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NATIONAL COUNCIL FOR AIR AND STREAM IMPROVEMENT

# LONG-TERM RECEIVING WATER STUDY
# FIELD METHODS COMPENDIUM - 2007

**TECHNICAL BULLETIN NO. 948**

**MAY 2008**

**by**
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NCASI wishes to acknowledge the contributions of the Long-Term Receiving Water Study Science Advisory Panel (SAP) in developing methods used in field monitoring. Although methods used in field monitoring were based on existing published methods, input and discussion with the SAP were important in the fine-tuning of methods for individual study locations. Members of the SAP include 1) Dr. Monique Dubé, University of Saskatchewan; 2) Mr. Barry Firth, Weyerhaeuser Company; 3) Mr. Tibor Kovacs, FPInnovations; 4) Dr. Wayne Landis, Western Washington University; 5) Dr. Wayne Minshall, Idaho State University; and 6) Dr. John Rodgers, Clemson University. Other input for methods came from the current technical support staff at NABF, including Bill Arthurs, Renee Ragsdale, Joan Ikoma, and former staff members Dr. Judy Dudley and Dr. Daniel McGarvey. We would also like to thank Dr. Fred Howell, University of Southern Mississippi, and Jan Napack from the NCASI West Coast Regional Center (WCRC) for photographs of the water quality sites in Mississippi and Oregon. Susan Easthouse assisted with document formatting and editing. Project oversight was carried out under the direction of Tim Hall, then NCASI Aquatic Biology Program Manager, and Camille Flinders, recently named Aquatic Biology Program Manager upon Tim Hall's retirement.

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## PRESIDENT'S NOTE

NCASI Technical Bulletin No. 841, issued in 2002, describes field methods used in the NCASI Long-Term Receiving Water Studies (LTRWS). These studies were initiated in 1998 and 1999, and are being conducted in representative U.S. receiving waters, including Codorus Creek in Pennsylvania, the Leaf River in Mississippi, and the McKenzie and Willamette Rivers in Oregon. The four locations represent a matrix of coldwater/warmwater streams, bleached/unbleached mill process types, geographic locations, and instream waste concentrations. Field methods employed in the LTRWS include procedures for the bioassessment of periphyton (algae), macroinvertebrates (aquatic insects), and fish communities as well as the characterization of instream water quality and habitat conditions.

As a long-term (10-20-year) study, an important element of the LTRWS is the incorporation of an adaptive management strategy, recognizing that knowledge gained as the study progresses may call for method changes in order to fulfill aquatic biology information needs while also optimizing the use of study resources. The current technical bulletin provides an update to the 2002 field methods, and reflects a variety of monitoring changes resulting from the practical experience and knowledge gained during the initial years of monitoring. These changes include an update to sampling sites based on channel changes to the two Oregon Rivers, and modifications of the substrate used for sampling periphyton on the Leaf River from sand to artificial substrates in order to reduce variability for chlorophyll measurements. An additional change was the reduction in sampling frequency from twice to once per year for Codorus Creek, based on the knowledge gained regarding temporal and spatial variability of the measured biological endpoints and related physical/chemical habitat variables.

The LTRWS receives important guidance from a six-person Science Advisory Panel (SAP), convened twice yearly since the beginning of the LTRWS, including the period of initial monitoring method development. Methods and method changes presented in the current technical bulletin are a reflection of the combined knowledge and expertise of both NCASI and the LTRWS SAP. Through 2007, the LTRWS has resulted in more than 30 NCASI reports and 14 peer-reviewed journal articles. The importance of a sound experimental design, as exemplified by the described field methods, is fundamental to the establishment of a valid knowledge base on the potential for effluent effects. The LTRWS will continue to use the adaptive management approach to modify, where necessary and appropriate, field methods used in the study.

Ronald A. Yeske

May 2008

## MOT DU PRÉSIDENT

Le bulletin technique n° 841 de NCASI, publié en 2002, décrit les méthodes de terrain utilisées dans le cadre des études à long terme des cours d'eau récepteurs (*Long-Term Receiving Water Studies, LTRWS*). NCASI a initié ces études en 1998 et 1999, dans certains cours d'eau récepteurs représentatifs situés aux États-Unis : le ruisseau Codorus en Pennsylvanie, la rivière Leaf au Mississippi et les rivières McKenzie et Willamette en Orégon. Ces quatre emplacements représentent une matrice comportant différentes variables : cours d'eau froide/tiède, types de procédé de pâte blanchie/non blanchie, différents emplacements géographiques et différentes concentrations dans les cours d'eau. Les procédures pour l'évaluation biologique du périphyton (algues), des macroinvertébrés (insectes aquatiques) et des communautés de poissons de même que la caractérisation de la qualité de l'eau des cours d'eau et des conditions des habitats comptent parmi les méthodes de terrain utilisées dans les LTRWS.

Puisque les LTRWS sont des études à long terme (d'une période de 10 à 20 ans), l'incorporation d'une stratégie de gestion évolutive constitue un élément important. En effet, les connaissances acquises tout au long des études peuvent entraîner des changements dans les méthodes afin de rencontrer les besoins en matière d'information sur la biologie aquatique tout en optimisant l'utilisation des ressources allouées aux études. Ce bulletin technique porte sur la mise à jour des méthodes de terrain de 2002 et présente une série de changements dans les méthodes de suivi, résultant à la fois de l'expérience pratique et des connaissances acquises pendant les premières années de suivi. Ces changements touchent la mise à jour des sites d'échantillonnage en fonction des modifications de l'écoulement dans les deux rivières de l'état de l'Orégon et le changement de substrats utilisés pour échantillonner le périphyton dans la rivière Leaf (le sable a été remplacé par des substrats artificiels afin de réduire les variations dans les mesures de chlorophylle). La réduction de la fréquence d'échantillonnage (qui est passée de deux fois l'an à une fois l'an) dans le ruisseau Codorus constitue un changement additionnel. Ce dernier changement s'appuie sur les connaissances acquises en matière de variabilités temporelle et spatiale des mesures terminales de l'évaluation biologique et des variables physiques et chimiques des habitats associés.

Un comité-conseil, constitué de six scientifiques (*Science Advisory Panel, SAP*) qui se réunissent deux fois l'an, guide les travaux des LTRWS depuis le tout début, même lors de la période initiale de développement des méthodes de suivi. Les méthodes présentées dans ce bulletin ainsi que les modifications apportées aux méthodes sont le reflet de la combinaison de connaissances d'expertises de NCASI et du comité SAP des LTRWS. Pendant l'année 2007, les LTRWS ont généré plus de 30 rapports NCASI et 14 articles scientifiques d'examen par les pairs. Il est fondamental que ce type de programme d'échantillonnage et de suivi, comme le démontre les méthodes de terrain décrites dans ce bulletin, soit conçu de façon judicieuse. En effet, ce programme permet d'établir une base de

connaissances valide sur les effets potentiels des effluents. Les LTRWS continueront de s'appuyer sur une approche de gestion évolutive afin de modifier, lorsque nécessaire et approprié, les méthodes de terrains utilisées dans les études.

Ronald A. Yeske

Mai 2008

# LONG-TERM RECEIVING WATER STUDY FIELD METHODS COMPENDIUM - 2007

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MAY 2008

## ABSTRACT

Methods are described for bioassessment and physical/chemical habitat measurements made in conjunction with the NCASI Long-Term Receiving Water Study (LTRWS) as incorporated at Codorus Creek (Pennsylvania), Leaf River (Mississippi), and McKenzie and Willamette River (Oregon) study locations. The current bulletin reflects an update to methods originally described in Technical Bulletin 841 and includes several changes resulting from knowledge and practical experience gained during the initial years of the LTRWS. Included are detailed descriptions for the collection and processing of water quality samples, including a detailed description and photographic record of each sample location. Bioassessment methods for periphyton, macroinvertebrate, and fish populations are also described, including a detailed description and photographic record of biological sample sites. Some site location changes are noted and documented based on river channel changes that have taken place since the study was initiated. Periphyton and macroinvertebrate method descriptions include the use of both natural and artificial substrate procedures as well as those for characterizing temperature, current velocity, solar radiation, substrate, and habitat parameters. Fish community assessment procedures are described for both boat and backpack electrofishing and include the process for obtaining fish morphometric data while maintaining fish in a healthy condition for their return to the receiving water. There are safety issues and procedures related to each of the field procedures that are also described.

## KEYWORDS

effluent, fish, habitat, macroinvertebrate, periphyton, water quality

## RELATED NCASI PUBLICATIONS

Technical Bulletin No. 841 (February 2002). *A compendium of field methods used in NCASI long-term receiving water studies.*

Technical Bulletin No. 752 (March 1998). *A compendium of stream and river monitoring methods*.

# COMPENDIUM DES MÉTHODES DE TERRAINS UTILISÉES DANS L'ÉTUDE À LONG TERME DES COURS D'EAU RÉCEPTEURS - 2007

BULLETIN TECHNIQUE N$^{O}$ 948
MAI 2008

## RÉSUMÉ

Ce bulletin décrit les méthodes d'évaluation biologique et de mesures des paramètres physique/ chimique des habitats réalisées par NCASI dans le cadre des études à long terme des cours d'eau récepteurs (*Long-Term Receiving Water Study, LTRWS*) au ruisseau Codorus (Pennsylvanie), à la rivière Leaf (Mississippi) et aux rivières McKenzie et Willamette (Orégon). Il présente une mise à jour des méthodes décrites originalement dans le bulletin technique n$^{o}$ 841 et inclut plusieurs changements résultant des connaissances et de l'expérience pratique acquises lors des premières années des LTRWS. Il présente également des descriptions détaillées de la collecte et du traitement des échantillons pour la qualité de l'eau de même qu'une description détaillée et des photos de chaque site d'échantillonnage. Les méthodes d'évaluation biologique du périphyton, des macroinvertébrés et des populations de poissons sont également décrites et le rapport contient une description détaillée et des photos des sites d'échantillonnage biologique. Les auteurs notent et documentent certains changements au niveau de l'emplacement des sites d'échantillonnage qui font suite aux changements de l'écoulement dans certaines rivières depuis que les études ont débuté. Les descriptions des méthodes d'évaluation du périphyton et des macroinvertébrés portent sur les procédures d'utilisation de substrats naturel et artificiel de même que sur les procédures de caractérisation de la température, de la vitesse du courant, de la radiation solaire, du substrat et des paramètres reliés aux habitats. Le bulletin décrit les procédures d'évaluation des communautés de poissons au moyen de dispositifs de pêche électrique installés sur un bateau et portatifs. Ces procédures présentent aussi le processus pour obtenir des données morphométriques sur les poissons tout en maintenant ces derniers dans des conditions optimales pour leur retour dans les eaux réceptrices. Enfin, le bulletin présente des informations sur les enjeux et les procédures de santé sécurité reliées aux méthodes de terrains.

## MOTS CLÉS

effluent, poisson, habitat, macroinvertébrés, périphyton, qualité de l'eau

## AUTRES PUBLICATIONS DE NCASI DANS CE DOMAINE

Bulletin technique n$^{o}$ 841 (février 2002). *A compendium of field methods used in NCASI long-term receiving water studies.*

Bulletin technique n$^{o}$ 752 (mars 1998). *A compendium of stream and river monitoring methods*.

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# LONG-TERM RECEIVING WATER STUDY FIELD METHODS COMPENDIUM - 2007

## 1.0 INTRODUCTION

In 1998, NCASI initiated Long-Term Receiving Water Studies (LTRWS) on Codorus Creek in Pennsylvania, and on the Willamette and McKenzie Rivers in Oregon. An additional study location, the Leaf River in Mississippi, was added in 1999. The scope and framework for the LTRWS have been described by Hall and Miner (1997) and a description of the study locations by Hall et al. (1999). The LTRWS, expected to extend over a 10 to 20 year period, includes collection of multiple samples each year of instream aquatic biota, including periphyton, macroinvertebrate, and fish populations. Additional monitoring elements include water quality monitoring of the study rivers and adjoining tributary streams, effluent chemical and biological characterization, and habitat assessment. An overview of monitoring parameters and an initial characterization of biological and chemical properties have been provided by Hall et al. (2000) and NCASI (2002). This report represents an update of the standard operating procedures (SOPs) for carrying out periphyton, macroinvertebrate, and fish sampling for the LTRWS as well as supporting field data collection for habitat and water quality characterization.

## 2.0 GENERAL OPERATIONAL APPROACH AND FIELD SAFETY

Biomonitoring occurs twice a year in the spring (April–June) following the hydrograph decline after winter and in the fall (August–November) during low flow. Beginning in 2007, Codorus Creek is sampled once a year in the spring. Sampling should not be carried out within two weeks of a period of unusual water level fluctuations. Sampling dates should be adjusted as necessary to provide for safe sampling conditions (e.g., avoid high water). River discharge should be checked at the appropriate USGS station website prior to field sampling. The following are the USGS websites for river discharge information including suggested discharges for sampling.

Leaf River (New Augusta): http://waterdata.usgs.gov/usa/nwis/uv?site_no=02474560. Discharge should preferably not exceed 2000 cfs.

Codorus Creek (Spring Grove): http://waterdata.usgs.gov/pa/nwis/uv?01574500. Discharge should preferably be less than 70 cfs.

Willamette River (Harrisburg): http://waterdata.usgs.gov/or/nwis/uv?141660001. Discharge should not exceed 12000 cfs (preferably less than 10000 cfs) for sampling.

McKenzie River (Vida): http://waterdata.usgs.gov/nwis/dv/?site_no=14162500. Multiply the discharge at Vida by a correction factor of 1.1916. The corrected discharge should preferably be less than 5000 cfs.

Normally, a field crew of three or four people will be needed to complete the biological sampling fieldwork in 4 to 5 d, allowing for 8 to 10 h work/d. The time may vary according to whether artificial substrates or natural substrates are being sampled and the extent to which sampling activities for periphyton, macroinvertebrate, and fish activities are combined. One member of the field crew for each field trip will be designated as the Field Crew Leader (FCL). Division of labor for field activities is at the discretion of the FCL. Staff will cross-check each other's field sheets and sample labels before leaving the site in order to make sure that all work has been completed. The field crew consists primarily of NCASI staff from the Northwest Aquatic Biology Facility (NABF) in Anacortes, Washington, but on some occasions, other personnel may be involved, including staff from other

NCASI locations, representatives from the host mill, students, or contractors. It is the responsibility of the FCL that non-NABF individuals assisting in field sampling be adequately trained in the sampling procedures they will be using and in all aspects of field safety related to their work.

A personal flotation device (PFD) is required for all boat-related sampling activities. The use of a PFD is also encouraged at any time that conditions present a potential threat to personal safety. Such conditions may exist, for example, during periods of high water and swift current, or in the process of loading or unloading gear from the boat. Each member of the field crew should have at least minimal swimming skills. Footwear should have non-slip soles. All field crew from NABF must be trained in first aid and CPR. A first aid kit should be available in the field as well as a map with directions to the nearest hospital. A cell phone must be carried in the field and be available for use in emergencies.

Wadeable habitat in riffle areas will be selectively sampled for periphyton and macroinvertebrates. Location positions should be confirmed with a GPS and any discrepancies noted on the field data sheet. Personnel unfamiliar with these locations should consult project files, and highway or topographic maps.

Periphyton samples are taken in concert with benthic macroinvertebrate samples. On the McKenzie River, Willamette River, and Codorus Creek, macroinvertebrates are sampled with a Hess sampler and periphyton is sampled from river rocks. On the Leaf River, macroinvertebrate communities are evaluated from suspended Hester-Dendy (HD) multi-plate artificial substrates and from natural woody debris. Periphyton is sampled from the top plate of each HD sampler for taxonomic identification and chlorophyll content, while sand periphyton is sampled at sandbars for taxonomic identification. Chlorophyll had initially been measured in sand periphyton as well, but this was discontinued after June 2003 because the samples were often below detection.

The sampling of fish populations is based on both boat and backpack electrofishing. Safety procedures outlined in Section 5.1 should be followed very carefully to minimize risks associated with these activities. Backpack electrofishing locations are generally in close proximity to those described for periphyton and macroinvertebrate samples. Backpack electrofishing is carried out on wadeable portions of the stream and operators should be extremely cautious of staying clear of hidden deep-water pools. Boat electrofishing locations are typically mid-channel river “run” locations or along the shoreline where the riverbank is steeply cut and the water deep. NCASI will rely on experienced riverboat operators for boat electrofishing and these activities should be suspended at any time when conditions are determined to be unsafe by either the boat captain or the FCL.

A qualified person extraneous to the field team will audit field procedures periodically. This audit will include some or all of the elements involved in monitoring, including sample collection methods and data recording, at one or more of the sample stations. This audit may occur with or without advanced notice.

## 3.0 LTRWS WATER QUALITY MONITORING

### 3.1 Overview

Water quality samples are collected and analyzed in support of the LTRWS at each study location. Samples are collected on the Willamette and McKenzie rivers by NCASI West Coast Regional Center (WCRC) personnel, by Glatfelter mill personnel on Codorus Creek, and by a contractor on the Leaf River. As of 2005, water quality sampling was reduced from monthly to six times per year with sample dates bracketing biological sampling dates. Grab samples are collected from either bridges (Figure 3.1) or along the shoreline (Figure 3.2) at each location, and either analyzed in the field or later at NABF or at the NCASI WCRC in Corvallis, Oregon for nutrients. Analysis parameters include pH, temperature, turbidity, color, conductivity, TOC, chemical oxygen demand (COD), hardness, total nitrogen, and total phosphorus. Ammonia, nitrate + nitrite, and orthophosphate analyses were discontinued in January 2000 but began again in April 2005. Hardness is measured twice a year (in May and September) in conjunction with samples collected for metals analyses. Samples collected for metals analyses are sent to a contract lab (Columbia Analytical Services, Inc. (CAS)).

**Figure 3.1** Water Quality Sampling at Long Tom River

**Figure 3.2** Water Quality Sampling at Harvest, McKenzie River

## 3.2 Water Quality Sample Methods

### 3.2.1 *Equipment*

Blue ice
Bubble wrap
Cell phone
Clipboard
Container for carrying bottles and equipment from car to river
Coolers
Data sheets on waterproof paper:

1) NCASI LTRWS Water Quality Monitoring Field Data Sheet (Appendices A–D)
2) NCASI LTRWS Monthly Temperature and pH Calibration Check (Appendices A and E)

FedEx labels for shipping samples to NABF in Anacortes, WA
FedEx labels for shipping samples for nutrient analyses to WCRC in Corvallis, OR
Gloves (PVC)
GPS (optional)
Packaging tape
pH meter (Hach sensION1 or equivalent) with automatic temperature compensation
pH meter (backup)
pH probes (refillable glass probe for the Hach sensION1 including backup probe) (Hach catalog # 51940-00)
pH buffers to calibrate meter (pH 7.00, pH 10.01, and pH 4.01)

Plastic bags for the data sheets
Potassium chloride filling solution for pH probe (4M KCl)
Reflective orange vest
Safety glasses
Sample bottles (all bottles are prelabeled with site names):

1) 250-ml Nalgene for WCRC nutrients (1 per site) (acid washed at WCRC)
2) 500-ml Nalgene for the NABF water quality (1 per site) (acid washed at NABF)
3) 125-ml amber glass bottles with preservative (1 per site) for TOC and COD (Codorus Creek and Leaf River only)

SOP for LTRWS Water Quality Monitoring Sample Collection
Stainless-steel bucket with rope
Storage solution for pH probe
Thermometer (mercury, NIST traceable) to check pH meter calibration
Thermometer (backup if the pH meter thermometer fails)
Writing implements

### 3.2.2 *Preparation*

1) Sample bottles for nutrients are acid cleaned at WCRC. They are soaked in 20% HCl overnight and rinsed at least six times with deionized water. The bottles are prelabeled and are shipped to the sampling locations in coolers containing blue ice and prelabeled FedEx forms. These samples are unpreserved.

2) Sample bottles for NABF water quality are acid cleaned at NABF following the same procedures described for the nutrient bottles. The bottles are prelabeled and shipped to the sampling locations with blue ice in coolers. Concentrated sulfuric acid (250 µl) is added at NABF to each 125-ml glass amber bottle for TOC and COD analyses. The samples collected in the 500-ml Nalgene bottles are unpreserved. FedEx shipping forms are prelabeled at NABF and data sheets are included in the sample kit.

3) Ice packs should be placed in the freezer when the sample coolers are received.

4) Within 24 h prior to sampling, the pH meter(s) are calibrated at the sampler's lab following the manufacturer's instructions. Perform a 3 point calibration with pH 7.00, pH 10.01, and pH 4.01 buffers. The pH slope for the Hach sensION1 should be -58 ± 3 mV. If the slope is not within this range, the probe should be replaced with the backup probe and calibrated. On Codorus Creek, the pH meter used is the Orion Model 250A+. The slope should be 95 to 105%.

5) The pH meter is also used to measure the river water temperature in the field. Compare the temperature reading in water of the pH meter to the reading of the mercury NIST traceable thermometer. The field meter should be within 0.5˚C of the NIST traceable thermometer. Fill out the "NCASI LTRWS Monthly Temperature and pH Calibration Check" data sheet (See Appendix A for Codorus Creek and Appendix E for all other rivers).

6) In May and September, extra river water samples are taken at one site upstream of the mill discharge, one downstream of the mill discharge, and at least one tributary stream on each river for total metals analyses. See Table 2.1 for sites sampled for metals analyses. The sample bottles already contain preservative and are ordered from CAS. There are two bottles for each site: a) a bottle containing nitric acid as preservative for total metals and b) a bottle containing hydrochloric acid as preservative for mercury analyses. The sample bottles will be

prelabeled at NABF and shipped in a separate cooler from the other bottles received from NABF. Included in the sampling kit will be a prelabeled FedEx form, a chain of custody form which will be filled out in advance at NABF, and ice packs. Freeze the ice packs when the sample kit is received. The samples collected for metals will be shipped directly to CAS. See Section 3.2.5 for shipping instructions.

**Table 3.1** Sample Sites for Metals Analyses

| River | Metals Sample Sites |
|---|---|
| Willamette River | Willamette Park, Cartney, and Long Tom |
| McKenzie River | Hayden, Harvest, and Mohawk |
| Codorus Creek | Martin, USGS, and Oil Creek |
| Leaf River | New Augusta, Wingate, Tallahala, and Bogue |

### 3.2.3 *Safety Precautions*

Because the river banks may be steep and slippery, footwear with good traction should be worn. Anyone working alone should be particularly aware of safety issues. If requested, NABF will provide a PFD for use when collecting samples. If the sample collector feels unsafe for any reason (high water, etc.), personal safety should take priority and sampling should not be done. Watch for traffic when sampling from bridges and wear a reflective orange vest. In an emergency, dial 911 on the cell phone. Hospital directions should be on hand for non-emergency treatment.

### 3.2.4 *Water Quality Sampling Methods*

At each of the stations, the following procedure should be used.

1) Park the vehicle in a safe place out of the flow of traffic.

2) Record the date, time, weather conditions and other comments on the "NCASI LTRWS Water Quality Monitoring Field Data Sheet" (See Appendices A–D).

