

chapter five

INVERTEBRATES

Coastal **wetlands** play host to a rich **diversity of invertebrates** that scurry, burrow, or swim in tidal creeks, mudflats, and **salt marshes**. Snails, mussels, crabs, and shrimp are the most conspicuous animals in tidal areas, and together often comprise the greatest proportion of animal biomass in these ecosystems. You can look under vegetation, turn over rocks, and wade in water to observe less conspicuous animals such as insects, mites, spiders, amphipods, isopods, and worms. Salt marshes also support a rich diversity of tiny (often microscopic) and reclusive animals that we rarely ever see.

Just like plants, salt marsh invertebrates display a wide range of tolerance for physical and chemical conditions such as salinity and tidal influence. **Species** will occupy different areas of the marsh depending on their tolerance for local conditions, and unlike plants, can migrate between different habitats with the ebb and flow of the tide. The **low marsh** and permanently flooded areas support species that require almost constant inundation, including most mussels, clams, shrimps, crabs, and bristle worms. The **high marsh** and marsh border support a variety of marine and terrestrial invertebrates. During high tide, terrestrial invertebrates either migrate toward the marsh border or crawl up vegetation, and many crustaceans migrate from the low marsh or tidal creek into the high marsh to forage.

Invertebrates perform the critical task of converting tough salt marsh grasses into a form more palatable for other organisms, allowing animals (including other invertebrates, fish, birds, and mammals) to benefit from the rich

productivity of salt marsh grasses. Thus, invertebrates are largely responsible for providing the food resources that help fuel salt marsh and marine ecosystems. The condition of the invertebrate community will ultimately influence the health of all salt marsh dependent animals.

Scientists recognize invertebrates as good **indicators** of changes in tidal flow, vegetation cover, **salinity regime**, nutrients, and dissolved oxygen. A number of invertebrates are also sensitive to pesticides and heavy metals. Many marine invertebrates are sedentary (stay in one place for their entire lives), and their populations reflect past and present environmental conditions at a particular location. In contrast, fish and birds are highly mobile and since they can leave an area if conditions become unfavorable, they are not as useful in documenting historical conditions.

Invertebrate communities may provide information as to how impacted a site may be, but often cannot reveal the source of that impact. A thorough **habitat assessment** will usually help pinpoint the reasons for an impaired invertebrate community. This chapter provides guidelines and methods for conducting a habitat assessment as part of an invertebrate **monitoring** program.

EQUIPMENT

You will need equipment to conduct the habitat assessment and collect, sort, and identify invertebrates. Table 1 lists the equipment you will need for each of these tasks, as



TABLE 1. INVERTEBRATE SAMPLING EQUIPMENT

FIELD EQUIPMENT		
D-Net (500 micron mesh size)	Spatula	Permanent Marker
Quadrat Frame, 18" x 18"	Baster	Ziploc Bags
Auger	Forceps	Labels
2 Buckets	Trowel	Clipboard and Pencils
# 30 (500 micron) Sieve	Magnifying Lens	Form 1: Field Sheet
Flagging Tape	Protective Gloves	Topographic Map
Flagging Stakes	Cooler	Aerial Photographs (if available)
300' Measuring Tape	Alcohol (ethyl or isopropyl, >90%)	Camera and Film
SORTING EQUIPMENT		
Bagged and Labelled Samples	Squeeze Bottle	Magnifying Lamp
Alcohol (ethyl or isopropyl, 70%)	Small Glass Jars (Baby Food Jars)	Forceps
#30 (500 micron) sieve	40 mL Vials and Caps	Form 2: Invertebrate Samples Record Sheet
Small Glass Beaker	Plastic Bucket	
	White Sorting Tray	
IDENTIFICATION AND COUNTING EQUIPMENT		
Vials with preserved samples	Petri Dish	Dissecting Microscope (10x-40x)
Alcohol (ethyl or isopropyl, 70%)	Ice Cube Container	Form 2: Invertebrate Samples Record Sheet
Small Glass Beaker	Probe	Form 3: Laboratory Bench Sheet
Squeeze Bottle	Forceps	Identification Manuals
Small Glass Jar (Baby Food Jar)	Pencils	
DATA ENTRY AND ANALYSIS EQUIPMENT		
Form 1: Field Sheet (completed)	Form 4: Invertebrate Data Sheet	Calculator
Form 3: Laboratory Bench Sheet (completed)	Form 5: Habitat Assessment Sheet	Computer with spreadsheet software

well as the necessary data sheets. Although the equipment list is extensive, resourceful people should be able to gather the materials they need at relatively low cost. The most expensive items are the D-net, auger, sieve, magnifying lamp, and dissecting microscope. Volunteer groups should try to seek an arrangement with local biological laboratories (such as high school, university, or state research labs) so that they do not have to purchase expensive items. Many of the materials required are common household items and can be donated from volunteers or the community.

SAMPLING METHODS

At this point, take the time to read the “Overview of Invertebrate Monitoring” text box that accompanies this section. Salt marsh invertebrate monitoring is more complex than the other monitoring techniques described in this manual and may require considerable preparation. It is wise to have a full understanding of your commitment before

going into the field. At times, you will not be able to follow the instructions exactly as they are described below. You may encounter unexpected conditions such as dangerous mud flats where you want to sample, or steep banks that make access into the stream difficult. Use common sense to modify the procedures if necessary, but try to conduct your sampling in the prescribed manner.

