff gages, or other estimation techniques during the winter months to minimize exposure to hazardous conditions for staff. Water quality samples and measurements made during very high flows may be made from anywhere within the reach.

Laboratory measurements

Sediment and water quality laboratory analyses will be conducted at laboratories that have Ecology-accreditation for those analyses, except those noted in Table 5. Ecology's Laboratory Accreditation Program maintains a searchable database that may be accessed from this website: www.ecy.wa.gov/programs/eap/labs/lab-accreditation.html. (Laboratory methods and reporting limits are listed in the *Quality Control* section).

Unless alternative methods are approved by Ecology in writing, the analytical methods listed per the QAPP shall be used by Permittees when monitoring as required by section S8B.2 of this permit. Any alternative method proposed by the Permittee must have a similar reporting limit or

must be justified as adequate for the likely range of concentrations. Permittees are not guaranteed approval of their alternative methods or reporting limits. Taxonomic identification will be 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.

Work performed by the City of Redmond is expected to meet the quality control requirements of the analytical methods stated in this QAPP. These requirements are summarized in the *Quality Control* section of this document.

Laboratory selection

Multiple laboratories will be needed to ensure sample completeness. Contracting for laboratories is a responsibility of the permittee. Permittees may consider their own or other permittee's laboratories, commercial laboratories, or Ecology's Manchester Environmental Laboratory (MEL). Laboratories for water and sediment parameters will be selected based on their current accreditation status with Ecology (www.ecy.wa.gov/programs/eap/labs/search.html) and their ability to achieve acceptable limits of detection for the parameters measured as part of this project.

Table 5 Selected laboratories for sample processing.

Laboratory Name	Analytical Purpose	Address	Phone
AmTest	Fecal Coliform, Orthophosphate and Turbidity	13600 NE 126th PL # C Kirkland, WA 98034	425.885.1664
Manchester Environmental Laboratory (MEL)	Water Quality testing for Ammonia, Chloride, Hardness, Nitrate-Nitrite-N, Total Nitrogen, Total Phosphorus, Total Suspended Solids. Sediment sample testing for metals, Total Organic Carbon, Percent Solids, Pesticides, Herbicides, Phthalates, PBDEs, PAH, PCBs	7411 Beach Drive East Port Orchard, WA 98366	360. 871.8800
Manchester Environmental Laboratory (MEL)	Water sample testing for chlorophyll a	7411 Beach Drive East Port Orchard, WA 98366	360. 871.8800
Rhithron Associates, Inc.[1]	Stream benthos and periphyton	33 Fort Missoula Road Missoula, Montana	406.721.1977

		59804	
AXYS Analytical Services Ltd.[2]	Sediment sample testing for Pharmaceuticals and personal care products (PPCPs) and hormones/steroids (H/S).	2045 Mills Road W. Sidney BC Canada V8L 5X2	+1 .250. 655.5800
Manchester Environmental Laboratory (MEL)	Sediment sample testing[3] for inter-laboratory comparison	7411 Beach Drive East Port Orchard, WA 98366	360. 871.8800

[1] Chlorophyll a may be done either at Rhithron or MEL or another Ecology approved laboratory. Rhithron must be used for benthos and periphyton to remain consistent with the RSMP.

[2] Axys Analytical is required for PPCP and H/S analysis.

[3] There will be an inter-laboratory comparison element for sediment chemistry samples if different laboratories are used between the Option 2 permittees and the RSMP. One set of samples for the inter-laboratory comparison must be sent to MEL.

General Field Sampling Procedures

This section describes field sampling procedures. This QAPP's appendices C through F provide detailed procedures for watershed health, water quality monitoring and quality control. Appendix G provides field forms that may be used in the field for data collection.

Scientific collection permit

Permittees are responsible for obtaining the necessary permits for sampling macroinvertebrates. The scientific collection permit is required by the Washington Department of Fish and Wildlife (http://wdfw.wa.gov/licensing/scp). Permittees are advised that it may take several months to process and acquire these permits.

Safety

The City of Redmond should develop a site-specific safety plan including at a minimum the following elements.

Sampling

Field activities should be conducted by at least two people. Activities can be parsed into tasks to be accomplished by one or more persons at a given time. A contact person will be designated at the office to which field personnel report at the end of each day at pre-designated times. Staff must carefully plan field activities and obtain permission to access private land. Staff may also notify the land owner which day they will be sampling.

Field and laboratory preservatives

Biological samples collected from streams must be preserved immediately following storage in containers. Inadequate preservation often results in (1) loss of prey organisms through consumption by predators, (2) eventual deterioration of the macroinvertebrate specimens, and/or (3) deformation of macroinvertebrate tissue and body structures, making taxonomic identification difficult or impossible.

The field preservative used for biological samples is 85% denatured ethanol. The preservative is typically prepared from a stock standard of 95% denatured ethanol. Flammability, health risks, protective equipment, and containment information are listed on warning labels supplied with the preservative container. Detailed information can be found with the Materials Safety Data Sheets (MSDS). Minimal contact with the 95% denatured ethanol solution is recommended.

For the water samples, several of the nutrient parameters require field preservation using hydrochloric acid (HCL) or sulfuric acid (H_2SO_4) (see Table 14). These jars can be ordered with preservative in them, and they must be handled carefully to avoid spilling the preservative during sample collection.

Equipment and maintenance

A list of equipment necessary to complete both watershed health and water quality monitoring field activities is presented in Appendix H of this QAPP.

Servicing of scientific instrumentation will follow the instrument manufacturers' methods and will be conducted as needed. General maintenance will consist of equipment inventories, inspections, testing, and replacement of worn, torn, or missing components.

Equipment decontamination and prevention of spread of aquatic invasive species

Field work will be conducted and equipment cleaned to prevent the spread of invasive species. Staff practices and equipment that contact multiple surface waters will, at a minimum, be cleaned according to Ecology's standard operating procedure (SOP) EAP070, *Minimizing the Spread of Aquatic Invasive Species* (Parsons et al., 2012). These procedures will be followed at the end of each work day or upon leaving a water body before entering another. Some areas are designated to be of "Extreme Concern"; these areas are shown in several maps at the following link: www.ecy.wa.gov/programs/eap/InvasiveSpecies/AIS-PublicVersion.html

All sediment material not retained for analyses or archiving will be rinsed near the sampling location with stream water. Also, the sediment scoop and bowl will be rinsed on-site. Used equipment will not be used at another site on the day of sampling, unless completely cleaned.

Any portion of the sampling equipment (nets, sample container holders, scoops, bowls), filters, or other materials coming into contact with the sample will be decontaminated prior to use or will be certified as pre-cleaned from the equipment source. Sampling equipment and containers will be prepared prior to the sampling event. Otherwise, cleaning will match the purpose of sampling. For example, the kick nets only need to be free of benthic macroinvertebrates, leaves, and sticks and air dried. Nets, buckets, funnels, and other general sampling equipment may be washed or rinsed with tap water and air dried.

Water sampling equipment

The containers used to collect nutrient samples should be washed in 10% HCl and triple rinsed in deionized water prior to collecting each sample (Ward, 2007a).

Sediment sampling equipment

The stainless-steel scoops and bowls used to collect sediments for organic analysis need to be properly cleaned using the following procedure. Clean implements will be stored in aluminum foil or polyethylene bags for transport to the field station. Stainless-steel sampling implements, including the spoons, bowls, and stirrers, will be cleaned sequentially:

1. Washing in non-phosphate detergent (Liquinox) and hot tap water.
2. Rinsing with hot tap water.
3. Rinsing with 10% nitric acid (if sampling for metals).
4. Rinsing with deionized water three times.
5. Air drying in clean area free of contaminants.
6. Rinsing with pesticide-grade acetone (if sampling for organics).
7. Air drying in clean area free of contaminants.

After drying, equipment will be wrapped in aluminum foil and stored in polyethylene bags until used in the field. Sampling equipment will be dedicated to a single site. Reuse will require cleaning as outlined in the procedure above, which is based on EPA guidelines (EPA, 1990).

Labeling samples

Labeling is used to identify where and when a sample was collected and the analyte(s) in that sample to be analyzed. Laboratory-prepared bottles will be labeled to identify the cleanliness and/or preservative contents for each bottle. Bottles will be either numbered or pre-labeled to ensure proper handling. Labels will be filled out in pencil or permanent pen, placed on sample containers. Sample labels will contain the following information:

- Site name and RSMP SITE_ID
- Analysis to be performed
- Date and time of sampling
- Sample ID or coding information
- Sample numbers (1 of 3, 2 of 3, and so on)
- Name/initials of field tech performing the sampling

This labeling information will be written in the chain-of-custody forms, which are discussed below.

Chain-of-custody procedures for samples

Chain-of-custody (COC) procedures are necessary to ensure thorough documentation of handling for each sample, from field collection to laboratory analysis. The purpose of this procedure is to minimize errors, maintain sample integrity, and protect the quality of data collected. A COC form will accompany each cooler of samples sent to a laboratory. Individuals who manipulate or handle these samples are required to log their activities on the form. When the laboratory receives a cooler of samples, it will assume responsibility for samples and maintenance of the COC forms. The laboratory will then conduct its procedures for sample receipt, storage, holding times, tracking, and submittal of final data to the responsible parties. Example COC forms for benthic and water or sediment chemistry samples are in Appendix L.

Holding times and Sample storage for transport

Holding times between sample collection and sample analysis in a laboratory are dependent on analysis type and method of analysis. Holding times and methods used are shown in tables 11, 12, 16, 19 and 20).

All collected samples will be stored in ice filled coolers during collection of multiple sites. Samples will be delivered to the lab in coolers with ample ice, dry ice or ice packs to keep samples cold and clean from contamination.

Watershed Health Monitoring

This section describes watershed health monitoring which refers to physical habitat, soil chemistry measurements, and biological community characterization. These measurements and samples will be collected once at each site between July 1 and October 15, 2015. The biological, physical habitat, and sediment chemistry parameters for watershed health monitoring are presented in Tables 6 and 7.

The RSMP analysis for PPCPs and H/S (personal care products and hormones and steroids) in small streams will be conducted on a sub-set of sites, those that number from 0-120 in the "ORDER" column in the master site list available at Ecology's RSMP website. No suitable sites within Redmond are included in sites ordered 1 – 120. As such, those parameters have been removed from Table 7 below.

Table 6. Biological and habitat parameters for watershed health monitoring.

Biological and Habitat Parameters
Aquatic macroinvertebrates (benthos)
Periphyton
Physical habitat (discharge, slope and bearing, wetted width, bankfull width, bar width, substrate size, substrate depth, shade, human influence, riparian vegetation, large woody debris).

Table 7. Sediment chemistry parameters for watershed health monitoring.

Sediment Parameters
Grain size estimation
Total organic carbon (TOC)
Percent Solids
Metals (arsenic, copper, lead, and zinc)
Polynuclear aromatic hydrocarbons[1,2]
Pesticides[1,3]
Phthalates[1,4]
Polybrominated diphenyl ethers (PBDEs)[1,5]
Polychlorinated biphenyls (PCBs)[1,6]

[1] Additional parameters recommended by the SWG, to the extent that funding becomes available. Permittees selecting Option 2 will monitor for these additional parameters.

[2] PAH compounds include: 1-methylnaphthalene, 2-methylnaphthalene, 2-chloronaphthalene, acenaphthylene, acenaphthene anthracene, benzo(a)anthracene, benzo(a)pyrene, benzo(b)fluoranthene, benzo(ghi)perylene, benzo(k)fluoranthene, dibenzo(a,h)anthracene, dibenzofuran, carbazole, chrysene, fluoranthene, fluorene, indeno(1,2,3-cd)pyrene, naphthalene, phenanthrene, pyrene, retene.

[3] Pesticides include: 2,4-D, triclopyr, carbaryl, and chlorpyrifos.

[4] Phthalates include: bis(2-ethylhexyl) phthalate, butyl benzyl phthalate, diethyl phthalate, dimethyl phthalate, di-n-butyl phthalate, and di-n-octyl phthalate.

[5] PBDEs include: 47, 49, 66, 71, 99, 100, 138, 153, 154, 183, 184, 191, 209.

[6] PCBs include: all 209 congeners.

Field activities and protocols for watershed health monitoring

The field activities and associated data collection protocols required for watershed health monitoring are described below. For each activity, the reader is also referred to specific appendices in this QAPP. The protocols in this QAPP have been tailored to the RSMP and based on Ecology's ambient biological monitoring program (Adams, 2010a) and Ecology SOPs for measuring physical habitat (Clinton, 2009; Kennedy, 2009; Werner, 2009a,b,c d).

The primary field data and samples collected during watershed health data collection event (DCE) include stream discharge, sediment chemistry, benthic macroinvertebrates, periphyton, and habitat. Water quality and chlorophyll a may also be monitored (as described in the *Water Quality Monitoring* section of this QAPP) during a watershed health DCE, or it may be completed during a separate site visit.

One site may be sampled in several workdays by a two-person field team (Table 8). The following activities must be completed in the listed order over the course of several days to avoid damage to biological specimens while sampling sediment or measuring habitat:

1. Site verification and layout
2. Benthos
3. Periphyton and Chlorophyll a sample collection
4. Sediment chemistry sample collection
5. Habitat measurements

Table 8. Typical daily work flow for a watershed health data collection event.

Activity	# Staff	Day of Sampling				
		1	2	3	4	5
Verification & Layout/Streamflow	1					
Benthos/periphyton sample collection	2					
Chlorophyll a sample collection	2					
Sediment sample collection	2					
Habitat measurements	2					

Watershed health monitoring is conducted according to a reach-wide sampling scheme (Hayslip, 2007) that uses equidistant transects set along a stream reach with a length equal to 20 times the bankfull width (10 upstream and 10 downstream) at the sample site. Benthos and sediment sampling along the reach are conducted systematically without consideration for habitat type. For example, the sampler might start collecting on the left bank at transect one, move to midstream at transect two and the right bank at transect three, and move back again until 8 square feet have been sampled from randomly chosen transects. A list of the field activities required for monitoring watershed health and the corresponding appendices of this QAPP that describe the procedures and protocols is given in Table 9.

Table 9. Field activities for watershed health monitoring, QAPP appendices describing the procedures, and where at a site the activities take place.

Activity	Appendix	Where[1]
Site verification and layout	C-1	Entire site
Benthos sample collection	D-1	Major transects
Periphyton sample collection	D-3	Major transects
Sediment chemistry sample collection	C-4	Near index station
Habitat measurements		
Bank measurements	C-5	Major transects
Substrate and depth measurements	C-6	Major transects
Shade measurements	C-7	Major transects
Estimating fish cover	C-8	Major transects
Human influence	C-9	Major transects
Riparian vegetation structure	C-10	Major transects
Measuring thalweg depth	C-11	Thalweg transect
Large woody debris tally	C-12	Major transect
Habitat unit descriptions	C-13	Thalweg transect
Side-channel descriptions	C-14	Thalweg transect
Width and substrate measurements	C-15	Minor transects
Measuring slope and bearing	C-16	All transects

[1] Transects and index stations are described in Appendix C-1.

Table 10 presents the same information according to which activities take place at the different transects and stations that are established during the site layout.

Table 10. Field procedures by station within a site (transects and index stations are described in Appendix C-1).

Major Transect	Minor Transect	Thalweg Transect	Near the Index station (Transect F) [6]
Slope[1]	Slope[1]	Slope[1]	In-situ measures
Bearing[2]	Bearing[2]	Bearing[2]	Sediment sampling
Wetted width	Wetted width	Thalweg depth	Discharge
Bankfull width	Bankfull width	Habitat unit presence	GPS coordinates [5]
Bar width	Bar width	Side channel presence	
Substrate sizes	Substrate sizes	Edge pool presence	
Substrate depths		Bar presence	
Fish cover by class			
Shade			
Human influence			
Riparian vegetation			
Benthos[3]			
Periphyton[3]			
Large woody debris [4]			
GPS coordinates [5]			

[1] Slope can be measured among any combination of convenient contiguous stations, as long as crews determine total elevation gain or average slope across the entire site length.
[2] Bearing: normally 20 measurements per site: one measurement at each major transect and one at each minor transect. Supplemental measurements sometimes are needed from intermediate thalweg transects.
[3] The benthos and periphyton samples are composite samples from eight randomly selected major transects.
[4] Large woody debris is tallied across the full length of the site, but records are kept for counts between major transects, on the *Thalweg Data Form*.
[5] Global Positioning System (GPS) is required at site coordinates (index station) and at two major transects for small streams (top and bottom of site).
[6] Except for GPS coordinates, these measurements can be done anywhere within the site, but near the index station (mid-reach, transect F) is preferred.