3) Assemble the following items to be carried to the river:

- Prelabeled 500-ml Nalgene bottle for the NABF sample
- Prelabeled 250-ml Nalgene bottle for WCRC nutrient sample
- Prelabeled 125-ml glass amber bottle for TOC and COD at NABF (sampled on the Leaf River and Codorus Creek only)
- Hach sensION1 pH meter or equivalent (measures both pH and temperature)
- Stainless-steel bucket with rope
- Clipboard, pencil, and data sheet

4) At sites where the samples are taken from bridges, sample on the upstream side of the bridge.

5) At the river access point, rinse the bucket in river water three times. Fill the bucket with river water and rinse the Nalgene sample bottles twice and then fill to overflowing. Do not rinse the 125-ml amber bottles, because they already contain preservative.

6) Fill the bucket again and measure the pH and temperature with the Hach sensION1 meter. Record all notes on the data sheet. If the pH readings are extremely low (below pH 6), the probe may be suspect, and the backup pH meter and manual thermometer should be used instead.

7) Return to the vehicle and make sure the data sheet is filled out. Place each bottle in its appropriate cooler (either for NABF or WCRC) with bubble wrap and ice packs.

8) Repeat at the next sampling station.

9) When taking samples for metals analyses during sampling in May and September, wear safety glasses and gloves when handling the sample bottles that contain either nitric or hydrochloric acids. Fill sample bottles to the top with no air space. Do not prerinse the bottles with river water since they already contain the acid preservative, and do not overfill the bottles.

10) If any questions arise about sampling, call 360–293–4748 (NABF).

### 3.2.5 *Shipping Water Quality Samples*

Place the data sheets in a plastic bag and place in the sample cooler. The cooler should be well packed with bubble wrap and ice packs. The FedEx form is filled out in advance at NABF, so only the weight needs to be filled out. Ship the samples priority overnight express via FedEx to:

NCASI
1219 Q Avenue
Anacortes, WA 98221
360–293–4748

The nutrient samples collected from the Willamette and McKenzie Rivers are hand delivered to WCRC for nutrient analyses. The nutrient samples collected on the Leaf River and Codorus Creek are shipped via FedEx Priority Overnight to:

NCASI
720 SW 4$^{th}$ Street
Corvallis, OR 97333
541–752–8801

Metals samples collected in May and September are shipped directly to CAS via FedEx priority overnight service. The cooler should contain the samples as described in the “Preparation” section as well as the chain of custody form. A chain of custody label should be affixed to the cooler. The FedEx label is filled out at in advance at NABF as follows:

Columbia Analytical Services
Jeff Christian
1317 South 13th Avenue
Kelso, WA 98626
360–577–7222

### 3.3 Water Quality Sample Sites: Codorus Creek

Codorus Creek water quality samples are collected from six bridge or shoreline locations (Table 3.2 and Figure 3.3). Figures 3.4 through 3.9 show upstream views taken from each water quality sampling location. Additional directions for reaching these locations are provided in Section 4.3.1.

**Table 3.2** Codorus Creek Water Quality Sample Locations and Descriptions

| N Latitude | W Longitude | Site Name and Location Description |
|---|---|---|
| 39°51’49” | 76°53’37” | **Oil Creek**<br>Located at the Highway 116 bridge. This is a supplemental station to address the potential influence this tributary stream may have on Codorus Creek water quality. |
| 39°51’45” | 76°53’23” | **Menges**<br>Bridge at Menges Mills on Colonial Valley Road above Spring Grove. |
| 39°52’43” | 76°51’13” | **USGS**<br>USGS check dam/gauge at the end of Hershey Road near Spring Grove (changed based on 2005 survey). |
| 39°53’14” | 76°50’10” | **Martin**<br>Bridge at Martin on Martin Road below Spring Grove. |
| 39°55’18” | 76°47’55” | **Graybill**<br>Bridge near Graybill on Highway 616. |
| 40°03’08” | 76°39’16” | **Furnace**<br>Bridge on Codorus Furnace Road below York. |

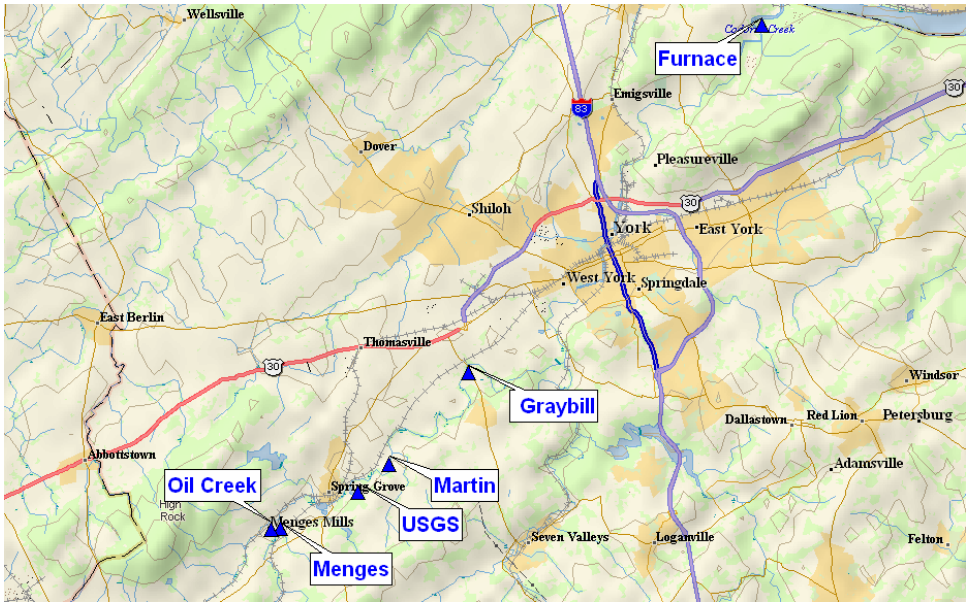


**Figure 3.3** Codorus Creek Water Quality Sites

**Figure 3.4** Oil Creek Water Quality Site

**Figure 3.5** Menges Mills Water Quality Site

**Figure 3.6** USGS Water Quality Site

**Figure 3.7** Martin Water Quality Site

**Figure 3.8** Graybill Water Quality Site

**Figure 3.9** Furnace Water Quality Site

### 3.4 Water Quality Sample Sites: Leaf River

Leaf River water quality samples are collected from seven bridge or boat launch locations (Table 3.3 and Figure 3.10). Figures 3.11 through 3.17 show downstream views taken from each water quality sampling location.

**Table 3.3** Leaf River Water Quality Sample Locations and Descriptions

| N Latitude | W Longitude | Site Name and Location Description |
|---|---|---|
| 31°13’57” | 89°05’03” | **Tallahala Creek**<br>Tallahala Creek Tributary sampled at the bridge on Old River Road. 0.6 river miles upstream of the confluence with the Leaf River Bridge on Old River Road near New Augusta. This is a supplemental station to address the potential influence this tributary stream may have on Leaf River water quality. |
| 31°13’39” | 89°05’09” | **Mahned**<br>Located at boat launch ramp north of the city of Mahned and 5.1 RM of the mill effluent discharge. Boat launch ramp is located on the north side of the river at the site of the old bridge crossing on Memorial CH Road east of Mahned and above New Augusta. |
| 31°13’18” | 89°03’12” | **New Augusta**<br>Located at the railroad bridge west of the city of New Augusta. 0.4 RM upstream of the mill effluent discharge. (Based on 2005 survey). |
| 31°13’34” | 88°59’28” | **Wingate**<br>Located at the bridge on Wingate Road. 4.8 RM below the mill effluent discharge. |
| 31°10’57” | 88°55’07” | **Beaumont**<br>Located at bridge on Highway 15 in Beaumont. 12.9 RM below the mill effluent discharge. |
| 31°07’38” | 88°49’02” | **McLain**<br>Located at bridge on River Road. East of the city of McLain. 24.6 RM below the mill effluent discharge. |
| 31˚15’43” | 89˚00’19” | **Bogue**<br>Tributary sampled at the bridge on Old Augusta Road. 3.2 RM upstream of the confluence with the Leaf River. This site was added in June 2003. |

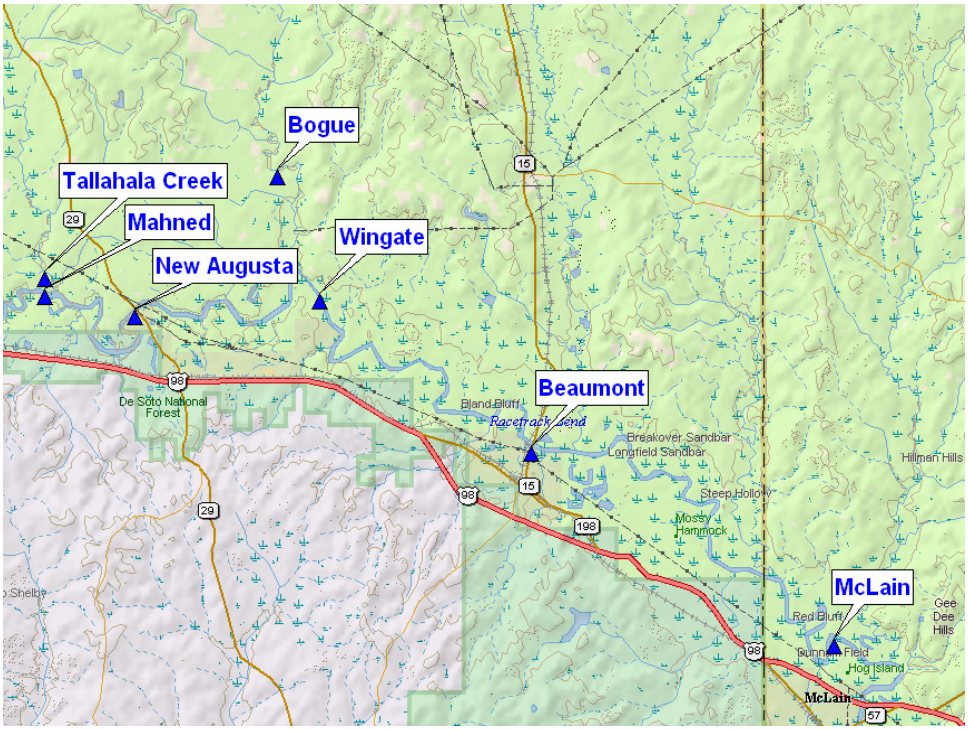


**Figure 3.10** Leaf River Water Quality Sites

**Figure 3.11** Tallahala Creek Water Quality Site

**Figure 3.12** Mahned Water Quality Site

**Figure 3.13** New Augusta Water Quality Site

**Figure 3.14** Bogue Water Quality Site

**Figure 3.15** Wingate Water Quality Site

**Figure 3.16** Beaumont Water Quality Site

**Figure 3.17** McLain Water Quality Site

### 3.5 Water Quality Sample Sites: McKenzie River

McKenzie River water quality samples are collected from six bridge or boat launch locations (Table 3.4 and Figure 3.18). Figures 3.19 through 3.24 show upstream views taken from each water quality sampling locations.

**Table 3.4** McKenzie River Water Quality Sample Locations and Descriptions

| N Latitude | W Longitude | Site Name and Location Description |
|---|---|---|
| 44°03’23” | 122°49’41” | **Hendricks Bridge**<br>Boat launch ramp at Hendricks Bridge State Wayside east of Springfield on Highway 126. 9.6 RM above the mill effluent discharge. (Based on 2005 survey.) |
| 44˚04’12” | 122˚54’21” | **Bellingers Landing**<br>Boat launch at Bellingers Landing located on Oak Point Road off of Camp Creek Road. 4.4 RM above the mill discharge. This site was added in April 2006. |
| 44°04’20” | 122°57’49” | **Hayden Boat Launch**<br>Hayden Bridge Landing (boat launch) just downstream of highway bridge on Marcola Road east of Springfield. 0.03 RM above the mill effluent discharge. |
| 44°05’37” | 122°57’20” | **Mohawk River**<br>Tributary sampled at Hill Road Bridge on Mohawk River north of Springfield (reached via Old Mohawk River Road). 1.6 RM upstream of the confluence with the McKenzie River and 3.1 RM upstream of Harvest Landing. This is a supplemental station to address the potential influence this tributary stream may have on McKenzie River water quality. |
| 44°04’43” | 122°59’54” | **Harvest Boat Launch**<br>Harvest Landing (boat launch) at end of Harvest Lane on NW edge of Springfield. 2.5 RM below the mill effluent discharge and 1.4 RM downstream of the confluence with the Mohawk River. |
| 44°06’44” | 123°02’47” | **Armitage Park**<br>Armitage State Park boat launch off of Coburg Road north of Eugene/Springfield. 7.2 RM below the mill effluent discharge. This site had previously been named Coburg. |

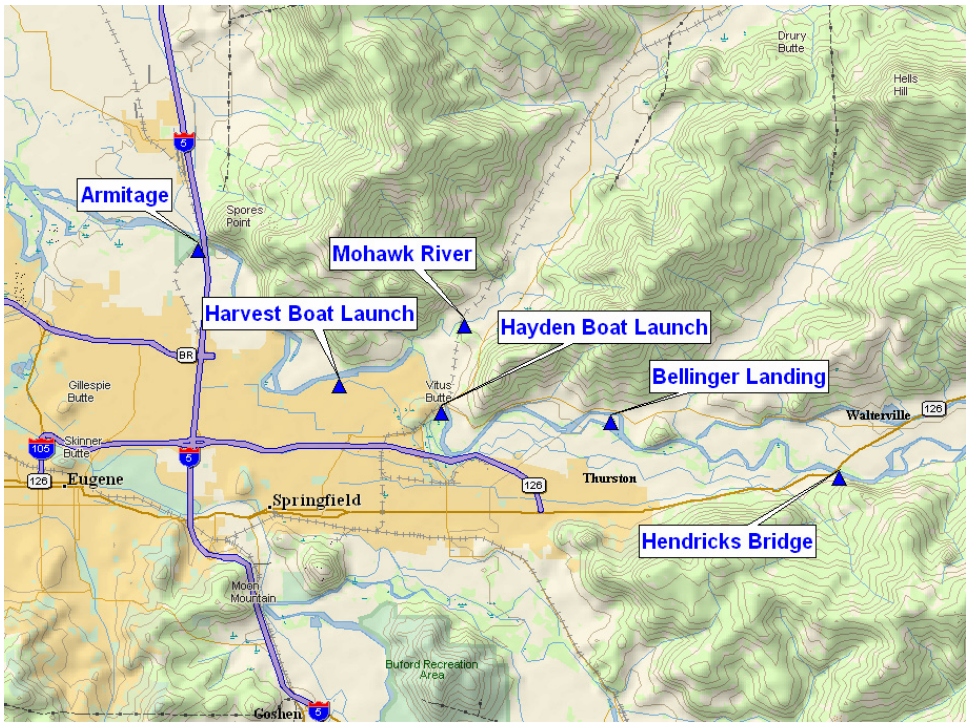


**Figure 3.18** McKenzie River Water Quality Sites

**Figure 3.19** Hendricks Water Quality Site

**Figure 3.20** Bellingers Water Quality Site

**Figure 3.21** Hayden Bridge Water Quality Site

**Figure 3.22** Mohawk Water Quality Site

**Figure 3.23** Harvest Water Quality Site

**Figure 3.24** Armitage Water Quality Site

### 3.6 Water Quality Sample Sites: Willamette River

Willamette River water quality samples are collected from six bridge or boat launch locations (Table 3.5 and Figure 3.25). Figures 3.26 through 3.30 show upstream views taken from each water quality sampling location.

**Table 3.5** Willamette River Water Quality Sample Locations and Descriptions

| N Latitude | W Longitude | Site Name and Location Description |
|---|---|---|
| 44°22’49” | 123°14’53” | **Long Tom**<br>Tributary sampled at Bundy Bridge which is accessed via Eureka/Bundy Road and Highway 99W south of Corvallis. 0.8 RM upstream of the confluence with the Willamette River and 1.4 RM above Intake. This is a supplemental station to address the potential influence this tributary stream may have on Willamette River water quality. |
| 44°16’23” | 123°10’21” | **Harrisburg**<br>Boat launch in Harrisburg reached by Highway 99E north of Eugene/Springfield. 13.4 RM above the mill effluent discharge. |
| 44°19’00” | 123°12’54” | **Cartney**<br>Located at McCartney Park boat launch at the end of Cartney Drive. Accessed via Highway 99E or Peoria Road north of Eugene/Springfield. 9.2 RM above the mill effluent discharge. |
| 44°27’20” | 123°12’32” | **Peoria**<br>Peoria Park boat launch just north of Peoria and accessed via Peoria Road/Highway 34 SE of Corvallis. 5.8 RM below the mill effluent discharge. |
| 44°26’42” | 123°12’11” | **New Peoria**<br>Located 0.8 RM upstream of the Peoria site. Alternate site to Peoria boat launch. Sampling was moved to this location from the Peoria boat launch, because the site became silted and was no longer in the main channel. New Peoria was sampled June 2004 to October 2005 but became inaccessible in 2006 due to changes brought on by winter high water. Sampling resumed at the Peoria boat launch because an alternative site could not be located. |
| 44°33’07” | 123°15’03” | **Willamette Park Launch**<br>Boat launch accessed via Crystal Lake Drive/Fisher Lane and Highway 99W just south of Corvallis. Located below the city of Corvallis and 15.1 RM below the mill effluent discharge. Site originally named “Corvallis/Fisher Lane”. |


**Figure 3.25** Willamette River Water Quality Sites

**Figure 3.26** Long Tom Water Quality Site

**Figure 3.27** Harrisburg Water Quality Site

**Figure 3.28** Cartney Water Quality Site

**Figure 3.29** Peoria Water Quality Site

**Figure 3.30** Willamette Park Water Quality Site

# 4.0 PERIPHYTON AND MACROINVERTEBRATE COMMUNITY ASSESSMENT

## 4.1 Overview

Periphyton and macroinvertebrate monitoring is carried out to assess whether there are differences upstream and downstream of pulp and paper mill effluent discharges. This evaluation includes long-term monitoring at multiple/upstream locations to allow for an assessment of natural seasonal and spatial variability.

Assessment endpoints for periphyton include chlorophyll *a* and taxonomic evaluation of the periphyton community. Periphyton populations are evaluated on natural riffle cobble substrates in wadeable stream sections for Codorus Creek, and the McKenzie and Willamette Rivers. On the Leaf River, periphyton is sampled from HD substrates for chlorophylls and taxonomy. Sand periphyton is also collected at sandbars on the Leaf River for taxonomic evaluation.

Assessment endpoints for macroinvertebrates include biomass and taxonomic evaluation. Macroinvertebrates are sampled from natural riffle cobble substrates on Codorus Creek and the McKenzie and Willamette Rivers and from HD substrates and woody debris in the Leaf River.

Sampling for macroinvertebrates and periphyton occurs together at the same sites. See Section 4.3 for site locations. Scientific collecting permits, which are renewed annually, are required for collecting macroinvertebrates on Codorus Creek and the Leaf River. These permits also include the collection of fish. The McKenzie and Willamette Rivers require collection permits for fish but not for macroinvertebrates. See Section 5.3 for details of the fish collection permits.

## 4.2 Sampling Time and Frequency

Periphyton and macroinvertebrate sampling was initiated on Codorus Creek and the McKenzie and Willamette Rivers in fall 1998 and continued on a quarterly basis for three years. Beginning in 2001, sampling was reduced to twice a year in the spring and fall. Beginning in 2007, sampling on Codorus Creek will be reduced to once a year in the spring. Sampling on the Leaf River was initiated in 2000 and occurs twice a year (spring and fall).

## 4.3 Sample Locations—Periphyton and Benthic Macroinvertebrates

### 4.3.1 *Codorus Creek*

Codorus Creek periphyton and benthic macroinvertebrate samples are collected from six sites (Table 4.1 and Figure 4.1) adjacent to bridge or highway crossings and can be accessed by car. These locations should generally be expected to remain stable over the duration of the study since Codorus Creek is a small stream with a stable streambed. Sample locations represent wadeable riffle areas, generally close to mid-channel. Figures 4.2 through 4.7 show upstream views from each sampling location. Specific sampling areas are generally indicated as being in either an upstream or downstream direction from the designated bridge crossing. The sampling area should be at least 50 m distant from the bridge, although this is not always possible. Location differences should be noted on the corresponding field data sheet (Appendix F).