Habitat Description

The ability of a salt marsh to withstand the effects of various **environmental stressors** depends upon **hydrology**, substrate, the shape and size of the marsh, and its resident **biological community**. Volunteers should fully describe habitat conditions and potential stressors to the marsh, and the best way to do this is by making careful field observations. Volunteers should conduct the habitat description during the growing season after the vegetation has become established, and preferably at the same time as the invertebrate sampling. The instructions below will guide you



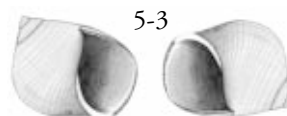
OVERVIEW OF INVERTEBRATE MONITORING

- **Set Goals and Objectives.**
- **Choose a Study Site, Reference Site(s), and Sampling Stations:** Visit the study site and reference site(s) well before conducting the invertebrate sampling.
- **Set Date for Field Work:** Marine invertebrates are always present, but the presence and abundance of some species changes throughout the year. Late summer is a good time to sample because invertebrate size, diversity, and abundance are maximum then. If you are planning a second collection effort, spring is good because certain taxa are present that are scarce in the summer.
- **Borrow or Purchase Equipment and Materials.**
- **Organize the Support Team:** A field team should consist of a team leader with experience in salt marsh ecology or invertebrate taxonomy, and field assistants to help with equipment, sampling and recording. For safety reasons, single individuals should never undertake invertebrate sampling alone.
- **Conduct the Habitat Assessment.**
- **Conduct the Invertebrate Sampling.**
- **Sort the Invertebrate Samples:** Ideally, volunteers will work in a laboratory environment equipped with at least one deep sink and workbench. A university or local high school biology laboratory is a perfect setting for sorting and identification.
- **Identify and Count Invertebrates.**
- **Perform Data Entry and Analysis.**
- **Complete the Habitat Assessment Score.**
- **Submit All Completed Forms and Invertebrate Samples to the Project Leader:** Be sure that the project leader has all the necessary contact details so that if questions arise they can be resolved expediently.

through the field data form and provide explanations as you go. Maps, aerial photographs, and accurate field observations are your “tools” for completing Form 1, and later Form 5, to compute a habitat condition score.

Complete the site identification section at the top of Form 1 (Appendix 1 of this chapter). List the names of all team members conducting the assessment. Use a **GPS** to record latitude and longitude of your site or estimate coordinates from topographic maps. Before leaving the marsh site, double check Form 1 to ensure you have recorded all information. Record observations on the following variables:

1. **Weather:** Tick the appropriate boxes to describe weather conditions in the 24 hours before invertebrate sampling, and on the day of the invertebrate sampling. Stormy weather or heavy rains can affect sampling conditions, turbidity, dissolved oxygen, and conductivity.
2. **Hydrology:** Tick the appropriate boxes to list the sources of water. Use tidal charts to determine the average tidal range during the year. Document any tidal restrictions and record any impediment to water movement.
3. **Marsh Vegetation:** Categorize the **abundance** of different types of marsh vegetation in the **wetland evaluation area (WEA)** using the following abundance descriptors: N = None, R = Rare, C = Common, and A = Abundant. Abundant vegetation usually indicates a healthy marsh, and marshes with a rich variety of vegetation types provide more habitats and feeding opportunities.
4. **Abundance of Food for Invertebrates:** Follow the same method as for the marsh vegetation. Aquatic invertebrates generally prefer to consume softer vegetation. The hard stemmed plants, such as *Spartina alterniflora* (smooth cordgrass) and *Phragmites australis* (common reed), provide good habitat even though they are a poor food source. Record the presence of fish because they prey on invertebrates and can affect their densities.



5. **Substrate:** Follow the same method as for the vegetation to record the relative abundance of the different substrate types, and record other observations such as oil slicks. Most water-dependant salt marsh invertebrates favor sandy and muddy substrates, though some (snails, barnacles) prefer solid surfaces. Solid substrates are often colonized by seaweed that in turn provides food and habitat for invertebrates.
6. **Impacts to Salt Marsh:** Record all observed and known impacts to the salt marsh WEA. Mark their location on the sketch or take photographs.
7. **General Water Quality:** (*Optional, but recommended*) Using a water quality analyzing system (see: “Salinity” chapter), record the water quality parameters indicated in the table. Measure water quality at each of three sampling stations, and if necessary, record other water quality observations. Tick the appropriate boxes if suspended materials or water odors are observed.
8. **Record Invertebrate Samples:** After completing the invertebrate sampling, ensure that there is a record of all collected, bagged, preserved, and labeled samples ready for return to the laboratory. Also, be sure to record all of the live invertebrates you identified and counted in the **quadrat samples**.
9. **Sketch of Marsh:** Use a topographic map and aerial photographs to assist you with the sketch. Include all of the elements listed at the top of the sketch area. Drawing the map will familiarize you with the surrounding land uses and roadways, the size and shape of the marsh and the related WEA, the stream pattern, ditches, vegetation types, location of restrictions, and other disturbances. It is important to mark the three sampling stations at each sampling site. Take photographs of the WEA to complement your sketch.

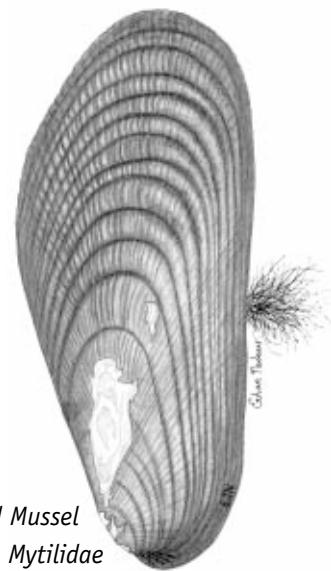
Collecting Samples

At this point, you have already selected your study site, reference site(s), and WEA using guidelines in Chapter Three and previous sections of this chapter. The WEA should be representative of the marsh condition and meet the overall goals and objectives of your study. Within the WEA, you must choose three locations to collect invertebrates, called **sample stations**. Here are some general recommendations for choosing sample stations and collecting samples:

- Be sure that your invertebrate sampling coincides with low tide.
- Flag a 300’ transect along the primary tidal creek if you are studying an estuarine marsh, or a 300’ transect along the bank if you are studying a salt marsh that borders an embayment.