Field quality control procedures

To ensure the quality and consistency of sample collections, equipment maintenance and sample collection protocols described in the appendices of this QAPP will be followed.

Sample holding times

Holding times are the maximum allowable length of time between sample collection and laboratory manipulation. Holding times are different for each analyte and are in place to maximize analytical accuracy and representativeness. Each sample collected will be packaged in a container and labeled accordingly. The packaged samples will be held in an ice filled cooler. If necessary, the permittee will coordinate with the analytical laboratory to ensure samples can be transported, received, and processed during non-business hours. Sample containers will be transported or sent by the field team to the analytical laboratory, following established sample

handling and chain-of-custody procedures. At the laboratory, samples may be further divided for analysis or storage.

Tables 11-12 list sample volumes, holding times, containers, and preservation requirements for sediment and biological samples collected during a watershed health DCE. Appendix D elaborates on the bottles and other equipment needed for biological samples.

Table 11. Sample containers, amounts, holding times, and preservation for sediment samples.

Analysis	Container[1]	Holding Time	Preservative[2]
Grain Size	8 oz plastic jar	6 months	Do NOT freeze or dry; Cool to ≤6°C
Percent solids	2 oz glass jar	7 days	Cool to ≤6°C
Total organic carbon (TOC)	2 oz glass jar	14 days/1 year if frozen	Cool to ≤6°C; PSEP standard (1986): may freeze at ≤18°C at lab
Metals (As, Cu, Pb, Zn)	4 oz glass jar[3]	6 months	Cool to ≤6°C
Pesticides – 2,4-D, triclopyr, carbaryl, chlorpyrifos	8 oz glass jar[3,4]	14 days	Cool to ≤6°C
PAHs	8 oz glass jar[3,5]	14 days/1 year if frozen	Cool to ≤6°C; PSEP standard (1986): may freeze at ≤18°C at lab
Phthalates (can be sent in same container as PAHs)	8 oz glass jar[3,5]	14 days/1 year if frozen	Cool to ≤6°C; PSEP standard(1986): may freeze at ≤18°C at lab
PBDEs	8 oz glass jar[3]	14 days/1 year if frozen	Cool to ≤6°C; PSEP standard(1986): may freeze at ≤18°C at lab
PCBs (congeners)	8 oz glass jar[3]	14 days/1 year if frozen	Cool to ≤6°C; PSEP standard(1986): may freeze at ≤18°C at lab

[1] No additional sample volume is needed for analysis and QC samples if the jar is filled, with the exception of PPCPs and H/S, check with laboratory. Some parameters may be sampled from same container, check with laboratory.

[2] Preservation needs to be done in the field, unless otherwise noted. Ice will be used to cool samples to approximately 4-6°C.

[3] Glass containers with Teflon-lined lids, certified clean by manufacturer or laboratory in accordance with OSWER Cleaning Protocol #9240.0-05 (MEL, 2008).

[4] All four pesticides can likely be combined in the same jar; check with laboratory.

[5] PAHs and phthalates can likely be combined in the same jar; check with laboratory.

Table 12. Sample containers, amounts, holding times, and preservation for biological samples.

Analysis	Container[1]	Holding Time	Preservative[2]
Periphyton - for species analysis	500 mL brown poly bottle	6 months	Lugol's iodine to 1%; Cool to ≤4°C
Periphyton - for chlorophyll *a*[3] analysis	500 mL poly vial	25 days, 2 days if filtered by lab	Field filtered 0.7 micron glass microfiber filter; folded in quarters and wrapped in foil then polyethylene bag; Cool to ≤4°C
Macroinvertebrate	3.8 L wide-mouth poly jars	Indefinitely	Field preserved with ethanol

[1] Replicate samples should be collected in additional container.

[2] Preservation should be done in the field, unless otherwise noted. Ice will be used to cool samples.

[3] Chlorophyll a may be sent to the water quality laboratory, as opposed to the benthos laboratory.

Documentation

Field data measurements will be recorded in the field; example field forms are provided in Appendices C, D, and G for biological, habitat, and sediment chemistry monitoring, respectively. These forms are used in other Ecology monitoring efforts, as print documents and taken into the field for recording. At this time, Ecology has developed new electronic field forms and software to improve field documentation for their Watershed Health Monitoring Program with completeness and data entry to the EIM database. The anticipated completion date is spring 2015. Once completed, the software will be freely distributed to the permittees conducting monitoring for the RSMP for use during the 2015 field season for habitat, benthos and sediment sampling. This software and data management are further discussed in the Data Management and Reporting section of this QAPP.

While development and distribution of the field software is intended, as a back-up the field forms provided in the appendices can be printed and used. Paper field forms should be printed on water-resistant paper. When completed, these field sheets will be entered into the above mentioned electronic forms at the office, and paper copies will be stored in an organized central filing location. Forms and documentation will include the station visit/maintenance sheet, meter calibration, and chain-of-custody forms. All entries on field documents will be made in pencil or permanent pen and will list the field technician name(s). Any errors or typos will be crossed out and rewritten by the technician who recorded the data. All corrections will be initialed and dated when made.

If field sampling or procedural errors are discovered, action will be taken to manage and correct those errors. Corrections may occur with corrective editing, relabeling, or, if warranted, flagging, discarding, and re-sampling. If a consistent error persists, an amendment to the sampling procedures may be required.

Watershed environmental health replicate samples

Composited benthic macroinvertebrate, periphyton, and sediment field replicate samples will be collected one time for up to 12 samples collected. Field replicates will be collected by splitting composited samples. All field replicates will be labeled similar to other samples, so that the

sample has its own unique number. These replicate samples will be submitted blind to the laboratory, with all other field samples.

Field blanks

Field blanks are not required for watershed health monitoring.

Table 13. Field quality control schedule for watershed health samples collected.

Field Sample Collected	Frequency	Control Limit	Corrective Action
Composited benthic macroinvertebrate, periphyton, and sediment field replicate	Once	Qualitative control – Assess representativeness, comparability, and field variability	Review procedures; alter if needed

Water Quality Monitoring

Water quality monitoring consists of collecting monthly year-round (October through September) measurements and water samples at each suitable sample site. Measurements and sample collection may be done from a bridge, by wading, or using a stream-side grab method, depending on the size of the stream and access, provided stream water at the specific monitoring location is well-mixed and representative.

Water quality parameters that are either measured in the field or in a laboratory are listed in Table 14.

Table 14. Water quality parameters to be monitored.

Parameter	Where measured
Ammonia	Laboratory
Chloride	Laboratory
Dissolved oxygen	Field
Hardness	Laboratory
Fecal coliform	Laboratory
Nitrate-Nitrite-N	Laboratory
pH	Field
Orthophosphate	Laboratory
Specific Conductance	Field
Temperature	Field
Total nitrogen	Laboratory
Total phosphorus	Laboratory
Total suspended solids	Laboratory
Turbidity	Laboratory

Field activities and protocols for water quality monitoring

The primary field activities conducted during a water quality data collection event includes measuring stream discharge and in-situ (field) water quality parameters, and collecting and processing water-quality samples for laboratory analyses.

Field activities for water quality monitoring and appendices within this QAPP that describe the procedures and protocols are shown in Table 15. Protocols for the RSMP water quality monitoring are adapted from Ecology's Watershed Health and Water Quality Index (WQI) programs are discussed in Appendix D and are based on multiple Ecology SOPs (Ward et al., 2001; Hallock and Ehinger, 2003; Ward, 2007a,b; Hallock, 2012).

Table 15. Field activities for water quality monitoring and the appendices describing the procedures.

Activity	Appendix
Site verification	C-1[1]
Streamflow measurement	C-3
Water-quality measurements and sample collection	D

[1] The site layout part of this appendix is not applicable to a water-quality DCE

Seasonal conditions should be considered during site verification, as the water quality portion of the monitoring program will occur monthly over an entire year and may include sampling during inclement weather or high-flow conditions. Field staff must always survey the sample location on the day of sampling for hazards for staff and equipment.

The City of Redmond will make a good faith effort to collect monitoring data described per QAPP requirements. If a water quality sample or measurement is missed on occasion, a second effort will be made to collect the sample with the same month. If a second attempt is also unsuccessful, the RSMP Coordinator will be notified, and a third attempt is not required.

Reasons a sample or measurement may not be made include, but are not limited to: a stream goes dry; the stream site cannot be accessed due to high flow conditions, vandalism, extreme climactic conditions, or monitoring equipment has a sudden failure. Flow measurements may need to be estimated using high water mark, staff gages, or other estimation techniques during the winter months to minimize the exposure hazardous conditions for staff. Water quality samples and measurements made during very high flows may be made from anywhere within the site reach.

Field quality control procedures for water quality monitoring

To ensure the quality and consistency of sample collections, equipment maintenance and sample collection protocols described in the appendices of this QAPP will be followed.

Sample holding times

Holding times are the maximum allowable length of time between sample collection and laboratory manipulation. Holding times are different for each analyte and are in place to maximize analytical accuracy and representativeness. Each sample collected will be packaged in a container and labeled accordingly. If necessary, staff will coordinate with the analytical laboratory to ensure samples can be transported, received, and processed during non-business hours. Sample containers will be transported or sent by the field team to the analytical laboratory, following established sample handling and chain-of-custody procedures. At the laboratory, samples may be further divided for analysis or storage. Table 16 lists sample volumes, holding times, containers, and preservation requirements for sediment and biological samples collected for water quality. Appendix D elaborates on the bottles and other equipment needed for biological samples.

Table 16. Sample containers, amounts, holding times, and preservation for water samples.

Analysis	Container[1]	Holding Time	Preservative[2]
Ammonia-N	125 mL poly bottle	28 days	H_2SO_4 to pH<2; cool to ≤4°C
Chloride	500 mL poly bottle	28 days	Cool to ≤4°C
Fecal coliform	250 or 500 mL autoclaved glass/poly bottle	24 hours (Hallock, 2007)	Fill bottle to shoulder, cool to ≤ 4°C
Hardness as $CaCO_3$	125 mL w/m poly bottle	6 months	H_2SO_4 to pH<2; cool to ≤6°C
Nitrate-Nitrite-N	125 mL poly bottle	28 days	H_2SO_4 to pH<2; cool to ≤4°C
Nitrogen, total	125 mL poly bottle	28 days	H_2SO_4 to pH<2; cool to ≤4°C
Orthophosphate	125 mL brown poly	2 days	Filter in field and cool to <4°C
Phosphorus, total	60 mL poly bottle	28 days	H_2SO_4 to pH<2; cool to ≤4°C or HCl to pH<2; cool to <°4
Total suspended solids (TSS)	1000 mL poly bottle	7 days	Cool to ≤4°C
Turbidity	500 mL poly bottle	2 days	Cool to ≤4°C

[1] Replicate samples should be collected in additional containers.
[2] Preservation should be done in the field, unless otherwise noted. Ice will be used to cool samples.

Field instrument quality control

In order to maintain the highest degree of data quality, field equipment will undergo routine cleaning, calibrations, and maintenance at the recommended frequency specified by each manufacturer and described in SOPs. Appendices B and F discuss field meters.

Documentation

Field data measurements will be recorded in the field. The software being developed for Watershed health monitoring will also be able to record any in-situ water quality measurements. As previously mentioned in the Documentation section for watershed health monitoring, staff will need to prepare paper field forms in case the software is not ready in time for the October 2014 start date. Appendix M contains an example field form to record in-situ water quality measurements made at each site. Forms and documentation will include the station visit/maintenance sheet, meter calibration, and chain-of-custody forms.

All entries on field documents will be made in pencil or permanent pen and will list the field technician name(s). Any errors or typos will be crossed out and rewritten by the technician who recorded the data. All corrections will be initialed and dated when made. If field sampling or procedural errors are discovered, action will be taken to manage and correct those errors. Corrections may occur with corrective editing, relabeling, or, if warranted, flagging, discarding,

and re-sampling. If a consistent error persists, an amendment to the sampling procedures may be required.

Composite/grab field replicate samples

A field quality control schedule is shown in Table 17. Grab and composited field replicate samples will be collected at a rate of 10% of the total samples collected for monitoring under the permit. Field replicates will be collected by splitting composited samples or by collecting additional grab samples. Parameters measured in the field sample also will be measured in the replicate sample for that particular site.

All field replicates will be labeled similar to other samples, so that the sample has its own unique number. These replicate samples will be submitted blind to the laboratory, with all other field samples.

Field blanks

The term *field blanks* includes equipment rinsate blanks, trip blanks, transfer blanks, or specific equipment blanks. Blanks serve as field audits to measure contamination. A single transfer blank for water-based parameters will be labeled with unique numbers and will accompany the first set of samples to the laboratory.

If field blank contamination is discovered, additional field blank samples may be used to determine the source of the contamination. Field blank samples collected to determine the contamination source may include:

- A field equipment blank collected from the filtration apparatus used to filter orthophosphate.
- A field transfer blank collected by pouring lab-provided deionized water into a clean sample bottle to determine whether field contamination is present, unrelated to the equipment.
- A field trip blank collected by transporting unopened bottles containing organic and metal-free, certified clean water from the laboratory into the field, and then returned it to the laboratory (bottles are not opened in the field). Trip blanks are used to determine whether any contamination occurs while traveling from field to laboratory.

Any field blank contamination will be reviewed by the field lead to determine if samples associated with the field blanks should be qualified based on the contamination. Sample results will be flagged with a *J* if they are less than, or equal to, 5 times the field blank concentration.

Table 17. Field quality control schedule for water quality samples collected.

Field Sample Collected	Frequency [1]	Control Limit	Corrective Action
Grab water field replicate	7 replicates per year or 10% of total samples[2]	Qualitative control – Assess representativeness, comparability, and field variability	Review procedures; alter if needed
Transfer blank	At least one sample a year	Blank analyte concentration should be below the reporting limit	Compare blanks for analyte to determine whether the sampling process is the source of contamination; re-evaluate decontamination procedures; evaluate results greater than 5x blank concentrations
Other blank samples for determining a contamination source	As needed	Blank analyte concentration should be below the reporting limit	Compare results from separated blanks to isolate the source of contamination; evaluate results greater than 5x blank concentrations

[1] This table is based in part on an EPA QA website accessed August 2013: www.epa.gov/earth1r6/6pd/qa/qadevtools/mod5_sops/sample_handling_preservation/appendix_b3.pdf

[2] Total samples refers to the total number of samples for each permittee who selects Option S8.B.1.b or S8.B.2 in the permit. The number of samples is dependent on the number of sites each permittee monitors.

Laboratory Quality Control Procedures

This section discusses the laboratory QC procedures that will be implemented to provide high quality data. Field QC procedures were previously described as part of the *Field Operations* sections of this QAPP. QC will be monitored throughout the duration of the study. The quality of raw, unprocessed, and processed data is subject to review according to established protocols in the *Measurement Quality Objectives* section of this QAPP.

Biotic samples

QC procedures for biotic samples are currently limited to field replicates precision and laboratory duplicates for accuracy for benthic macroinvertebrates and periphyton. Contract laboratories will make every effort to ensure accurate identification of specimens. More information on laboratory QC procedures is provided in Appendix F-2 and F-3.