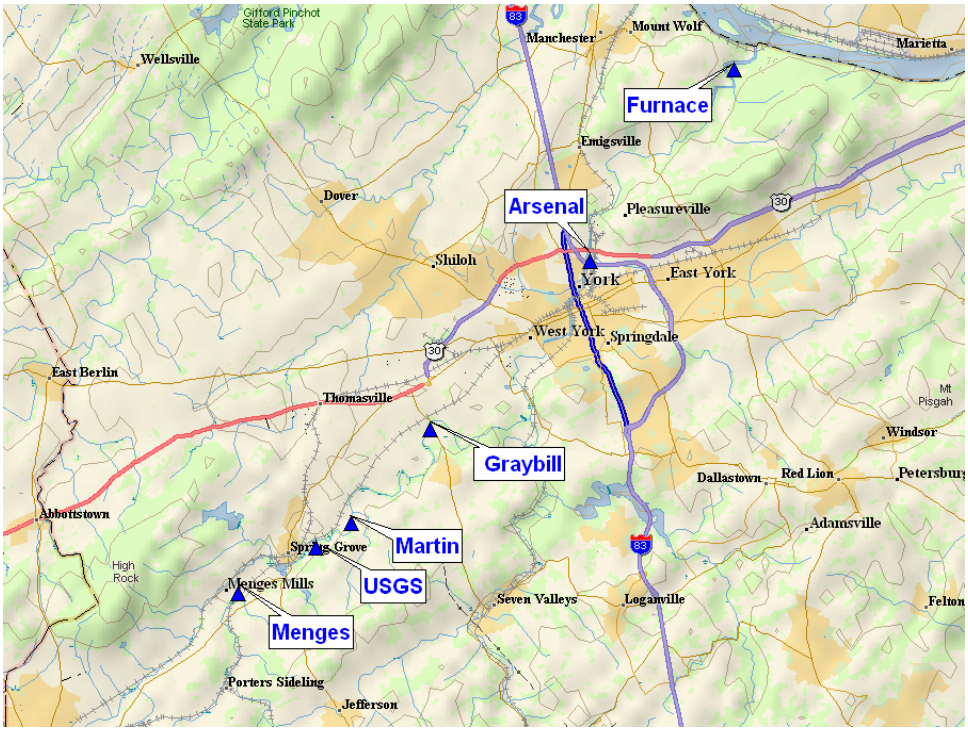


**Figure 4.1** Codorus Creek Periphyton and Benthic Macroinvertebrate Sampling Locations

**Table 4.1** Codorus Creek Locations for Macroinvertebrate and Periphyton Sampling

| N Latitude | W Longitude | Waypoint | Site Name and Location Description |
|---|---|---|---|
| 39°51'45" | 76°53'23" | MENG | **Menges**—Menges Mill<br>3.5 RM (5.6 km) above the effluent discharge. From the Old Forge Technical Center, turn left onto Highway 116 (Main Street). Turn right (west) onto Colonial Valley Rd. Follow the road until it crosses the creek and park on the left along the road. The site is on private property. Seek out the property owner for permission to sample on each trip. Sample station is upstream of the bridge. |
| 39°52'44" | 76°51'11" | USGS | **USGS**—USGS Gauging Station<br>0.4 RM (0.6 km) above the effluent discharge. From the Old Forge Technical Center turn right onto Highway 116 (Main Street) and turn right onto Rockery Road. Continue straight on Rockery Road after the stop sign. Turn left onto Hershey Rd., which is located just before the effluent secondary treatment system. There is a locked gate blocking the road, and Doug Brodhecker of Glatfelter has the key. Drive to the end of the road and park. Sample station is about 20 to 27 m below the USGS check-dam and gauge. |
| 39°53"16" | 76° 50'10" | MART | **Martin**—Martin Bridge<br>1.2 RM (1.9 km) downstream of the effluent discharge. From the Old Forge Technical Center, turn right onto Highway 116 (Main Street). At the stop light, turn right onto Sprenkle Road. Turn right at Martin Road and stop before crossing the bridge over the creek. Park in the field along the road. Sample station is approximately 23 m downstream of bridge in the 1st riffle. |

(Continued on next page)

**Table 4.1** Continued

| N Latitude | W Longitude | Waypoint | Site Name and Location Description |
|---|---|---|---|
| 39°55'18" | 76°47'56" | GRAY | **Graybill**—Graybill Bridge<br>6.0 RM (9.7 km) below the effluent discharge. From the Old Forge Technical Center, turn right onto Highway 116. Turn right at the light onto US 30 East. Turn right onto Highway 616 at the light. Just before the highway crosses the creek, turn right onto Graybill Rd and park on the left. Sample station is approximately 23 to 27 m upstream of the bridge. |
| 39°58'59" | 76°43'25" | ARSE | **Arsenal**—Arsenal Bridge just below York.<br>15.4 RM (24.8 km) below the effluent discharge. From Spring Grove on Highway 30, turn right (north) onto Toronita Street at the light. Take an immediate right onto San Carlos Street. Park in the parking lot behind "The Hop". The sampling station is upstream of the Highway 30 Bridge. It is necessary to cross the creek to get to the sampling area. If it is difficult to cross the creek due to high water, continue straight on Torinita and turn left on Arsenal Road (Highway 30) at the light. Continue across the bridge and make an immediate right onto Loucks Mill Road at the end of the bridge. Turn right and park in the area in front of a locked gate. This property is government property (U.S. Army Corp of Engineers). Leave a note on the car window explaining the reason for parking there. Walk down the bank to the creek. |
| 40°03'09" | 76°39'20" | FURN | **Furnace**—Furnace Bridge<br>24.1 (38.8 km) RM below the effluent discharge. From Highway 30 in York, turn N onto North Sherman Street at the stop light. (This would be a left turn coming from the direction of Spring Grove.) Stay on Sherman Street until the "T" intersection with Codorus Furnace Rd. Turn right and follow Codorus Furnace Rd. Immediately after it crosses the creek it will turn right. Park at the furnace located on the left. The sampling station is immediately across the road. The landowner (Michael Wolgemuth, 717–266–4573) has provided his permission to access and work on his land. |

**Figure 4.2** Menges Periphyton and Benthic Macroinvertebrate Sampling Site

**Figure 4.3** USGS Periphyton and Benthic Macroinvertebrate Sampling Site

**Figure 4.4** Martin Periphyton and Benthic Macroinvertebrate Sampling Site

**Figure 4.5** Graybill Periphyton and Benthic Macroinvertebrate Sampling Site

**Figure 4.6** Arsenal Periphyton and Benthic Macroinvertebrate Sampling Site

**Figure 4.7** Furnace Periphyton and Benthic Macroinvertebrate Sampling Site

### 4.3.2 *Leaf River*

Leaf River periphyton and benthic macroinvertebrate samples are collected from six sites which are reached by boat. Sampling sites consist of two types: pool locations used for the placement of HD samplers (Table 4.2), and beach sandbars (Table 4.3) where sand is collected for sand periphyton identification. GPS waypoint should be used to verify each sample location. Pool and sandbar locations are in close proximity to one another (Figure 4.8). The exact sample location may change depending on river level. Location differences should be noted on the corresponding field data sheet (Appendix H). Figures 4.9 through 4.14 show upstream views from each sandbar sampling location.

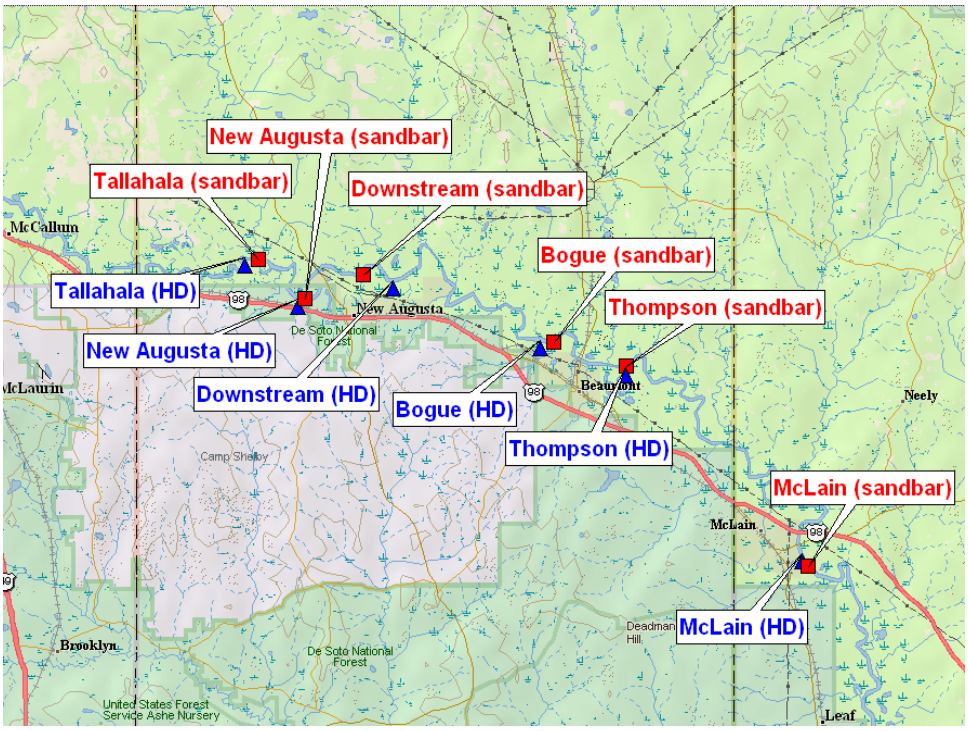


**Figure 4.8** Leaf River Periphyton and Benthic Macroinvertebrate Sampling Locations

**Table 4.2** Leaf River Pool Locations for Hester-Dendy Plates

| N Latitude | W Longitude | Waypoint | Site Name and Location Description |
|---|---|---|---|
| 31°13’38.7” | 89°05’26.3’ | TALLHD | **Tallahala**—Tallahala Creek confluence ~5.5 RM (8.9 km) above discharge, upstream of Tallahala Creek. HD samplers placed in 2 m deep pool on the right bank side facing upstream. |
| 31°12’33.8” | 89°03’48.6” | NAUGHD | **New Augusta**—Near New Augusta Bridge ~2.4 RM (3.9 km) above discharge between New Augusta Bridge and G.P. Railroad Bridge. HD samplers placed in 3 m deep pool on the left bank side facing upstream. |
| 31°13’02.7” | 89°00’53.0” | DOWNHD | **Downstream**—Downstream of mill discharge ~2.5 RM (4.0 km) below discharge. HD samplers placed in 2 m deep pool on the left bank side facing upstream. |
| 31°11’29.0 | 88°56’22.4” | BOGHD | **Bogue**—Bogue Homo Creek confluence ~10.7 RM (17.2 km) below discharge, below Bogue Homo Cr. HD samplers placed in 2 m deep pool on the left bank side facing upstream. |
| 31°10’46.0” | 88°53’45.8” | THOMHD | **Thompson**—Thompson Creek confluence ~14.3 RM (23 km) below discharge, downstream of Thompson Creek. HD samplers placed in 3 m deep pool on the left bank side facing upstream. |
| 31°05’55.3” | 88°48’22.9” | MCLAHD | **McLain**—McLain Bridge ~28 RM (45 km) below discharge, downstream of McLain Bridge. HD samplers placed in 2 m deep pool on the left bank side facing upstream. |

**Table 4.3** Leaf River Shore/Sandbar Locations for Periphyton Sand Samples and Natural Substrate Macroinvertebrates

| N Latitude | W Longitude | Waypoint | Site Name and Location Description |
|---|---|---|---|
| 31°13'37.9" | 89°05'30.7" | TALLBP | **Tallahala—**Tallahala Creek confluence ~0.8 RM (1.3 km) above the Tallahala Creek confluence and ~5.5 RM (8.9 km) above the mill discharge. This location is also ~ ¼ mile above location of the old Mahned highway bridge. Sampling sandbar is on the left bank facing upstream. |
| 31°12'33.8" | 89°03'33.9" | NAUGBP | **New Augusta**—Near New Augusta Bridge ~2.2 RM (3.5 km) above the discharge. Upstream of the New Augusta Bridge and ~ ½ mile below the railroad bridge. Sampling sandbar is on the right bank facing upstream. |
| 31°13'10.5" | 89°01'47.8" | DOWNBP | **Downstream**—Downstream of mill discharge ~1.1 RM (1.8 km) below discharge and~ ½ mile above confluence with Gum Branch Creek. Sampling sandbar is on the right bank facing upstream. |
| 31°11'25.8" | 88°55'57.3" | BOGBP | **Bogue**—Below confluence of Bogue Homo Creek Downstream of Bogue Homo Creek confluence and ~11.2 RM (18 km) below mill discharge. Sampling sandbar is on the right bank facing upstream at Racetrack Bend, ~2 miles above Beaumont highway bridge. |
| 31°10'48.0" | 88°53'45.1" | THOMBP | **Thompson**—Below Thompson Creek Confluence About ½ mile below Thompson Creek confluence and ~14.3 RM (23 km) below mill discharge. Sample location is on the right bank facing upstream at Longfield Sandbar. |
| 31°05'35.4" | 88°48'10.5" | MCLABP | **McLain**—Below McLain Bridge ~28.6 RM (46 km) below mill discharge, ~ 4.1 miles (6.6 km) downstream of the McLain Bridge. Sample location is on the right bank facing upstream. |

Note: Based on October 2001 survey

**Figure 4.9** Tallahala Sampling Site

**Figure 4.10** New Augusta Sampling Site

**Figure 4.11** Downstream Sampling Site

**Figure 4.12** Bogue Sampling Site

**Figure 4.13** Thompson Sampling Site

**Figure 4.14** McLain Sampling Site

### 4.3.3 *McKenzie River*

McKenzie River periphyton and benthic macroinvertebrate samples are collected from five wadeable riffle sites near shoreline and are accessed by boat (Figure 4.15 and Table 4.4). These indicated locations represent electronic GPS "pins" from which the general sample area is located. The actual area from which samples are obtained will differ according to river level and will also change somewhat over time due to the gradual shifting of the riffle channel. Distances and orientation from the GPS "pin" to the sample location should be indicated on the corresponding field data sheet (Appendix F). Figures 4.16 through 4.20 show upstream views from each sampling location.

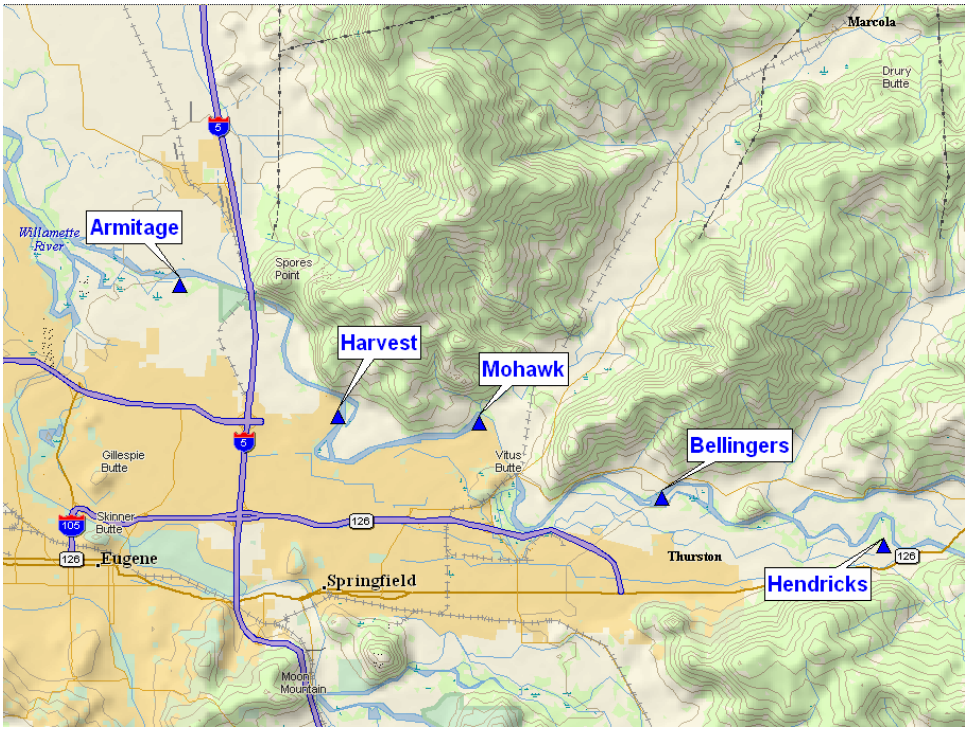


**Figure 4.15** McKenzie River Periphyton and Benthic Macroinvertebrate Sampling Locations

**Table 4.4** McKenzie River Sample Site Locations for Periphyton and Macroinvertebrates

| N Latitude | W Longitude | Waypoint | Site Name and Location Description |
|---|---|---|---|
| 44°03’29.9” | 122°50’54.9” | HEND2 | **Hendricks—**Hendricks Bridge<br>Approximately 1.0 RM (1.6 km) downstream of Hendricks Bridge Park. Located on the left bank facing upstream. 8.6 RM (13.8 km) upstream of the mill effluent discharge. Replaced HEND1 and Walterville in May 2002. This site is only accessible by drift boats. |
| 44°03’55.7” | 122°54’15.5” | BELL4 | **Bellingers**—Bellingers Landing<br>South bank gravel beach above Bellingers Landing Boat Launch (right bank facing upstream). Approximately 0.4 RM upstream of Bellingers Landing (boat launch). 4.6 RM (7.4 km) above the mill effluent discharge. The Bellingers site has been located at four different gravel bars (all less than 0.9 RM of each other). The sampling site has been relocated due to accessibility difficulties or changes at the gravel bars (i.e., decreasing size of gravel bar, poor flow, increase in water depth). Sampling at BELL4 began in May 2007. |
| 44°05’08.9” | 122°58’25.3” | MOHA1 | **Mohawk**—Mohawk River confluence<br>Located just upstream of the confluence with the Mohawk River on the right bank facing upstream. 1.1 RM (1.8 km) below the mill effluent discharge. |
| 44°05’13.6” | 123°01’03.1” | HARV1 | **Harvest**—Harvest Lane<br>Located on the right bank facing upstream on a gravel beach 2.2 RM (3.5 km) below Harvest Lane. 4.7 RM (7.6 km) below the mill effluent discharge. |
| 44°06’59.2” | 123°04’00.0” | ARMI2 | **Armitage**—Armitage Park<br>South bank gravel bar downstream of Armitage Park. Located on the right bank facing upstream. 1.1 RM (1.8 km) below Armitage Park. 8.3 RM (13.4 km) below the mill effluent discharge. It is 0.4 RM downstream of ARMI1 which it replaced as of August 1999. |

**Figure 4.16** Hendricks Periphyton and Benthic Macroinvertebrate Sampling Site

**Figure 4.17** Bellingers Periphyton and Benthic Macroinvertebrate Sampling Site

**Figure 4.18** Mohawk Periphyton and Benthic Macroinvertebrate Sampling Site

**Figure 4.19** Harvest Periphyton and Benthic Macroinvertebrate Sampling Site

**Figure 4.20** Armitage Periphyton and Benthic Macroinvertebrate Sampling Site

### 4.3.4 *Willamette River*

Willamette River macroinvertebrate and periphyton stations are wadeable riffle areas near the shoreline and are accessed via boat (Table 4.5 and Figure 4.21). These indicated locations represent electronic GPS "pins" from which the general sample area is located. The actual area from which samples are obtained will differ according to river level and will also change somewhat over time due to the gradual shifting of the riffle channel. Distances and orientation from the GPS "pin" to the sample location should be indicated on the corresponding field data sheet. Figures 4.22 through 4.27 show upstream views from each sampling location.

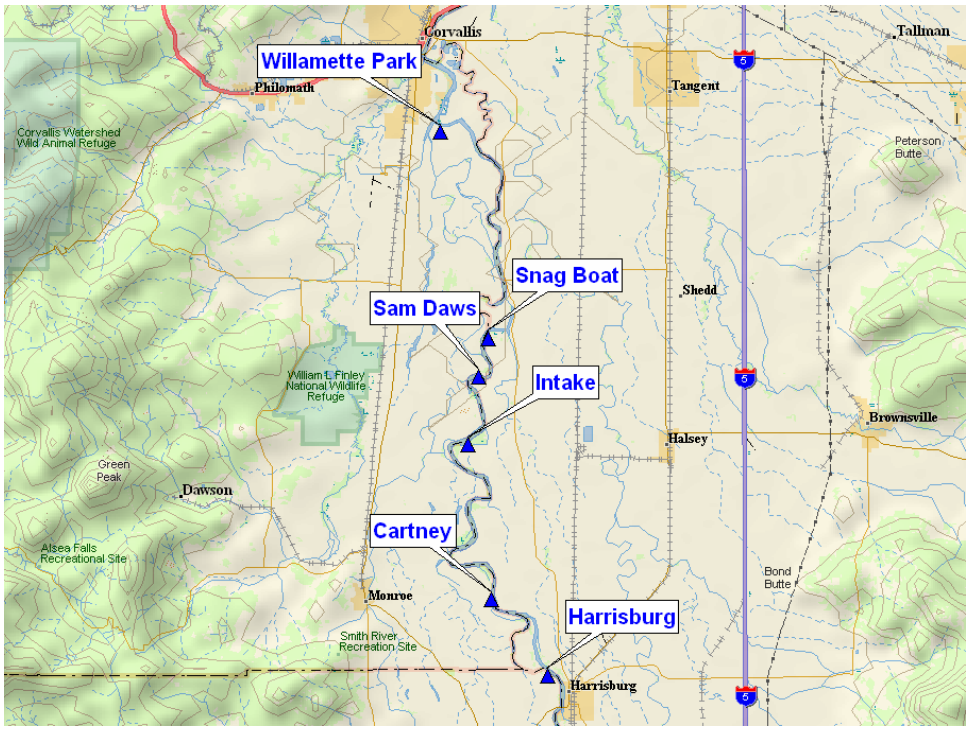


**Figure 4.21** Willamette River Periphyton and Benthic Macroinvertebrate Sampling Locations

**Table 4.5** Willamette River Sample Site Locations for Periphyton and Macroinvertebrates

| N Latitude | W Longitude | Waypoint | Site Name and Location Description |
|---|---|---|---|
| 44°17’03.4” | 123°10’59.2” | HARR1 | **Harrisburg**—Harrisburg<br>Below the city of Harrisburg on the right bank facing upstream. 12.4 RM (20 km) above the mill effluent discharge. |
| 44°19’06.3” | 123°13’03.6” | CART1 | **Cartney**—Cartney Park<br>Across channel from McCartney Boat Launch on right bank facing upstream. 9.0 RM (14.5 km) above the mill effluent discharge. |
| 44°23’14.7” | 123°13’56.9” | INTAK1 | **Intake**—Mill water intake<br>Above mill water intake/outfall on the right bank facing upstream. 0.6 RM (1 km) above the mill effluent discharge. |
| 44°25”03.7” | 123°13’33.7” | SAM2 | **Sam Daws**—Sam Daws Bend<br>Below the mixing zone on the left bank facing upstream. 1.8 RM below the mill effluent discharge. |
| 44°26’05.1” | 123°13’11.1” | SNAG2 | **Snag Boat—**Snag Boat Bend<br>Located on the right bank facing upstream. 3.7 RM (6.0 km) below the mill effluent discharge. |
| 44°31’35.8” | 123°14’58.8” | WILL2 | **Willamette Park—**Willamette Park<br>Located on the left bank facing upstream. 12.5 RM (20.1 km) below the mill effluent discharge. |

**Figure 4.22** Harrisburg Periphyton and Benthic Macroinvertebrate Sampling Site

**Figure 4.23** Cartney Periphyton and Benthic Macroinvertebrate Sampling Site

**Figure 4.24** Intake Periphyton and Benthic Macroinvertebrate Sampling Site

**Figure 4.25** Sam Daws Periphyton and Benthic Macroinvertebrate Sampling Site

**Figure 4.26** Snag Boat Periphyton and Benthic Macroinvertebrate Sampling Site

**Figure 4.27** Willamette Park Periphyton and Benthic Macroinvertebrate Sampling Site

## 4.4 Field Data Sheets—Codorus Creek, McKenzie/Willamette Rivers

Periphyton and macroinvertebrate sampling occur at the same time, and consequently, they share in many aspects of site characterization. Field data sheets should be completed for each sample location and sample date. There are two field data sheets (Appendices F and G) for periphyton and macroinvertebrates described in Sections 4.4.1 and 4.4.2.

### 4.4.1 *Periphyton and Macroinvertebrate Sampling Data Sheet*

**Site Information**

**River**: Name of the river (e.g., Codorus Creek, McKenzie River, Willamette River)

**Site Name**: Site name as provided in Section 4

**Date/Time**: Date and time of sample collection

**Latitude and Longitude of Sample Location:** Coordinates of sample location as determined with a Garmin GPS 12, 12 channel GPS (or equivalent). Data entry should be made in the degrees ($^{o}$), minutes (‘), seconds (“) format with either N latitude or W longitude indicated for the appropriate coordinate. GPS coordinates are not required for Codorus Creek.

**Sampling Team**: Initials of sample team members

**Bank Width**: The distance (m) from the water edge at the sample area to the water edge on the opposite stream bank as measured by the Bushnell Yardage Pro 1000 range finder (or equivalent optical range finder) or a measuring tape. The range finder should be used in the “standard” targeting mode with an expected manufacturer’s based precision of +/- 1 m. The range finder cannot measure distances <15 m.

**Position of sample from “pin”:** Distance to the middle of the 20 m run from the GPS pin and upstream/downstream direction relative to the pin as measured by the Bushnell Yardage Pro 1000, or equivalent optical range finder. This is not required for Codorus Creek.