Two volunteers conducting invertebrate sampling in a quadrat.
Photo: Ethan Nedeau



Ribbed Mussel
Family *Mytilidae*



- Choose three sample stations near the beginning, middle, and end of the transect. Be sure to choose sample stations that are representative of local conditions. Place flags at each location so that you will remember where you took the samples.
- If you are working along a tidal creek, first check the direction in which the water is flowing. Begin sampling at the downstream location against the flow of water and work “upstream” against the flow so that you do not disturb other unsampled stations above. If you are working along an embayment, it does not matter what order you collect samples.

Three types of samples are collected at each station: quadrat (or plot) samples at the top of the bank, **D-Net samples** in the stream or bay, and **auger samples** in the stream or bay (Figure 1). These three methods will later form one composite invertebrate sample for each station. Use the instructions below to collect these samples.

Quadrat Sampling

Quadrats are used to sample invertebrates that exist on the upper edge of the estuarine stream bank or seaward marsh edge. You should expect to find crabs, mussels, barnacles, amphipods, isopods, flies, spiders, grasshoppers, and mites in this habitat. It is useful to have one person do the sampling while another person records the results on Form 1. You should use protective gloves for this sampling technique.

Use the following procedure:

1. Place the quadrat on the bank near the water's edge at a location that is typical of the bank condition.
2. Methodically work the hands backwards and forwards across the surface of the ground within the frame, and identify, count, and record every living invertebrate that you encounter. Since barnacles are usually too numerous to count, record their abundance with the following notation: + = rare, ++ = common, and +++ = abundant.
3. Repeat this procedure at the other sampling stations.

D-Net Sampling

D-Nets are used to collect invertebrates from shallow water environments at low tide, either in tidal creeks or embayments. Using this method, you should expect to collect molluscs, polychaete worms, amphipods, isopods, and other organisms requiring constant inundation with seawater. At least two people (ideally three) are needed to conduct D-Net sampling. Use the following procedure:

1. If working in a tidal creek, note the direction of the tidal movement, and face against the flow.
2. Before entering the water, look for all the different habitat types, such as banks and vegetated margins, different substrate types, **woody debris**, and floating **alga mats**, and try to collect from each of these habitats. Enter the water gently so that you do not frighten and disperse swimming organisms.

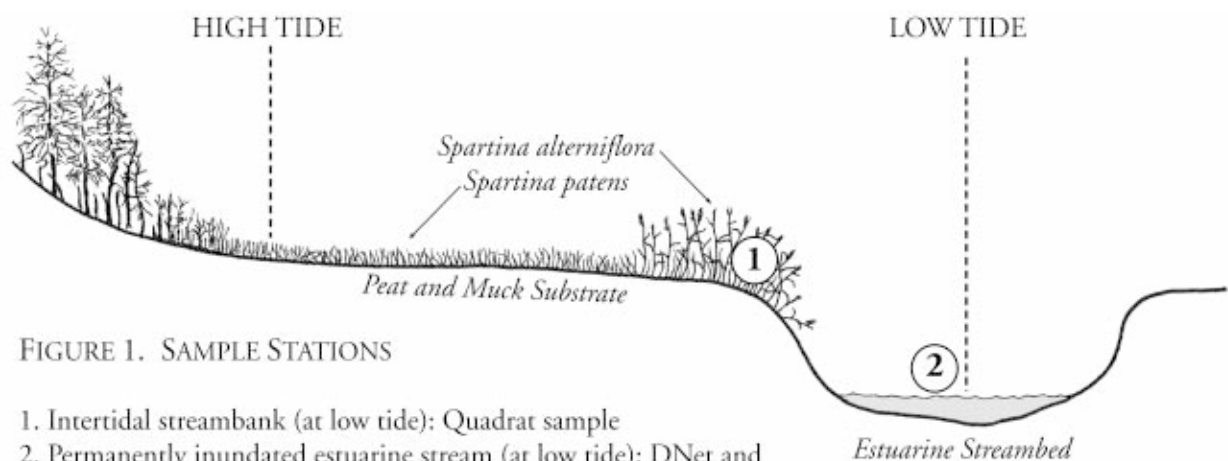


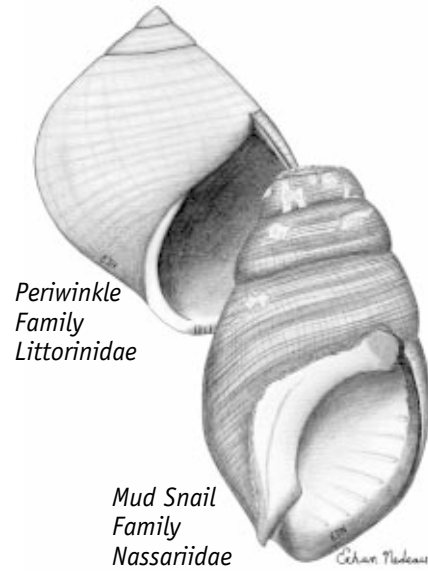
FIGURE 1. SAMPLE STATIONS

1. Intertidal streambank (at low tide): Quadrat sample
2. Permanently inundated estuarine stream (at low tide): DNet and auger samples





Collecting a D-Net sample in a tidal creek. Photo: Ethan Nedeau



3. Place the flat side of the D-Net on the surface of the substrate in approximately 0.3 meters (14 inches) of water, and hold the net perpendicular to the substrate as you walk 10 strong and even paces toward the water flow, pulling the D-Net through and over different habitats. If working in an embayment, you will not have the current to contend with and it is important that you maintain your momentum as you collect the sample. Many invertebrates are good swimmers and try to escape the net.
4. Bring the net containing the sample to the surface for retrieval. Gently swish the net back and forth in the stream to allow fine silt and sand to pass through the mesh, being careful not to lose organisms.
5. Place the contents of the inverted net over a bucket half filled with water and wash all debris and invertebrates off the net and into the bucket.
6. Use forceps to remove any organisms that remain on the net, and place these in the bucket.
7. Pour the contents of the bucket through a standard US No. 30 brass sieve to remove the water.
8. Place the contents of the sieve into a resealable plastic bag, and make sure that no invertebrates are left on the sieve.
9. If you have a large number of snails and crabs in your sample, you should identify them in the field, record the numbers of each **family** (and if possible, species) on the field sheets, and then return them back to the water alive.