Water and sediment samples

This section discusses QC procedures that will be implemented by the contracted analytical laboratory to provide high quality chemical and physical analyses that meet these QAPP requirements. Contract laboratories will make every effort to meet sample holding times and target reporting limits for all parameters. Laboratory QC procedures and results will be closely monitored throughout the duration of the permit-mandated sampling. The quality of laboratory data is subject to review via the established protocols in the *Measurement Quality Objectives* section. A typical schedule for laboratory QC samples is shown in Table 18 and, at a minimum, includes:

- Laboratory duplicates
- Matrix spikes
- Matrix spike duplicates
- Method/instrument blanks
- References (lab standards/surrogate standards/internal standards)

Table 18. Laboratory quality control schedule for monitoring.

Quality Control Sample[1]	Analysis Type	Frequency[2]	Corrective Action
Laboratory Duplicates	inorganic	5% of total samples or 1 per batch (method-specific)	Evaluate procedure; ID contaminant source; reanalyze or qualify affected data
	conventional		
	organics		
Matrix Spikes	inorganic	For metals at least 1 sample per year, otherwise 5% of total samples or 1 per batch	Evaluate procedure and assess potential matrix effects; reanalyze or qualify data
	conventional	5% of total samples or 1 per batch	
	organics	5% of total samples or 1 per batch	Evaluate lab duplicates/standards recoveries and assess matrix effects; evaluate or qualify affected data
Matrix Spike Duplicates[3]	inorganic	For metals at least 1 samples per year, otherwise 5% of total samples or 1 per batch	Evaluate procedure and assess potential matrix effects; reanalyze or qualify data
	conventional	5% of total samples or 1 per batch	
	organics	5% of total samples or 1 per batch	
Method / Instrument Blanks	inorganic	5% of total samples or 1 per batch (method-specific)	Blank concentration is defined as the new reporting limit. Evaluate procedure; ID contaminant source; reanalyze blanks or qualify sample data (<5-10x blank concentration).
	conventional		
	organics		
References (lab control standard, surrogate, and internal standards)	inorganic	5% of total samples or 1 per batch (method-specific)	Evaluate lab duplicates/matrix spike recoveries; assess efficiency of extraction method; evaluate or qualify affected data
	conventional		
	organics		

[1] Quality control samples may be from different projects for frequencies on a per-batch basis.
[2] Frequencies may be determined from the study number of samples collected by the permittee.
[3] Matrix spike duplicates may be used to satisfy frequencies for laboratory duplicates.

Instrument calibration

The instrumentation used by the chosen laboratories will meet or exceed manufacturers' specifications for use and maintenance. Maintenance of this equipment will be conducted in a manner specified by the manufacturer or by the QA guidelines established by the chosen laboratory.

Duplicate/splits

Laboratory duplicate samples will be analyzed regularly to verify that the laboratory's analytical methods are maintaining their precision. The laboratory should perform "random" duplicate selection on submitted samples that meet volume requirements. After a sample is randomly selected, the laboratory should homogenize the sample and divide it into two identical "split" samples. To verify method precision, identical analyses of these lab splits should be performed and reported. Some parameters may require a double volume for the parameter to be analyzed as the

laboratory duplicate. Matrix spike duplicates may be used to satisfy frequencies for laboratory duplicates.

Matrix spikes and matrix spike duplicates

Matrix spike samples are triple-volume field samples (per parameter tested) to which method-specific target analytes are added or spiked into two of the field samples, and then analyzed under the same conditions as the field sample. A matrix spike provides a measure of the recovery efficiency and accuracy for the analytical methods being used. Matrix spikes are typically analyzed in duplicate (matrix spike/matrix spike duplicate [ms/msd]) to determine method accuracy and precision. Matrix spikes will be prepared and analyzed at a rate of 1 pair/20 (five percent) samples collected or one pair for each analytical batch, whichever is most frequent. Use of ms/msd at the frequency of 5% of the total number of samples is common practice. For the purposes of permit monitoring, these frequencies meet the expectations.

Blanks and standards

Laboratory blanks are useful for instrument calibrations and method verifications, as well as for determining whether any contamination is present in laboratory handling and processing of samples.

Laboratory standards

Laboratory standards (reference standards) are objects or substances that can be used as a measurement base for similar objects or substances. In many instances, laboratories using digital or optical equipment will purchase from an outside accredited source a solid, powdered, or liquid standard to determine high-level or low-level quantities of a specific analyte. These standards are accompanied by acceptance criteria and are used to test the accuracy of the laboratory's methods. Laboratory standards are typically used after calibration of an instrument and prior to sample analysis.

Surrogate and internal standards

Surrogate standards are used to process and analyze extractable organic compounds (PAHs, phthalates, and pesticides). A surrogate standard is added before extraction, and it monitors the efficiency of the extraction methods. Internal standards are added to organic compounds and metal digests to verify instrument operation when using inductively coupled plasma mass spectrometry (ICP-MS) analysis.

Method blanks

Method blanks are designed to determine whether contamination sources may be associated with laboratory processing and analysis. Method blanks are prepared in the laboratory using the same reagents, solvents, glassware, and equipment as the field samples. These method blanks will accompany the field samples through analysis.

Instrument blank

An instrument blank is used to "zero" analytical equipment used in the laboratory's procedures. Instrument blanks usually consist of laboratory-pure water and any other method-appropriate reagents, and they are used to zero instrumentation.

Inter-laboratory comparison

There is a recognized need to conduct an inter-laboratory comparison study if multiple laboratories are used for the RSMP and permittees conducting monitoring under this QAPP. Permittees will collect replicate samples to support this comparison as follows: Water and sediment replicate samples collected under the RSMP and this QAPP will be sent to two of the participating laboratories. Under this QAPP for permittees conducting monitoring, the contracted laboratory will be sent one of the replicates, and the other replicate samples will be sent to the other determined by the RSMP. Since the laboratories used by the Redmond and the RSMP are the same, then the inter-laboratory comparison requirement is nullified. This inter-laboratory comparison does not apply to the fecal coliform samples.

Measurement quality objectives (MQOs) for laboratory samples

This section refers to the MQOs, the acceptance thresholds for water data collected under the Water Quality Index monitoring, and sediment data collected under Watershed Health monitoring. MQOs specifically are used to address instrument and analytical performance.

MQOs established for stormwater permit monitoring are based on guidance from multiple sources, including EPA, Ecology, and laboratory experience. Tables 19 and 20 represent how data will be verified prior to reporting results. Failure to meet the MQOs may result in data being qualified or rejected.

Table 19. Measurement quality objectives (MQOs) for water chemistry and chlorophyll *a* for periphyton.

Water Parameters	Methods in Water MQO[3]	Reporting Limit Target	Field/Lab Replicate (RPD) [1]	Matrix Spike[2] (% Recovery)	Lab Duplicate (RPD) [1]	Control Standard/ Surrogate (% Recovery)
		Sensitivity	Bias and Precision	Bias and Accuracy	Bias and Precision	Bias and Accuracy
Ammonia-N	SM 4500-NH_3H	0.01 mg/L	≤10%	75-125	≤20%	80-120
Chloride	EPA 300.0	0.1 mg/L	≤20%	75-125	≤20%	90-100
Dissolved Oxygen*	Electrode meter ±0.2 mg/L	n/a	n/a	n/a	n/a	n/a
Fecal coliform	SM 9222D	1 cfu/100 mL	≤20%	n/a	n/a	n/a
Nitrate+Nitrite-N	SM 4500-NO_3^-I	0.01 mg/L	≤10%	75-125	≤20%	80-120
Hardness as CaCO3	SM 2340B or EPA 200.7	0.3 mg/L	≤10%	75-125	≤20%	85-115
pH*	Electrode Meter ± 0.2 std. units	n/a	≤10%	n/a	n/a	n/a
Orthophosphate	SM 4500 PG or SM 4500PE	0.003 mg/L	≤10%	75-125	≤20%	80-120
Specific conductance* (conductivity)	Electrode meter ±5 us/cm at 100 us/cm	15umhos/cm	≤10%	n/a	≤20%	90-110
Temperature*	Electrode meter ± 0.2°C	n/a	≤10%	n/a	n/a	n/a
Total nitrogen (TN)	SM 4500-N-B	0.025 mg/L	≤10%	75-125	≤20%	80-120
Total phosphorus (TP)	SM 4500PE or SM 4500-P H	0.005 mg/L	≤10%	75-125	≤20%	80-120
Total suspended solids (TSS)	SM 2540D	1 mg/L	≤15%	n/a	≤5%	90-110
Turbidity	EPA 180.1	0.1 NTU	≤15%	n/a	≤20%	90-110
Chlorophyll *a* – in periphyton[4]	SM 10200H(3)	0.1 ug/L	≤50%	n/a	≤20%	n/a

*Field-measured parameters follow manufacturer's website guidelines for meter calibrations.
EPA: Environmental Protection Agency method (http://water.epa.gov/scitech/methods/cwa/methods_index.cfm).
SM: *Standard Methods for the Examination of Water and Wastewater* (www.standardmethods.org).
[1] The relative percent difference (RPD) must be less than or equal to the indicated percentage for values that are greater than 5 times the reporting limit. RPD must be ±2 times the reporting limit for values that are less than or equal to 5 times the reporting limit.

[2] For inorganics, the *Laboratory Program Functional Guidelines* state that the spike recovery limits do not apply when the sample concentration exceeds the spike concentration by a factor of 4 or more (EPA, 2010).
[3] MQOs are based on Hallock (2012) and SOP EAP033 (Swanson, 2007).
[4] MQOs are based on Adams (2010a) and MEL (2008) for chlorophyll *a* content, once field-filtered from a periphyton slurry sample.

Table 20. Measurement quality objectives (MQOs) for chemical analysis of sediments.

Sediment Parameters for Bioassessment	Analysis Methods in Sediment MQO	Reporting Limit Target	Lab Replicate (RPD) [1]	Matrix Spike[2] (% Recovery)	Matrix Spike Duplicate (RPD) [1]	Control Standard/ Surrogate (% Recovery)
		Sensitivity	Bias and Precision	Bias and Accuracy	Bias and Precision	Bias and Accuracy
Grain Size on <2mm sieved sediments	PSEP, 1986. Sieve and pipette or ASTM D422	Sensitivity = 1.0%	≤20%	n/a	n/a	n/a
Percent Solids	SM 2540 G	Sensitivity = 0.1%	≤20%	n/a	n/a	n/a
Total Organic Carbon	PSEP (1986, with 1997a,b updates), combustion/CO^2 (MEL, 2008)	Sensitivity = 0.1%	≤20%	n/a	n/a	80-120
Metals (As, Cu, Pb, Zn)	EPA Method 200.8 (ICP/MS)	(0.1, 0.1, 0.1, 5.0) mg/Kg dw	≤20%	75-125	≤20%	85-115
2,4-D, triclopyr	EPA 8151A (GC-MS/MS)	6.7 ug/Kg dw	n/a	40-130	≤40%	40-130
Chlorpyrifos and Dichlobenil	EPA 8270D (GC-MS/MS)	6.7 ug/Kg dw	n/a	40-130	≤40%	40-130
Diuron and Carbaryl	EPA 8321B (LC-MS/MS)	6.7 ug/Kg dw	n/a	30-130	≤40%	40-130
Phthalates	EPA 8270D (GC/MS)	70 ug/Kg dw	Compound specific ≤40%	40-150	≤40%	40-150[3]
Polybrominated diphenyl ethers (PBDEs)	EPA 1614	2 ng/Kg dw[4]	≤20%	50-150	25-150%[5]	50-150
Polycyclic aromatic hydrocarbon (PAH) compounds	EPA 8270D (GC/MS)	70 ug/Kg dw	Compound specific ≤40%	50-150	≤40%	50-150[3]
Polychlorinated biphenyls (PCBs)	EPA Method 1668A	20 ng/Kg dw[4]	≤20%	50-150	≤40%	25-150[6]

EPA: Environmental Protection Agency Method (http://water.epa.gov/scitech/methods/cwa/methods_index.cfm).
SM: *Standard Methods for the Examination of Water and Wastewater* (www.standardmethods.org).
PAH compounds include: 1-methylnaphthalene, 2-methylnaphthalene, 2-chloronaphthalene, acenaphthylene, acenaphthene anthracene, benzo(a)anthracene, benzo(a)pyrene, benzo(b)fluoranthene, benzo(ghi)perylene, benzo(k)fluoranthene, dibenzo(a,h)anthracene, dibenzofuran, carbazole, chrysene, fluoranthene, fluorene, indeno(1,2,3-cd)pyrene, naphthalene, phenanthrene, pyrene, and retene.

Pesticides include: 2,4-D, triclopyr, carbaryl, and chlorpyrifos.
Phthalates include: bis(2-ethylhexyl) phthalate, butyl benzyl phthalate, diethyl phthalate, dimethyl phthalate, di-n-butyl phthalate, and di-n-octyl phthalate.
PBDE congeners include: 47, 49, 66, 71, 99, 100, 138, 153, 154, 183, 184, 191, 209.
PCB congeners include: all 209 congeners.
PSEP: Puget Sound Estuary Program (PSEP) Protocols, www.psparchives.com/our_work/science/protocols.htm.
RPD: Relative percent difference.

[1] The relative percent difference (RPD) must be less than or equal to the indicated percentage for values that are greater than 5 times the reporting limit. RPD must be ±2 times the reporting limit for values that are less than or equal to 5 times the reporting limit.
[2] For inorganics, the *Laboratory Program Functional Guidelines* state that the spike recovery limits do not apply when the sample concentration exceeds the spike concentration by a factor of 4 or more (EPA, 2010).
[3] Organic material is not removed prior to analysis.
[4] Triplicate QC samples; MQO given for the relative standard deviation of the triplicates.
[5] Semivolatile surrogate recoveries are compound specific. MQOs are based on Johnson (2005) and Dutch et al. (2010).
[6] Varies with congener. PBDE and PCB MQOs are based on Johnson (2010) and Dutch et al. (2010).
[7] Except 20-200% for 13C12DeBDE; see EPA Method 1614.
[8] Applies to most congeners; see EPA Method 1668A.

Data Management

Permittees will be collecting and managing data from field observations/measurements and laboratory analysis of field samples. All data will be managed and stored by the permittees. Post-processed data will be finalized and incorporated into annual reports and electronic reports. Reports and data will be submitted to Ecology in the format required.

Field data

Permittees will need record field water quality and watershed health data using field forms. If necessary, paper field forms should be printed on waterproof paper and kept in a three-ring binder during sampling.

Ecology intends to provide to permittees electronic field data collection software that will assist permittees to assure completeness in the field for benthos and habitat monitoring, and with loading this data to Ecology's Watershed Health database in EIM. Because timing (anticipated completion date of spring 2015) of the software development may not suit monitoring needs under this QAPP, recording on field forms is required.

Laboratory data

Chemical and bacteria laboratory data will be sent to the permittee from each laboratory following analysis. Reporting times may vary depending on holding time but should not exceed six months of the documented sampling date. Laboratory reports will be reviewed by the permittees for errors or missing data; permittees may implement corrective actions. Finalized electronic laboratory data will be loaded to Ecology's EIM database by permittees and saved in the permittee's data file.

Data storage

All field forms, photographs, electronic data, and laboratory electronic or printed data generated for this project will be stored by the permittees in an organized filing system for electronic or paper files. These files may be sought by Ecology within the permit term. Location, measurement, and sample result data will be evaluated through the data verification process, outlined earlier in this QAPP. Acceptable results will be used by scientists to prepare a summary report and entered into Ecology's EIM database.

Benthos data will be loaded into King County's Puget Sound Stream Benthos database (http://www.pugetsoundstreambenthos.org/), as well as the Watershed Health section of Ecology's EIM database. Periphyton data, habitat data, and any field-measured data will be entered into the Watershed Health section of EIM.

Data Verification and Usability

Data verification

Data verification involves examining the data for errors, omissions, and compliance with quality control (QC) acceptance criteria.

