# APPENDIX F

# SURBER-SAMPLER PROTOCOLS

## Sample Collection

The objective of Surber sampling is to collect a minimum of three replicate Surber samples, and to remove and preserve all individual benthic macroinvertebrates from each replicate sample.

## Records

In addition to other sample-labeling requirements specified in this appendix, maintain the following records.

### *Field Logbook*

For each sample event, enter all relevant information in the field logbook, including the date and time of sample collection, the location of the sample site (station ID), the name of each collector, the method of collection, the number and type of samples collected, the number of sample containers, and the preservative used.

### *Sample-Tracking Logbook*

Maintain a sample-tracking logbook that contains the information described in Chapter 11. This logbook documents when samples arrive at the laboratory or headquarters facility, when each sample enters each processing step, and who has custody or responsibility for it.

### *Laboratory Bench Sheets*

Maintain laboratory bench sheets where specimen identification and enumeration occur. These sheets document the raw counts of individuals for each taxon and contain notes relevant to identification and enumeration.

## Where to Collect Samples

Collect samples only in riffle-type habitats with depths < 0.3 m. If there are multiple riffles within a reach, inspect and evaluate each for substrate characteristics and microhabitat heterogeneity. Evaluate substrate characteristics according to the following priorities:

1. cobble and gravel
2. debris jams
3. sand
4. bedrock

For example, if one riffle among several riffles in a reach contains primarily cobble and gravel substrate and all the rest contain primarily bedrock, collect the sample in the riffle that contains cobble and gravel substrate. If all of the riffles contain primarily bedrock or sand, inspect each one for available microhabitats, such as pockets of gravel or debris jams. If these types of microhabitats are present, collect the sample from one or more riffles, spending most of the sampling time in these microhabitats.

**Consider alternate sampling methods if the riffles are essentially homogeneously bedrock or sand.** For example, the runs and glides in the reach must be evaluated as potential alternative candidate habitats for collecting either RBP snag or RBP kicknet samples. See Chapter 5 for details.

# Collecting a Surber Sample

To collect a Surber sample, firmly push the sampler down on the substrate with the net mouth facing upstream. Lift larger rocks individually and scrub them off at the mouth of the net. Thoroughly disturb the remaining sediment by repeatedly digging and stirring as deeply as possible, allowing the current to sweep organisms and detritus into the bag net. Collect and preserve a total of three individual replicate Surber samples by following these procedures for each replicate.

Collect the three replicates in a manner that represents the longitudinal and cross-sectional heterogeneity of the riffle. For example, collect the first replicate at the lower end of the riffle a suitable distance away from the right bank; collect the second midstream about halfway up the riffle; and collect the third a suitable distance from the left bank at the upper end of the riffle. If the riffle is large, it may be desirable to establish transects and use a random-number generator to decide where to locate each replicate.

## *Field Processing and Preserving a Surber Sample*

The objective of a Surber sample is to count and identify every individual benthic macroinvertebrate collected in a known area. Since sorting usually takes several hours to several days, it must be done in the laboratory. Inspect the complete sample under magnification to ensure that all individuals are counted and identified.

Transfer the entire sample from the net to one or more sample containers and preserve it in 10 percent formalin—one part full-strength formalin and nine parts water. Alternatively, if the sample is to be sorted soon after reaching the laboratory, it may be preserved in 95 percent ethanol.

Use enough preservative to cover the sample. To ensure adequate preservation of benthic macroinvertebrate collections, do not fill sample containers more than half full of sample. The amount of preservative should at least equal the volume of organic material, including detritus. If there is too much organic matter in the jar, the sample may begin to decompose before processing.

### Safety

Avoid breathing formalin fumes! Formalin is corrosive to the eyes, skin, and respiratory tract. Wear safety glasses and latex gloves when working with this suspected carcinogen. Always work in a well-ventilated area or under a hood when preparing formalin solutions.

Alcohol is highly flammable. Take care in storage and handling.

Check the safety data sheet for alcohol and formalin solutions for proper handling requirements.

### Labeling the Sample Container

Place a label in each sample container that includes, at minimum, the following information. Use pencil or waterproof ink on paper with a high rag content for each label.

- station number and location description
- date and time of collection
- collection method (for example: Surber or quantitative snag)
- sample-container replicate number (for example: *1 of 3*, *2 of 3*, or *3 of 3*), if needed
- preservative used
- name of each collector

Repeat this labeling process for each individual replicate sample. For example, if three replicate Surber samples or quantitative snag samples are collected at a site, there must be three separate jars or three separate sets of jars with a single jar or set of jars corresponding to an individual replicate sample.