10. Label the resealable plastic bag (see instructions below) and record the sample information on the field sheet.
11. Once you complete a sample, wash out the D-Net to remove all remaining debris.
12. Repeat the procedure at the two other sample stations, placing each sample into a separate resealable plastic bag. It is important that you collect all samples in a consistent manner.

Auger Sampling

An auger, or corer, is used to collect a sediment or substrate sample from the stream or embayment. Using this method, you should expect to collect a variety of worms, snails, clams, amphipods, isopods, and other organisms that live on or within the substrate. You should collect the sample in a location that was not disturbed by D-Net sampling. Use the following procedure to collect auger samples:

1. Hold the auger perpendicular to the water surface above the point from which the sample will be taken.
2. Push the auger downward into the sediment until the bucket of the auger is half embedded in the substrate. Turn the auger handle to help force the auger into the substrate.
3. Carefully pull the auger out of the sediment and quickly place the sieve beneath the auger so that none of the sample is lost. Keep the sieve under the auger as you return to the bank, where you should empty the remaining auger contents into the sieve.



4. Remove fine sediment from the sieve by carefully placing it face up in the water (being careful that the water does not cover the top!) and gently swirling the contents so that the fine sediment passes through the sieve. This important step will greatly decrease sorting time.
5. Place sieve contents into a resealable plastic bag, seal, label (see instructions below), and record the sample information on the field sheet.
6. Clean the auger by swishing it back and forth in the water.
7. Repeat the procedure at the other two sampling stations, placing each sample into a separate resealable plastic bag. It is important that you collect all samples in a consistent manner.

Sample Bagging and Labeling

You must properly bag and label all samples so that everybody knows how, where, and by whom a sample was collected. A sample without a label is worthless, and it would be unfortunate if valuable field time were wasted because of improper bagging or labeling procedures. Use the following procedures to label samples:

1. Using a permanent ink marker, label all samples with the following information: sample number, field site identification, sampling station number, date, names of collectors, sampling method, and the **preservative** used (Figure 2). This can be done before going into the field.
2. Flood all bagged samples with 90% or higher concentration alcohol or similar preservative and seal carefully.
3. Record the sample numbers on the field sheet (See: Form 1).
4. Place bagged samples in a cooler with ice to prevent heating in hot weather.
5. Store samples in an air-conditioned laboratory (or similar workspace) or a refrigerator for no longer than two weeks before sorting.

Sample Sorting

After you collect samples and return to your indoor working area or laboratory, you must begin the painstaking process of removing invertebrates from the sand, silt, and peat substrate. Many invertebrates are less than

Sample Number: #4
 Field Site Identification: WEA MSBP-R
 Sampling Station: #2
 Date: April 20, 2002
 Names of Collectors: Tom Hopkins, Bev O'Halloran
 Sampling Method: D-Net
 Preservative Used: 90% ethyl alcohol

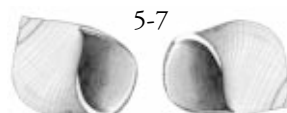
FIGURE 2. SAMPLE LABEL

5mm long and pale in color, making them difficult to see among the sediment. Sample sorting is a very important part of your study, and it is imperative that you remove all of the organisms in your sample (or sub-sample — see #4 below) so that your data are not biased and your conclusions are reliable. You might have to be patient and thorough, but the data you generate will be well worth the effort. Use the following guidelines for sorting samples:

1. Empty the contents of a sample into the standard US #30 sieve. You should place the sieve over a bucket so that sediment is not washed down the drain.
2. Gently rinse the sample under tap water to remove fine organic detritus, silt, and clay. Place the sieved sample into a white sorting tray (one small handful at a time, if necessary). You must be careful to remove any organisms that may be stuck on the sieve.
3. Place the sorting tray under a desk light or magnifying lamp, and using the magnifying lens and forceps, remove invertebrates from the sediment and place them into a large (40mL) vial two-thirds filled with 70% or higher concentration of alcohol.



Volunteers sorting and identifying invertebrates. Photo: Anna Hicks



RAPID BIOASSESSMENT SUBSAMPLING PROTOCOL

(100-Organism Count Technique)