**Site Observations**

**Sky**: Sky conditions (clear sky, partly cloudy, or overcast) during sampling

**Wind**: Estimated wind speed (mph) and direction (e.g., N, NW) during sampling using a Davis Instruments “TurboMeter” anemometer (or equivalent)

**Precipitation**: Designate the form of precipitation encountered during sampling. Drizzle is differentiated from rain by the lack of drop patterns on the water surface.

**Water Clarity and Aesthetics**: Guidelines for recording water conditions are as follows:

| | |
|---|---|
| Clear | The stream bottom is easily viewed without any obvious cloudiness or turbidity. |
| Turbidity | The streambed is obscured due to cloudiness or particulate matter in the water. |
| Color | The water has no obvious apparent coloration (None) or is tinged with green or brown. |
| Foam | Foam or froth appears on the water surface, along the shoreline, or on objects in the water. |
| Odor | The presence of musty effluent or other types of odor. |

**Riparian Shading**: Record whether the sample location, at the time of sampling, is fully exposed to sunlight (Exposed), partially shaded (Partial Shade), or completely shaded (Shaded).

**Conductivity**: Conductivity measurements are made with a Hach sensION5 (or equivalent) conductivity meter and probe. Measurements should be taken at a location considered representative of the general conditions at the site (e.g., current velocity and water depth). Conductivity measurements should be recorded as µS/cm. The Hach conductivity meter and backup meter (Cole Parmer CON 5 or equivalent) should be checked and calibrated at NABF prior to use in the field. See the lab manual for calibration instructions. Reported accuracy of the conductivity meter is $\pm$ 4 µS/cm.

**Temperature**: Temperature is also measured with the Hach sensION5 at the same time as conductivity measurements are taken. The meter should be checked at the NABF laboratory with a NIST traceable mercury thermometer prior to use in the field. Readings on the meter should be within 0.5˚C of the mercury thermometer readings.

**Turbidity:** Turbidity is measured with the Hach 2100P Turbidimeter. Check the turbidity meter with the Hach StablCal 20 NTU standard at the NABF lab before and after the sampling trip. See the lab manual for instructions.

**Light**: Light measurement should be measured with a Li-Cor Model LI-250 light meter with an underwater quantum sensor probe (Figure 4.28). Measurements should be recorded as photosynthetically active radiation (PAR) in units of µmol m $s^{-1}$ $m^{-2}$. The correct calibration multiplier should be entered into the light meter prior to taking light readings. The current multipliers (210.08 for air and 277.31 for underwater) are based on the November 2006 calibration and will change with future calibrations. The probe should be returned to Li-Cor every two years for recalibration. Surface measurements should be made with the dry probe surface held just above the water surface and underwater measurements made with the probe surface 15 cm below the water surface. In each case the probe should be oriented skyward in a position perpendicular to the earth's surface. In order to reduce measurement variability, light measurements should be taken only at stations where sky conditions are either completely clear or completely overcast. Light measurements should be taken in an area representative of the sample area used for biological samples.

**Figure 4.28** Taking Light Measurements on Codorus Creek

**Periphyton Samples**

**Quantitative** (chlorophylls): Take length (mm) and width (mm) measurements of each of five rocks along with the total volume and filtered slurry volume for each rock. When extra subsamples are taken at a site for QA/QC, identify the filtered volumes of the subsamples as either "a" or "b".

**Qualitative** (taxonomy): 20 ml of slurry from each rock for a total of 100 ml per station

**Observations**

**Periphyton**: (filamentous beds, sloughing algae, bleached algae, macrophytes, moss etc.)

**General**: (Livestock, fish mortalities, bank erosion, insect hatches, surface sheen, etc.)

### 4.4.2 *Benthic Macroinvertebrate Sampling and Current Velocity Measurement Data Sheet*

**River**: Name of the river

**Site Name**: Site name

**Date/Time**: Date and time of sample collection

**Sampling Team**: Initials of sample team members

**Sampling Gear and Mesh Size:** Hess

**Sample time:** 2 to 3 min processing time for sampling with the Hess (note if different)

**Five replicates:** Note if number of replicates is different from standard of five

**Substrate Characteristics %**: Record the relative contribution at the surface of the streambed of silt, sand, gravel, cobble, boulder, and bedrock. Subjective guidelines for defining substrate types are as follows:

| | |
|---|---|
| Silt | Very fine material (0.004–0.06 mm diam) has a "greasy" feel when rubbed between fingers |
| Sand | Material (0.06–2.0 mm diam) has a gritty texture when rubbed between fingers |
| Gravel | Mixture of rounded course material from 2 to 64 mm d |
| Cobble | Stones from 64 to 256 mm d |
| Boulder | Rounded stones over 256 mm d or large slabs more than 256 mm in length |
| Bedrock | Solid rock forming a continuous surface |

**Current Velocity**: Velocity is measured with the Marsh-McBirney Model 2000 Flow-mate flow meter within a meter of each replicate and at a depth where the sample was taken. See Section 4.14.4 for details.

**General Comments/Observations**: Large or rare taxa or other observations

## 4.5 Field Data Sheets—Leaf River

The field data sheets for the Leaf River differ from the other rivers in that both shallow and deep-water habitat must be characterized to describe conditions from which sand samples are collected for sand periphyton identification and from which the HD samplers are placed. Most physical habitat and site characteristic measurements for the Leaf River follow the same guidelines as described for completing field records for the other LTRWS locations. Additional field observations for the Leaf River include recording the coordinates of the pool used for placement of the HD samplers, as well as pool depth and current velocity at the depth of sampler placement. There are two field data sheets (Appendix H) for periphyton and macroinvertebrates as described in Sections 4.5.1 and 4.5.2. The first page is based on measurements and observations at sandbar locations where natural periphyton from sand is sampled for taxonomic identification. The second page refers to measurements taken at the pools where the HD samplers are deployed for macroinvertebrates and periphyton sampling. The second page also contains information regarding the macroinvertebrate natural substrate samples.

### 4.5.1 *Leaf River Periphyton and Macroinvertebrate Sampling Data Sheet (Sandbar)*

**Site Information**

**River**: Leaf River

**Site Name**: (Tallahala, New Augusta, Downstream, Bogue, Thompson, McLain)

**Date/Time**: Date and time of sample collection

**Sampling Team**: Initials of sample team members

**Latitude and Longitude of sample location:** GPS reading at site of sand periphyton collection

**Bank Width**: The distance (m) from the water edge straight out from the GPS pin position to the water edge on the opposite stream bank as measured by the Bushnell Yardage Pro 1000 range finder

**Site Observations (See Section 4.4.1 for details)**

**Sky**:

**Wind**:

**Precipitation**:

**Water Clarity and Aesthetics**:

**Substrate Characteristics:**

**Conductivity**:

**Temperature**:

**Turbidity:**

**Current Velocity:** Velocity is measured within a meter of the vicinity and at the depth where sand periphyton samples are taken for taxonomic identification**.**

**Position of sample location from "pin":**

**Light**:

**Sand Periphyton Samples**

Qualitative (taxonomy) sampling consists of three scoops of sand composited into one sample and preserved with Lugol's Solution.

**Observations**

Periphyton: (filamentous beds, sloughing algae, bleached algae, macrophytes, moss, etc.)
General: (Livestock, fish mortalities, bank erosion, insect hatches, surface sheen, etc.)

### 4.5.2 *Leaf River Periphyton and Macroinvertebrate Sampling Data Sheet (Pool) Page Two*

**Site Information** (based on HD sampler locations)

**River**: Leaf River

**Site Name**: Site name (Tallahala, New Augusta, Downstream, Bogue, Thompson, McLain)

**Date/Time**: Date and time of sample collection

**Sampling Team**: Initials of sample team members

**Macroinvertebrate Hester-Dendy Samples**

**Latitude and Longitude**: GPS location of pool where HD samplers deployed

**Conductivity and Temperature:** Conductivity and temperature measurements are made with Hach sensION5 (or equivalent) conductivity meter and probe which are calibrated at NABF (see lab manual).

**Turbidity:** Turbidity is measured with the Hach 2100P Turbidimeter.

**Pool Depth (cm**): Depth of the pool where HD samplers deployed as measured with the depth finder (Speedtech Instrument or equivalent)

**Current Velocity (m/s):** Current velocity as measured with the Marsh-McBirney Model 2000 Flow-Mate portable flowmeter at the depth of the upper HD sampler plate

**Date of HD deployment, number deployed and number harvested:**

**Macroinvertebrate-Natural Substrate Sample**

**Latitude and Longitude of location sample location:** GPS location if different from native sand sample location

**Description of substrate sampled (log, stick, branch-approximate size)**

**Time (< 10 min):** Time taken to collect macroinvertebrates from substrate if <10 min

**Periphyton Samples from Hester-Dendy Plates**

Quantitative **(chlorophylls)**: Total and filtered volume of slurry from each top HD sampler plate (3 replicates). Subsamples are designated as "a" or "b" at one site chosen for QA/QC.

Qualitative **(taxonomy):** 20 ml aliquot of slurry from each top HD sampler plate (3) composited into one sample

### 4.6 Photographic Record

A photographic record should be made of each macroinvertebrate and periphyton gravel or sandbar sample location for each sample date. Photographs should be taken with a digital camera (Olympus Stylus 410 or equivalent). See the camera manual for instructions. The date and time should be set (M–D–Y), so this information will be included with the downloaded picture.

Three photographs should be taken for each sample location: one directly upstream, one from the shore in a perpendicular direction toward the opposite stream bank, and one directly downstream. It is helpful to take the photos in this order, so the first picture at each site is always the upstream view and the third picture is the downstream view. For Codorus Creek, photographs should be taken in the immediate biological sample area. For the McKenzie, Willamette, and Leaf Rivers, photographs should be taken from the GPS "pin" position. Other pictures of interest can also be taken: field sampling methods, changes in habitat, fish photos, etc.

### 4.7 Temperature Logger Deployment

Temperature data loggers were deployed at each sample location through 2005. They are no longer deployed on the Willamette, McKenzie, or the Leaf River because loggers deployed in the spring were generally found out of water at retrieval in the fall, or lost due to high water. Temperature loggers (Stowaway Tidbit) are deployed at all Codorus Creek macroinvertebrate/periphyton sites with the exception of Arsenal, which lacks a location for secure deployment. See the NABF lab manual for data logger initiation and download instructions.

The temperature loggers continuously collect data at the sites and are usually attached to root wads with plastic zip ties. Labels indicating "stream study" with the NCASI contact phone number should be attached by zip tie to each logger to discourage vandalism. On each sampling trip, the loggers collecting data are exchanged with newly initiated loggers during the algae/macroinvertebrate sample collection. The date and time of deployments as well as latitude, longitude, GPS waypoint, and descriptions of the locations are recorded on the "Codorus Creek Temperature Logger Location Data Sheet" (Appendix I). The data sheet from the previous sampling trip should also be taken on each trip in order to locate the data loggers.

### 4.8 General Field Equipment List for Periphyton and Macroinvertebrate Collection

The following is a list of equipment used during periphyton and macroinvertebrate collection. Equipment specific to macroinvertebrate or periphyton sampling is described in Section 4.9.1 and Section 4.9.2.

Batteries: AAA, AA, C, D, 9V, 357 silver oxide watch batteries

Biomonitoring methods

Camera (digital, Olympus Stylus 410 or equivalent), rechargeable batteries, charger, memory card

Cell phone

Conductivity meter (Hach sensION5 or equivalent) (2)

Depth finder (Speedtech Instrument or equivalent) (Leaf River only)

Field Safety Manual (Part 3 from NABF Chemical Hygiene and Safety Plan)

First aid kit (including CPR barrier)

Flow meter (Marsh-McBirney Model 2000 Flow-mate) with pole and stand

GPS (2)

Light meter-LiCor (probe, meter, attachment pole)

Measuring tape (reel, meter)

Multi-tool

MSDS for 10% neutral buffered formalin and Lugol's solution

PFDs

Range finder (Bushnell Yardage Pro 1000 or equivalent)

Scientific collecting permit for specific river

Turbidity meter (Hach 2100P Turbidimeter)

Waders (including backup)

Wading boots

Weights with flagging and floats for marking sites

Windmeter (Davis Instrument "Turbometer")

### 4.9 Periphyton Sample Collection—Natural Riffle Substrate (Codorus, McKenzie, Willamette Rivers)

Periphyton samples are collected from natural river cobble on Codorus Creek and the McKenzie and Willamette Rivers. Sample locations for natural substrate periphyton samples should represent water depths from 20 to 40 cm. All samples should be collected within a 20 m (or less) stretch of streambed with all wadeable habitat types represented in general proportion to their distribution. All sampling should take place in a progression from downstream to upstream. Best professional judgment should

be used to avoid locations within the 20 m area that clearly represent extreme or unusual conditions (e.g., areas of unusual depth, current velocity, or substrate type). The 20 m stretch can be measured either with the range finder or meter measuring tape.

Five specific locations within the 20 m sample area should be marked at the initiation of sampling with weighted colored markers with attached colored streamers and floats. These markers should be broadly distributed within the 20 m area, but individual markers should be relocated if water depth exceeds 40 cm or if unusual conditions (e.g., current velocity, substrate type) are represented. Depth and current velocity measurements, as well as macroinvertebrate and periphyton samples, should be taken within 1 m of the markers.

Since periphyton and macroinvertebrate sampling occur simultaneously, the sampling substrates should not be disturbed by different activities of the field staff. The algae field personnel measure conductivity, temperature, turbidity, and light at each site, while the macroinvertebrate field personnel measure current velocity and depth.

### 4.9.1 *Periphyton Quantitative Sampling Methods (Chlorophylls)*

Five "fist-sized" rocks should be randomly selected within the 20 m sample area and within 1 m of the markers. Although the primary objective is to collect random rocks, effort should be made to assure that these rocks are also representative of the general sample area. Extremes should be avoided, including rocks that are in unusual depth or current velocity positions within the sample area or rocks that have unusually dense or sparse periphyton growth.

*Equipment*

Aluminum foil squares (5 per site, extra 5 for each QA/QC site)

Bottles, screw cap, 1-pt Nalgene to mix periphyton samples (2)

Caliper for measuring rocks (waterproof, digital) (2)

Clipboard

Cooler for dry ice

Container, plastic with screw cap lid (1-gal) for tap water

Data sheets on *Rite in the Rain* ("Periphyton and Macroinvertebrate Sampling Data Sheet", 1 per site)

Dry ice

Filter forceps

Filter apparatus (filter holder and 1-L vacuum flask) (2 sets)

Filters (glass fiber, 1.0 µm particle retention, 47-mm) (5 per site, extra 5 for each QA/QC site)

Graduated cylinders, plastic (25-ml, 50-ml, 100-ml, 250-ml) (2 each)

Hand pump (2)

Labels (waterproof, prelabeled, 5 per site, 10 for each QA/QC site)

Markers (waterproof Sharpies), mechanical pencils, lead refills

Plastic bags (resealable, 1-gal) for foil-wrapped chlorophylls, labeled with river name and parameter (periphyton chlorophylls) (1 per river)

Scissors (2)

Squirt bottles for tap water (1-L) (2)

Storage containers (2-qt plastic) to scrape and rinse algae into (2)

Tap water

Tape (clear, tear by hand) (2 rolls)

Toothbrushes and penknives for scraping algae (2 each)

*Sampling Method*

1) Collect the first rock (replicate 1) within 1 m of the most downstream marker. Place the rock into a shallow, small plastic container and immediately transport to the streamside for processing. Periphyton should be removed from the entire rock surface by gentle scraping with a knife blade combined with brushing with a toothbrush (Figure 4.29). If there are long strands of algae on the rocks, be sure to include all of the strands in the sample. Use a pair of scissors to clip the strands of algae into small pieces, so the sample is as homogeneous as possible. Detached algae are rinsed from the rock into the container, using a squirt bottle of tap water. Care should be taken to use sufficient water to thoroughly rinse material from the rock without generating an excess volume of periphyton slurry. Work quickly to minimize the exposure of the sample to sunlight.


**Figure 4.29** Periphyton Sampling at Martin, Codorus Creek

2) Once the substrate is cleaned of periphyton, the material should be rinsed into a graduated cylinder (using the graduated cylinder whose size most closely matches the sample volume). Record the total volume on the data sheet. Transfer the slurry back and forth between the graduated cylinder and a screw cap plastic jar to ensure that the sample is mixed and that all

is transferred to the jar. The jar should then be shaken and 10 to 15 ml of the well-mixed slurry poured into a 25-ml graduated cylinder. Set aside the remaining slurry for the periphyton identification sample which is described in Section 4.9.2. This slurry should be processed for periphyton identification before sampling the next rock.

3) Filter 10 to 15 ml aliquots of the periphyton slurry with the goal to filter as much of the slurry as possible but avoiding filter clogging (Figure 4.30). Filtering involves the use of a vacuum flask, hand vacuum pump, and filter apparatus. Filters used for chlorophyll samples are 47-mm glass fiber filters (1.0 µm particle retention). If the initial aliquot of 10 to 15 ml filters quickly, mix the remaining slurry again and measure out and filter additional slurry in order to ensure the chlorophylls are within detection limits. Record the volume of filtered slurry on the data sheet.

**Figure 4.30** Filtering Periphyton Samples at Furnace, Codorus Creek

4) After filtration, fold the filter into quarters using the forceps. Place the filter on a square of foil, and fold the foil around the filter. Place a waterproof label onto the foil-wrapped sample indicating sample type (periphyton chlorophylls), river, sample site, replicate number, date, total and filtered volumes, and initials of the sampler. The label should be prelabeled at NABF with all information except the date, the total and filtered volumes, and initials of the sampler. Place clear tape over the label to protect the writing.

5) Place the foil-wrapped filters in a plastic bag labeled as “Periphyton Chlorophylls” with the name of the river and immediately freeze by placing in a cooler with dry ice. See Section 4.12 for sources of dry ice.

6) As part of QA/QC for subsampling and chlorophyll methods, one site will be selected on each river where a second subsample is taken from each slurry. These sites should be chosen in advance in order for the labels to be prelabeled at the lab. Labels should indicate parameter (periphyton chlorophylls), river, sample site, replicate number, designations as "a" or "b" to differentiate subsamples, date, total and filtered volume, and sampler initials. The sites chosen for the extra set of subsamples will be different on each sampling trip, so all sites will eventually be checked over time.

7) Measure the maximum length and width of each rock with a digital caliper and record these measurements on the field data sheet. Measurements should be recorded to the nearest 1 mm. The estimated surface area of each rock sampled will be determined later at NABF based on these measurements and a site specific regression model providing predictive relationships between true area and caliper measurements (Dudley, Arthurs, and Hall 2001).

8) Carefully rinse toothbrushes, scraping knives, graduated cylinders, filter apparatus, plastic tub, and mixing jars between samples with tap water. Again check equipment for cleanliness at the start of sampling at each station.

9) Repeat the procedure for the remaining four rock replicates, working downstream to upstream.

10) Samples are transported or sent back to NABF on dry ice and are stored in a -20˚C freezer until analyses (3-week holding time). See Section 4.13 for shipping instructions.

### 4.9.2 *Periphyton Qualitative Sampling Methods (Taxonomic Identification)*

Samples for periphyton chlorophylls and periphyton taxonomic identification are collected at the same time. The slurries collected during periphyton chlorophyll sampling are also used for taxonomic identification. See Section 4.9.1 for periphyton chlorophyll sampling equipment and methods.

*Equipment*

Bottles, screw cap, 125 ml Nalgene for periphyton identification (prelabeled, 1 per site)

Container to transport algae ID samples (cooler or Rubbermaid Action Packer)

Container, plastic with screw cap lid (1-gal) for tap water

Electrical tape

Gloves, PVC

Graduated cylinders, 25-ml, plastic (2)

Lugol's solution

Markers (waterproof Sharpies)

Safety glasses

Squirt bottles for tap water (1-L) (2)

Storage containers (2-qt) with plastic with lids for secondary containment of samples and Lugol's solution (3)

Tap water

Tape, clear, tear by hand (2 rolls)

Transfer pipettes for Lugol's solution (minimum of 2)

*Sampling Method*

1) One 125-ml Nalgene bottle per site should be prelabeled at NABF with a waterproof label indicating sample type (periphyton ID), the river name, site name, and preservative. The sample date is recorded in the field.

2) A 20-ml aliquot from each of the five rock periphyton slurries described in Section 5.7.2 should be measured using a 25-ml graduated cylinder and composited in a prelabeled bottle. The original sample slurry should be agitated prior to the removal of the 20-ml aliquot.

3) After compositing all five 20-ml subsamples of slurries into the sample bottle, Lugol's solution is added as a preservative. Lugol's solution is made in advance at NABF (see the lab manual) and is added to the samples with a transfer pipette to create a dark tea color solution. Safety glasses and PVC gloves should be worn when handling Lugol's solution, because it is acidic and contains iodine.

4) Tape the lids of the sample bottles with electrical tape and place clear tape over the bottle label. Samples are stored in the dark at room temperature.

5) Rinse the graduated cylinder and mixing bottles with tap water between samples.

6) Samples are shipped or transported back to NABF in secondary containment and are repackaged and shipped out to the consulting taxonomist. See Section 4.13 for shipping instructions.

### 4.10 Periphyton Sample Collection—Native Sand (Leaf River)

Native sand samples are collected twice a year on the Leaf River in conjunction with other sampling for a determination of species composition. Samples should be collected at the sandbar locations (Table 4.3) at a water depth of 20 to 40 cm. The sample area should be representative of the free-flowing perimeter of the main channel of the river. Backwater areas or other areas of stagnant flow should be avoided. Water quality measurements of conductivity, temperature, light, and current flow are taken at the location of the sand periphyton samples.

*Equipment*

Clipboard

Conical centrifuge tubes, screw cap, polypropylene, 50-ml, prelabeled, (1 per site)

Data sheets on *Rite in the Rain* ("Leaf River Periphyton and Macroinvertebrate Sampling Data Sheet", Appendix H, page H1) (1 per site)

Electrical tape

Gloves, PVC

GPS

Lugol's solution

Safety glasses

Secondary containment for Lugol's solution (2-qt plastic container with lid)

Secondary containment, 1-L Nalgene, wide mouth

Transfer pipettes

*Sampling Method for Native Sand*

1) A 50-ml conical centrifuge tube should be prelabeled at NABF with sample type (sand periphyton identification), river name, and site. Sample date is filled out in the field.

2) Because this sample will be used for determining relative abundance, sampling can be done using the cap from the 50-ml centrifuge tube used to hold the native sand sample (exact surface area isn't important). Take at least three scoops of sand from the surface of the sand substrate and composite into one prelabeled 50-ml centrifuge tube.

3) To preserve the sample, add Lugol's solution with a transfer pipette to create a dark tea color solution. Lugol's solution is made in advance at NABF (see the lab manual). Wear PVC gloves and safety glasses when handling the Lugol's solution. Invert the centrifuge tube several times to ensure that the Lugol's solution is mixed in with the sand.

4) Seal the lid of the tube with electrical tape, and store the samples at room temperature in the dark.

5) The samples will be shipped back to NABF with the field equipment and then shipped out to the taxonomist. See Section 4.13 for shipping instructions.