# *Tracking Requirements for Surber Samples*

Upon return to the laboratory, assign a unique sample tracking number to the jars containing the Surber or snag samples, according to the sequence in the sample tracking logbook. For example, an instance of numbering may look like *BM 040 13*, where *BM* refers to 'benthic macroinvertebrate,' *040* refers to sample number 40, and *13* refers to the year 2013.

The sample log will contain the following information.

- sample tracking number
- collection date and time
- station number and location description
- name of each collector
- collection methods
- name of person conducting subsampling procedure in the lab, if different from collector
- number of jars in the sample

Once the sample tracking number has been assigned, affix a label with the number to the outside of the container. Wrap the label with clear tape to ensure that the label will not come off. Do not put the label on the container lid.

# *Laboratory Processing for Surber Samples*

The objective of processing a benthic macroinvertebrate sample in the laboratory is to count and identify every individual benthic macroinvertebrate collected in the Surber sampler. Process each of the three replicate samples individually. Place all of the individual benthic macroinvertebrates from each replicate Surber sample in a separate vial. Once sorting is complete, there will be three separate vials, each containing all of the specimens from each individual replicate. This sorting is critical because it allows for evaluation of the variability between replicates.

## Rinsing a Sample

Thoroughly rinse the sample using a No. 30 or smaller (≤ 595 µm) sieve to remove preservative and fine sediments. Place rinsed sample in a shallow white pan.

## Safety

To reduce your exposure to formalin, rinse the sample with water in a sieve with mesh size ≤ 595 µm under a vent hood, or—if a hood is not available—in an area with good ventilation. Transfer it to alcohol before sorting. Follow your organization's plan for collection and disposal of hazardous formalin and alcohol waste.

## Sorting a Sample

Put 1 to 2 cm of water in the bottom of the pan to disperse the contents as evenly as possible. Pick **all macroinvertebrates** visible to the unaided eye from the sample and place them in a sample bottle or vial containing 70 percent ethanol or isopropyl alcohol.

After thoroughly inspecting the sample and removing all macroinvertebrates visible to the unaided eye, place small portions of the remaining sample in a petri dish and inspect them using a dissecting scope. Repeat this process until you have inspected the entire replicate sample under magnification.

## Labeling a Sample

Label the sample bottle or vial containing the benthic macroinvertebrates obtained using this procedure with the following information. Use pencil or waterproof ink on paper with a high rag content for each label.

- station number and location description
- sample tracking number
- date and time of collection
- collection method (for example, Surber, snag, Ekman dredge)
- container-replicate number (for example, *1 of 3*, *2 of 3*, *3 of 3*)
- preservative used
- name of each collector
- name of person subsampling, if different from collector

Repeat this labeling process for each of the three replicate samples.

Affix a label with the sample-tracking number and container-replicate number to the outside of the container. Make sure the container is dry, and wrap it with clear tape to ensure the label will not come off. Do not put the label on the container lid.

# Quantitative Snag Samples

The method for collecting snag samples, as described below, is the primary collection method in riffles or runs when the predominant substrate type is sand or silt.

## *Selecting Snags*

Snags are submerged pieces of woody debris (for example, sticks, logs, or roots) that are exposed to the current. Optimally, snags are 0.5 to 2.5 cm in diameter and have been submerged in the stream for a minimum of two weeks. Moss, algae, or fungal growth on the snags can be taken as evidence that the snag has been in the stream long enough to allow colonization by benthic macroinvertebrates.

## *Collecting a Sample*

For quantitative snag samples, collect woody debris accumulated in debris piles or jams **in areas exposed to good flow**. Use lopping shears to cut off sections of the submerged woody debris. The section should be of a length appropriate to fit in a 1 qt mason jar. Avoid depositional zones (pools) and backwater areas. Place a D-frame net immediately downstream of the snag, while cutting, to minimize loss of macroinvertebrates. Place snag samples directly into the mason jars containing 10 percent formalin. Collect enough snag material to fill two 1 qt mason jars.