1. Thoroughly rinse sample in a 500-micron screen or the sampling net to remove fine sediments. Any large organic material (whole leaves, twigs, algal or macrophyte mats) should be rinsed, visually inspected for invertebrates, and discarded.
2. Place sample contents in a large flat pan with a light-colored (preferably white) bottom. The bottom of the pan should be marked with a numbered grid pattern, each block in the grid measuring 5 x 5 cm. (Sorting using a gridded pan is only feasible if the organism movement in the sample can be slowed by the addition of club soda or tobacco to the sample. If the organisms are not anesthetized (or preserved), 100 organisms should be removed from the pan as randomly as possible.) A 30 x 45 cm pan is generally adequate, although pan size ultimately depends on sample size. Larger pans allow debris to be spread more thinly, but they are unwieldy. Samples too large to be effectively sorted in a single pan may be thoroughly mixed in a container with some water, and half of the homogenized sample placed in each of two gridded pans. Each half of the sample must be composed of the same kinds and quantity of debris and an equal number of grids must be sorted from each pan, in order to ensure a representative subsample.
3. Add just enough water to allow complete dispersion of the sample within the pan; an excessive amount of water will allow sample material to shift within the grid during sorting. Distribute sample material evenly within the grid.
4. Use a random numbers table to select a number corresponding to a square within the gridded pan. Remove all organisms from within that square and proceed with the process of selecting squares and removing organisms until the total number sorted from the sample is within 10 percent of 100. Any organism that is lying over a line separating two squares is considered to be in the square containing its head. In those instances where it is not possible to determine the location of the head (worms for instance), the organism is considered to be in the square containing the largest portion of its body. Any square sorted must be sorted in its entirety, even after the 100 count has been reached. In order to lessen sampling bias the investigator should attempt to pick smaller cryptic organisms as well as the larger more obvious organisms.
5. After 100 or more organisms have been removed, check the entire contents for any taxonomic group that has been missed. Pick out one representative of each previously missed group. This ensures a complete record of taxa richness.

Source: Plafkin et al., 1989, modified from Hilsenhoff, 1987. #5 is a modification made by the authors of this manual.

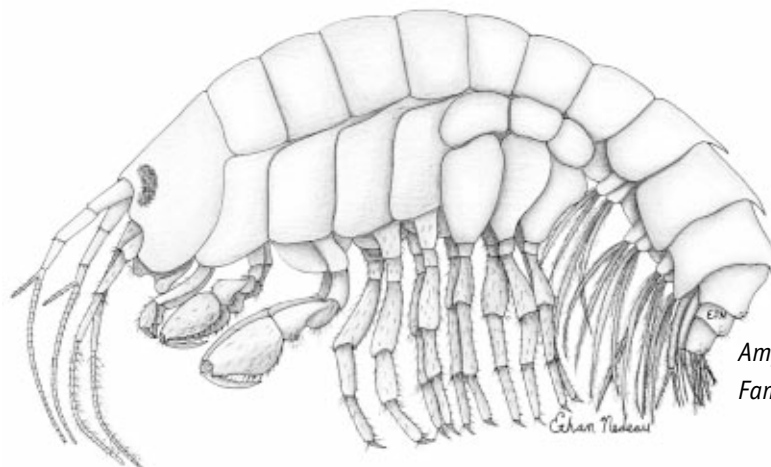
4. If you think there are approximately 100 or less organisms in the sample, then remove all organisms. If you think there are well over 100 organisms, you may use the 100-organism **sub-sample** procedure (see textbox above). This sub-sampling method is appropriate for volunteers with limited time and financial resources. If time and resources are available, volunteer groups should conduct a full count.
5. After you have finished sorting, have a second person scan the debris in the sorting tray to double check your work. Tightly seal and label each vial (two for each sampling station: one for D-Net and one for auger), and register the sample on the Invertebrate Samples Record Sheet (Form 2, Appendix 2 of this chapter).
6. Samples can be identified and counted any time after the sorting has been completed, but should not be left for more than six months because alcohol in the vial sometimes evaporates and ruins the sample.



Identifying and Counting Samples

Identification can be enjoyable because you get the chance to look closely at organisms, observe their fascinating shapes, and spend time looking through identification guides and learning about each organism and its close relatives. It is very satisfying to know the difference between Capitellidae, Spionidae, and Nereidae — all different types of marine worms — especially considering that you probably did not even know these words existed before you became a volunteer monitor! Identification takes practice, and people working in the Northeast are fortunate to have two outstanding identification manuals for marine invertebrates entitled *Marine Animals of Southern New England and New York* (Weiss, 1995) and *A Practical Guide to the Marine Animals of Northeastern North America* (Pollock, 1998). Use the following guidelines to identify and count invertebrates:

1. Create a **composite sample** for each station by pouring the vial contents of the D-Net sample and auger sample into one petri dish. Make sure that no organisms remain in the vials.
2. Place the petri dish under the dissecting scope set at 10X magnification, and in a deliberate, systematic manner, scan back and forth, identifying organisms as you go. You may need to increase the magnification to see finer details.
3. Using Weiss (1995), Pollock (1998), and other references, identify the invertebrates to family level.
4. Record and count each **taxon** on the Laboratory Bench Sheet (Form 3, Appendix 3 of this chapter).
5. Immediately after you identify and record a specimen, return it to a labeled vial two-thirds filled with 70% or higher concentration alcohol. There should be one vial per sample station per sample date.
6. Label and safely pack the vials for return to your project coordinator so that someone can reexamine specimens or identify them to a lower level of taxonomy at some future date.
7. If you have doubts about an organism's identity, consult with a marine invertebrate specialist. Place the specimen in question in a separate vial with alcohol and a complete label. Send the specimens to a specialist for verification, and add to your records later. Alternatively, arrange to have a **taxonomist** present during an identification session to provide assistance.
8. Record the completion of this process for each sample on the Invertebrate Sampling Record Sheet (Form 2). Form 2 traces the sample collection, sorting, and identification to this stage.
9. On the Laboratory Sheet (Form 3), add the data from the quadrat sample taken from the same sampling station. Enter the total number of organisms for each family or **taxonomic group**, the number of different types of taxa you identified, and the resulting total abundance for the completed composite sample.
10. Repeat this process for the remaining two sample stations, using a separate Form 3 for each station.
11. Once volunteers have finished collecting, sorting, and identifying samples for a site, there will be three completed copies of Form 3 (one for each sample station).
12. Samples and the registration sheet (Form 2) are to be returned to the project leader for **archival action**. Similarly, return Forms 1, 3, 4, and 5 to the project leader once they are completed. Project leaders should keep copies (hard or floppy disk) for their own records and as a safety backup.