6) In the same vicinity as the sand sampling, take conductivity, temperature, light, and current velocity measurements, and record on page one of the field data sheets ("Leaf River Periphyton and Macroinvertebrate Sampling Data Sheet", Appendix H, page H1).

## 4.11 Periphyton Sampling—Artificial Substrates (Leaf River)

HD samplers are used to sample for both periphyton and macroinvertebrates, since the Leaf River is a shifting sand bottom river lacking in natural riffle/cobble substrate. See Section 4.15 for details of sampler construction, deployment, and retrieval. The top PVC plate of the HD sampler serves as a substrate for periphyton which is sampled for taxonomic identification and chlorophyll. The remaining plates are sampled for macroinvertebrates. See Section 4.17 for macroinvertebrate preservation instructions.

### 4.11.1 *Processing of HD Plates for Periphyton (Chlorophylls and Taxonomy)*

After the HD plates are retrieved, periphyton is sampled from the top PVC plates (three replicates per site) for chlorophylls and taxonomic identification. The samples are either processed on the boat or taken to the associated sandbar. To process each of three replicates collected from HD top plates for periphyton, tilt the top plate over a small plastic storage container while keeping the lower plates over the macroinvertebrate sample jar. The methods for processing periphyton from the plates for chlorophyll and periphyton identification are identical to those described in Section 4.9.1 and Section 4.9.2 for natural riffle substrates. Work quickly to prevent the loss of macroinvertebrates from the lower plates. No caliper measurements are needed for the HD samplers. It may be necessary to reduce the aliquot of slurry from each plate for the periphyton identification composite from 20 ml to 10 to 15 ml, because the total volume of the slurry is usually less than that scraped from cobble. QA/QC subsamples of each replicate are taken at one site.

## 4.12 Dry Ice Sources for Chlorophyll Samples

Chlorophyll samples are stored and shipped on dry ice. Dry ice should be purchased prior to sampling.

### 4.12.1 *Dry Ice Source for Codorus Creek Chlorophyll Samples*

Rutters Farm Stores (Main office) (717–848–9827)
2100 North George Street
York, PA
Hours: 7:30 am to 4:30 pm (M–F)

Dry ice may not be immediately available during sampling on Codorus Creek. A liquid nitrogen dewar is often used to store the samples during sampling, and the samples are later transferred to dry ice for shipping. The nitrogen dewar is stored at the Glatfelter Old Forge Technical Center, and Doug Brodhecker, chemist at Glatfelter, can fill it. Give him at least a week's advance notice.

### 4.12.2 *Dry Ice Sources for Leaf River Chlorophyll Samples*

There are two sources of dry ice in Hattiesburg. Saverite is the preferred source, because the dry ice is sold in blocks, while Nick's dry ice is a powdered form, which is not as long lasting.

1) Saverite Grocery Warehouse
4400 Hardy Street
Hattiesburg, MS
Phone: 601–268–2929
Hours: 6 am–11 pm daily

2) Nick's Ice House
2106 Hardy Street
Hattiesburg, MS
Phone: 601–544–5987
Hours: After 3 pm

### 4.12.3 *Dry Ice Sources for Willamette and McKenzie River Chlorophyll Samples*

Dry ice may be purchased in Anacortes on the day of travel to Oregon and can also be purchased at locations in Corvallis, Eugene, and Springfield, OR.

1) Safeway (Anacortes, WA)
911 11th Street
Phone: 360–293–5393

2) Food Pavilion (Anacortes, WA)
1615 Commercial Avenue
Phone: 360–588–8181

3) Albertsons (Corvallis, OR)
2005 NW Circle Boulevard
Phone: 541–752–5538
Hours: 6 am to 12 am (daily)

4) Crystal Dry Ice Company (Eugene, OR)
3894 Roosevelt Boulevard
Phone: 541–485–2436
Hours: M–F 8 am to 12 pm and 1 pm to 5 pm; Sat. 9 am to 12 pm

5) Main Street Muffler & Brake (Springfield, OR)
2309 Main Street
Phone: 541–744–6646
Hours: 9 am to 5 pm (M–F)
Call ahead of time to make sure dry ice is on hand.

### 4.13 Shipping Periphyton Samples

Periphyton chlorophyll samples and periphyton taxonomy samples from either Codorus Creek or the Leaf River will be shipped back to NABF. Periphyton samples from the McKenzie and Willamette Rivers will be transported by NABF staff. Ship samples to:

NCASI
1219 Q Avenue
Anacortes, WA 98221
Phone: 360–293–4748

#### 4.13.1 *Shipping Periphyton Chlorophyll Samples*

Periphyton chlorophyll samples will be shipped back on dry ice to NABF via FedEx Priority Overnight Service. A small cooler (approximate size 14" x 9" x 13") completely filled to the top with approximately 15 pounds of dry ice is sufficient to ensure the samples remain frozen during overnight transport. Two dry ice shipping labels should be filled out and attached to the sample cooler. Be sure that the box for dry ice is checked on the FedEx air bill and fill out the weight of the dry ice in kilograms. Someone at NABF should be designated in advance to receive and transfer samples to one of the garage freezers. Provide tracking information to the sample receiver and insure the samples for $500.

#### 4.13.2 *Shipping Periphyton Taxonomy Samples*

Samples for periphyton taxonomic identification will be shipped back to NABF with other equipment (Ground FedEx or FedEx Express). Place the samples in secondary containment. The periphyton will be repackaged at NABF and shipped via FedEx ground to the algal taxonomist:

Yangdong Pan
Environmental Sciences and Resources
Portland State University
218A SBII
1719 SW 10th Avenue
Portland, OR 97201
503–725–4981
Email: bwyp@pdx.edu

Include with the samples an analysis request which has a list of the sample sites, sample collection dates, date samples were shipped for identification, and the NABF address to send results. Place copies of this request into the Periphyton/WQ data file for each river as a record that the samples have been shipped out for analyses. Insure samples for $500. Dr. Pan should be notified via email prior to shipment, and he should be provided with the shipment tracking number.

### 4.14 Macroinvertebrate Sample Collection—Natural Substrate (Cobble)

For Codorus Creek, McKenzie River, and Willamette River, macroinvertebrates are evaluated for biomass and taxonomy on natural riffle cobble. See Section 4.3 for site locations.

### 4.14.1 *Sample Station Characteristics*

The same general sample area should be used for macroinvertebrate sampling that is used for periphyton samples on natural riffle cobble. The overall sample area should extend over a 20 m area of river bed and represent water depths ranging from 20 to 40 cm. Field personnel who sample for macroinvertebrates report observations of substrate characteristics on the field data sheet. As described for periphyton sampling, five specific locations within the 20 m sample area should be marked at the initiation of sampling with weighted colored markers with attached colored streamers and floats. These markers should be broadly distributed within the 20 m area, but individual markers should be relocated if water depth exceeds 40 cm or if unusual conditions (e.g., current velocity, substrate type) are represented. Depth and current velocity measurements, as well as macroinvertebrate and periphyton samples, are taken within 1 m of the markers.

### 4.14.2 *Macroinvertebrate Sample Collection—Hess Sampler*

*Equipment*

Bottles, plastic screw top (1 pt), prelabeled (5 per site)

Bubble wrap for shipping containers

Clipboard

Data sheets on *Rite in the Rain* waterproof paper ("Benthic Macroinvertebrate Sampling and Current Velocity Measurement Data Sheet", Appendix G) (1 per site)

Digging tool (trowel) (2)

Electrical tape

Eyewash (portable)

Filter netting apparatus and extra mesh (250-µm mesh)

Forceps

Formalin, 10% neutral buffered containing rose bengal, (2 x 4-L per river)

Garbage bags to double bag invertebrate samples

Gloves, nitrile (used when handling formalin)

Gloves, rubber, gauntlet type for Hess sampling (2 pairs)

Hess

Markers (waterproof Sharpies), mechanical pencils, lead refills

Pan, white

Rubber bands to hold up gloves

Safety glasses

Secondary containment buckets for formalin

Shipping containers for samples (8-gal Rubbermaid Action Packers)

Spatula, rubber

Spill pillows

Squirt bottle (1-L Nalgene)

Stopwatch (2)

Weights, flagging, and floats for marking sites (5)

*Sampler Operation*

Samples to be collected at each sampling station will include five Hess samples stored as separate replicates. The Hess sampler (Wildlife Supply Company Model 16-C22) is 33 cm diam x 40 cm deep and is equipped with a 243 μm mesh net.

Sampling begins at the downstream end of the sampling reach. The Hess sampler should be oriented with the stream flow (opening of net facing upstream) (Figure 4.31). Before beginning a sample, it should be determined that the sampler has formed a good seal with the substrate. Because the Hess is a cylinder, it can easily be seated into most substrates by pushing down while twisting. If seating the Hess proves difficult (most likely due to large diameter substrate material), select a different position within the same general sample area.

**Figure 4.31** Hess Sampling at Furnace, Codorus Creek

*Sample Collection*

Hess sample time should be based on 2 to 3 min of effort with the substrate worked to a depth of 10 cm either by hand or with a digging tool. Smaller particles (i.e., up to pebble size) need only be stirred. Larger particles (i.e., cobble and larger), however, will need to be scraped individually (by hand or with garden tools—whatever works best for that particular rock), as macroinvertebrates frequently reside in depressions on these larger rocks. The overall sample effort should be timed with a stopwatch and recorded in minutes on the data sheet.

*Sample Processing*

When removing the Hess from the sampled substrate, rinse the collected materials from the net down into the removable cup. Once the bottom edge of the Hess has cleared the substrate, pull the bottom of the Hess up in a "scooping" motion to flush the net. The entire Hess sampler should be held up, with the tip of the net pointed down, to allow the net to drain. If too much material (macroinvertebrates and/or debris) remains on the sides of the net, repeat this rinsing process by

dipping the Hess into the water, lifting, and allowing the net to drain. Once the sample material has been flushed into the cup at the end of the Hess net, transfer it to a screw top plastic storage jar (≈500 ml). The sample volume (including rinse water) should be kept to a minimum in order to minimize the amount of formalin preservative that will be required. See Section 4.17 for sample preservation instructions. Perform a final inspection of the net by turning it inside-out and removing any remaining specimens with forceps (caddisflies and beetles in particular tend to cling to the net).

### 4.14.3 *Substrate Characteristics*

A member of the macroinvertebrate sampling crew also makes an estimate of the substrate characteristics for that site, and this is recorded on the "Benthic Macroinvertebrate Sampling and Current Velocity Measurement Data Sheet" (Appendix G). See Section 4.4.2 for descriptions of these substrate characteristics.

### 4.14.4 *Current Velocity and Water Depth*

Current velocity is another parameter collected during field operations to characterize environmental conditions and is normally measured by the macroinvertebrate component of the field crew. Velocity readings are recorded on the "Benthic Macroinvertebrate Sampling and Current Velocity Measurement Data Sheet" (Appendix G). Current velocity measurements should be taken within 1 m of the sample site location markers. These measurements are usually taken after a replicate has been collected to avoid substrate disturbance in the actual area of macroinvertebrate collection.

Water depth for the five sample locations is measured with a meter stick and current velocity is measured to the nearest 0.01 m/s with a Marsh-McBirney Model 2000 Flow-Mate portable flow meter. Velocity measurements should be made with the probe 10 cm above the stream bed and oriented upstream toward the water flow. One measurement should be made adjacent to each of the five locations used for macroinvertebrate and periphyton samples. The flow meter should be returned to the factory for calibration every three years or at any time when symptoms of malfunction appear.

## 4.15 Macroinvertebrate Sample Collection—Natural Substrate (Woody Debris)

On the Leaf River, macroinvertebrate collections are made from submerged natural woody debris (tree branches, logs, etc.) in addition to the HD samplers (Figure 4.32). These natural substrate samples are used to provide supplemental taxonomic information for those organisms that might not colonize the HD samplers but might nevertheless be important to the overall river ecosystem. Biomass determinations are not made on these samples. Sample locations for natural substrate macroinvertebrate collections should be in the same vicinity as periphyton native sand samples. However, it is sometimes necessary to use the boat to collect these samples, if woody debris is not available at the sandbar.


**Figure 4.32** Sampling Macroinvertebrates from Natural Substrate on the Leaf River

*Equipment*

Bottles, screw top (125-ml Nalgene, prelabeled) (1 per site)

Clipboard

Data sheets on *Rite on the Rain* paper (Appendix H: "Field Data Sheets for Periphyton and Macroinvertebrate Sampling from the Leaf River" (page 2) (1 per site))

Electrical tape

Eyewash (portable)

Forceps

Formalin (10% buffered neutral formalin) containing rose bengal (500 ml)

Gloves, nitrile

Markers (waterproof Sharpies), mechanical pencils, lead refills)

Measuring tape

Pruning saw

Shipping containers (8-gallon Rubbermaid Action packers—also used for HD samplers) (3)

Safety glasses

Secondary containment buckets for formalin

Spatula (rubber)

Spill pillows for formalin

Squirt bottle (1-L Nalgene)

Stopwatch (2)

Tray or collection pan (white)

Zip ties for shipping containers

*Method for Sampling Macroinvertebrates from Natural Substrates*

1) 125-ml Nalgene screw cap bottles should be prelabeled at NABF with sample type (natural substrate macroinvertebrates), river name, sample location, and date.

2) An arm-sized (approximate) submerged piece of wood (tree branch or section of log) should be removed from the stream and placed in a white collection tray or pan. A single piece of wood should be used for each of the Leaf River sample locations. A pruning saw or lopping shears may be helpful in the removal of woody debris from the river.

3) Collect macroinvertebrates from the surface of the wood over a 10 min period. Some macroinvertebrates should become visible over this time span as they emerge from crevices in the wood.

4) Measure the dimensions of the wood with a measuring tape and record measurements on the field data sheet.

5) Organisms collected should be placed in a prelabeled sample bottle (125-ml screw top Nalgene bottle). Add 10% buffered neutral formalin containing rose bengal. See Section 4.17 for details of sample preservation. The bottle lid should be secured with electrical tape for transport and shipment. These samples are sent back to NABF and later sent to the taxonomist along with the HD samples for identification.

## 4.16 Macroinvertebrate Sample Collection—Artificial Substrate (HD Samplers)

Hester-Dendy samplers are used on the Leaf River since it is a shifting sand bottom river lacking in natural riffle/cobble substrate. The HD samplers are constructed at NABF and consist of seven 76 mm x 76 mm x 4 mm thick fiberboard plates, and one PVC plate on top with the same dimensions. Periphyton is collected from the top PVC plate of each of three samplers at each site for chlorophylls and taxonomy, and the remaining plates are sampled for macroinvertebrates. Each sampler is assembled with a stainless-steel eyebolt and washers to provide spacing between the plates ranging in a decreasing gradation from 10 to 2 mm. See the NABF lab manual for details of construction.

### 4.16.1 *Deployment of HD Samplers*

The plates are deployed over a period of five to six weeks and are attached by tethers to concrete anchor blocks at fixed locations in pools on the riverbed. Stations for anchoring substrates represent 1.7 to 2.3 m deep pools and should represent roughly equivalent conditions with respect to current and light exposure. HD samplers are anchored to the streambed with anchor blocks and suspended from a tether and float as indicated in Figure 4.33. The components of the anchor system may vary, but in general, the primary anchor cable should be made of stainless-steel aircraft cable, and the tether to the sampler should be made of plastic-coated cable. The marker buoys may also vary but are typically Styrofoam fishing floats.

The HD samplers, when suspended from the float/tether, should be approximately 50 cm below the water surface. A total of six HD samplers are placed in each of the river pool locations. Three of these are attached to the marker buoy, and three are attached to a nearby snag as redundancy in the event the buoy samplers are vandalized. Three replicates are analyzed for macroinvertebrates and periphyton. Identification tags with the contact phone number should be attached to the samplers.

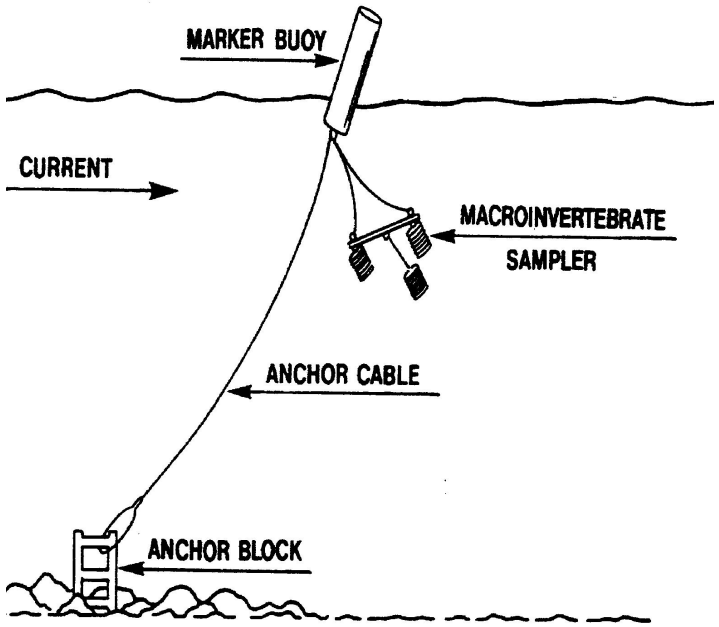


**Figure 4.33** Leaf River HD Sampler Anchor, Tether, and Float System (Mischuk, Howell, and Howell 2000)

### 4.16.2 *Retrieval of HD Samplers for Periphyton and Macroinvertebrates*

A colonization time from 5 to 6 weeks should be allowed before the HD samplers are retrieved. Site locations are located using GPS positions. Exercise caution when retrieving the HD samplers, because this procedure occurs from a boat and the plates are at mid-pool locations. Because boat electrofishing often occurs in the same vicinity where the HD samplers are deployed, the HD samplers should be retrieved before boat electrofishing begins.

*Equipment for HD Multi-plate Retrieval*

Bottles, screw top (1-L Nalgene, wide mouth, 11 cm diam), prelabeled (3 per site)

Bubble wrap for shipping containers

Clipboard

Conductivity meter (Hach sensION5 or equivalent)

Data sheets on *Rite on the Rain* waterproof paper ("Leaf River Periphyton and Macroinvertebrate Sampling Data Sheet") (page 2) (see Appendix H) (1 per site)

Depth finder (Speedtech Instruments or equivalent)

Flow meter (Marsh-McBirney Model 2000 Flow-Mate portable flowmeter)

GPS

Markers (waterproof Sharpies), mechanical pencils, lead refills

Scientific collecting permit (Leaf River)

Shipping containers (8-gallon Rubbermaid Action packers) (3)

Wire cutters

Yardstick (folding)

*Retrieval of HD Plates*

1) Locate the buoy to which three replicates of HD plates are attached. If the buoy cannot be located, look for the second set of plates which should be attached to a nearby snag (redundancy set).

2) Tie the boat up to the buoy or snag. The HD plates should be placed individually in 1-L Nalgene wide-mouth (11 cm diam) screw top containers prelabeled with a waterproof marker with river, sample location, replicate number, and sample date. Before pulling up the plates, open the sample containers and line them up in a row. Then, carefully lift up the bar holding the multiplates and line the three sets of multiplates over the three sample jars. Snip off the wires attaching the multiplates to the bar with wire cutters. Work quickly to prevent the loss of macroinvertebrates.

3) Add enough river water to each container to cover the top plates to prevent them from drying out, but do not pour water directly onto the top plate, since it will be sampled later for periphyton. The macroinvertebrates should be preserved with 10% buffered neutral formalin after the plates are processed for periphyton. See Section 4.11 for periphyton processing and Section 4.17 for macroinvertebrate preservation.

4) Fill out the "Leaf River Periphyton and Macroinvertebrate Sampling Data Sheet" (page 2). See Section 4.5.2 for other details of the data sheet.

5) Measure pool depths at the HD deployment site with a hand-held sonar depth finder and record to the nearest 1 ft.

6) Measure the water depth to the upper surface of the top most HD with a meter stick.

7) Measure current velocity at the uppermost HD sampler to the nearest 0.01 m/s with a Marsh-McBirney Model 2000 Flow-Mate portable flowmeter. Velocity measurements should be made with the probe oriented upstream toward the water flow.

8) Measure conductivity and temperature with the Hach sensION5 conductivity meter (or equivalent).

### 4.17 Macroinvertebrate Sample Preservation

Macroinvertebrates are preserved with 10% buffered neutral formalin containing rose bengal. Approximately 25 mg of rose bengal or enough to produce a light pink color should be added to the 4-L 10% buffered neutral formalin bottles before use. Wear safety glasses, PVC gloves, and use a metal spatula when adding rose bengal to the formalin.

The Chemical Hygiene/Field Safety Plan (CHSP) should be consulted for safe handling procedures for formalin. Standard procedures include secondary containment vessels for formalin transport and the use of safety glasses and nitrile gloves for working with formalin during sample processing. A field eyewash bottle should also be available, and the expiration date on the eyewash should be checked on each trip.

Formalin is added to sample jars by carefully pouring directly from the bottle into the sample bottle. For secondary containment, place the sample bottle into a small plastic container when pouring the formalin. The volume of formalin in the sample bottle should be at least 50% formalin to river water for all samples whether collected from natural substrates or consisting of HD plates. For the HD samplers, it is important that there is enough volume to cover all the plates. It may be necessary to carefully pour off some of the water in order to add sufficient formalin to the containers with the plates.

HD sample bottle lids should be secured with duct tape prior to shipment, while electrical tape can be used for the bottles containing natural substrate macroinvertebrates. Electrical tape is too narrow to seal the HD sample bottle lids and leakage may occur.

### 4.18 Shipping Macroinvertebrate Samples

Codorus Creek samples should be sent directly to the consulting taxonomist from the field, while samples from the Willamette and McKenzie Rivers should be transported back to NABF and then shipped to the taxonomist. Leaf River HD samples and woody debris samples will be sent back to NABF where the HD samplers will be processed before shipment to the taxonomist. See the NABF lab manual for processing instructions.

Effort should be made to split different sample locations and rivers between shipping containers to minimize potential losses during shipping. Line the bottom of the shipping containers (Rubbermaid Action Packer boxes) with bubble wrap. For added containment, place the sample bottles into double garbage bags. Add enough bubble wrap so the sample bottles remain upright during shipment. Information including NCASI address and phone number should be included inside the shipping containers so that in the event of shipment loss or misplacement, there is a better opportunity of sample recovery. Use zip ties to secure the lids of the action packers. FedEx shipment tracking numbers should be retained in order to allow for package tracking if needed. Each shipping container should be insured for $500.

Ship the samples to the taxonomist, David Goldhammer, Benthic Aquatic Research Services via FedEx Ground or FedEx Express at the following address:

Benthic Aquatic Research Services
174 Trabecca Circle
Hot Springs, AR 71913
Telephone: 501–276–2033
Email: aquaticinsects@hotsprings.net

David Goldhammer should be notified via email prior to shipment and be provided with the shipment tracking numbers. Macroinvertebrate samples contain <10% formalin and are not regulated under Department of Transportation regulations as Dangerous or Hazardous goods. Consequently, no external package labeling is required nor is it necessary to notify the shipper of the formalin content of the shipment.