## *Laboratory Processing for Quantitative Snag Samples*

Using a squirt bottle, wash the surface of the snags and collect the dislodged benthic macroinvertebrates and associated debris in a sorting tray. Carefully inspect the snag, including cracks, crevices, and under loose bark, for any remaining macroinvertebrates. Use a dissecting microscope, if necessary, to ensure that all organisms are removed from the snags. A soft-bristled brush may be appropriate for removing the macroinvertebrates from the snag surface, taking care not to damage the organisms. Once all macroinvertebrates are removed from the snags, follow the procedures outlined in "Laboratory Processing for Surber Samples," above.

Before discarding snags, measure their length and diameter of the snags in order to calculate their surface area. This allows expression of the results as individuals per unit area of snag surface.

## *Laboratory Procedures for Identification of Specimens Collected Using a Surber Sampler or Quantitative Snag Samples*

Use all appropriate references, a stereo dissecting microscope, and a compound phase-contrast microscope to identify organisms to the appropriate taxonomic level listed below.

- Insecta, identify to genus, except leave Chironomidae at family
- Oligochaeta, leave at Oligochaeta
- Hirudinea, leave at Hirudinea
- Hydracarina, leave at Hydracarina
- Isopoda, identify to genus
- Amphipoda, identify to genus
- Nematoda, leave at Nematoda
- Ostracoda, leave at Ostracoda
- Palaemonidae, identify to genus
- Cambaridae, leave at Cambaridae
- Gastropoda, identify to genus
- Turbellaria, identify to family
- Pelecypoda, identify to genus

**Maintain a separate count of individuals and list of taxa for each replicate Surber sample to allow an evaluation of variability between replicates**.

Chapter 11 gives complete required and recommended references for identifying freshwater macroinvertebrates.

# *Data Evaluation for Surber Samples*

Calculation of the BIBI is based upon the combined results, counts, and number of individuals from all three replicates. Evaluate data in accordance with the draft BIBI metric criteria as shown in Table F.1. The BIBI criteria were derived for three bioregions (central, east, and north) that overlap ecoregions as defined by Omernik and Gallant (1987). Figure F.1 illustrates the three bioregions, whose boundaries all coincide with ecoregion lines:

**Central bioregion.** The region composed of Ecoregions 23, 24, 27, 29, 30, 31, and 32, which includes a disjunct portion of Ecoregion 27 in the Texas panhandle and an isolated fragment of Ecoregion 32 in southeastern Texas.

**East bioregion.** The region encompassing Ecoregions 33, 34, and 35.

**North bioregion.** The region consisting of Ecoregions 25 and 26.

The BIBI was designed for definitive evaluation of quantitative data on benthic macroinvertebrates and is applicable for lotic-erosional habitats under low-flow hydrological regimes. Regional criteria include 11 metrics that integrate structural and functional attributes of macroinvertebrate assemblages to assess biotic integrity. The method was designed to determine ALUs using a Surber sampler. Report metric scoring on a form as shown in Appendix C or on a comparable form.

The draft criteria set includes the following 11 metrics.

1. **Taxa richness.** This metric is the total number of benthic macroinvertebrate taxa. Macroinvertebrates are identified to the lowest taxonomic level possible, generally genus or species, and the number of taxonomic categories are counted. In general, relatively lower taxa richness values reflect lower biotic integrity. Decreases in taxa richness may result from disturbance of physicochemical factors.

2. **Diptera taxa.** This metric is the total number of benthic macroinvertebrate taxa within the order Diptera. It reflects the condition of the most ecologically diverse insect order in aquatic ecosystems. This metric usually reflects the order with the highest number of species present. The Diptera taxa usually increase with increasing perturbation.

3. **Ephemeroptera taxa.** This metric is the total number of benthic macroinvertebrate taxa within the order Ephemeroptera. It reflects the status of one of the more environmentally sensitive aquatic insect orders, making it a valuable indicator of ambient conditions. A decrease in Ephemerpotera taxa usually indicates increasing stream perturbation.

4. **Intolerant taxa.** This metric is the total number of intolerant benthic macroinvertebrate taxa. Analysis of tolerance and intolerance conforms to the protocol of Fore et al. (1996), where the most and least tolerant taxa are used. The tolerant-taxa metric is expressed as a percentage of total abundance, and the intolerant-taxa metric as taxa richness—the optimal approach according to Karr et al. (1986) and Fore et al. (1996). Designation of tolerant and intolerant taxa is based primarily on information in Lenat (1993), as outlined in Table B.6, Appendix B. Tolerant taxa are defined as those having tolerance values $\geq 8.5$, and

intolerant taxa, values ≤ 4.0. This metric embodies the axiom that sensitive organisms seldom are numerically abundant, yet their presence provides valuable insight into environmental suitability (Fore et al. 1996).