Amphipoda
Family Gammaridae



DATA ENTRY

Even though the project scientist or leader usually analyzes the data, volunteers can perform a number of tasks associated with data entry. If you followed the sampling, identification, and counting procedures outlined above, you will have three completed copies of Form 3 for each site, which will then be combined into a single Invertebrate Data Sheet (Form 4, Appendix 4 in this chapter) for each site. Volunteers should transfer invertebrate data onto Form 4 using the instructions below. Data entry and analysis is ideally suited for a spreadsheet program such as Microsoft Excel. The instructions below include figures that show spreadsheet format and use real data to illustrate key aspects of data entry. Figure 3 is set up similar to a spreadsheet with column and row identifiers (letters for columns and numbers for rows), so that any cell can be identified. For example, cell D12 is located in column D and row 12.

Data Entry Instructions

1. Record all pertinent information (site identifiers, processing dates, names of volunteers) on Form 4.

2. Transfer the number of organisms in each taxonomic group for each composite sample (the last column of Form 3) into columns D(1), D(2), and D(3) of Form 4. For example, if 23 individuals of the family Hydrobiidae were collected at Station 1, then you would enter 23 into cell C6 in Figure 3.
3. Sum D(1), D(2), and D(3) for each taxonomic group (class, order, or family), divide by three, and enter this value into column \bar{D} . This is the Taxa Average. For example, the value “6.7” in cell F3 of Figure 3 represents the average number of Acteonidae collected at the three sampling stations.
4. For each order, sum column \bar{D} to compute Order Subtotal (Figure 3). For example, the value “21.7” in cell F16 of Figure 3 represents the average number of Isopoda collected at the three sampling stations.
5. Transfer each Order Subtotal into the second column of the box “Composition of Major Groups” on page two of Form 4 (see Table 2). The sum of the column “Number” represents the Average Number

	A	B	C	D	E	F	G
1	TAXA	FG	D(1)	D(2)	D(3)	\bar{D}	%
2	GASTROPODA						
3	Acteonidae	G	5.0	12.0	3.0	6.7	6.2
4	Cerithiidae	G	0.0	0.0	0.0	0.0	0.0
5	Columbellidae	G	1.0	5.0	1.0	2.3	2.2
6	Hydrobiidae	DF	23.0	12.0	6.0	13.7	12.6
7	Littorinidae	G	7.0	4.0	34.0	15.0	13.9
8	Melampodidae	G	0.0	0.0	0.0	0.0	0.0
9	Nassariidae	G	9.0	4.0	6.0	6.3	5.8
10	Subtotal					44.0	40.6
11	ISOPODA						
12	Idoteidae	DF/C	31.0	8.0	14.0	17.7	16.3
13	Janiridae	DF/SC	0.0	0.0	0.0	0.0	0.0
14	Limnoriidae	DF	3.0	7.0	2.0	4.0	3.7
15	Other	DF/SC	0.0	0.0	0.0	0.0	0.0
16	Subtotal					21.7	20.0
17	SPIONIDA						
18	Spionidae	DF	24.0	17.0	81.0	40.7	37.6
19	Other	DF	2.0	3.0	1.0	2.0	1.8
20	Subtotal					42.7	39.4

FIGURE 3: EXAMPLE DATA ENTRY SPREADSHEET

The numbers along the side (1-20) and letters along the top (A-G) are used to identify individual cells within the spreadsheet. For instance, cell “F6” refers to the average value of the family Hydrobiidae, which is 13.7. This figure only shows partial data, and a realistic spreadsheet would include several additional taxonomic groups.

Column labels are as follows:

Taxa: Class, Order, or Family

FG: Feeding Group (see Table 3 for abbreviations)

D(1): The number of organisms in sample station #1

D(2): The number of organisms in sample station #2

D(3): The number of organisms in sample station #3

\bar{D} : Taxa Average

%: Taxa Percent Composition



of Organisms for the site (stations 1-3 combined). In Table 2, the value “108.3” represents the average number of organisms collected at the three sampling stations.

6. In the box “Composition of Major Groups” (Table 2), divide each Order Subtotal by the Average Number of Organisms, multiply by 100, and enter this value into the column “Percent.” This represents the percentage of the entire sample comprised by each Order, and the sum of the column should be 100%. In Table 2, to compute the percent contribution of the order Isopoda, divide 21.7 by 108.3 and multiply by 100 to reach 20.0%.
7. In Figure 3, divide Taxa Average by Average Number of Organisms (from Table 2) and multiply by 100 to compute Taxa Percent Composition for the entire sample. Enter these values into the column “%.” For example, in Figure 3 the percent composition of the family Idoteidae is determined by dividing its taxa average (17.7; Cell F12) by the Average Number of Organisms (108.3, from Table 2) and multiplying by 100 to reach 16.3% (Cell G12).
8. Sum column “%” for each order to compute Order Percent Composition for the entire sample, which is entered as “Subtotal” in column “%” of Figure 3. The Order Percent Composition in Figure 3 should be the same as the percent composition in Table 2. For example, the percent Gastropoda in Table 2 and Figure 3 (Cell G10) are both 40.6.
9. Complete the box “Composition of Feeding Groups” by summing the Taxa Percent Composition for each **feeding group** (see Table 3). Feeding group is indicated in column FG of Figure 3. The **Mixed Feeding Group** is used for families that have more than one feeding group (e.g. SF/DF). For example, in Figure 3 the **deposit feeders** (DF) are the Hydrobiidae (12.6), Limnoriidae (3.7), Spionidae (37.5), and Spionida Other (1.8), for a combined percentage of 55.6%. This value is entered into the appropriate line on Table 3. If you have counted correctly, the sum of percentages should be 100.
10. Locate the summary box “Introduced Species” on Form 4. You may not be able to complete this box because it requires species-level identification of three common introduced species — the green crab