## 5.0 FISH COMMUNITY ASSESSMENT

### 5.1 Overview

Fish community assessment is carried out to determine whether there are measurable differences in fish community structure upstream or downstream of the LTRWS mill effluent discharge. Procedures for both boat and backpack electrofishing used in the LTRWS were derived from those developed by Dr. Leo Bodensteiner (1998a, 1998b) at Western Washington University. Fish sampling is based on a combination of boat (Figure 5.1) and backpack (Figure 5.2) electrofishing on the McKenzie, Willamette, and Leaf Rivers to assure that the fish community assessment is as thorough as possible. Backpack electrofishing is carried out in relatively shallow water along the shoreline whereas boat electrofishing extends to deeper water areas along the shore and mid-channel. Codorus Creek fish samples are based entirely on backpack electrofishing since the Codorus, due to shallow water, is generally not navigable by boat. In addition to species identification, length, weight, and external health assessments are made on individual fish.

**Figure 5.1** Boat Electrofishing on the McKenzie River

**Figure 5.2** Backpack Electrofishing on Codorus Creek

## 5.2 Electrofishing Safety

Electrofishing represents potential hazards for both boat- and backpack-based activities. Since electrofishing is either boat-based or involves wearing a relatively heavy backpack shocker unit, the use of a suitable PFD is mandatory. An automatic inflation PFD is recommended for backpack shocker use since the bulk of a conventional PFD makes equipment use difficult and uncomfortable. The following excerpts from the NABF Chemical Hygiene/Safety Plan (CHSP) provide guidelines for safe electrofishing.

### 5.2.1 *Electrofishing Safety Methods excerpted from the American Fisheries Society, 1983. Electrofishing by LB. Reynolds in Fisheries Techniques, pp. 147–163, and Smith-Root, Inc. 1997. Electrofishing Manual*.

Backpack Electrofishing Safety:

- ✓ Before each operation, check the frame emergency release to ensure it is in working order and check that the tilt switch shuts off power if the unit is tipped more than 45 degrees.
- ✓ Wear hip boots or chest waders with non-skid soles.
- ✓ Beware of turbid water that can hide unseen subsurface obstacles and sudden drop-offs.
- ✓ Shut off your electrofisher before entering or leaving a stream.
- ✓ If you develop leaks in your boots, waders or gloves, stop work immediately and get dry clothing.
- ✓ Operate slowly and carefully. Footing in most streams is poor, and most falls often occur when operators are hurrying.

Boat Electrofishing Safety:

- ✓ Ground the generator to the boat hull.
- ✓ Be sure that all of the metal parts on the boat are bonded to each other electrically.
- ✓ Run all cables through electrical conduit or use a heavy-duty rubber-covered cord recommended for wet locations.
- ✓ Make all electrical connections in watertight junction boxes.
- ✓ One dip netter will have a foot switch to control the output.
- ✓ Rubber-soled shoes, rubber boots, hip or chest waders should be worn with non-skid surfaces while electrofishing on the boat.
- ✓ The person driving the boat will not take part in netting. The operator must be constantly aware of the netters in the electrical field.
- ✓ Always be sure that all personnel are clear of the electrodes before turning on the power.
- ✓ DC current is used and the amperage is varied by the crew leader and boat operator depending on water conditions.
- ✓ Operation of electrofishing equipment near other people, pets or livestock in or on the water or shore is not permitted.
- ✓ Operations should cease if water or weather conditions are threatening.

### 5.2.2 *Electrofishing Safety Checklist from the U.S. Geological Survey*

Electrical Equipment

- ❑ Electrical connections secure and protected
- ❑ Gages and wiring in proper working condition
- ❑ "Deadman switch" in operating condition
- ❑ Anodes in good condition; attached to handles securely (wadeable streams)

Ancillary Equipment

- Fire extinguisher—fully charged (for boat operations)
- First aid kit present
- Dip net handles constructed of nonconductive material

Crew Members

- Trained in electrofishing operation
- Wearing rubber gloves (inspected for leaks)
- Wearing chest waders (inspected for leaks) with nonslip soles (wadeable streams)
- Wearing hip boots (inspected for leaks) (non-wadeable streams)

### 5.3 Sampling Time and Frequency

Backpack and boat electrofishing are carried out twice a year (spring and fall) on the Willamette and McKenzie Rivers. Backpack and boat electrofishing are carried out once a year in the fall on the Leaf River. Beginning in 2007, fish sampling on Codorus Creek was reduced from twice a year to once a year in the spring. Dates should be adjusted as necessary to provide for safe sampling conditions (e.g., avoid high water). Sampling should not be carried out when Secchi readings are <20 cm since turbid water would adversely impact catch efficiency and safety.

As of 2004, there are restriction dates on the fish permit for boat electrofishing on the McKenzie and Willamette Rivers in order to avoid the salmon spawning runs. See Section 5.4.3 for details.

### 5.4 Scientific Collection Permits

All four rivers require scientific collecting permits for fish. The collection permit for Codorus Creek and the Leaf River include the collection of macroinvertebrates and fish. Permits are not required on the McKenzie or Willamette River for the collection of macroinvertebrates.

#### 5.4.1 *Codorus Creek*

Application for the annual renewal of the collecting permit for fish and macroinvertebrates is through the Pennsylvania Fish and Boat Commission (PFBC). The annual report summary of fish sampling is due prior to permit renewal or by January 30 of the following year. It can be reported online anytime during the collection year. Application for the collection permit can also be done online at the same website (http://www.scientificcollector.state.pa.us/).

Fishing licenses and trout stamps are required for all members of the sampling team and must be in their possession along with a copy of the collecting permit during sampling. Both the licenses and trout stamps can be obtained at the PFBC website (http://sites.state.pa.us/PA_Exec/Fish_Boat/mpag1.htm). Licenses should be purchased prior to renewal of the collecting permit, because the permit requires the fishing license numbers of each of the field staff. Licenses should be worn when sampling for fish or macroinvertebrates.

Currently, there are two collecting permits, each with a different primary collector listed. One permit lists Bill Arthurs as the primary collector on the permit and the other permit lists Joan Ikoma. One of these two must be part of the field crew since other members of the field staff are listed on the permit as assistants.

Notify the PFBC of the sample dates at least a week in advance of sampling. Call 717–486–7087 (Southcentral Region) and ask the information to be relayed to Officer David Keller or other contact as designated by PBFC.

### 5.4.2 *Leaf River*

Application for renewal of the "Administrative Scientific Collection Permit" for macroinvertebrates and fish (boat and backpack electrofishing) as issued by the Mississippi Museum of Natural Science typically occurs in May of each year. The permit is effective in June and expires one year from date of issuance. A complete report of collections (fish and macroinvertebrates) is due within 15 d of permit expiration and is typically sent in with the application renewal.

Each field person must have a copy of the permit in possession during sampling for either macroinvertebrates or fish. Local conservation officers must be given at least 2 d advance notice of electrofishing. Provide sample dates, sample gear, sampling locations, and request that the appropriate conservation officers be notified.

Phone: 601–928–3720 (District 6 office for Perry and Greene Counties)

### 5.4.3 *McKenzie and Willamette Rivers*

Two collecting permits are required for fish sampling on the McKenzie and Willamette Rivers. The National Marine Fisheries Service (NMFS) issues the Section 10 permit required for boat electrofishing on the McKenzie and Willamette Rivers. Annual reports are due by January 31 of each year and work on each succeeding year is based on the approval of the report. The Section 10 permit is valid for five years, and the current permit expires December 31, 2009.

The "Scientific Taking Permit—Fish" and the 4(d) attachment are issued from the Oregon Department of Fish and Wildlife (ODFW) and are required for both backpack and boat electrofishing on both rivers. These permits are applied for annually and also require an annual collection report due in December of each year.

Each member of the field team must have a copy of all permits when fish sampling. ODFW officials should be notified of the sampling dates at least two weeks in advance to allow ODFW staff to observe boat electrofishing if they so choose. The following are the current ODFW officials who should be contacted.

| | |
|---|---|
| Steve Mayomac (Willamette River): | Phone number: 541–757–4186 ext. 249<br>Email: Steven.R.Mamoyac@state.or.us |
| Jeff Ziller (McKenzie River): | Phone number: 541–726–3515 ext. 26<br>Email: Jeffrey.S.Ziller@state.or.us |

There are permit-based restriction dates when boat fishing cannot occur to avoid the salmon spawning runs. Boat electrofishing cannot occur from May 1 to July 1 and from September 1 to October 15.

The NMFS Section 10 permit indicates that if the water temperature exceeds 70˚F (21.1˚C) at the sample site during boat electrofishing, the permit holder must stop handling listed fish to minimize handling stress. To prevent stress and/or mortalities on unlisted fish as well, NABF staff will extend this requirement to all fish species, with boat electrofishing suspended if water temperatures exceed 70˚F. For backpack electrofishing, the NMFS permit requires that the permit holders follow the NMFS "Guidelines for Electrofishing Waters Containing Salmonids Listed under the Endangered Species Act." This guide indicates that no backpack electrofishing should occur when water temperatures exceed 64.4˚F (18˚C). Backpack electrofishing should consequently occur in the

mornings when water temperatures are cooler. Before fish sampling trips, USGS websites should be checked for river temperatures. See Section 2.0 for USGS website addresses.

The permits provide limits on fish mortalities and take. The Endangered Species Act (ESA) defines the term "take" to mean "to harass, harm, pursue, hunt, shoot, wound, kill, trap, or attempt to engage in any such conduct." Thus, NMFS instructs us to report not only ESA-listed fish that were netted but also those not netted but shocked (i.e., seen while shocking). For non-listed species, we use "take" to mean those fish caught and counted. Diligently monitor the take and remain aware that the permit limit for ESA-listed fish is for all seasons, all sites, and both rivers combined. These specified take limits can be changed annually. No mortalities are allowed for ESA-listed wild or hatchery adult Chinook salmon, wild adult steelhead, or juvenile wild steelhead. See Table 5.1 for a summary of take limits and authorized unintentional mortalities.

Fish mortalities will be recorded on the Fish Mortality Data Sheet (Appendix L) at each sample site. See Section 5.8 for information on the fish mortality data sheets. If any mortality of mountain whitefish occurs, scales will be taken from these fish and sent to Steve Jacobs, ODFW Fish Research, Corvallis, Oregon as this is a requirement of the Oregon Scientific Taking Permit. See Section 5.9 for more details.

If an adult Chinook salmon or steelhead is encountered during boat electrofishing, stop electrofishing and allow it to escape the shock field. Since Chinook salmon (wild and hatchery) and wild steelhead are ESA-listed, do not net these fish. It is likely that the fish cannot be identified as wild or hatchery, so the assumption will be that it is a wild salmonid and should be recorded as such. These encounters count toward the take limits even if the fish are not netted. Only net the fish if it appears distressed and needs reviving. It should be aided in its recovery and released immediately.

Similar procedures apply to backpack electrofishing, except that the fish of concern are primarily juvenile Chinook and juvenile steelhead. They should be counted and immediately released without measurements or health assessments. The permit allows a combined total of 70 juvenile Chinook for backpack and boat electrofishing. Only one (combined total) juvenile wild steelhead is allowed via combined backpack and boat electrofishing.

Any take or mortality that exceeds the limits allowed for non-listed species is reported at the end of the year in the annual report. Contact regulatory officials if the take or mortality limits for non-listed fish are exceeded before sampling can be completed for the year. There are no take limits on species not included in Table 5.1, but the mortality must not exceed 5%.

**If there are any mortalities of adult Chinook salmon (wild or hatchery) or wild steelhead mortalities (juvenile or adult), all electrofishing must stop, and regulatory officials should be notified as soon as possible (preferably the same day). Regulatory officials should also be contacted immediately if the take limits for any listed fish are exceeded.** These contacts are currently as follows:

Shelly Miller at ODFW (backpack electrofishing): 503–947–6254

Mary Hanson (Endangered Species Act Coordinator, ODFW): 503–947–6253

Gary Rule at NMFS (boat electrofishing): 503–230–5424

**Table 5.1** Current (as of 2007) Permit Take Limits for Boat and Backpack Electrofishing

| Method | Species | Life Stage | Production | Limit | Authorized Unintentional Mortality | ESA-listed |
|---|---|---|---|---|---|---|
| Boat Electrofishing | Chinook | Juvenile | Wild | 60* | 3 | Yes |
| | Steelhead | Adult | Wild | 1* | 0 | Yes |
| | Steelhead | Juvenile | Wild | 1* | 0 | Yes |
| Combined Limits: Boat & Backpack Electrofishing | Chinook | Adult | Wild | 13 | 0 | Yes |
| | Chinook | Juvenile | Wild | 70 | 3 | Yes |
| | Chinook | Adult | Hatchery | 15 | 0 | Yes |
| | Chinook | Juvenile | Hatchery | 70 | 0 | Yes |
| | Steelhead | Adult | Wild | 1 | 0 | Yes |
| | Steelhead | Juvenile | Wild | 3 | 0 | Yes |
| | Steelhead | Adult | Hatchery | 10 | 0 | No |
| | Cutthroat | Adult | N/A | 60 | 5% | No |
| | Rainbow trout | Adult | N/A | 40 | 5% | No |
| | Rainbow trout | Juvenile | N/A | 8 | 0 | No |
| | Brook lamprey | Adult | N/A | 2 | 0 | No |
| | Brook lamprey | Juvenile | N/A | 20 | 0 | No |
| | Other lamprey | Adult | N/A | 22 | 5% | No |
| | Mountain whitefish | Adult | N/A | 225 | 5% | No |

*This represents the boat take limit but counts toward the combined boat and backpack electrofishing limit.

## 5.5 Boat Electrofishing Sites

Sample locations for boat electrofishing are described in Sections 5.5.1, 5.5.2, and 5.5.3 for the Leaf, McKenzie, and Willamette rivers respectively. Due to shallow water depths, boat electrofishing is not carried out on Codorus Creek.

### 5.5.1 *Leaf River*

Sample locations generally correspond to those described in Section 4.3.2 for macroinvertebrate and periphyton sampling with additional detail provided below (Table 5.2). Boat electrofishing locations are selected that represent good riparian and submerged habitat diversity and are in areas with deep water adjacent to the river shoreline. These locations are approximate and judgment should be used in selecting an area representing optimum but representative fish habitat. Sampling on the Leaf River proceeds in a direction from downstream to upstream. Boat electrofishing sites are shown in Figure 5.3.

**Table 5.2** Leaf River Boat Electrofishing Sample Locations

| N Latitude | W Longitude | Waypoint | Site Name and Location Description |
|---|---|---|---|
| 31°13'38" | 89°05'26" | TALLBT | **Tallahala Creek**<br>Near the confluence of Tallahala Creek |
| 31°12'33" | 89°03'37" | NAUGBT | **New Augusta**<br>Above the New Augusta Bridge |
| 31°12'03" | 89°00'54" | DOWNBT | **Downstream**<br>Downstream of mill discharge |
| 31°11'29" | 88°56'22" | BOGBT | **Bogue**<br>Downstream of the Bogue Homo confluence |
| 31°10'45" | 88°54'12" | THOMBT | **Thompson**<br>Downstream of Thompson Creek confluence |
| 31°05'23" | 88°47'58" | MCLABT | **McLain**<br>Downstream of McLain Bridge |

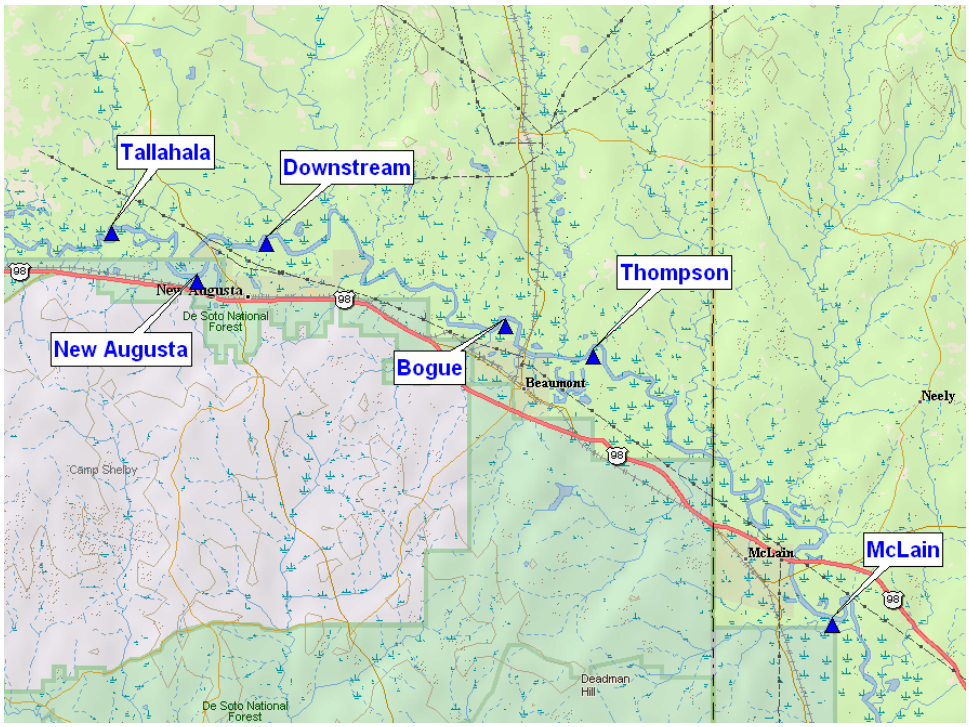


**Figure 5.3** Leaf River Boat Electrofishing Locations

### 5.5.2 *McKenzie River*

The McKenzie River is boat electrofished in an upstream to downstream direction in order to facilitate boat transport in this high-energy system. Sample locations generally correspond to those described in Section 4.3.3 for macroinvertebrate and periphyton sampling, with additional detail provided below (Table 5.3). Boat electrofishing sites are shown in Figure 5.4.

**Table 5.3** McKenzie River Boat Electrofishing Locations

| N Latitude | W Longitude | Waypoint | Site Name and Description |
|---|---|---|---|
| 44˚03’13.0” | 122˚49’21.5” | HENDBT | **Hendricks** The sampling area is upstream and downstream of Hendricks Bridge. Sampling begins ≈ 1.4 RM upstream of the macroinvertebrate/algae site. |
| 44° 03’35.9” | 122°53’39.4” | BELLBT | **Bellingers** Sampling begins ≈ 0.8 RM upstream of the macroinvertebrate/algae site. |
| 44°05’09.1” | 122°58’30.6” | MOHABT | **Mohawk** Sampling begins ≈ 0.05 RM downstream of the macroinvertebrate/algae site. |
| 44˚05’27.0” | 123˚01’19.5” | HARVBT | **Harvest** Sampling begins ≈ 0.3 RM downstream of the macroinvertebrate/algae site. |
| 44°06’46.2” | 123°02’52.0” | ARMIBT | **Armitage** Sampling begins ≈ 1.0 RM upstream of the macroinvertebrate/algae site. Sampling occurs upstream and downstream of the Armitage Bridge. |

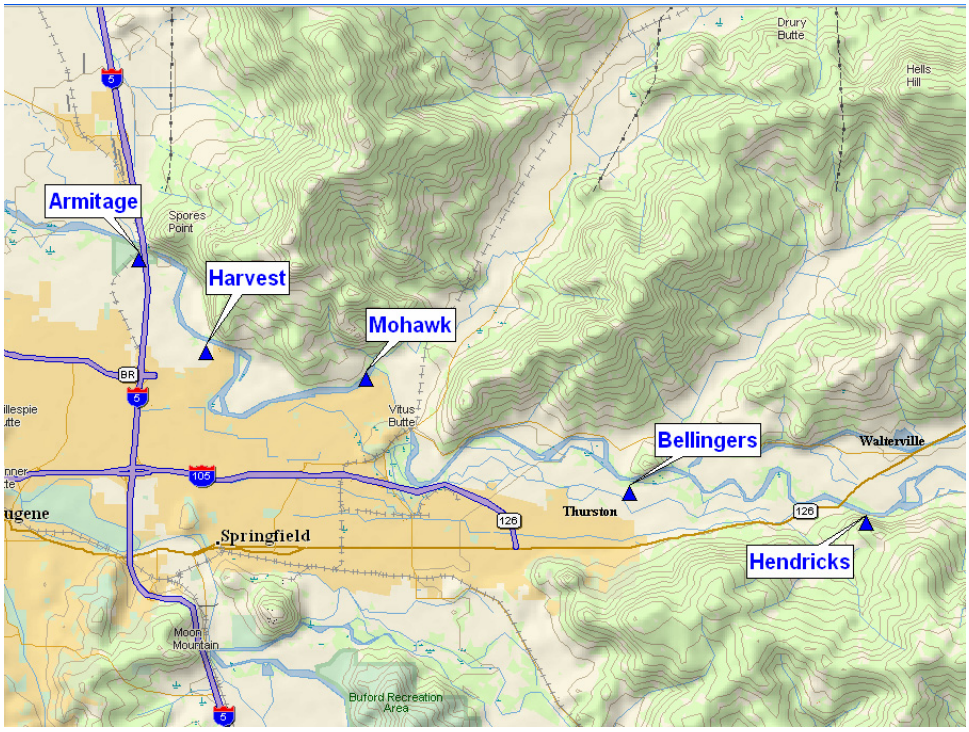


**Figure 5.4** McKenzie River Boat Electrofishing Locations

### 5.5.3 *Willamette River*

The Willamette River is boat electrofished in an upstream to downstream direction in order to facilitate boat transport in this high-energy system. Most boat electrofishing sites are near the corresponding macroinvertebrate and periphyton sites as described in Section 4.3.4, with additional detail provided below (Table 5.4). The Peoria site is located near the backpack electrofishing site. GPS positions are the approximate locations where boat electrofishing begins at each site. Boat electrofishing sites are shown in Figure 5.5.

**Table 5.4** Willamette River Boat Electrofishing Sample Locations

| N Latitude | W Longitude | Waypoint | Site Name and Description |
|---|---|---|---|
| 44°16’28.7” | 123°10’28.5” | HARRBT | **Harrisburg**<br>Sampling begins ≈ 0.8 RM upstream of the macroinvertebrate/algae site. |
| 44°19’10.8” | 123°13’04.3” | CARTBT | **Cartney**<br>Sampling begins ≈ 0.1 RM downstream of the macroinvertebrate/algae site. |
| 44°23’14.5” | 123°13’47.7” | INTABT | **Intake**<br>Sampling begins ≈ 0.1 RM upstream of the macroinvertebrate/algae site. The sampling area is upstream of the mill water intake and effluent discharge. |
| 44°24’35.2” | 123°13’41.8” | SAMBT | **Sam Daws**<br>Sampling begins ≈ 0.6 RM upstream of the macroinvertebrate/algae site. |
| 44°31’04.6” | 123°13’41.9” | PEORBT | **Peoria**<br>Sampling begins ≈ 0.03 RM downstream of the backpack electrofishing site. |
| 44°31’52.1” | 123°15’05.0” | WILLBT | **Willamette Park**<br>Sampling begins ≈ 0.4 RM upstream of the macroinvertebrate/algae site. |


**Figure 5.5** Willamette River Boat Electrofishing Locations

## 5.6 Backpack Electrofishing Sites

Backpack electrofishing sites for Codorus Creek, Leaf River, McKenzie River, and Willamette River should be located using GPS positions as described in the following sections.