Field staff will verify field results after measuring and before leaving the site. They will keep field notes to meet the requirements for documentation of field measurements. The field lead will ensure that field data entries are complete and error-free. The field lead also will check for consistency within an expected range of values, verify measurements, ensure measurements are made within the acceptable instrumentation error limits, and record anomalous observations. The project manager will verify field data to ensure that:

- Data are consistent, correct, and complete, with no errors or omissions.
- Results of QC samples accompany the sample results.
- Established criteria for QC results were met.
- Data qualifiers are properly assigned where necessary.
- Data specified in the Sampling Process Design were obtained.
- Methods and protocols specified in this QAPP were followed.

The project manager at the taxonomic laboratory will verify all taxonomic results, and the laboratory will verify all analytical results prior to reporting.

Corrective actions for inadequate data

If discrepancies in the data are found, there are two options for correction, depending on when the problem is identified.

1. If the problem is identified before the end of the index period (July 1 to October 15), a review of the protocols and SOPs outlined in the appendices of this document is required. After this review, a repeat site visit may be made to re-collect the sample. This may occur if the data set is incomplete or incorrectly collected. Due to the inter-related nature of chemical and biological conditions, problems identified in the chemical or biological data should be addressed by again collecting the entire suite of chemical and biological analyses parameters. Because the habitat is mostly constant within an index period, if the data in question is related to habitat, only the missing habitat information needs to be collected. Before the second sampling, the investigator must review the SOPs and the appendices of this document to understand the protocols. Equipment should be cleaned and recalibrated and checked for proper function.
2. If the problem is identified after the index period, the data should be flagged and the problem explained in a comment in the database. This will allow the Ecology investigator, as well as external users of these data, to know how these data may be used in projects. If the data are

incomplete, or if some data standard was not met, the data will not be used to meet the objectives of the study design.

Data usability assessment

Data usability assessment follows verification. This involves a detailed examination of the data package using professional judgment to determine whether the quality objectives have been met. The project manager examines the complete data package to determine compliance with procedures outlined in the QAPP and SOPs. The project manager also ensures that the MQOs have been met and determines if the quality of the data are usable for the project objectives.

Stream Monitoring Reports

An annual report is required to provide a summary of the previous year's monitoring activities and results. A detailed monitoring report is due to Ecology as an attachment to the permittee's annual report due on March 31 of each year 2016, 2017, and if needed, 2018. Annual reports require a G19 signature.

The stream monitoring report will include a complete discussion of the monitoring effort. The report must include:

- Site selection confirmation documentation,
- Summary of monitoring activities for water quality index, habitat, benthos and sediment chemistry, and
- A cumulative water quality and watershed health results summary for each site.

Table 21 outlines the required monitoring report elements. These monitoring reports shall be submitted electronically, as MS Word documents.

Table 21. Reporting requirements.

Category	Reporting Requirement
Site Confirmation	Documentation of the site confirmation process, including desktop evaluation and field visits for each of the required number of assigned sites.
	List of sites disqualified and specific reasons for disqualification.
	List of final sites. In a table, provide final GPS coordinates for each site and the distances from the initial GPS locations provided in the Master Sample; and indicate the Strahler stream order.
Ancillary Site Information	Description of land use adjacent to the reach sampled.
	Description of the upstream land use of the basin contributed to the site sampled.
	Area (in square miles) of the basin draining to the site.
	Approximate (to the nearest 5%) percentages of industrial, commercial (including multi-family residential), residential (3 categories: 5 or more dwelling units per acre; 3-4 dwelling units per acre; 1-2 dwelling units per acre), agricultural, and forest land uses in the basin area draining to each site.
Water Quality Monitoring	Sample information (dates, times, locations).
	Tabular water quality data. Water Quality Index (WQI) monitoring results shall be provided in an Excel spreadsheet cross-tab format (see Appendix N). This format is designed to facilitate WQI calculations by the permittee or Ecology.
	Summary results for each monitored parameter at each site and for all of the sites.
	Computation of the WQI for each site, if feasible.
Watershed Health Monitoring	Sample information (dates, times, locations).
	Benthos, habitat, periphyton, and sediment monitoring results for each site and for all of the sites.
	B-IBI score for each site using Puget Sound Stream Benthos tools and 0-100 score methodology.
Concerns	Narrative description of any deviations from this QAPP, including any delays, problems, and resolutions in conducting required monitoring activities.
Costs	A summary of monitoring costs for each required monitoring program component.

References

Web links to resources

EPA's aquatic resource monitoring - frequently asked questions

Survey sampling: www.epa.gov/nheerl/arm/sursampfaqs.htm
Survey design: www.epa.gov/nheerl/arm/surdesignfaqs.htm
Data analysis: www.epa.gov/nheerl/arm/dataanalysisfaqs.htm

EPA's data analysis resources and tools for surveys

General statistical books on survey designs:
www.epa.gov/nheerl/arm/bibliography.htm#generalsurveydesignbooks

Monitoring Data Analysis and Reporting:
www.epa.gov/nheerl/arm/analysispages/monitanalysisinfo.htm

Presentations on statistical analysis processes: www.epa.gov/nheerl/arm/presents.htm

Statistical tools for data analysis (Software for R):
www.epa.gov/nheerl/arm/analysispages/software.htm

Example analysis software for EMAP West Wadeable Stream Data
www.epa.gov/nheerl/arm/analysispages/r10_work/r10_intro.htm

Stressor Identification Guidance:
http://water.epa.gov/scitech/swguidance/standards/criteria/aqlife/biocriteria/stressors_index.cfm

Biological Indicators of Watershed Health: www.epa.gov/bioindicators

Ecology's relevant resources for biological, habitat, and water quality sampling

Status and Trends Statewide Monitoring:
http://www.ecy.wa.gov/programs/eap/stsmf

Stream Biological Monitoring:
www.ecy.wa.gov/programs/eap/fw_benth

River and Stream Water Quality Monitoring:
www.ecy.wa.gov/programs/eap/fw_riv/rv_main.html

River and Stream Water Quality Index:
www.ecy.wa.gov/programs/eap/fw_riv/docs/WQIOverview.html

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Table 19. Measurement quality objectives (MQOs) for water chemistry and chlorophyll *a* for periphyton.

Water Parameters	Methods in Water MQO[3]	Reporting Limit Target	Field/Lab Replicate (RPD) [1]	Matrix Spike[2] (% Recovery)	Lab Duplicate (RPD) [1]	Control Standard/ Surrogate (% Recovery)
		Sensitivity	Bias and Precision	Bias and Accuracy	Bias and Precision	Bias and Accuracy
Ammonia-N	SM 4500-NH_3H	0.01 mg/L	≤10%	75-125	≤20%	80-120
Chloride	EPA 300.0	0.1 mg/L	≤20%	75-125	≤20%	90-100
Dissolved Oxygen*	Electrode meter ±0.2 mg/L	n/a	n/a	n/a	n/a	n/a
Fecal coliform	SM 9222D	1 cfu/100 mL	≤20%	n/a	n/a	n/a
Nitrate+Nitrite-N	SM 4500-NO_3^-I	0.01 mg/L	≤10%	75-125	≤20%	80-120
Hardness as CaCO3	SM 2340B or EPA 200.7	0.3 mg/L	≤10%	75-125	≤20%	85-115
pH*	Electrode Meter ± 0.2 std. units	n/a	≤10%	n/a	n/a	n/a
Orthophosphate	SM 4500 PG or SM 4500PE	0.003 mg/L	≤10%	75-125	≤20%	80-120
Specific conductance* (conductivity)	Electrode meter ±5 us/cm at 100 us/cm	15umhos/cm	≤10%	n/a	≤20%	90-110
Temperature*	Electrode meter ± 0.2°C	n/a	≤10%	n/a	n/a	n/a
Total nitrogen (TN)	SM 4500-N-B	0.025 mg/L	≤10%	75-125	≤20%	80-120
Total phosphorus (TP)	SM 4500PE or SM 4500-P H	0.005 mg/L	≤10%	75-125	≤20%	80-120
Total suspended solids (TSS)	SM 2540D	1 mg/L	≤15%	n/a	≤5%	90-110
Turbidity	SM2130	0.1 NTU	≤15%	n/a	≤20%	90-110
Chlorophyll *a* – in periphyton[4]	SM 10200H(3)	0.1 ug/L	≤50%	n/a	≤20%	n/a

*Field-measured parameters follow manufacturer's website guidelines for meter calibrations.

must be justified as adequate for the likely range of concentrations. Permittees are not guaranteed approval of their alternative methods or reporting limits. Taxonomic identification will be 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.

Work performed by the City of Redmond is expected to meet the quality control requirements of the analytical methods stated in this QAPP. These requirements are summarized in the *Quality Control* section of this document.

Laboratory selection

Multiple laboratories will be needed to ensure sample completeness. Contracting for laboratories is a responsibility of the permittee. Permittees may consider their own or other permittee's laboratories, commercial laboratories, or Ecology's Manchester Environmental Laboratory (MEL). Laboratories for water and sediment parameters will be selected based on their current accreditation status with Ecology (www.ecy.wa.gov/programs/eap/labs/search.html) and their ability to achieve acceptable limits of detection for the parameters measured as part of this project.

Table 5 Selected laboratories for sample processing.

Laboratory Name	Analytical Purpose	Address	Phone
AmTest	Fecal Coliform	13600 NE 126th PL # C Kirkland, WA 98034	425.885.1664
Manchester Environmental Laboratory (MEL)	Water Quality testing for Ammonia, Chloride, Hardness, Nitrate-Nitrite-N, Total Nitrogen, Total Phosphorus, Total Suspended Solids, Orthophosphate and Turbidity Sediment sample testing for metals, Total Organic Carbon, Percent Solids, Pesticides, Herbicides, Phthalates, PBDEs, PAH, PCBs	7411 Beach Drive East Port Orchard, WA 98366	360. 871.8800
Manchester Environmental Laboratory (MEL)	Water sample testing for chlorophyll a	7411 Beach Drive East Port Orchard, WA 98366	360. 871.8800
Rhithron Associates,	Stream benthos	33 Fort Missoula Road	406.721.1977

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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/psmonitoring/ps_monitoring_docs/SWworkgroupDOCS/SWGfinalreportoct292010.pdf). 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 (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. O 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 also 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.

Sampling and visit precision

Sampling precision measures the extent of random variability among replicate measurements of the same property and is a data quality indicator (USGS, 1998). Sampling precision is estimated by keeping the eight replicate samples separate at 10% of the reaches sampled annually. Sampling precision is calculated using the relative standard deviation (RSD) among the replicate samples and should be < 20% in reference streams when using the taxa richness metric (Plotnikoff, 1992). Staff expects collections of macroinvertebrates and periphyton from multiple sample locations to have similar community structure in reference streams.

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.

Once a year, the project manager will provide a list of relevant SOPs and protocols to all support staff. The project manager will conduct sampling with each support staff team once each year. At this time, the staff will verify, by signature, that applicable SOPs and protocols were reviewed. 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 17-18 indicate the target quantitation limit established from experience at Manchester Environmental Laboratory (MEL) or local laboratories for the given analysis. Other laboratories may be able to achieve better 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 three sample duplicates is calculated as the relative standard deviation (RSD), which is expressed as a percentage:

Equation 2: Relative Standard Deviation

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

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 is 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 17-18. 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

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

Where:

$\%\mathrm{R}$ = 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.

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 the 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): JW

Status and Trends Program - GPS Positions Form

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).

Draft

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

DCE:	WAM0600-000018-DCE-2009-0701-09:00 (Site Number; YY; MMDD; HH:MM)		
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.

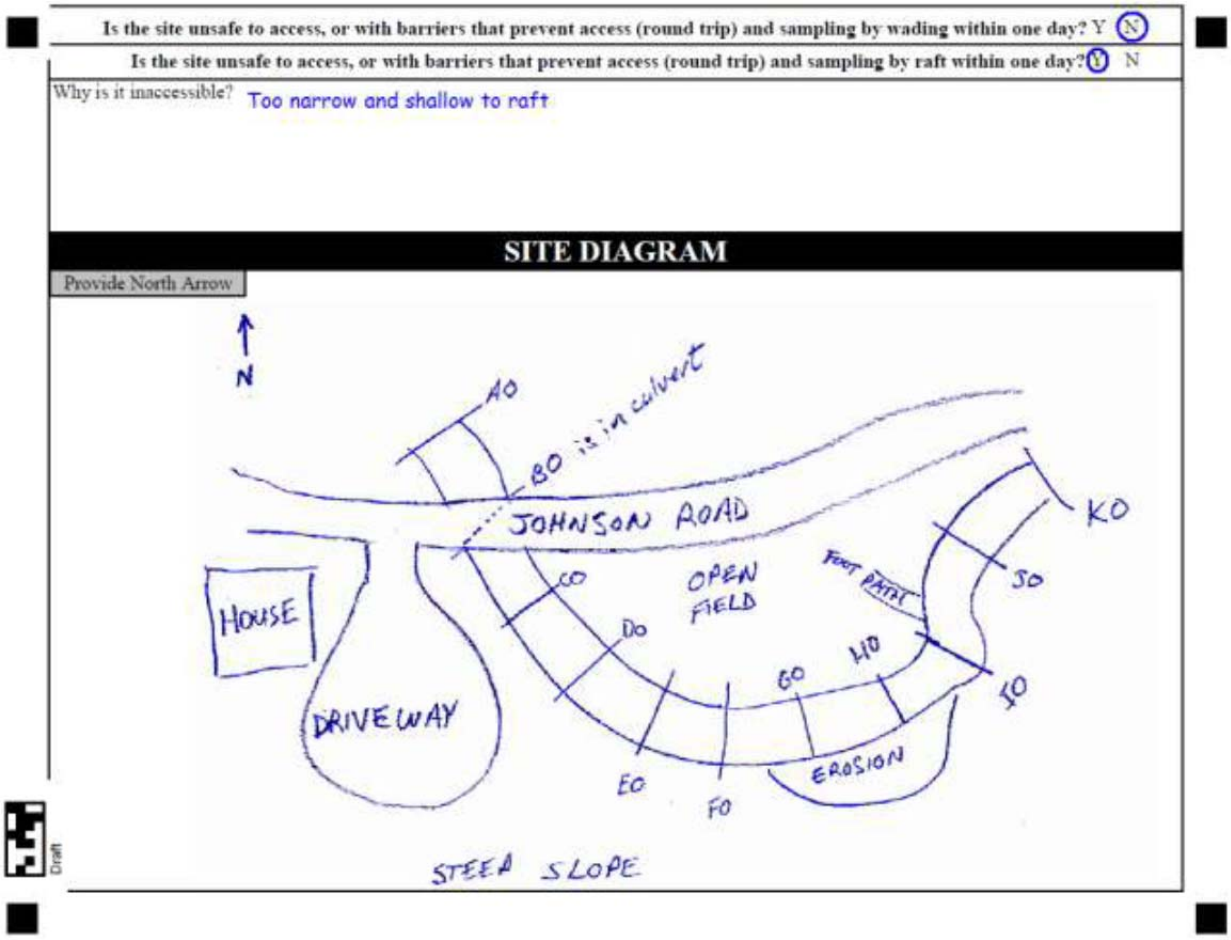


Figure C-1.3. The back side of the Site Verification Form, with example data.

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.