5. **Percent EPT taxa.** This metric is the ratio of the number of individuals within the orders Ephemeroptera (mayflies), Plecoptera (stone flies), and Trichoptera (caddis flies) to the total number of individuals in the sample multiplied by 100. In general, this metric tends to decrease with increasing disturbance of physicochemical factors, as most taxa in these orders are pollution sensitive.

6. **Percent Chironomidae.** This metric is the ratio of the number of individuals in the family Chironomidae to the total number of individuals in the sample multiplied by 100. Chironomidae are relatively ubiquitous in aquatic habitats, and many of the species are facultative or pollution tolerant. Excessive representation within the community often reflects environmental perturbation.

7. **Percent tolerant taxa.** This metric is the ratio of the number of individuals classified as tolerant taxa to the total number of individuals in the sample multiplied by 100. See Table B.6, Appendix B. Refer to the intolerant-taxa metric (no. 4) for further discussion.

8–10. **Percent grazers, percent gatherers, and percent filterers.** This metric is the ratio of the number of individuals in the grazer, gatherer, and filterer FFGs to the total number of individuals in the sample multiplied by 100. Community trophic structure is assessed following the convention of Minshall (1981), in which six FFGs are used:

- **grazers**—scrapers of periphyton, piercers of living macrophyte tissues or filamentous algal cells
- **gatherers**—gatherers of deposited FPOM
- **filterers**—filterers of suspended FPOM
- **miners**—burrowers in deposited FPOM
- **shredders**—chewers, miners, and borers of living macrophyte tissues or CPOM
- **predators**—piercers, engulfers, and parasites of living animal tissues

FFG assignments are mainly based on information in Merritt and Cummins (1996)—insects—and Pennak (1989)—non-insects. Some investigators employ only five FFGs, typically lumping gatherers and miners into a single group (collector-gatherers). For the present index, gatherers and miners are treated separately to maximize functional feeding resolution. Taxa categorized as collector-gatherers by Merritt and Cummins (1996) are differentiated on the basis of described habit. Taxa having habits other than burrowing (sprawling, climbing, clinging) are considered gatherers; burrowers are regarded as miners. For some taxa, the literature presents multiple indications for trophic relationships and habit. In these cases, the number of individuals in the taxon was apportioned among appropriate FFGs.

11. **Percent dominance**. The ratio of the number of individuals in the three most abundant taxa to the total number of individuals in the sample multiplied by 100. In general, domination of a community by relatively few taxa may indicate environmental stress, and a high-percentage contribution by a few taxa often represents an imbalance in community structure.

# *Pool, Reservoir, or Lake: Protocols for Sampling by Ekman, Ponar, Petersen, or Van Veen Dredge*

Methodologies for assessing ALUs have not been developed for Texas depositional habitats such as reservoirs and pools. Any private group, such as a consulting firm, considering an assessment using reservoir or pool benthic macroinvertebrates must consult closely with TCEQ personnel before planning the study.

# *Sample-Collection Procedures*

The Ekman dredge is the preferred tool for collecting benthic macroinvertebrate samples from lentic or depositional habitats, such as pools or reservoirs whose bottom is primarily mud, silt, or fine sand. Use of the Ekman dredge is considered quantitative sampling and collection and processing should be similar to those for Surber samples. In pools or reservoirs with substrates composed of gravel, hard sand, or clay, a Ponar or Van Veen dredge may be necessary. Before using any of these devices, inspect it carefully to ensure that all parts are in good operational condition. The following collection methods refer to the Ekman dredge but, with only minor exceptions, apply to the Ponar and Van Veen as well.

## Collecting a Sample

Collect a minimum of four Ekman dredge samples, each placed and preserved in a separate sample container, according to the following procedures.

Before collecting the sample, thoroughly rinse the dredge in ambient water. Once it has been cleaned, use the line (or pole in shallower pools) to lower the dredge to the bottom. Avoid lowering the sampler too rapidly, as this could cause a pressure wave that can disturb the topmost sediment or give a directional signal to invertebrates capable of retreating from the sample area. Once the Ekman reaches the bottom, and you have determined that the line is vertical and taut, drop the messenger. After the dredge jaws are triggered, retrieve the closed dredge at a moderate speed (< 1 m/sec). At the water's surface, make sure the jaws are closed and the surface layer of fine silt is intact. Water must cover the sediment sample in the dredge. Do not drain the water off, as this may cause the loss of organisms. Bring the dredge on board and empty it into a large container, such as a large plastic tub. Collect the remaining replicates in the same way, placing each into a separate tub.