TABLE 2. COMPOSITION OF MAJOR GROUPS

ORDER	NUMBER	PERCENT
Gastropoda	44.0	40.6
Isopoda	21.7	20.0
Spionida	42.7	39.4
Total	108.3	100.0

TABLE 3. COMPOSITION OF FEEDING GROUPS

FEEDING GROUP	PERCENT
Predator (PR)	0.0
Deposit Feeder (DF)	55.6
Grazer (G)	28.0
Omnivore (OM)	0.0
Scavenger (SC)	0.0
Suspension Feeder (SF)	0.0
Mixed (M)	16.3
Total	100%

(*Carcinus maenas*), common periwinkle (*Littorina littorae*), and Japanese crab (*Hemigrapsus sanguineus*). Remember that this chapter has recommended family-level identification. If you have identified and counted these species, you can record the average number of individuals and percent composition in the sample following the same procedure you did for the box “Percent Composition of Feeding Groups.”

11. Complete the box “Percent Insects, Spiders, and Mites” on Form 4 by summing Taxa Percent Composition for the Insecta (insects), Aranea (spiders), and Acarina (mites).
12. Fill out the box “Summary” (see Table 4) using the following instructions:

Line 1: Enter the Average Number of Organisms (Table 2, value = 108.3).

Line 2: Count the number of different taxonomic groups present in the samples (whether to class, order, or family) and enter that figure into line two. Using data in Figure 3, there are nine different taxonomic groups.

Line 3: Find the highest value in the third column of the box “Composition of Major Groups” and enter the value into line three (Table 2, value = 40.6 [Gastropoda]).

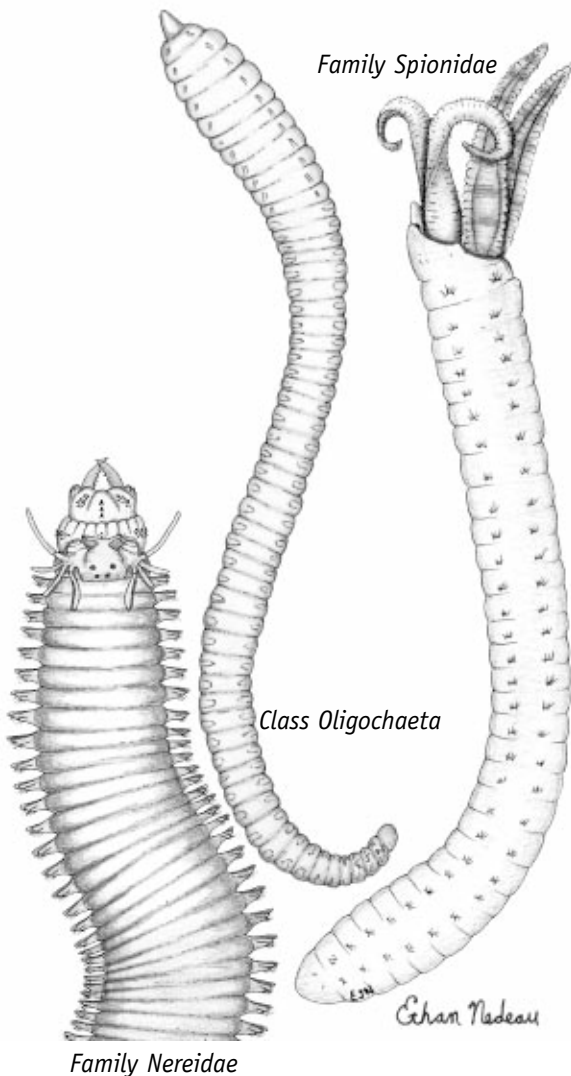


TABLE 4. SUMMARY

METRIC	VALUE
Average Number of Organisms	108.3
Taxonomic Richness	9
% Contribution of Dominant Taxonomic Group	40.6%
% Contribution of Dominant Feeding Group	55.6%
% Insects, Spiders, and Mites	0%

Line 4: Find the highest value in the second column of the box “Composition of Feeding Groups” and enter the value into line 4 (Table 3, value = 55.6 (Deposit Feeder).

Line 5: Enter the combined percentage of Insects, Spiders, and Mites.



DATA ANALYSIS AND COMPARISON

You have completed Data Entry and by doing so you have computed some important community **metrics** for your study site and reference site. This section discusses what each of these metrics means and how they are used to compare a study site with a reference site.

Average Number of Organisms

This is the average number of organisms that were collected in three composite samples from a site. This is sometimes called “average abundance” or “average density.” This is not a very meaningful value. Scientists have found that the average total number of organisms, or abundance, usually does not respond in a consistent way to environmental impact. The results are always highly variable. Some impacts, such as an increase in nutrients, may promote an increased abundance of organisms, whereas other impacts, such as heavy metals, may cause a decreased abundance of organisms. Even though this figure is not very useful as a metric, it is needed to calculate other more reliable metrics.

Taxonomic Richness

This is the number of different types of organisms that were collected at the site. If you identified all taxa to the family level, then this would be the number of families. It is more likely that you only identified some difficult organisms to the class or order level. Taxonomic richness usually represents the number of groups that were identified to the lowest possible level. For example, if the final taxa list looked like this:

- Class Insecta
- Class Turbellaria
- Unknown Polychaeta
- Family Spionidae
- Family Capitellidae
- Family Gammaridae
- Family Idoteidae

then the taxonomic richness for your sample would be seven, even though you could only identify four of these to the family level. High taxonomic richness is usually associated with favorable conditions with various types of microhabitats. Taxonomic richness is usually lower in disturbed areas because sensitive species are lost and certain habitats are eliminated. The exception occurs with a mild disturbance that can create more habitats or niches than previously existed. In this case, taxonomic richness is likely to increase.