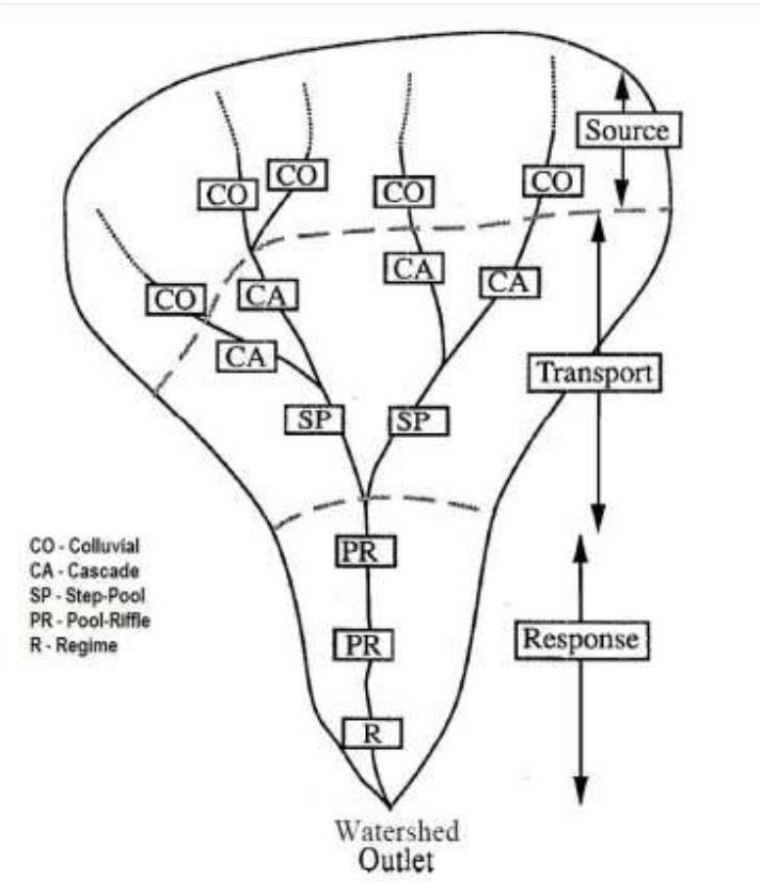


Figure C-1.4. Idealized positions (aerial view) of bed form types within a watershed.

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

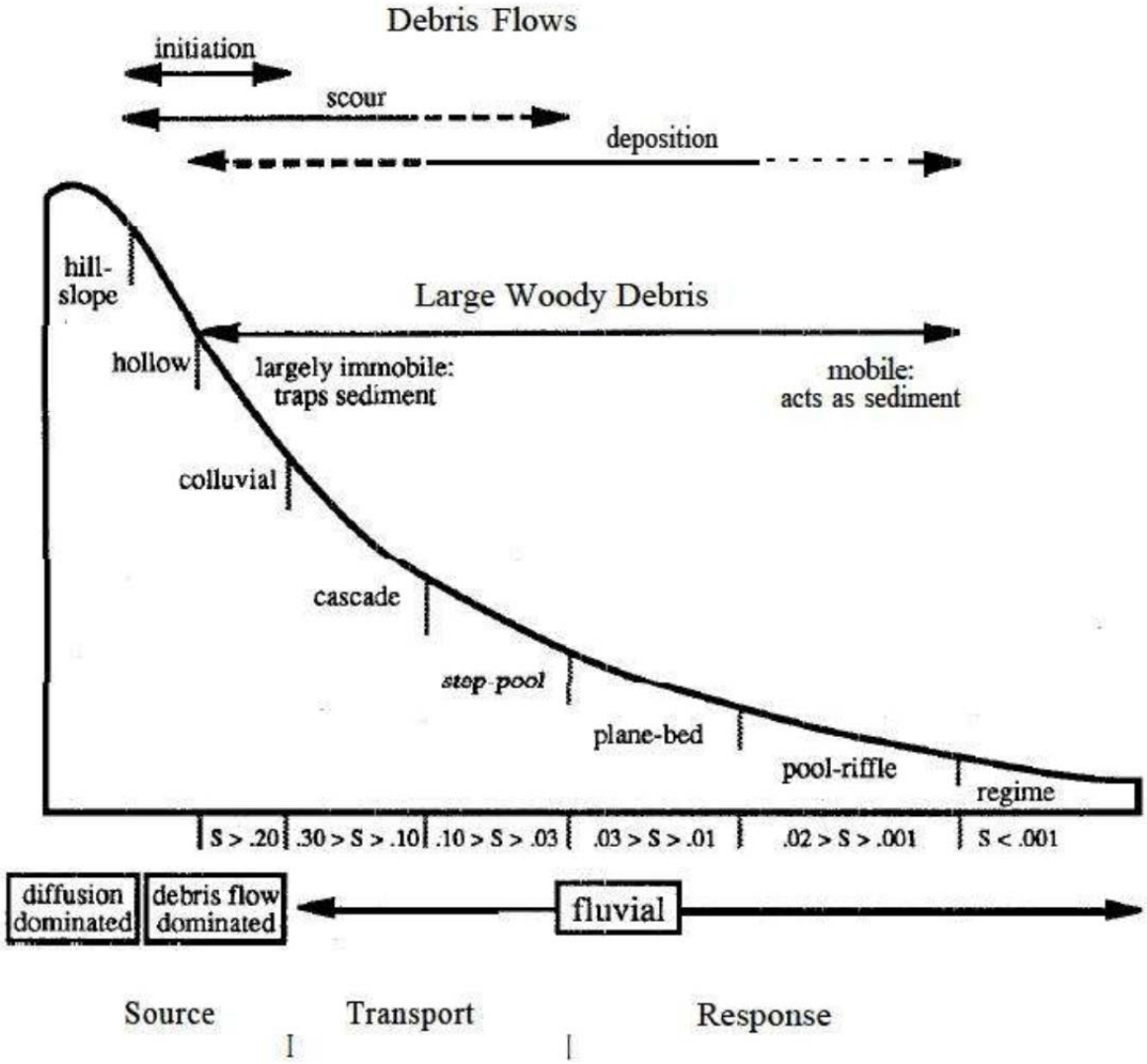


Figure C-1.5. Idealized positions (plan view) of bed form types within a watershed.

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.

<u>Thalweg transects</u>

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 1/5th 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

Station	Thalweg Transect	Major Transect	Minor Transect	Distance from Bottom (Bankfull Widths*)
D3	Yes			6.6
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

Station	Thalweg Transect	Major Transect	Minor Transect	Distance from Bottom (Bankfull Widths*)
I3	Yes			16.6
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. Hach HQ40d (equipped with an LDO, Temperature and pH probe and Hach SensION 5 (conductivity and temperature probe) are used. 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 Stream Sampling Protocols for Monthly Sampling & the Generation of the WQI (WS_SOP_013, City of Redmond) shall be used instead. The calibration techniques are discussed in the manufacturer's websites.

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.
- Hach HQ40d and Hach SensION5
- Hach Manuals (Hach 1999; May 2009; Hach 2004).

Summary of procedure

Calibrate the instrument before sampling. Measure the stream twice.

Verify quality control

<u>Prior to sampling</u>

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:** _____

Meter Model: _____ **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, turbidity 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). Follow procedures discussed in Stream Sampling Protocols for Monthly Sampling & the Generation of the WQI (WS_SOP_013, City of Redmond).

Reviewed by (Initials): JS

Status and Trends - Chemistry and Sampling Form

DCE: WAM0600 - 000001 - 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		
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-2.1. The *Chemistry and Sampling Form*, with examples of in-situ data records.

C-3. Estimating discharge in small streams

Purpose and scope

This method describes how to collect field data necessary for estimating instantaneous discharge (in cubic feet per second) during each data collection event (DCE) for Watershed Health monitoring. Flow data are used to verify the categorical stream order for wadeable streams, based on the time of sampling, which represents approximate base-flow conditions.

Personnel responsibilities

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

Equipment, reagents, supplies

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

Discharge is normally measured near the index station ("X") where there is uniform (non-turbulent) flow. It can be done at any time during the data collection event, except prior to sampling for water, sediment, or invertebrates. For method references see SOP EAP024 (Kardouni, 2013) and Kaufmann (2006). For operation of the flow meter, refer to the manufacturer's manual.

For calculation of discharge (from meter data), refer to USBR (2001). The QWIN program created by Larsen (2005) applies the USBR Midsection Method. Example data and online access to QWIN are provided by PSNS&IMF (2006). Conditions may not always allow for use of flow meters. In these cases, discharge can be estimated through alternate methods. Examples of alternate methods are:

- Velocity of floats (e.g., oranges).
- Time to fill a bucket of known volume (e.g., discharge off of a hanging culvert).
- Retrieval of data from co-located gages or models.

Velocity-area method

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 C-3.1. 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).

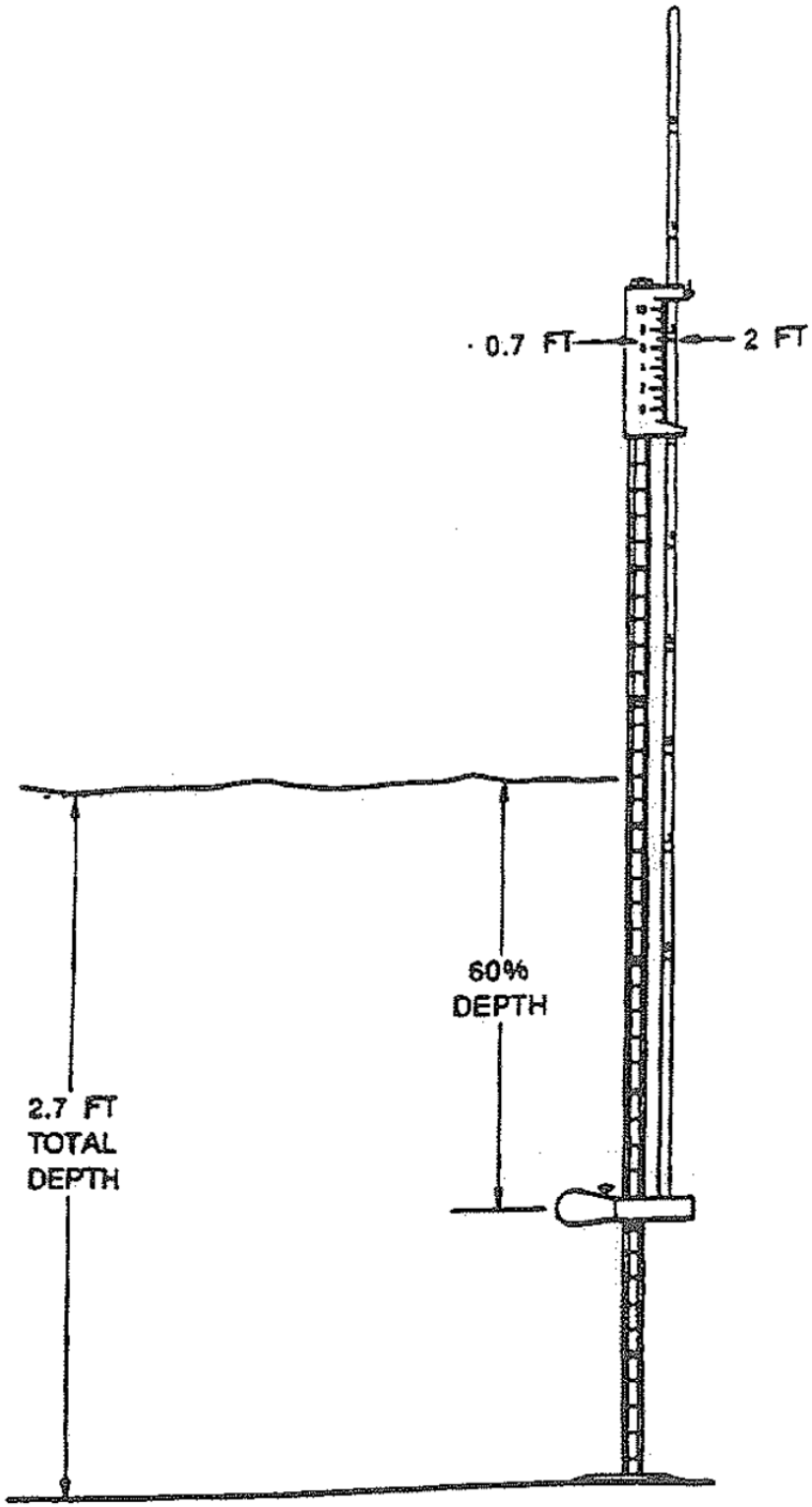


Figure C-3.2. Setting the wading rod at 60% depth when at a station that is 2.7 meters deep.

Alternate methods

Conditions may not always allow for use of flow meters. In these cases, discharge can be estimated through alternate methods such as those listed below. At the bottom of the *Discharge Worksheet* (Figure C-3.1), record which alternate method was used. If an alternate method other than the timed float was used, record the estimated discharge value (cfs).

Examples of alternate methods are:

- Velocity of timed floats (e.g., oranges, plastic golf balls, sticks).
- Time to fill a bucket of known volume (e.g., discharge off of a hanging culvert).
- Obtaining collocated gage data or model data.

Timed float

In the absence of a current meter, you can time the transport of a neutrally-buoyant object 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.

<u>Velocity</u>

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.

<u>Cross-sectional area</u>

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.

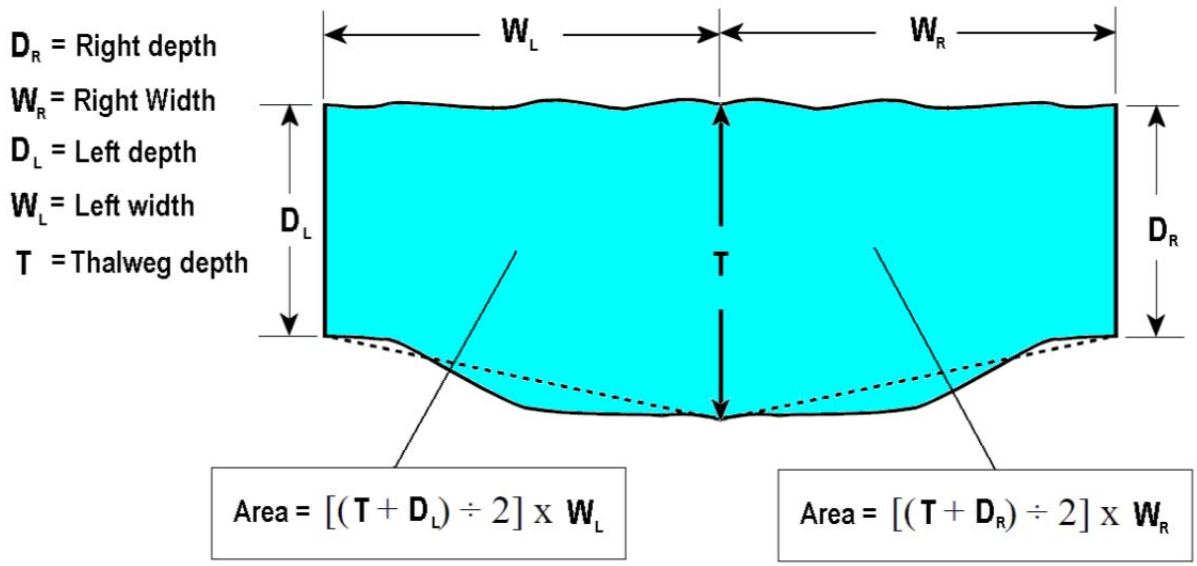


Figure C-3.3. The cross-sectional area of a stream segment as estimated by calculating the area of component trapezoids, centered on the thalweg.

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.

<u>Discharge</u>

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 C-3.1. 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 data

Record discharge (cfs) at the bottom of the Discharge Worksheet (Figure C-3.1) and note the data source, next to “Describe alternate method”.

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 to three different samples. One sample will be sieved to less than 63 µm and analyzed for metals (arsenic, copper, lead, and zinc). A second 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.) and total-organic carbon (TOC). Optionally, a third sample may be collected and sieved to less than 2.0 mm and analyzed for percent particle-size distribution less than 63 µm (sand/silt) by a laboratory. This laboratory analysis for grain size is optional, but is likely going to be an element of the RSMP sediment monitoring. The grain size estimation in the field (stated below) is required, if the laboratory analysis is not undertaken by the permittees.

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 C-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)
▪ Glass, 500-1,000 mL for organics
▪ Plastic, 500-1,000 mL for metals
▪ Plastic, 500-1,000 mL for particle-size analyses (optional for grain size)
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
8. Rinsing with pesticide-grade hexane
9. 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.

<u>Metals samples</u>

- 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.

<u>Organics samples</u>

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): PS

Status and Trends - Chemistry and Sampling Form

DCE: WAM0600 - 000001 - 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		
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.