## Rinsing the Sample

Insert the dredge into the mouth of a bucket with a No. 30 or smaller sieve (mesh size ≤ 595 µm) and open the jaws of the dredge to allow all of the material collected in it to fall into the sieve bucket. It may be necessary to rinse any remaining material into the sieve. After rinsing, thoroughly inspect the Ekman and place any remaining invertebrates or other material contained in it in the sieve bucket. Wash fine sediments from the sample by submerging the mesh of the sieve bucket in the pool or reservoir and gently wash it, taking care to minimize destruction of soft-bodied organisms.

Repeat this process three more times to produce a total of four separate replicate benthic macroinvertebrate samples.

## Preserving the Sample

Empty the washed contents of the sieve bucket into a clean, wide-mouthed bottle. Transfer the entire sample from the sieve bucket to one or more sample containers and preserve it in

10 percent formalin. Alternatively, if the sample is to be sorted shortly after reaching the laboratory, preserve it in 95 percent ethanol.

Use enough preservative to cover the sample. To ensure adequate preservation of benthic macroinvertebrate collections, fill sample containers no more than half full with the sample, so the amount of preservative is at least equal to the volume of organic material, including detritus. Avoid placing too much sample in one jar. If there is too much organic matter in the jar, the sample may begin to decompose before processing.

## Labeling the Sample Container

Place a label in each sample container that includes, at minimum, the following information.

- station number and location description
- date and time of collection
- collection method (for example, Ekman or Van Veen dredge)
- container replicate number (for example, *1 of 4* or *2 of 4*)
- preservative used
- name of each collector

Repeat this labeling process for each individual replicate sample. If four replicate Ekman dredge samples are collected at a site, there must be four separate jars or four separate sets of jars. Each jar, or set of jars, corresponds to each individual replicate. Upon returning to the laboratory, assign a unique sample tracking number to each individual replicate sample according to the sequence in the benthic macroinvertebrate sample-tracking logbook. Follow the procedures outlined in "Tracking Requirements for Surber Samples."

# *Laboratory Procedures for Identification of Specimens Collected in Pools or Reservoirs Using an Ekman, Ponar, or Van Veen Dredge*

Use all appropriate references, a stereo dissecting microscope, and a compound phase-contrast microscope to identify organisms to the appropriate taxonomic level listed below. Chapter 11 gives a complete list of required and recommended references on identifying freshwater macroinvertebrates.

- Insecta, identify to genus, except leave Chironomidae at family
- Oligochaeta, leave at Oligochaeta
- Hirudinea, leave at Hirudinea
- Hydracarina, leave at Hydracarina
- Isopoda, identify to genus
- Amphipoda, identify to genus
- Nematoda, leave at Nematoda
- Ostracoda, leave at Ostracoda

- Palaemonidae, identify to genus
- Cambaridae, leave at Cambaridae
- Gastropoda, identify to genus
- Turbellaria, identify to family
- Pelecypoda, identify to genus

**Maintain a separate count of individuals and list of taxa for each replicate dredge sample to allow an evaluation of variability between replicates**. Methodologies for assessing ALUs have not been developed for Texas freshwater depositional habitats, including pools and reservoirs. Before any biological monitoring on this type of water body, it is imperative to coordinate this work with the TCEQ and the TPWD. As methodologies and metrics are established, this manual will be updated to reflect those changes.

# Voucher Specimens

**Retain at least one representative of each benthic macroinvertebrate taxon collected as a voucher specimen for at least five years or until the conclusion of any applicable regulatory decision (whichever is longer) to allow verification of identification if necessary.** Voucher specimens serve as long-term physical proof that confirm the names applied to organisms stored in SWQMIS. Voucher specimens ensure the credibility of TCEQ biological data by documenting the identity of the organisms and making them available for review by the general scientific community.

Take the following into consideration when storing voucher specimens:

- long-term maintenance of wet (alcohol-preserved) and mounted specimens
- adequate quantity and quality of space to store specimens
- an effective mechanism for locating and retrieving specimens upon request
- personnel experience in invertebrate taxonomy

The organization maintaining voucher specimens must have a history that indicates it will be able to preserve the specimens into the future (USGS 2000). This could include in-house provisions for sample maintenance or archiving in a university or museum natural-history collection.