### 5.6.1 *Codorus Creek*

Codorus Creek backpack electrofishing locations are described in Table 5.5 and shown in Figure 5.6. Additional detailed information about reaching these sample locations can be found in Section 4.3.1.

**Table 5.5** Codorus Creek Backpack Electrofishing Sample Locations

| N Latitude | W Longitude | Waypoint | Site Name and Location Description |
|---|---|---|---|
| 39°51'46.2" | 76°53'23.6" | MENGBP | **Menges**<br>Sampling begins just upstream of the bridge. Sample both sides of the stream and continue upstream just beyond the riprap at the bend. |
| 39°52'34.6" | 76°51'18.3" | USGSBP | **USGS**<br>This site is upstream of the USGS bug/algae site and upstream of the dam. Instead of turning onto Hershey Road for the bug/algae site, turn left at the road just before Hershey (no road name). The pool/riffle immediately in front of the parking lot marks the upstream end of the sampling area. Sampling begins as far downstream as safe depth allows. The stream becomes too deep upstream of the bend. Both sides of the stream are sampled. |
| 39°53'24.8" | 76° 50'09.0" | MARTBP | **Martin**<br>From the bridge, hike (downstream) along the edge of the farm field until it curves to the left. Access the creek through the riparian brush. Sample the two riffle/pools below the large logjam and one pool above it. Do not approach too closely to the logjam, as it is deep in this vicinity. Sample both sides of the stream. |
| 39°55'18.3" | 76°47'56.3" | GRAYBP | **Graybill**<br>Begin sampling upstream of the bridge. Sample the three-pool/riffle complexes upstream of the bridge. Beware of deep pools upstream on the right bank. Both sides of the stream are sampled. |
| 39°59'02.5" | 76°43'24.1" | ARSEBP | **Arsenal**<br>Sample downstream of the bridge. Sample the stretch from the large pipe upstream of the sewage outfall moving upstream to the rock outcrops on both sides of the stream. |
| 40°03'08.7" | 76°39'20.7" | FURNBP | **Furnace**<br>Sampling is upstream of the bridge. Walk downstream along the side channel and begin sampling upstream in the main channel on the left bank. The right bank is typically too deep. |

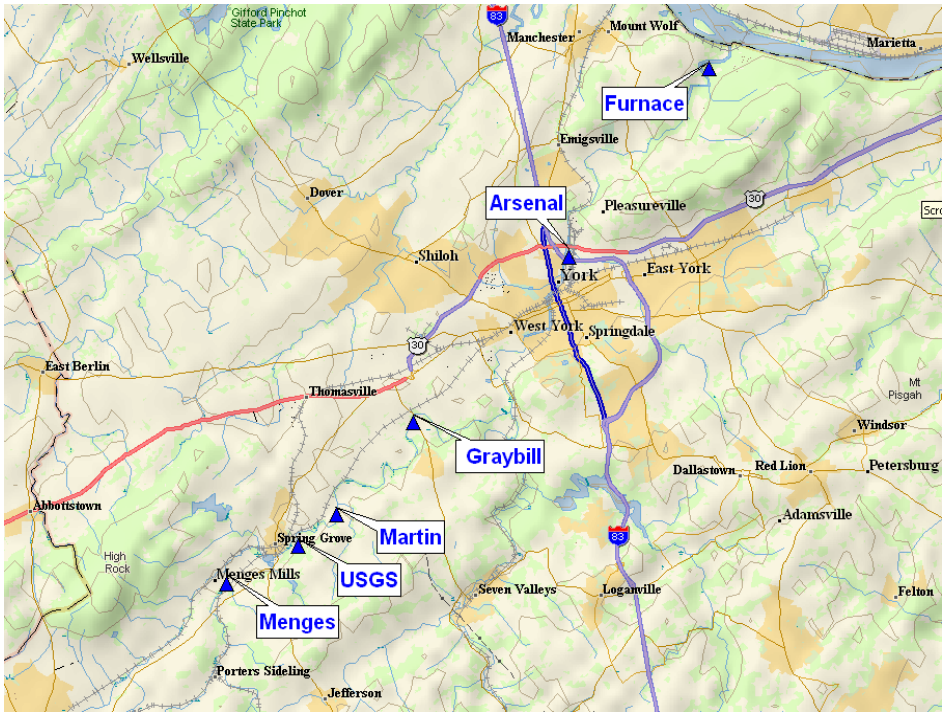


**Figure 5.6** Codorus Creek Backpack Electrofishing Locations

### 5.6.2 *Leaf River*

Leaf River backpack electrofishing sample sites are listed in Table 5.6. These locations are on the same sandbars used for periphyton sampling (see Figure 4.8). Additional detailed information about reaching these locations can be found in Section 4.3.2.

**Table 5.6** Leaf River Backpack Electrofishing Sample Locations

| N Latitude | W Longitude | Waypoint | Site Name and Location Description |
|---|---|---|---|
| 31°13’38.4” | 89°05’30.3” | TALLBP | **Tallahala Creek**<br>Upstream of the confluence with Tallahala Creek |
| 31°12’33.4” | 89°03’47.5” | NAUGBP | **New Augusta**<br>Upstream of the New Augusta Bridge |
| 31°13’11.8” | 89°01’47.1” | DOWNBP | **Downstream**<br>Downstream of mill discharge |
| 31°11’25.6” | 88°55’56.9” | BOGBP | **Bogue**<br>Downstream of the Bogue Homo confluence |
| 31°10’47.9” | 88°53’44.9” | THOMBP | **Thompson**<br>Downstream of Thompson Creek confluence |
| 31°05’35.3” | 88°48’09.3” | MCLABP | **McLain**<br>Downstream of McLain bridge |

### 5.6.3 *McKenzie River*

McKenzie River backpack electrofishing locations are described in Table 5.7 and shown in Figure 5.7. Additional detailed information about reaching these sample locations can be found in Section 4.3.3.

**Table 5.7** McKenzie River Backpack Electrofishing Sample Locations

| N Latitude | W Longitude | Waypoint | Site Name and Location Description |
|---|---|---|---|
| 44°03’29.9” | 122°50’54.9” | HEND2 | **Hendricks**<br>Approximately 1.0 RM (1.6 km) downstream of Hendricks Bridge Park.. Located on the left bank facing upstream. 8.6 RM (13.8 km) upstream of the mill effluent discharge. This site is only accessible by drift boats. |
| 44°04’55.7” | 122°55’15.5” | BELL4 | **Bellingers**—Bellingers Landing<br>South bank gravel beach above Bellingers Landing Boat Launch (right bank facing upstream). Approximately 0.4 RM upstream of Bellingers Landing (boat launch). 4.6 RM (7.4 km) above the mill effluent discharge. The Bellingers site has been located at four different gravel bars (all less than 0.9 RM of each other). The sampling site has been relocated due to accessibility difficulties or changes at the gravel bars (ie.decreasing size of gravel bar, poor flow, increase in water depth). Sampling at BELL4 began in May 2007. |
| 44°05’08.9” | 122°58’25.3” | MOHA1 | **Mohawk**<br>Located just upstream of the confluence with the Mohawk River on the right bank facing upstream. 1.1 RM (1.8 km) below the mill effluent discharge. |
| 44°04’37.7” | 123°00’39.6” | HARBP | **Harvest**<br>Located on the right bank facing upstream 0.8 RM (1.3 km) below Harvest Lane. 3.3 RM (5.3 km) below the mill effluent discharge. 1.4 RM (2.3 km) upstream of the periphyton and macroinvertebrate sampling site. |
| 44°06’59.2” | 123°04’00.0” | ARMI2 | **Armitage**<br>South bank gravel bar downstream of Armitage Park. Located on the right bank facing upstream. 1.1 RM (1.8 km) below Armitage Park. 8.3 RM (13.4 km) below the mill effluent discharge. |

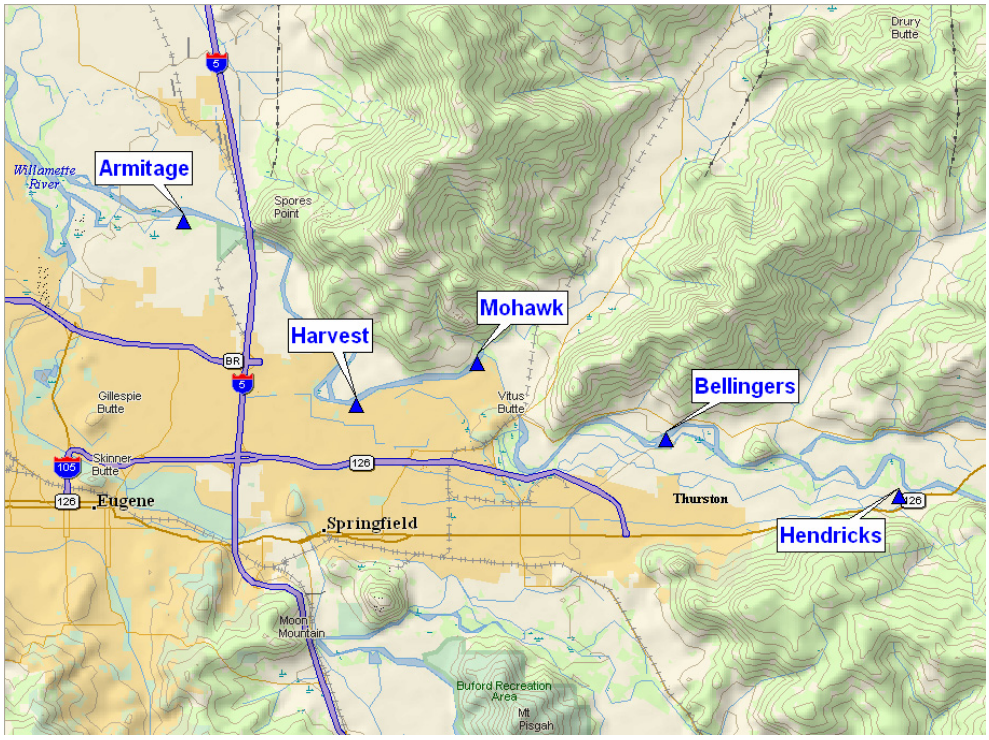


**Figure 5.7** McKenzie River Backpack Electrofishing Locations

### 5.6.4 *Willamette River*

Willamette River backpack electrofishing locations are described in Table 5.8 and shown in Figure 5.8. Additional detailed information about reaching these sample locations can be found in Section 4.3.4.

**Table 5.8** Willamette River Backpack Electrofishing Sample Locations

| N Latitude | W Longitude | Waypoint | Site Name and Location Description |
|---|---|---|---|
| 44°17'03.4" | 123°10'59.2" | HARR1 | **Harrisburg**<br>Below the city of Harrisburg on the right bank facing upstream. 12.4 RM (20 km) above the mill effluent discharge. |
| 44°19'06.3" | 123°13'03.6" | CART1 | **Cartney**<br>Across channel from McCartney Park Boat Launch on the right bank facing upstream. 9.0 RM (14.5 km) above the mill effluent discharge. |
| 44°23'14.7" | 123°13'56.9" | INTAK1 | **Intake**<br>Located above the mill water intake/outfall on the right bank facing upstream. 0.6 RM (1 km) above the mill effluent discharge. |
| 44°31'02.0" | 123°13'41.3" | PEORBP | **Peoria**<br>Located on the left bank facing upstream 4.9 RM (7.9 km) downstream of the Peoria boat launch, and 10.7 RM (17.2 km) below the mill effluent discharge. This site is accessible by car. |
| 44°31'35.8" | 123°14'58.8" | WILL2 | **Willamette Park**<br>Located on the left bank facing upstream. 12.5 RM (20.1 km) below the mill effluent discharge. |

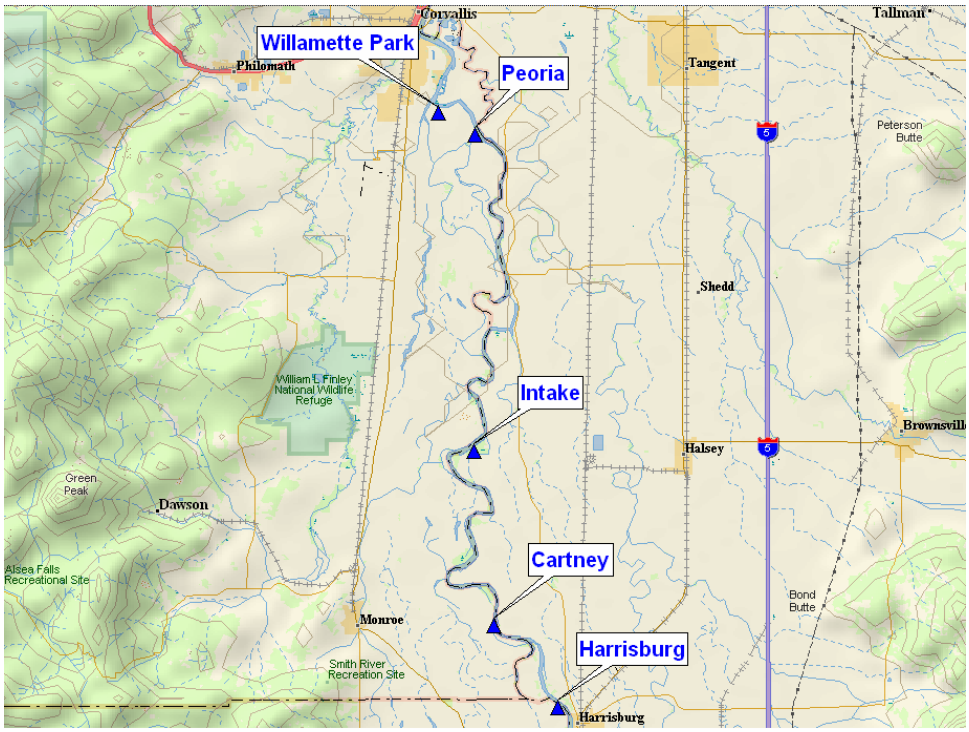


**Figure 5.8** Willamette River Backpack Electrofishing Locations

## 5.7 Electrofishing Data Sheet

The same data sheet (Electrofishing Data Sheet, Appendix J) is used for both backpack and boat electrofishing as follows:

**River**:

**Site**:

**Run**: 1, 2, or 3 (3 runs for backpack electrofishing, 1 run for boat electrofishing)

**Seconds/Distance (m):** Target of 600 s per run for backpack electrofishing
Target of 1000 s/500 to 1000 m for boat electrofishing on the Willamette and McKenzie Rivers
Target of 30 min for boat electrofishing on the Leaf River

**Crew:**

**General comments:**

**Date:**

**Time**

**Boat/Backpack: (circle one)**

**Number of netters:** (2)

**Conductivity:** Conductivity measurements are made with a Hach sensION5 (or equivalent) conductivity meter and probe. Conductivity measurements should be recorded as µS/cm. The conductivity meter should be calibrated at NABF with conductivity standards prior to use in the field.

**Water Temperature:** Temperature measurements are made with the Hach sensION5 conductivity meter (or equivalent) and are taken at the same time as the conductivity readings. Calibration is checked at NABF with a NIST traceable mercury thermometer.

**Secchi Depth:** Depth in meters at which the Secchi disk cannot be seen

**Water Clarity Comments:** Clear, turbid, cloudy, etc.

**GPS Coordinates**: GPS coordinates of backpack fishing vicinity
Beginning and ending GPS coordinates of boat electrofishing vicinity

**Species**: Species

**TL (mm):** Total length

**SL (mm**): Standard length

**Wt (g)**: Weight

**Fish External HAI**: Appendix K contains the codes for the external health assessment.

**Comments:** Identify salmonid fish as wild (adipose fin is not clipped or tagged) or hatchery (adipose fin is clipped or tagged) and adult or juvenile. Include other health assessment comments which might not be included in the health assessment categories.

### 5.8 Fish Mortality Data Sheet

Fishing permits on the Willamette and McKenzie Rivers require the reporting of fish mortalities. All species mortalities that occur while either boat or backpack electrofishing should be recorded at the conclusion of processing fish at each sample site on the Fish Mortality Data Sheet (Appendix L). These mortalities include those resulting from handling as well as from electrofishing. Record the date, river, site, species, adult or juvenile, wild or hatchery, mortality count, and method (boat or backpack electrofishing). Check to ensure that the mortalities do not exceed authorized unintentional mortality limits (Table 5.1).

### 5.9 Boat Electrofishing Method

Boat electrofishing on the Leaf River is carried out with the assistance of the staff and the electrofishing boat and equipment of either a private contract firm or the Mississippi Department of Environmental Quality (MSDEQ). A second boat (chase boat) is driven by Dr. Fred Howell, professor emeritus at the University of Southern Mississippi (USM). Arrangements for boat electrofishing are handled through Dr. Howell, who is under contract with NCASI for several aspects of data collection and sampling activities in conjunction with the Leaf River. Typically, boat electrofishing requires a three-member NABF team.

Boat electrofishing on the McKenzie and Willamette Rivers is carried out through the use of the NCASI electrofishing boat and is driven by Adam Helfrich of Helfrich River Adventures. Boat electrofishing on these rivers requires a three-member NABF team.

#### 5.9.1 *Equipment*

Aerators (2) and D batteries (8)

Balance (Ohaus Model CW-11 top loading) and C batteries (12)

Balance (A&D SK-WP Washdown Bench Scale) and D batteries (6) (Backup balance)

Balance tare weights (500 g, 1000 g, 2000 g, 5 kg)

Buckets to fill and empty live well (5-gal) (2)

Clipboard (encased) and writing implements

Conductivity meter (Hach sensION5) and backup meter (Cole Parmer CON 5)

Container for weighing fish

Data sheets on *Rite in the Rain* waterproof paper (Electrofishing Data Sheet, Appendix E)

Electrofishing boat (including anodes, generator and live well)

Electrofishing log book

External HAI Quick Reference Card

Envelopes for mountain whitefish scales (McKenzie and Willamette Rivers only)

Forceps

GPS (2)

Lineman's gloves (3 pairs)

Local or regional fish identification books, taxonomic keys and lists

Mortality data sheets (Fish Mortality Data Sheet, Appendix G)

Multi-tool

Measuring board

Nets (long handled, non-conductive) (2)

Net (medium sized to net fish out of live well)

Pesola scales, (0-20 g, 0-100 g, 0-500 g, 1000 g) (backup for the top load balance)

Secchi disk

Scientific collection permit

Turbidity meter (Hach 2100P Turbidimeter)

**5.9.2** ***Procedures***

1) Boat electrofishing locations should be found using a GPS. See Section 5.5 for site locations.

2) Measure conductivity and temperature with the Hach sensION5 conductivity meter (or equivalent) at each site location. Boat electrofishing should not occur on the McKenzie or Willamette Rivers if river temperatures exceed 70˚F (see Section 5.4.3 for permit requirements.)

3) Measure the Secchi depth and turbidity (Hach 2100P Turbidimeter) at each site location. Sampling should not be carried out when Secchi readings are <20 cm since turbid water would adversely impact catch efficiency and safety.

4) The date, site, time, method, conductivity, temperature, Secchi depth, turbidity, clarity, electrofishing settings (voltage, amperage, frequency, and duty cycle), time duration, GPS coordinates (beginning and end), sampling site descriptions (left bank, right bank), and personnel identification are recorded in the appropriate electrofishing log (McKenzie and Willamette Electrofishing Log or the Leaf River Electrofishing Log). The fish data sheets ("Electrofishing Data Sheet"—Appendix J) should also be completed at each sample location. See Section 5.7 for more data sheet details.

5) The total number of netters should always be two to provide for a uniform sample effort.

6) The live well on the boat should be filled two-thirds of the way with fresh water and aerated using two battery operated aerators. For the Leaf River, there is a live well on the electrofishing boat. Two coolers serve as live wells on the chase boat. All live wells should be aerated.

7) Channel border habitat from 0.9 to 1.6 m deep should be electrofished. Sample locations should be designated as left bank or right bank when facing upstream and these site descriptions should be included in the electrofishing log.

8) There are some differences in boat electrofishing methods between the Leaf River and the McKenzie/Willamette Rivers.

    a. For the Leaf River, private contractors or MSDEQ will be operating the electrofishing boat, and electrofishing settings will be based on their professional experience as well as on settings used in the past at each site. Two boats are utilized for boat electrofishing: an electrofishing boat which carries the primary netter, and a chase boat carrying a second netter which follows behind the electroshocking boat and nets any fish that are not captured by the primary netter. Cumulative electrofishing time is 30 min at each location.

    b. For the McKenzie/Willamette Rivers, 1000 s of cumulative electrofishing time is targeted at each sample location. The distances sampled should be at least 500 to 1000 m. These distances can be estimated by recording beginning and ending GPS positions. Consult the McKenzie/Willamette Rivers Electrofishing Log for settings used in the past at each sample site. Set the electrofishing mode on DC at 60 Hz. Amperage should range between 0.5 to 2 amp and should never exceed 2 amp. The amperage is adjusted by varying the range of the voltage. The range of voltage should be initially set at 50% of the high range. The range of voltage will vary at each site and will be adjusted as needed to achieve 0.5 to 2 amp. Start at 0.5 amp and increase the amperage, if it appears the fish are swimming away. After starting the generator and setting the timer to 0, the boat is maneuvered upstream past the upstream-most point of the run. The boat is then directed into position downstream with the current. When the upstream point of the run is reached, the boat driver will engage the electrical current to the probes and check the generator output. If settings are acceptable, then proceeding downstream with the current, the two netters will net all possible fish, transferring them to the live well. Hand signals are used to notify the boat driver when the electrical current should be shut off.

9) The actual duration of electrofishing is displayed on the electroshocker control panel and is recorded in the electrofishing log and on the “Electrofishing Data Sheet.” Depending on the number of fish captured, the electrofishing activity may be suspended during the time period to process the fish (weights, lengths, external assessments) before continuing to sample. Reset the timer on the boat shocker at the start of each sampling.

10) Samples should be sorted in the field. Fish are identified to species. External health assessment, total length, standard length, and weight are recorded on the Electrofishing Data Sheet (Appendix J). External HAI procedures are modeled on those described by Goede (1993). See the “Fish External HAI Codes” (Appendix K) for the health assessment codes.

11) Fish mortalities (all species) on the McKenzie or Willamette Rivers should be recorded at the conclusion of processing fish at each sample site on the Fish Mortality Data Sheet (Appendix L). See Section 5.8 for instructions.

12) As required by the Oregon Scientific Taking Permit, scales should be taken from all mountain whitefish mortalities on the McKenzie or Willamette Rivers. Remove five to six scales from each fish with a pair of forceps and place into a labeled scale envelope. Label the envelope with mountain whitefish, river name, site, date, and include weight and length measurements of the fish. These scale samples should be sent to Steve Jacobs, ODFW Fish Research, 28655 Hwy 34, Corvallis, OR 97333–2227.

13) All fish sampled on the McKenzie or Willamette Rivers must be released. A few representative species from the Leaf River may be collected for a reference collection (see Section 5.11).