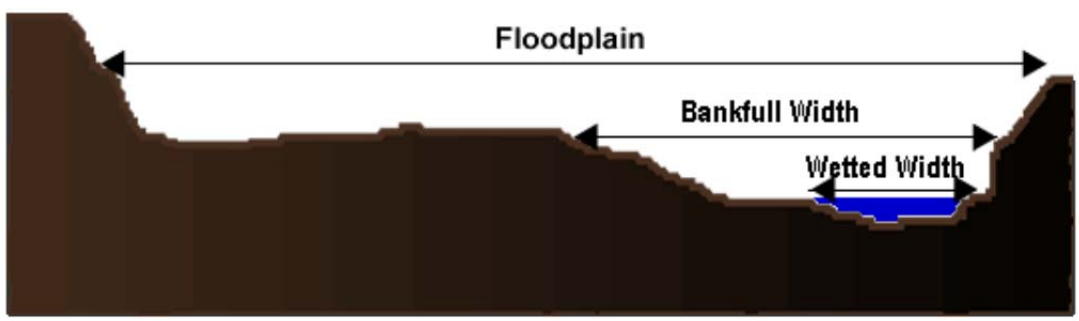


Figure C-5.3. Diagram of widths at the transect (Modified from Endreny 2009).

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).

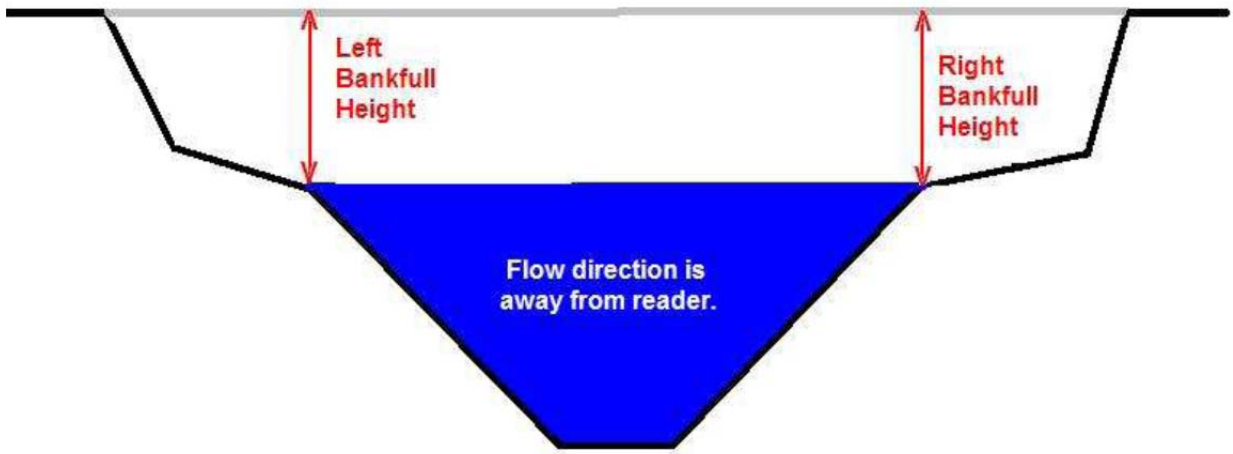


Figure C-5.4. Diagram of the left and right bankfull height measurements.

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.

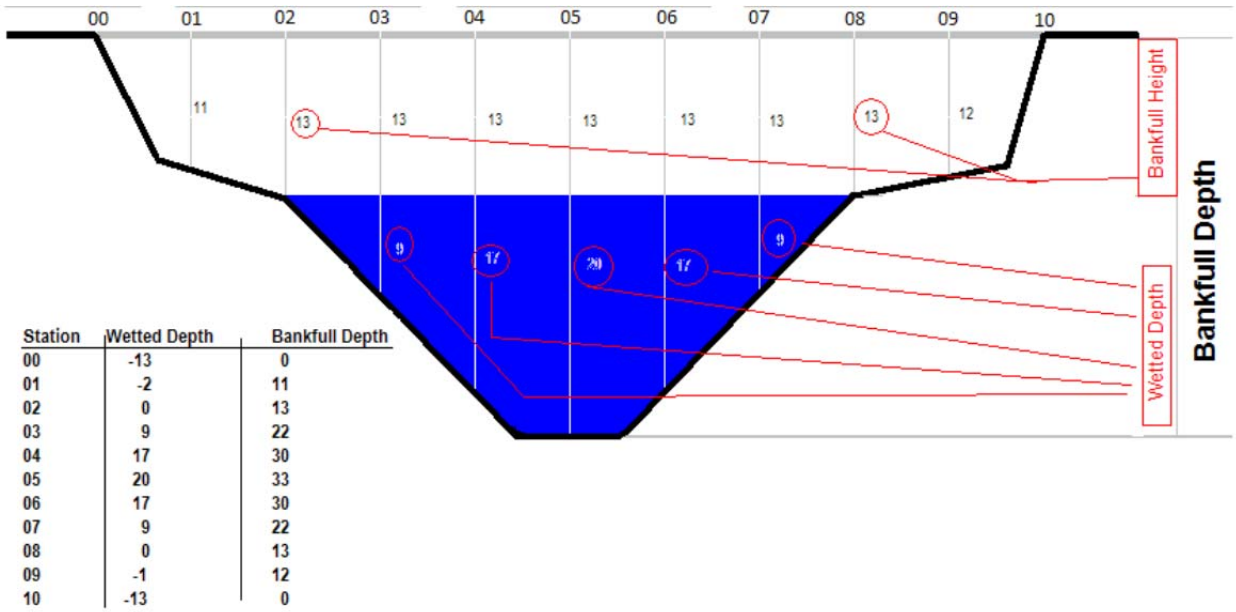


Figure C-6.3. Transect diagram showing example data for wetted depth, bankfull depth, and bankfull height. The bankfull depth equals the wetted depth plus average bankfull height.

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.

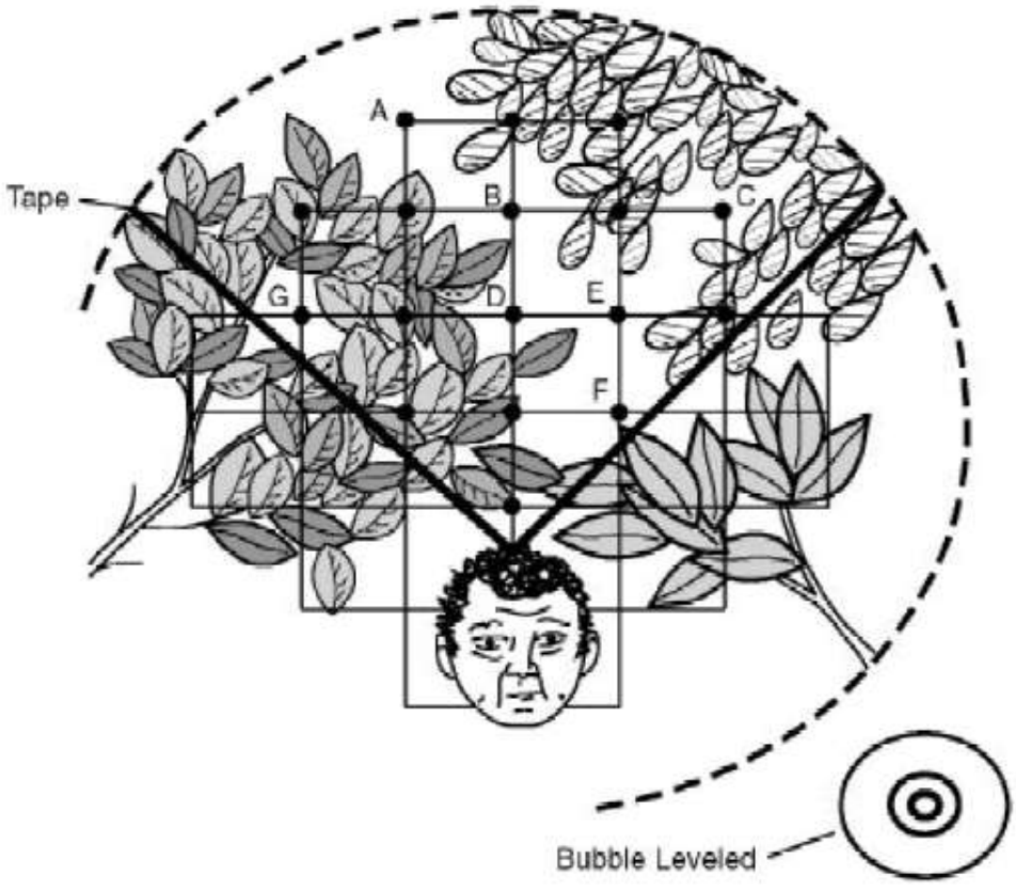


Figure C-7.2. An example reading from a modified convex densiometer. It shows 10 of 17 intersections with shade (a score of "10").

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.

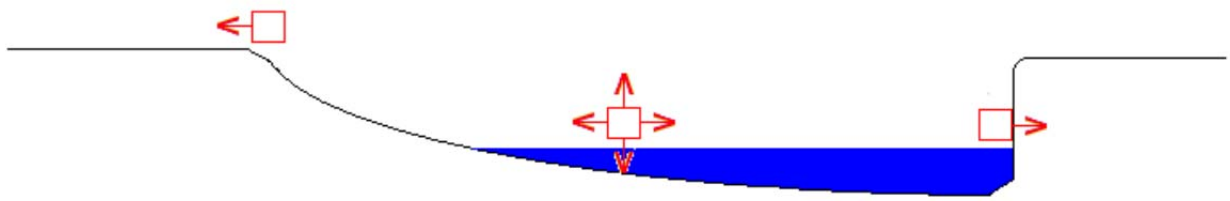


Figure C-7.3. Stations for densiometer measurement on each major transect. The densiometer is held level, and 30 cm above water for wet stations and 30 cm above ground for dry stations.

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.

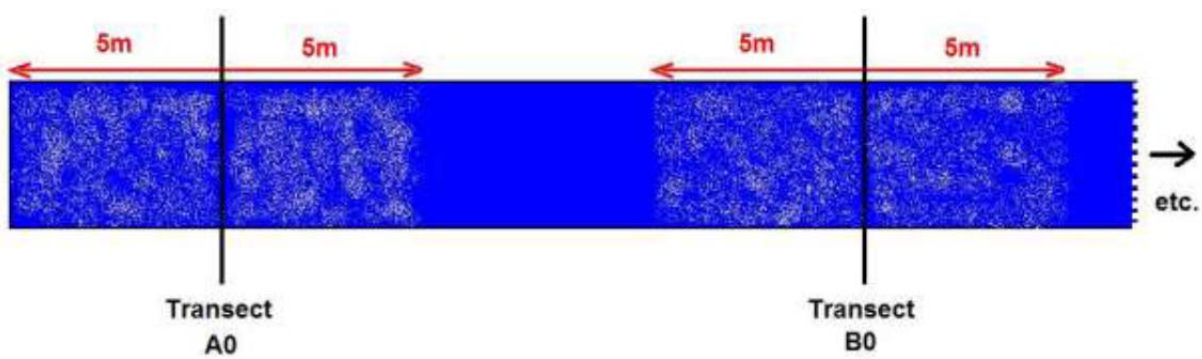


Figure C-8.1. Diagram of fish cover plots at each major transect of the main channel.

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.

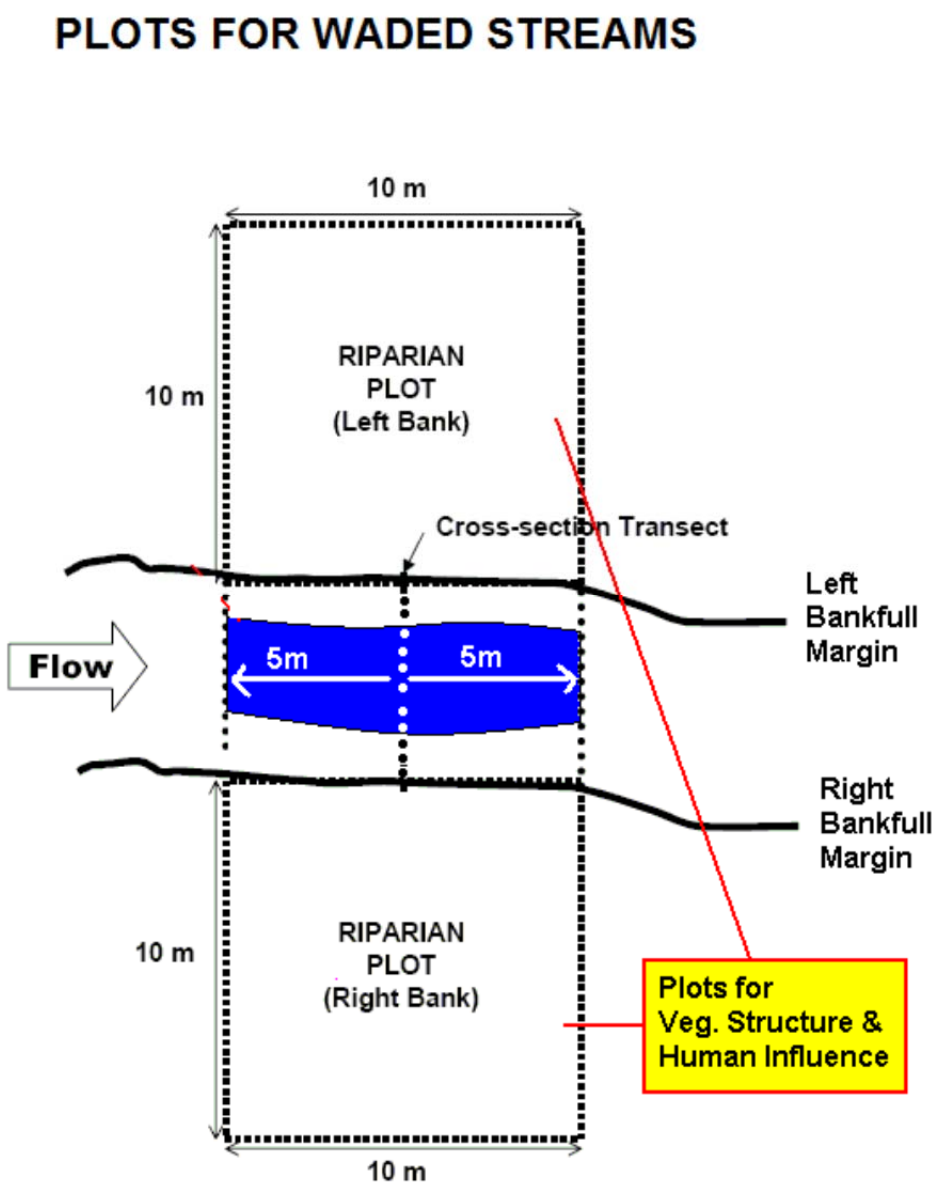


Figure C-9.3. Riparian plots.

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.