**Table F.1.** Metrics and scoring criteria for Surber samples—Benthic Index of Biotic Integrity. (Davis, 1997.) (Footnotes appear on following page.)

| | Metric | Scoring Criteria | | |
|---|---|---|---|---|
| | | **5** | **3** | **1** |
| **Central bioregion**<br><br>**(Ecoregions: 23, 24, 27, 29, 30, 31, and 32)** | 1. Total taxa | > 32 | 32–18 | < 18 |
| | 2. Diptera taxa | > 7 | 7–4 | < 4 |
| | 3. Ephemeroptera taxa | > 4 | 4–2 | < 2 |
| | 4. Intolerant taxa | > 8 | 8–4 | < 4 |
| | 5. % EPT taxa | > 30 | 30.0–17.4 | < 17.4 |
| | 6. % Chironomidae | [a] | < 22.3 | ≥ 22.3 |
| | 7. % Tolerant taxa | [a] | < 10.0 | ≥ 10.0 |
| | 8. % Grazers | > 14.9 | 14.9–8.7 | < 8.7 |
| | 9. % Gatherers | > 15.2 | 15.2–8.8 | < 8.8 |
| | 10. % Filterers | [a] | > 11.9 | ≤ 11.9 |
| | 11. % Dominance (3 taxa) | < 54.6 | 54.6–67.8 | > 67.8 |
| **East bioregion**<br><br>**(Ecoregions: 33, 34, and 35)** | 1. Total taxa | > 30 | 30–17 | < 17 |
| | 2. Diptera taxa | > 10 | 10–6 | < 6 |
| | 3. Ephemeroptera taxa | [b] | > 3 | ≤ 3 |
| | 4. Intolerant taxa | > 4 | 4–2 | < 2 |
| | 5. % EPT taxa | > 18.9 | 18.9–10.8 | < 10.8 |
| | 6. % Chironomidae | [a] | < 40.2 | ≥ 40.2 |
| | 7. % Tolerant taxa | < 16.0 | 16.0–24.3 | > 24.3 |
| | 8. % Grazers | > 9.0 | 9.0–5.2 | < 5.2 |
| | 9. % Gatherers | > 12.5 | 12.5–7.3 | < 7.3 |
| | 10. % Filterers | [a] | > 16.3 | ≤ 16.3 |
| | 11. % Dominance (3 taxa) | < 57.7 | 57.7–71.6 | > 71.6 |

| | Metric | Scoring Criteria | | |
|---|---|---|---|---|
| | | **5** | **3** | **1** |
| **North bioregion (Ecoregions: 25 and 26)** | 1. Total taxa | > 33 | 33–19 | < 19 |
| | 2. Diptera taxa | > 14 | 14–8 | < 8 |
| | 3. Ephemeroptera taxa | [b] | > 2 | ≤ 2 |
| | 4. Intolerant taxa | > 3 | 3–2 | < 2 |
| | 5. % EPT taxa | > 14.4 | 14.4–8.2 | < 8.2 |
| | 6. % Chironomidae | < 36.9 | 36.9–56.2 | > 56.2 |
| | 7. % Tolerant taxa | < 14.1 | 14.1–21.5 | > 21.5 |
| | 8. % Grazers | [b] | > 5.4 | ≤ 5.4 |
| | 9. % Gatherers | [a] | > 14.9 | ≤ 14.9 |
| | 10. % Filterers | > 12.2 | 12.2–7.1 | < 7.1 |
| | 11. % Dominance (3 taxa) | < 68.1 | 68.1–84.5 | > 84.5 |

[a] The discriminatory power was less than optimal for this bioregion, so the metric was assigned only two scoring categories.

[b] The median value for this bioregion was less than the metric-selection criterion (< 5.5 for taxa richness metrics; < 12 for percentage metrics expected to decrease with disturbance), so the metric was assigned only two categories.