## 5.10 Backpack Electrofishing Method

Sample locations for backpack electrofishing are provided in Sections 5.6. Samples should be collected from the vicinity of all habitat features within an identified pool/riffle complex. Due to depth limitations of the backpack electrofishing unit, depths >0.8 m should not be sampled.

### 5.10.1 *Equipment*

Aerators (2) and D batteries (8)

Backpack electrofishing nets (2)

Backpack electrofishing unit (Smith-Root LR-24, battery powered) with cathode and two anodes

Balances, digital (Scout Pro) (2) with spare batteries (4 x AA)

Batteries for backpack electrofishing unit, charged (2)

Buckets, 5-gal plastic (4)

Calibration weights for balances (100 and 200 g)

Cellular telephone

Charging unit for backpack electrofishing unit

Clipboard (encased) and writing implements

Conductivity meter (Hach) and backup meter (Cole Parmer)

Containers (2-qt, plastic) for sorting fish (8)

Data sheets on *Rite in the Rain* paper (Electrofishing Data Sheet)

Electrofishing log book

External HAI Quick Reference Card

First aid kit

Fishing licenses with trout stamps (required for Pennsylvania)

Folding table

Forceps and small rulers

GPS (2)

Instruction manual for backpack electrofishing unit (Smith-Root LR-24)

Key or lock combinations to storage lockers

Local or regional fish identification books, taxonomic keys and lists

Lid from 1 pint bottle—used to place fish on while weighing

Lineman's gloves (3 pairs)

Master Mechanic tool kit (or equivalent)

Multi-tool

Nets (small, for handling fish)

Pesola scales, 0 to 20g, 0 to 100g, 0 to 500g, 1000 g (for weighing large fish)

PFDs (3)

Secchi disk

Scientific collection permits

Tape measure (for measuring larger fish)

Turbidity meter (Hach 2100P Turbidimeter)

Waders (including backup waders) and wading boots

Wind screen for balances

**5.10.2** ***Procedures***

1) Fully charge the batteries (2) for the battery-powered electrofisher in advance of the day backpack electrofishing begins. Connect the charger to the battery and then the charger to AC power. The length of time needed for recharging depends on the amount of discharge of the battery. The charger should be allowed to complete its full cycle as indicated by the green "Ready" LED. When disconnecting the battery from the charger, unplug the AC supply to the charger first to eliminate the risk of gas explosion due to arcing. Keep batteries fully charged to increase battery life and avoid the complete discharge of the batteries. If a battery is completely discharged, the battery should be recharged as soon as possible. Batteries stored at NABF should be charged monthly. Avoid storage in exceedingly warm temperatures. A battery charger and two batteries for the battery-powered backpack electrofisher remain at the mill in Pennsylvania with Doug Brodhecker who can be called upon to charge the batteries prior to the arrival of NABF staff.

2) Measure Secchi depth, turbidity, conductivity, and temperature at mid-depth in the middle of the stream at each sample location before beginning to electrofish. Conductivity and temperature measurements may be taken with the same field meter used in recording conditions for periphyton and macroinvertebrate sample stations. Sampling should not be carried out when Secchi readings are <20 cm since turbid water would adversely impact catch efficiency and safety. Record these measurements in the electrofishing log book and the "Electrofishing Data Sheet" (Appendix J).

3) The quick setup option for the Smith-Root Model LR-24 battery powered electrofisher automatically sets the output voltage, waveform, and duty cycle based on the water conductivity (see the manual for details). It uses the default waveform of 30 Hz 12% duty cycle, and adjusts the output voltage as necessary to reach 25 W average power output. Based on the response of the fish, adjustments can be made to either increase or decrease the voltage. Turn the voltage down if it is taking the fish too long to recover (>15 s) and turn up the voltage if the fish are swimming away. If fish do not show taxis (swimming toward the anode), increase the duty cycle by 10%, press the Enter key and try again. Repeat this step until taxis occurs. If the duty cycle is increased to the maximum and no taxis occurs, reduce the cycle to 12% and increase the frequency by 10 Hz, press Enter, and try again. If

necessary, increase the voltage before increasing the frequency again. Frequency should never exceed 60 Hz. Refer to the electrofishing log book for electrofishing settings used in the past at each site.

4) The anode ring should be kept free of corrosion with a Scotch Brite pad when corrosion is evident, as indicated by discoloration and formation of oxide nodules. A backup anode ring should be taken along on each trip.

5) Sample locations should be designated as downstream, middle, and upstream at each sample site. After starting the generator and setting the timer to 0 s, sampling should begin at the downstream end of a location and proceed upstream. This is a three-person approach where one person will carry the electrofisher and the fish-holding bucket containing 5 to 10 cm of water, and the other two people will each carry a dip net. The netters will work with one person on each side of the electrofisher. All fish should be netted as the person with the electrofisher moves the anode in the vicinity of stream bank and stream bottom features, including isolated larger substrate, roots, logs or other material. Netted fish should be placed into a fish-holding container (5-gal plastic bucket) carried by the electrofisher. There should be three buckets for three runs, and each bucket should be labeled with the run number. After each run, an aerator should be added to the fish-holding container.

6) Sample time should be based on approximately 600 s (10 min) of effort for each of three runs. Record the duration of each run.

7) Record the date, time, site, method, conductivity, temperature, Secchi depth, turbidity, clarity, electrofishing settings (V, amp, W, Hz, and duty cycle), duration of each run, GPS coordinates, and personnel identification in the electrofishing log book. For each run, fill out a fish data sheet ("Electrofishing Data Sheet"—Appendix J). Note any problems encountered.

8) Fish should be sorted and identified to species in the field after all three runs are completed, and each run should be processed separately.

9) Fish are weighed, measured for total and standard length, and health assessed (external). See "Fish External HAI Codes" (Appendix K). Fish are weighed on the Scout Pro digital balance which weighs up to 200 g and reads to 0.01 g. The balances are calibrated annually by a professional, North West Instrument Services, Oak Harbor, Washington, and the certificates of calibration are on file at NABF. The Scout Pro balance has a shipping lock under the pan which is locked during transport and which must be unlocked before weighing. Calibration can be affected by changes in location, temperature, and rough handling of the balance. Before weighing fish at each site, check the calibration with the 100 g weight. The weight should read 100 ± 0.5 g. If it reads outside this range, recalibrate using the linear calibration method which requires a 100 and 200 g weight. See the instruction manuals for calibration instructions. Do not store equipment on top of the balance during transport and always have a second balance on hand along with extra batteries. Larger fish will need to be weighed using the Pesola scales.

10) Fish identification, external health assessment, standard length, total length, and weight are recorded on the "Electrofishing Data Sheet" (Appendix J).

11) For the McKenzie and Willamette Rivers, fish mortalities (all species) should be recorded at the conclusion of processing fish at each sample site on the "Fish Mortality Data Sheet (Appendix L). See Section 5.8 for details.

11) All fish are released except for those collected for a reference collection (see Section 5.11).

### 5.11 Fish Reference Collection

A few representative fish species from Codorus Creek and the Leaf River are collected and preserved for the reference collection maintained at NABF. The Oregon collecting permits do not allow fish to be kept for a reference collection. Fish are preserved in the field using 10% formalin after they have been identified, and lengths, weights and external HAI assessments have been recorded.

#### 5.11.1 *Equipment*

Alka-Seltzer tablets

Bottles, plastic screw top, (1-pt) (12)

Borax

Bubble wrap for shipping containers

Clipboard (encased) and writing implements

Containers, plastic (2 qt.) for anesthetizing fish

Data sheets on *Rite in the Rain* waterproof paper ("Electrofishing Data Sheet") (50)

Electrical tape

Eyewash (portable)

Formalin (37–40% solution) (1-L bottle)

Garbage bags (double bag fish vouchers in action packers) (2)

Gloves, nitrile (for working with formalin)

Markers (waterproof Sharpies)

Safety glasses

Secondary containment bucket for formalin

Shipping container for samples, 8-gallon Rubbermaid Action Packer (1)

Spill pillows

Teaspoon

Zip ties

#### 5.11.2 *Procedures*

1) The CHSP should be consulted for safe handling procedures for formalin. Formalin is a known carcinogen and is extremely toxic. Avoid breathing the fumes and always wear safety glasses and nitrile gloves when working with formalin. Use secondary containment vessels for formalin transport. Spill pillows should be stored in the secondary containment bucket. A field eyewash bottle should also be available, and its expiration date should be checked on each trip. Sample bottle lids should be secured with electrical tape prior to shipment.

2) Formalin oxidizes to make formic acid, which will decalcify bones and eventually cause the specimen to become brittle. To prevent these problems, buffer the formalin. Add one tsp of household borax to a 1-L bottle of stock 36 to 38% formalin. Avoid an excess of borax buffer since that might produce an edema-like puffiness.

3) Use a permanent marker to label the sample bottle and lid with the river name, site name, and date collected.

4) Place the specimens in a 2-qt plastic container with river water and add several Alka-Seltzer tablets to make the fish drowsy.

5) Fill the 1-pt plastic sample bottle to the 50 ml mark with the buffered formalin (36–38%). The bottles are pre-marked at NABF with a 50 ml mark and a 500 ml mark.

6) Place the anesthetized fish into the sample bottle. Do not crowd the fish.

7) Fill the bottle to the 500 ml mark with river water to make a 10% formalin solution.

8) Secure the lid with electrical tape and gently mix the bottle.

9) All fish vouchers are transported to NABF via FedEx Ground. Line the bottom of the shipping container (Rubbermaid Action Packer) with bubble wrap. For added containment, place sample bottles into double garbage bags. Add enough bubble wrap so the samples remain upright during shipment. Information including NABF address and phone number should be placed inside the shipping container so that in the event of shipment loss or misplacement, there is a better opportunity of sample recovery. FedEx shipment tracking numbers should be retained in order to track the packages.

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# APPENDIX A

## NCASI LTRWS Water Quality Monitoring Field Data Sheet:

## Codorus Creek

Date: _____

Data collected by: _____

Weather conditions: _____

_____

| Station Name | Time | pH | Temperature ($^{o}$C) | Conductivity (µS/cm) |
|---|---|---|---|---|
| Oil Creek | | | | |
| Menges | | | | |
| USGS Station | | | | |
| Martin | | | | |
| Graybill | | | | |
| Furnace | | | | |

Comments (River level, turbidity, floating debris, etc.):

_____

_____

_____

_____

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## NCASI LTRWS Monthly Temperature and pH Calibration Check

## Codorus Creek

Date: _____ Analyst: _____

Time: _____

| Calibrated Thermometer Reading (˚C) | Field Meter Temperature Reading (˚C) |
|---|---|
| | |

pH Meter: Orion Model 250A+

| pH Standards | | | Slope |
|---|---|---|---|
| 7.00 | 10.01 | 4.01 | |
| | | | |

# APPENDIX B

## NCASI LTRWS Water Quality Monitoring Field Data Sheet:

## Leaf River

Date: _____

Data collected by: _____

Weather conditions: _____

_____

| Station Name | Time | pH | Temperature ($^{o}$C) | DO (mg/L) | Conductivity (µs/cm) |
|---|---|---|---|---|---|
| Tallahala Creek Bridge | | | | | |
| Mahned Boat Launch | | | | | |
| New Augusta Bridge | | | | | |
| Wingate Bridge | | | | | |
| Bogue Homo Creek | | | | | |
| Beaumont Bridge | | | | | |
| McLain Bridge | | | | | |

Comments (River level, turbidity, floating debris, etc.):

_____

_____

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# APPENDIX C

## NCASI LTRWS Water Quality Monitoring Field Data Sheet:

## McKenzie River

Date: _____

Data collected by: _____

Weather conditions: _____

_____

| Station Name | Time | pH | Temperature ($^{o}$C) |
|---|---|---|---|
| Hendricks Bridge | | | |
| Bellingers Landing | | | |
| Hayden Boat Launch | | | |
| Mohawk River | | | |
| Harvest Boat Launch | | | |
| Armitage Park | | | |

Comments (River level, turbidity, floating debris, etc.):

_____

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# APPENDIX D

## NCASI LTRWS Water Quality Monitoring Field Data Sheet:

## Willamette River

Date: _____

Data collected by: _____

Weather conditions: _____

_____

| Station Name | Time | pH | Temperature ($^{o}$C) |
|---|---|---|---|
| Harrisburg Boat Launch | | | |
| Cartney Boat Launch | | | |
| Peoria Park Launch | | | |
| Willamette Park Launch | | | |
| Long Tom River | | | |

Comments (River level, turbidity, floating debris, etc.):

_____

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# APPENDIX E

## NCASI LTRWS Monthly Temperature and pH Calibration Check

Date: _____ Analyst: _____

Time: _____

| Calibrated Thermometer Reading (˚C) | Field Meter Temperature Reading (˚C) | pH Slope |
|---|---|---|
| | | |

Notes:

- Calibrations should be made within 24 h prior to sampling.
- pH slope following calibration should be -58 $\pm$ 3 mV.
- Temperature readings of the field meter should be within 0.5˚C of the calibrated thermometer reading.

# APPENDIX F

## PERIPHYTON AND MACROINVERTEBRATE SAMPLING DATA SHEET

| River: | | Site Name: |
|---|---|---|
| Date:<br>Time: | Latitude of sample location:<br>_____°_____'_____" | Longitude of sample location:<br>_____°_____'_____" |
| Sampling Team: | | Bank width (m):<br>(water edge to water edge, out from pin) |
| Position of sample from "pin":<br>(approximate distance from GPS pin and upstream/downstream direction) | | |

**Site Observations**

| *Sky* | *Wind* | *Precipitation* |
|---|---|---|
| Clear _____ Partly cloudy_____<br>Overcast _____ | Speed (mph) _____<br>Direction _____ | Fog _____ Drizzle_____<br>Rain _____ Snow _____ |

| *Water Clarity and Aesthetics* | | *Riparian Shading* | *Conductivity, Temperature, Turbidity* |
|---|---|---|---|
| Clear _____ | Turbidity _____ | Exposed _____ | Conductivity (µS/cm) _____ |
| Color _____ (none) | Foam _____ | Partial Shade _____ | Temperature (˚C) _____ |
| _____ (green) | Odor _____ | Shaded _____ | Turbidity (NTU) _____ |
| _____ (brown) | | | |

Light—PAR (µmol m $s^{-1}$ $m^{-2}$)

Multiplier in air = 210.08 Multiplier in water = 277.31

Air _____ Water_____

Note: all depths should be standardized to 6". Measurements should be made when the sky is either completely clear or completely overcast.

**Periphyton Samples (Rocks should be from 20 to 40 cm depth)**

**Quantitative (Chlorophylls)**

| | Rock 1 | Rock 2 | Rock 3 | Rock 4 | Rock 5 |
|---|---|---|---|---|---|
| Length (mm) | _____ | _____ | _____ | _____ | _____ |
| Width (mm) | _____ | _____ | _____ | _____ | _____ |
| Total vol. (ml) | _____ | _____ | _____ | _____ | _____ |
| Filt. vol. (ml) | a _____ | a _____ | a _____ | a _____ | a _____ |
| | b _____ | b _____ | b _____ | b _____ | b _____ |

**Qualitative** (Taxonomy) 20 ml of slurry from each rock for a total of 100 ml per station

**Observations**

**Periphyton:**

**General:**

# APPENDIX G

## BENTHIC MACROINVERTEBRATE SAMPLING AND CURRENT VELOCITY MEASUREMENT DATA SHEET

| RIVER: | SITE NAME: |
|---|---|
| Date and Time: | Sampling Team: |

| Sampling Gear:<br>Hess—All Rivers<br>Surber—back-up | Mesh Size (μm):<br>Hess—243<br>Surber—250 | Sample time: 2–3 min processing time | Five replicates |
|---|---|---|---|

Substrate Characteristics—%

silt____ sand _____ gravel_____ cobble_____ boulder____ bedrock____

Note:
Current velocity—Probe should be 10 cm (4 inches) above substrate
Water depth for Hess samples should be between 20 and 40 cm (8 to 15.5 inches)

| | Replicate 1 | Replicate 2 | Replicate 3 | Replicate 4 | Replicate 5 |
|---|---|---|---|---|---|
| Depth (in) | _____ | _____ | _____ | _____ | _____ |
| Velocity (m/s) | _____ | _____ | _____ | _____ | _____ |
| Process Time (min) | _____ | _____ | _____ | _____ | _____ |

Note: Replicate number for macroinvertebrates should correspond with that of the periphyton rock sampled at the same location.

General Comments/Observations, including any large or rare taxa:

# APPENDIX H

## LEAF RIVER PERIPHYTON AND MACROINVERTEBRATE SAMPLING DATA SHEET

**Site Information (based on location of periphyton natural substrate beach sample site)**

| | | |
|---|---|---|
| Site name: | Date:<br><br>Time: | Sampling Team: |
| Latitude of sample location<br>_____ º _____ ' _____ " | Longitude of sample location<br>_____ º _____ ' _____ " | Bank Width (m):<br>(water edge to water edge) |

**Site Observations (based on location of periphyton natural substrate beach sample site)**

| | | |
|---|---|---|
| Sky<br>Clear _____ Partly cloudy _____<br>Overcast _____ | Wind<br>Speed (mph) _____<br>Direction _____ | Precipitation<br>Fog_____ Drizzle_____<br>Rain_____ Snow_____ |
| Water Clarity and Aesthetics<br>Clear ____ Turbid _____<br>Color ____ (none) Foam _____<br>____ (green) Odor _____<br>____ (brown) | Riparian Shading<br>Exposed _____<br>Partial shade _____<br>Shaded _____ | Substrate Characteristics—%<br>silt ____ sand ____<br>gravel ____ cobble ____<br>boulder ____ bedrock ____ |
| Conductivity,Temperature,Turbidity<br><br>Conductivity (µS/cm) _____<br>Temperature (ºC) _____<br>Turbidity (NTU) _____ | Current Velocity (probe should be 10 cm (4 inches) above substrate)<br><br>Depth (m) _____<br>Velocity (m/sec) _____ | |
| Position of sample location from "pin":<br>(approximate distance from GPS pin and upstream/downstream direction) | | |
| Light—PAR (µmol m $s^{-1}$ $m^{-2}$)<br>Multiplier in air = 210.08 Multiplier in water = -277.31<br>Air _____ Water _____<br>Note: all depths should be standardized to 6 inches. Measurements should only be made at times when sky is either completely clear or completely overcast. | | |

**Sand Periphyton Samples**

| |
|---|
| **Qualitative (taxonomy)**—3 scoops composited to one and preserved with Lugol's Solution |

**Observations**

| |
|---|
| **Periphyton** (filamentous beds, sloughing, bleached algae, macrophytes, moss, etc.):<br><br>**General** (livestock, fish mortalities, bank erosion, insect hatches, surface sheet, etc.): |

| LEAF RIVER PERIPHYTON AND MACROINVERTEBRATE SAMPLING<br>DATA SHEET<br>PAGE TWO | | |
|---|---|---|
| Site name: | Date:<br><br>Time: | Sampling Team: |

**Macroinvertebrate—HD Samples**

| | | |
|---|---|---|
| Latitude of sample location:<br><br>_____ º _____ , _____ ” | Longitude of sample location:<br><br>_____ º _____ , _____ ” | Conductivity (µS/cm) Temperature (˚C) Turbidity (NTU)<br><br>_____ _____ _____ |
| Pool Depth (cm): | HD Depth (cm)(@upper HD): | Current Velocity (m/sec)(@upper HD): |
| Date of HD deployment: | Number of HDs deployed: | Number of HDs harvested: |

**Macroinvertebrate—Natural Substrate Sample**

| | | |
|---|---|---|
| Latitude of sample location (only if different from periphyton native sand sample location)<br><br>_____ º _____ , _____ ” | Longitude of sample location<br><br>_____ º _____ , _____ ” | Description of substrate sampled (log, stick, branch—approximate size):<br><br>Time (if < 10 min.) _____ |

**Periphyton Samples from HD Plates**

| **Quantitative (chlorophylls)** | Plate 1 | Plate 2 | Plate 3 |
|---|---|---|---|
| Total volume (ml) | _____ | _____ | _____ |
| Filtered volume (ml) | a_____ | a_____ | a_____ |
| | b_____ | b_____ | b_____ |

**Qualitative (taxonomy):** 20 ml of slurry from each HD top plate for a total of 60 ml per station

## APPENDIX I

## CODORUS CREEK TEMPERATURE LOGGER LOCATION DATA SHEET

| Site | Serial# | Date/Time | Latitude | Longitude | GPS Waypoints | Site Description |
|---|---|---|---|---|---|---|
| Furnace | | | | | | |
| Graybill | | | | | | |
| Martin | | | | | | |
| USGS | | | | | | |
| Menges | | | | | | |

# APPENDIX J

## ELECTROFISHING DATA SHEET

Page _____ of _____

**River:**
**Site:**
**Run:**
**Seconds/Distance(m):**
**Crew:**
**General Comments:**

**Date:**
**Time:**
**Boat/Backpack:**
**Number of Netters:**
**Conductivity:**
**Water Temperature:**

**Secchi depth:**
**Water Clarity Comments:**
**GPS Coordinates:**
**Latitude:** _____º _____' _____"
**Longitude:** _____º _____' _____"

| Species | TL (mm) | SL (mm) | Wt (g) | *Deformities* | *Petechiae* | *Eyes* | *Fins* | *Gills* | *Lesions* | *Opercles* | *Parasites* | *Tumors* | *Vent* | Comments (MUST include wild/hatchery and adult/juvenile for all salmonids) |
|---|---|---|---|---|---|---|---|---|---|---|---|---|---|---|
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# APPENDIX K

## FISH EXTERNAL HAI CODES

| Deformities | |
|---|---|
| H | Head |
| V | Vertebrae |
| B | Body |

| Petechiae | |
|---|---|
| M | Mild petechiae |
| S | Severe petechiae |

| Eyes | |
|---|---|
| S1, S2 | Swollen |
| H1, H2 | Hemorrhagic—bleeding |
| B1, B2 | Blind—opaque |
| M1, M2 | Missing |
| OT | Other |

| Fins | |
|---|---|
| M | Mild Erosion |
| S | Severe erosion |

| Gills | |
|---|---|
| F | Frayed—ragged appearance |
| C | Clubbed—swollen lamellae tips |
| M | Marginate—light distal margin |
| P | Pale— entire lamellae pale |

| Lesions | |
|---|---|
| M | Mild—few lesions |
| S | Severe—many lesions |

| Opercles | |
|---|---|
| M | Mild shortening |
| S | Severe shortening |

| Parasites | |
|---|---|
| M | Mild—few parasites |
| S | Severe—many parasites |

| Tumors | |
|---|---|
| M | Mild—few tumors |
| S | Severe—many tumors |

| Vent | |
|---|---|
| I | Inflamed (swollen & red) |
| R | Red |
| S | Swollen |

# APPENDIX L

## FISH MORTALITY DATA SHEET

| Date | River | Site | Species<br>Adult (A) or Juvenile (J)<br>Wild (W) or Hatchery (H) | Mortality Count | Method: Boat or BP |
|---|---|---|---|---|---|
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