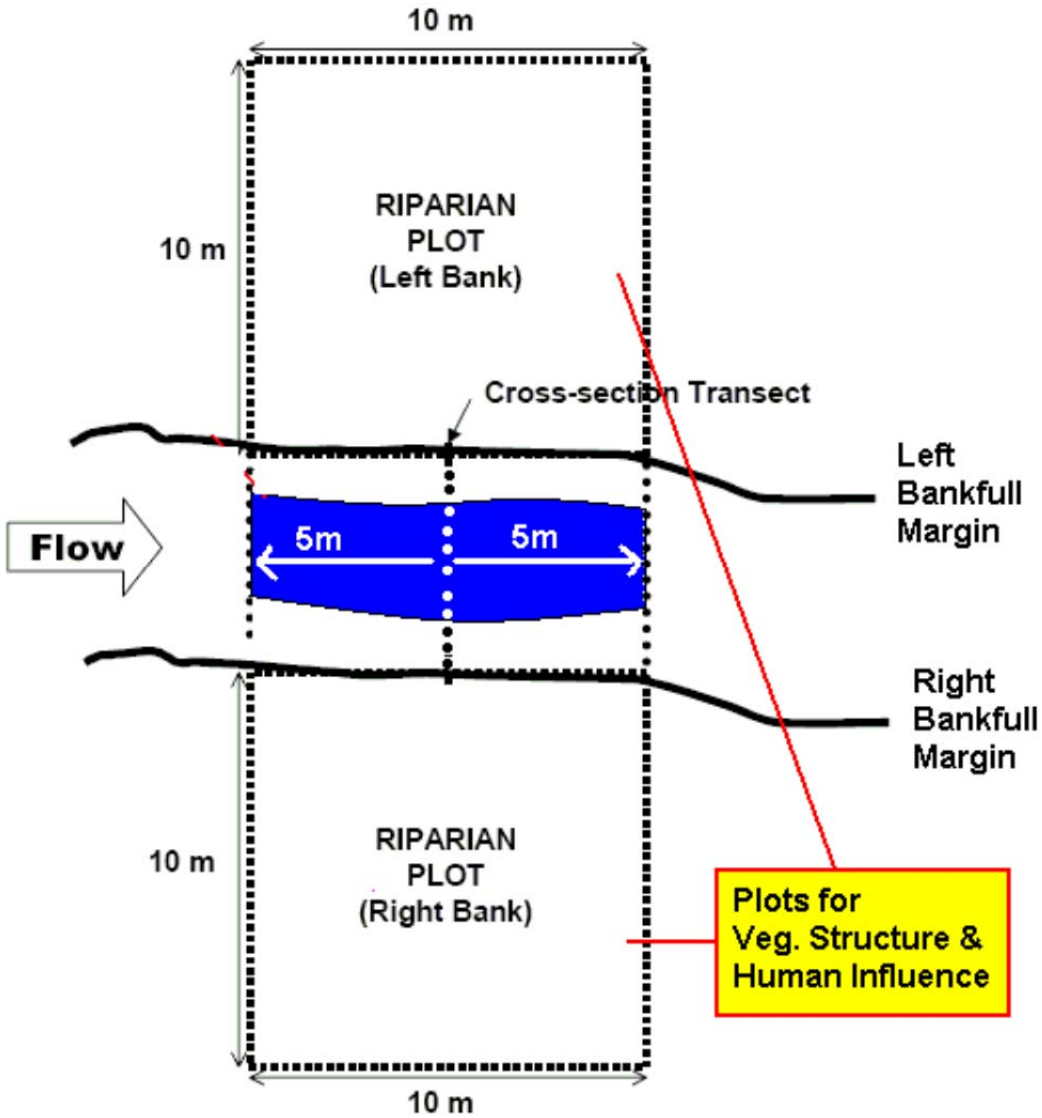


Figure C-10.2. Riparian plots.

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.

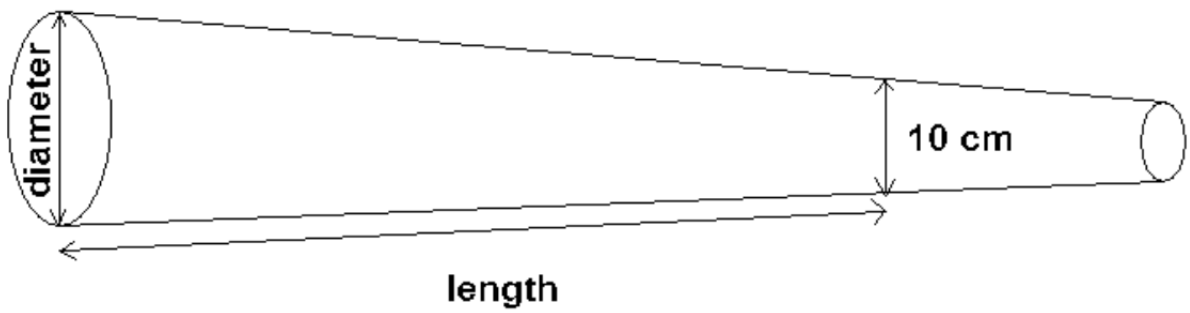


Figure C-12.1. Diagram of how to estimate the dimensions of a log.

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	𝍷	2	
30-60 cm	𝍷	1	𝍷	3	𝍷	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.

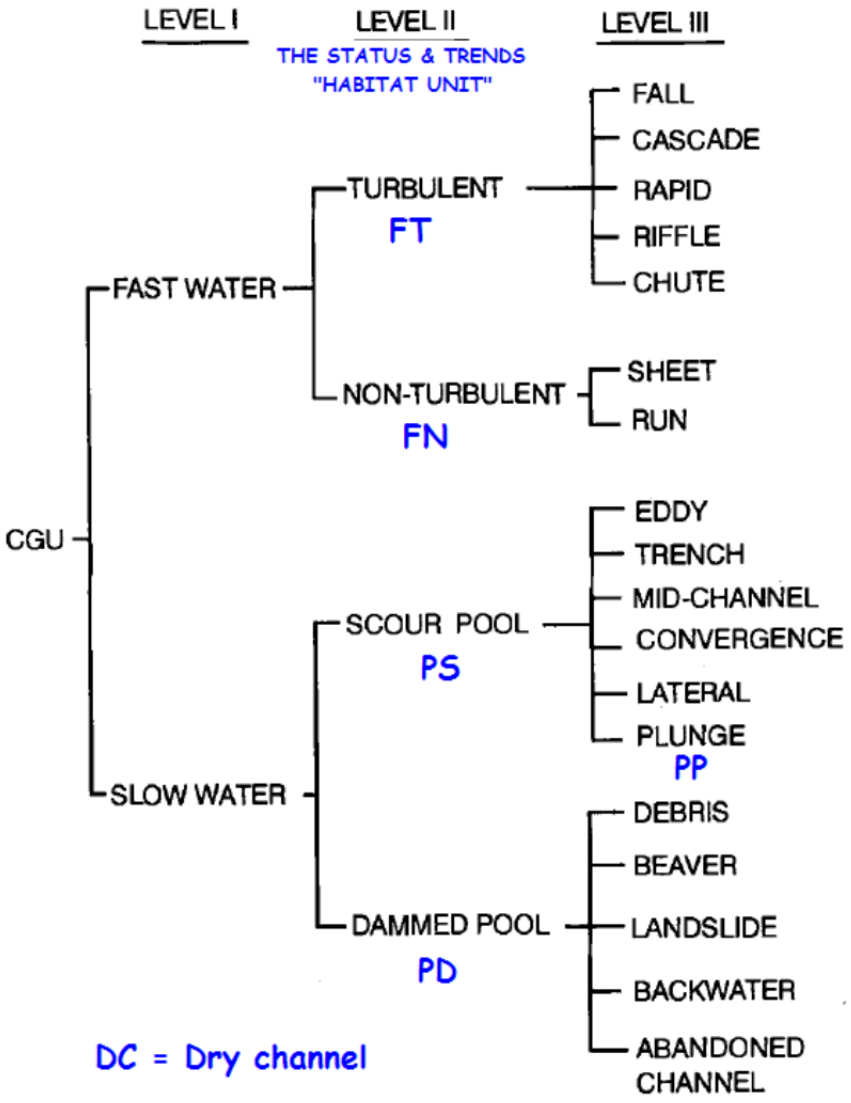


Figure C-13.1. Categories of channel geomorphic units (CGU) described by Hawkins et al. (1993) and their three levels of resolution.

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 need 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).

Wet Width (m.x)	BF Width (m.x)	Bar Width (m.x)
4.2	5.3	0.3

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.

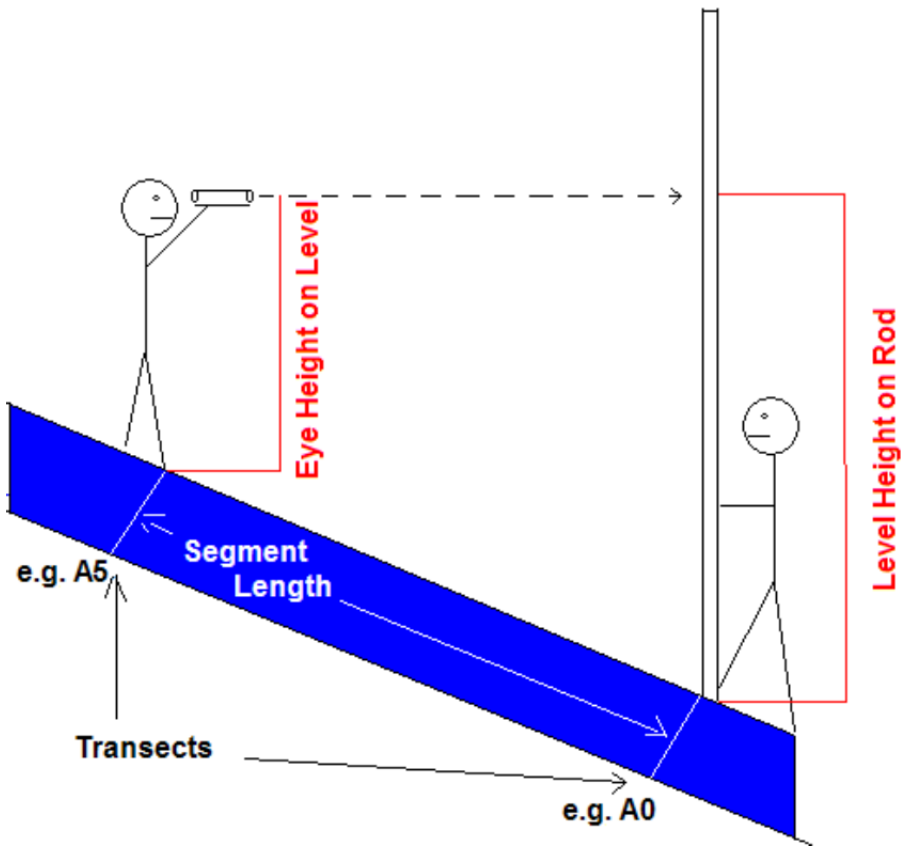


Figure C-16.1. Crew positions when measuring the slope and bearing.

Status & Trends - Slope and Bearing Form

Site Number YY MMDD HH MM

DCE: W A M 0 6 0 0 - 0 0 0 1 5 2 - D C E - 2 0 0 9 - 0 7 1 5 - 1 0 : 0 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
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	228	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 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 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.

<u>Flowing water stations</u>

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-ft2 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): JS

Status and Trends - Chemistry and Sampling Form

DCE: WAM0600 - 000001 - 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): F 0

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): F 0

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): F 0

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.

TBD-Per personal communication with Brandi Lubliner email dated 7/10/2014

~~D-3. Field sampling periphyton in small streams~~

~~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. Washington State's biological monitoring program follows Collyard (2010) for standard sampling procedure. 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.~~
- ~~Aluminum foil.~~
- ~~Wash bottle.~~
- ~~Plastic funnel.~~
- ~~One gallon Ziploc bag.~~
- ~~Large plastic bowl or tray to catch rinsate.~~
- ~~95% Ethanol (add 3 parts by volume for each part sample).~~
- ~~Deionized or distilled water.~~
- ~~Label (waterproof) for jar exterior.~~
- ~~Soft-lead pencil.~~
- ~~Clear tape.~~
- ~~Electrical tape.~~
- ~~Pocket knife.~~
- ~~Wading gear.~~

~~Summary of procedure~~

~~*Sampling method for epilithic (coarse substrate) habitats*~~

~~Periphyton sampling can be performed on-site with macroinvertebrate sampling, after site verification and layout. One sample is collected at each of 8 major transects and are added to a single composite sample jar for the site.~~

1. ~~Eight samples are randomly collected from each reach. A periphyton sample is taken at each major transect where macroinvertebrate samples are taken. Samples will be collected in close~~

~~proximity to (but not within) the randomly selected macroinvertebrate sample locations. See *Benthos Sampling* above for description of selecting random sample locations.~~

2. ~~Carefully remove a rock from each of the eight 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 only one rock per randomly selected sample location if the diameter of the first rock selected is equal to or exceeds 7.5 cm. If the diameter of the first rock selected is less than 7.5 cm, select a second rock. If possible, select rocks that are similar with respect to size, depth, and exposure to sunlight. Gently place the rocks (as they were oriented in the stream) in a plastic tray; do not stack rocks 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.~~

3. ~~Measure water depth and velocity at each of the eight locations using a top-setting rod and velocity meter and record on the datasheet. NOTE: Additional measurements of depth and velocity are not required if the sampler is already measuring these parameters for the macroinvertebrate sample. Assuming the sun is directly overhead, determine the relative degree of riparian shading (e.g., shaded, partial, or full sun) at each randomly selected sample location and record on the datasheet.~~

4. ~~Scrub only the upper surface of each rock with a firm-bristled toothbrush using a circular motion. In circumstances where rocks are much greater than 10 cm (medium to large cobbles), firmly brush only a portion of the upper rock surface around 10 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 deionized or distilled water. Use rinse water sparingly, but be thorough. Collect rinsate in the plastic tray containing any filamentous algae or mosses. Repeat for the remaining rocks. 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 500 mL Nalgene® sample bottle.~~

5. ~~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.~~

6. ~~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 microalgal 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 a total of eight pieces of woody snag material from the same number of different locations throughout the reach. Select pieces greater than 1 cm in diameter; these have likely been submerged for most of the year to allow for sufficient periphyton colonization but 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. ~~Measure water depth and velocity at the point where each of the eight woody snags 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 of the eight sample.~~

4. ~~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 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 section of wood with a rinse bottle containing deionized or distilled 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 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 500 mL Nalgene® sample bottle.~~

5. ~~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) = $\sum_{i=1}^{n}(\pi)(di)(li)$~~
   ~~Where,~~
   ~~n = number of discrete collections~~
   ~~p = 3.1416~~
   ~~di = mean diameter of each woody section, in centimeters~~
   ~~li = 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.~~

6. ~~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 epipsammic (pea gravel/sand) and epipelic (silt) habitats~~

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

1. ~~Select a total of five different locations, in shallow organically-rich depositional zones that consist of either pea gravel, sand, or silt substrate.~~

2. ~~At each 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 deionized or distilled water into a 500 mL Nalgene® sample bottle. Use rinse water sparingly, but be thorough. Combine all five discrete sample collections in the 500 mL Nalgene® sample bottle. Repeat at the remaining sample locations. 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.~~

9. ~~The total sample surface area for all five discrete samples collected with a 47 mm Petri dish is 85 cm^2. Record the sampled surface area on the datasheet.~~

10. ~~Measure water depth and velocity at the point where each of the five 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 of the five sample locations 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, epipsammic, 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 five sections of emergent vegetation from the same number of different locations throughout the reach. Each section represents a stem or leaf (no roots); however, all five 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, allowing for sufficient periphyton colonization but not smothered by bottom sediments.~~

~~7. Carefully remove an approximately 10 to 20 cm long section of each stem or leaf with pruning shears or a 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.~~

~~8. Measure water depth and velocity at the point where each of the five sections of emergent vegetation were removed using a top-setting rod and velocity meter and record on the datasheet.~~

~~9. 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~~

~~deionized or distilled 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 500 mL Nalgene® sample bottle.~~

10. ~~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) = $\sum_{i=1}^{n}(\pi)(di)(li)$~~

    ~~Where,~~
    ~~n = number of discrete collections~~
    ~~p = 3.1416~~
    ~~di = mean diameter of each cylindrical stem section, in centimeters~~
    ~~li = length of each cylindrical stem section, in centimeters~~

11. ~~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) = $\sum_{i=1}^{n} 3(wi)(li)$~~

    ~~Where,~~
    ~~n = number of discrete collections~~
    ~~wi = mean width of each triangular stem section, in centimeters~~
    ~~li = length of each triangular stem section, in centimeters~~

12. ~~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 datasheets.~~

13. ~~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, epipsammic, epipelic, or epiphytic-emergent vegetation), 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 five samples of submerged vegetation from the same number of different locations throughout the reach. 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 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.~~