# Metrics and Scoring for Surber Samples for Benthic Macroinvertebrates by Bioregion: Central, East, or North

| | | | | | | |
|---|---|---|---|---|---|---|
| **Stream Name:** | | | | | | |
| **Date:** | | **Collectors:** | | | | |
| **Location:** | | | | | | |
| **County:** | | **Ecoregion #:** | | | | |
| **Type of assessment:** | | | UAA | ALA | ALM | RWA |

| **Metric** | **Value** | **Score** |
|---|---|---|
| 1. Total taxa | | |
| 2. Diptera taxa | | |
| 3. Ephemeroptera taxa | | |
| 4. Intolerant taxa | | |
| 5. % EPT taxa | | |
| 6. % Chironomidae | | |
| 7. % Tolerant taxa | | |
| 8. % Grazers | | |
| 9. % Gatherers | | |
| 10. % Filterers | | |
| 11. % Dominance (3 taxa) | | |
| Aquatic life use point score ranges: | Exceptional:<br>High:<br>Intermediate:<br>Limited: | > 40<br>31–40<br>21–30<br>< 21 |
| **Total Score:** | | |
| **Aquatic-Life Use:** | | |

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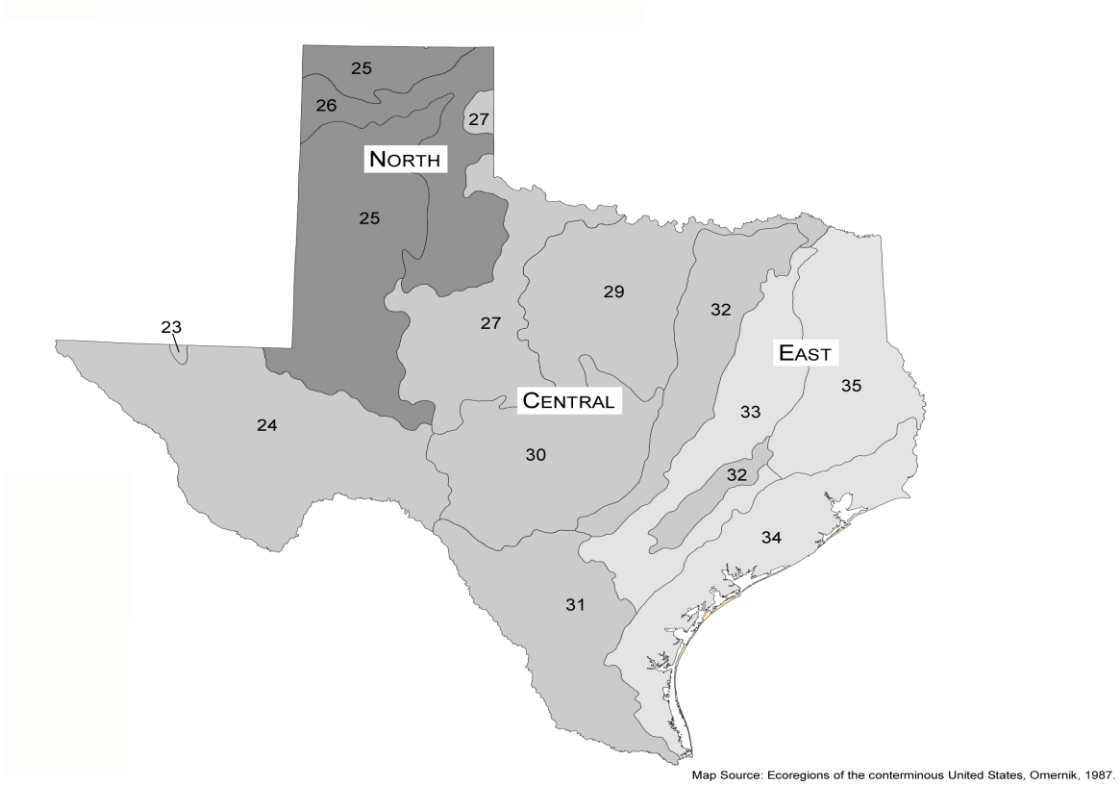
**Note:** This form should be used as part of the biological monitoring packet. If you chose to use another format, all information must be included.

**Figure F.1.** Macrobenthic bioregions (North, Central, East) and Level III ecoregions of Texas for use with Surber BIBI.




**Level III Ecoregions of Texas**

23 Arizona–New Mexico Mountains
24 Southern Deserts
25 Western High Plains
26 Southwestern Tablelands
27 Central Great Plains
29 Central Oklahoma–Texas Plains
30 Central Texas Plateau
31 Southern Texas Plains
32 Texas Blackland Prairies
33 East Central Texas Plains
34 Western Gulf Coastal Plain
35 South Central Plains