5. ~~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.~~

6. ~~Measure water depth and velocity at the point where each of the five samples of submerged vegetation were removed using a top-setting rod and velocity meter and record on the datasheet.~~

7. ~~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. Repeat for remaining samples. Within the plastic tray, separate the sample segments from the algal/moss segments.~~

8. ~~Fill a 1000 mL Nalgene® bottle with 300 mL of deionized or distilled water. Place sample segments in the Nalgene® bottle and cap the bottle. Shake the Nalgene® bottle vigorously for 30 seconds to dislodge attached periphyton. Open the Nalgene® bottle, remove individual segments and rinse segments with a bottle containing deionized or distilled water. Use rinse water sparingly, but be thorough. Collect rinsate in the 1000 mL Nalgene® bottle. Repeat for remaining sample segments. Set aside rinsed sample segments. Once all sample segments~~

~~have been processed, place algal/moss segments in the 1000 mL Nalgene® bottle. Any dislodged periphyton remaining in the plastic tray should be inserted through a funnel into the 1000 mL Nalgene® 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.~~

9. ~~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) = $\sum_{i=1}^{n}(\pi)(di)(li)$~~

   ~~Where,~~
   ~~n = number of discrete collections~~
   ~~p = 3.1416~~
   ~~di = mean diameter of each cylindrical stem section, in centimeters~~
   ~~li = 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.~~

10. ~~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 datasheets.~~

    ~~[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.~~

11. ~~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 or distilled 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 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 disposable serological volumetric glass pipette and dispense onto the middle of the wetted glass microfiber filter.~~
5. ~~Filter the aliquot 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 or distilled 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. Remove the filter from the funnel base with forceps and wrap in a small piece of aluminum foil. Place the aluminum foil wrapped filter in a separate 47 mm Petri dish.~~

   e. ~~Seal the sides of the Petri dish with plastic tape and label the Petri dish with the following required 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. Insert the labeled Petri dish(s) in a resealable plastic bag and place in a cooler containing dry ice. About 4.5 kg (10 pounds) of dry ice is needed for a small cooler (< 2 gal). Insulate the cooler with newspaper to minimize sublimation of dry ice. NOTE: Wet ice can be used if dry ice is not available. Make a note on the datasheet when wet ice is used.~~

~~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. Contact laboratory personnel to make them aware of plans to ship (via overnight shipping service) coolers containing dry ice and frozen subsamples. Make sure you disclose to the carrier the amount of dry ice in the cooler prior to shipping.~~

~~6. 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:~~

~~500 mL ID subsample, add 25 mL Lugol's solution.~~
~~400 mL ID subsample, add 20 mL Lugol's solution.~~
~~250 mL ID subsample, add 12 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].~~

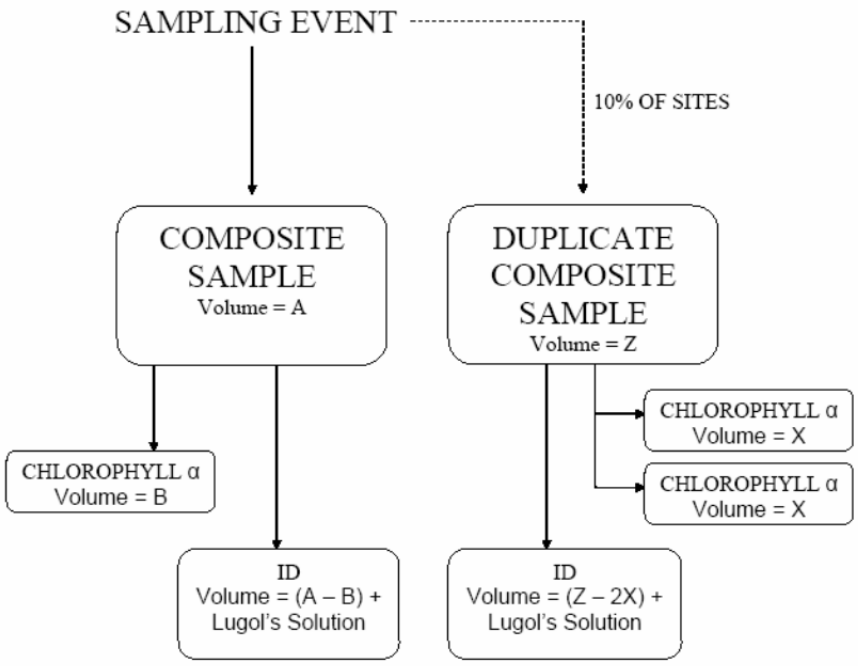
~~*Sample preservative-Lugol's solution*~~

~~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 Epipsammic/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.~~



~~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	
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

DCE: WAM0600 - 000001 - 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 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.

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.

~~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). Separate bench sheets will be used for algae and diatom counts, and these will be included in the data delivery.~~

~~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.~~

~~*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*~~

~~Taxa lists for each sample should be reported in a taxa- density matrix (expressed as abundance per cm^2). Surface area sampled must be reported on the chain-of-custody form. 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.~~

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.
- Turbidity will be collected in-situ with a 1-ml glass vial and measured with a Hach 2100p turbidity meter.
- 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 by the hand-dipped method 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 pH 7 and pH 10 standards. A linearity check will then be performed with a third pH 4 buffer.
- 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.
- 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.
- For turbidity, calibration must be conducted quarterly with factory supplied StablCal Stabilized Formazin Standards in 20, 100 and 800 NTU values. Prior to use, on the day of sampling, calibration checks are done with the factory supplied Gelex standards and follow the steps in the Procedure Manual provided with the Hach 2100P Portable Turbidimeter (Hach 2004)

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, hardness, nitrate-nitrite, total nitrogen, and total phosphorus) will require chemical preservation to maintain the integrity of the samples and prevent them from degrading prior to laboratory analysis.

Filtration is required for orthophosphate within 15 minutes after sample collection. Samples for orthophosphate will be filtered using a filtering set-up that pulls the sample through a filter using vacuum pressure created by either a peristaltic or hand pump. Prior to filtering the sample, an aliquot of deionized water will be passed through the filter to rinse the filter and container. 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.

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.

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.

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 (Hach HQ40d Portable Meter with the LDO Probe and pH301 probe and the Hach SensION5). It also includes the instrument used for measuring water velocity (YSI Flowtracker).

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 (YSI Flowtraker Handheld ADV® 2002).
- Five-gallon bucket (for flow meter zero-adjust).
- Hach Manuals (Hach 1999, Hach 2006a, Hach 2006b).
- pH 7 buffer (7.00) – Cole Palmer.
- pH 4 buffer (4.01) – Cole Palmer.
- pH 10 buffer (10.01) – Cole Palmer.
- Conductivity Standard (100 uS) – Cole Palmer.
- Conductivity Standard (1,000 uS) – Cole Palmer.
- Conductivity Standard (alternate as available).
- Deionized water (DI).
- Tap Water.
- Lab tissues (e.g., KimWipes®).
- Barometer from Hobolink® installed at Redmond Senior Center (https://www.hobolink.com/p/a055f30ffdc79010d4508b35b4cd2de3)
- Gelex standards for the Hach 2100p Turbidity Meter

Summary of procedure

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

Calibration of Hach 2100p, Hach HQ40d, YSI FlowTracker Handheld ADV and Hach sensION5 will be conducted based on the Hach manuals. Use the *Calibration Form* (Table F-1.1) to record calibrations and quality control checks. Each day, calibrate conductivity (COND), pH, dissolved oxygen (DO), turbidity (calibration check each day of use and calibration done quarterly with factory prepared standards) and velocity. The DO can be calibrated in Redmond's facility 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. Turbidity (Hach 2100p)
2. COND (Hach SensIon5)

3. pH (Hach pH301 probe with Hq40d meter)
4. DO (Hach LDO probe with Hq40d meter)
5. 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 Probes and the Flow Meter Sensor
Hach 1999, 2002 and YSI 2005

Rinse Probes 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			

<u>Calibrate conductivity to bracket expected field conductivity:</u>

1. Follow guidelines as set forth in the Hach SensION 5 Conductivity probe. Record data on the sheet.

Figure F-1. Temperature compensation with pH measurement.

APPLICATION NOTE

TEMPERATURE COMPENSATION WITH pH MEASUREMENT

Is there a temperature compensation table for pH measurement in samples?

The temperature coefficient of a sample is normally not known. Therefore no table exists correlating sample pH with temperature, as known from pH buffer solutions. That is why no exact temperature compensation can be made with sample measurements.

In order to correct the pH value of a sample to the calibration temperature, the following formula is commonly used in pH meter software.

$$S(T\ sample) = S(T\ cal) * \frac{T(sample) + 273.15}{T(cal) = 273.15}$$

S = slope
T = temperature °C
cal = calibration

With the new calculated slope S(T sample) from the mV signal, the pH of the sample can be calculated at sample temperature T(sample). A linear relationship is assumed between sample pH and temperature.

Example:

Calibration was done with pH buffers 4.01 and 7.00 at 24°C. The samples have been stored cool and now the measurement is done at 10°C.

The corrected pH value is calculated with slope (24°C) = -58,0 mV/pH and offset = 0.0mV:
Slope (10°C) = slope(24°C) * (10 + 273.15) / (24 + 273.15)
Slope (10°C) = -58.0 * (283.15) / (297.15)
Slope (10°C) = -55.28 mV/pH

pH value of the sample (measured potential +100 mV)
= 7 – 100 mV / -58.0 mV/pH = pH 5.28 (not corrected),
= 7 – 100 mV / -55.28 mV/pH = pH 5.19 (corrected)

The difference of 0.09 pH shows how important it is to precisely measure and correct for temperature.

FOR TECHNICAL ASSISTANCE, PRICE INFORMATION AND ORDERING:
Tel: 800-227-4224 | E-Mail: techhelp@hach.com
To locate the HACH office or distributor serving you, visit: www.hach.com

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In the interest of improving and updating its equipment, Hach Company reserves the right to alter specifications to equipment at any time.

HACH®
Be Right™

Buffers: a Hach 7.00, b Hach 4.01, c Hach 10.01.

From: http://www.hach.com/intellical-phc301-standard-refillable-ph-electrode-1-meter-cable/product-downloads?id=7640512232&callback=pf

Calibrate pH to bracket expected field pH

1. Measure 50ml of deionized water into a clean beaker with a clean stir magnet. Add the pH 7 buffer powder into the water. Place breaker on stir plate. Enter the theoretical pH value units into the Hach Hd40q meter. Theoretical values are based on temperature of the standard and are explained in figure below.
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 instrument). Be sure this percent slope matches the criteria described on the form. Otherwise, recalibrate.

Calibrate DO using water-saturated air

1. Fill the glass stopper bottle 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 D.O. probe into glass bottle and gently rest it on the cup.
4. Wait 5 minutes, making sure that temperature stabilizes.
5. Determine local barometric pressure (mm Hg) and record onto form.
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 probe 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-1.2. 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

Office and field diagnose the meter

Refer to YSI FlowTracker (pages 21-22).

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.*

Twice-seasonal checks

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

- pH 7 standard (6.97) – e.g., Hach buffer pillows.
- pH 4 standard (4.10) – e.g., Hach buffer pillows.
- pH 9 standard (9.15) – e.g., Hach buffer pillows.

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-1.3. 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 Hach'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 Rhithron. 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
Permission?:	Y N
Sampled?:	Y N
Wade or Raft?:	W R

If not sampled, why not?

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

General Notes

Draft

Figure G-1. Site Verification Form.

Is the site unsafe to access, or with barriers that prevent access (round trip) and sampling by wading within one day? Y N

Is the site unsafe to access, or with barriers that prevent access (round trip) and sampling by raft within one day? Y N

Why is it inaccessible?

SITE DIAGRAM

Provide North Arrow

Draft

Figure G-2. Site Diagram Form.

Reviewed by (Initials): _____

Status and Trends - Chemistry and Sampling Form

Site Number | YY | MMDD | HH : MM

DCE: W A M 0 6 0 0 - _ _ _ _ - D C E - 2 0 _ _ - _ _ _ _ - _ _ : _ _

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):

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 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

Draft

Discharge: ___,___.___cfs

Notes (discharge)

Figure G-4. Discharge Worksheet.

Major Transect Form

Reviewed by (Initials):_____

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

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	

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	

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.

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: W A M 0 6 0 0 - - - D C E - 2 0 - - :

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:

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:

Side Channel Number	Width (m.x)	Side Channel 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

Reviewed by (Initials):

Status and Trends Program - GPS Positions Form

Site Number YY MMDD HH : MM

DCE: W A M 0 6 0 0 - - D C E - 2 0 - - :

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

22965

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	No. 2 pencils
	General	Pencil sharpener
	General	Waders/Boots
	General	Backpacks
	General	This manual
	General	Clip boards (3): Habitat, Chemistry, Biology
	General	Camera
	General	Calculator
	General	Field notebook
	General	Digital Calipers
	Forms and labels	Site Verification Form x Site Diagram (1)
	Forms and labels	*Chemistry and Sampling Form* x Discharge Worksheet (1)
	Forms and labels	Major Transect Form x Thalweg Data Form (11)
	Forms and labels	*Slope and Bearing Form* x GPS Positions (1)
	Forms and labels	Calibration Form (1)
	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
	Calibration	Meter(s), components, maintenance kit
	Calibration	Meter Manuals
	Calibration	Quality control calibration standard (QCCS)
	Calibration	pH 7 buffer (7.00) - e.g., Hach
	Calibration	pH 4 buffer (4.01) - e.g., Hach
	Calibration	pH 10 buffer (10.01) - e.g., Hach
	Calibration	pH 7 standard (6.97) - e.g., Hach
	Calibration	pH 4 standard (4.10) - e.g., Hach
	Calibration	pH 9 standard (9.15) - e.g., Hach
	Calibration	Conductivity Standard (100 uS) - e.g., VWR 23226-589
	Calibration	Conductivity Standard (1,000 uS) - e.g., VWR 23226-603
	Calibration	Conductivity Standard (alternate as available)
	Calibration	Deionized water (DI)
	Calibration	Tap water
	Calibration	Lab tissues (e.g., KimWipes®)
	Calibration	Barometer
	In-situ	Hach meter with LDO and pH probes,
	In-situ	Hach Conductivity Probe
	In-situ	Hach 2100p Turbidity Meter
	In-situ	Hach 1ml glass vial with cap and oil
	Discharge	Flow meter
	Discharge	Flow meter manual
	Discharge	Batteries
	Discharge	Wading rod (top setting)
	Benthos sampling	Wide-mouth polyethylene jar (128 oz or 3.8 L)
	Benthos sampling	D-Frame kick net with handle and frame
	Benthos sampling	95% Ethanol (add 3 parts by volume for each part sample)

Check Box (√)	Category	Item
	Benthos sampling	Clear tape
	Benthos sampling	Electrical tape
TBD	~~Periphyton sampling~~	~~Firm bristle toothbrush~~
TBD	~~Periphyton sampling~~	~~Plastic tray or bowl~~
TBD	~~Periphyton sampling~~	~~Rinse bottle~~
TBD	~~Periphyton sampling~~	~~Aluminum foil~~
TBD	~~Periphyton sampling~~	~~Wide-mouth polyethylene jar (1 L)~~
TBD	~~Periphyton sampling~~	~~95% Ethanol~~
TBD	~~Periphyton sampling~~	~~Gallon Ziploc bags~~
TBD	~~Periphyton sampling~~	~~Plastic funnel~~
TBD	~~Periphyton sampling~~	~~Distilled or deionized water~~
TBD	~~Periphyton sampling~~	~~Pocket knife~~
	Habitat	GPS
	Habitat	Maps
	Habitat	Measuring rod
	Habitat	50-m tape
	Habitat	Laser rangefinder
	Habitat	Hand level with monopod
	Habitat	Clinometer
	Habitat	PVC ring with 10-cm ID
	Habitat	Modified convex densiometer
	Habitat	Flagging
	Habitat	Permanent marker
	Habitat	Calipers
	Habitat	Field notebook
	Habitat	Compass

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				