Percent Composition of Taxonomic Groups

The types of organisms and the percent composition of major groups of organisms can reveal important information about the health of a community because groups respond differently to environmental pollution and disturbance. Some species are very sensitive to disturbance, and a community with large numbers of **sensitive** organisms is probably very healthy. Other species are extremely **tolerant** of pollution and often increase in abundance in polluted habitats. For example, studies have shown that the abundance of the families Palaemonidae, Spionidae, Nereidae + Nephtyidae, and Capitellidae increase with eutrophication. Table 5 suggests that the study site is more eutrophic than the reference site, even though Nereidae + Nephtyidae did not respond as expected.

Percent Composition of Dominant Taxonomic Group

A healthy community should have a balanced composition of taxa consisting of at least three co-dominant groups. Usually no single group should greatly dominate the rest of the community. One or two groups usually dominate stressed communities, either because sensitive species are eliminated or because certain groups respond in a positive way to pollution or disturbance. If the percent composition of the dominant taxonomic group is 36% in the reference site and 78% in the study site, then it is possible that conditions at the study site favor one type of organism that out competes the others.

TABLE 5. COMPOSITION OF TAXONOMIC GROUPS

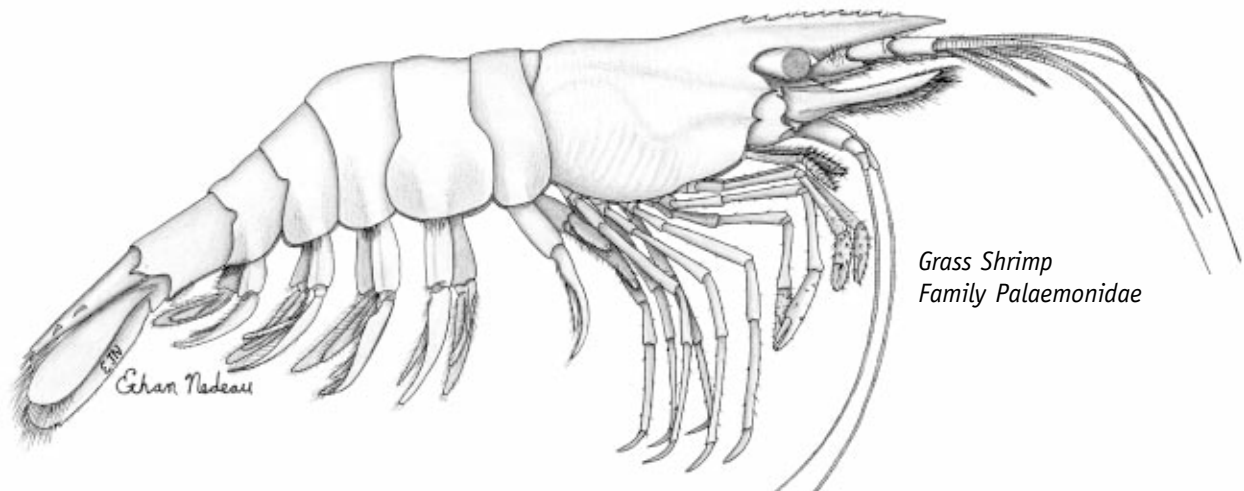
TAXA	PERCENT COMPOSITION	
	REFERENCE	STUDY
Palaemonidae	2	5
Spionidae	0	15
Nereidae + Nephtyidae	4	1
Capitellidae	3	30
Percent Abundance	12	48

Percent Composition of Trophic Groups

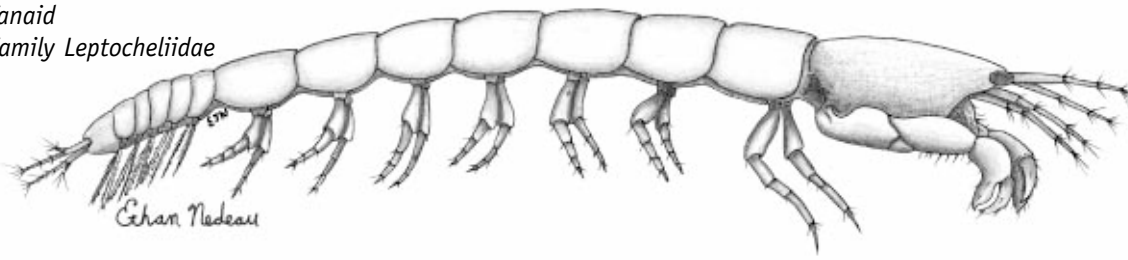
This metric is very similar to the one above except that it indicates the balance between different feeding groups in a community. A healthy community should have a well-balanced composition of different trophic groups, although **detritivores** and **suspension feeders** are usually dominant in salt marshes. Many invertebrate **predators**, such as the blue crab and the clamworm, live a long time and their presence in a water body usually indicates good water quality, especially because predators are subject to the effects of **biomagnification** — the increasing concentration of a pollutant in body tissues as food passes along a food chain. With increasing impact, the proportion of predators is expected to decline and the proportion of detritivores to increase.

Percent Contribution Dominant Trophic Group

A healthy community should have a balanced composition of trophic groups, and no single group should greatly dominate the community. The percent contribution of the



Tanaid
Family Leptocheliidae



dominant trophic group can be a reliable indication of the feeding opportunities that exist in a community, and provide insight about wetland condition.

Introduced Species

There is a growing concern that species introduced from other areas of the world find ideal conditions in New England. Often they do not have natural predators or diseases that were present in their native habitats, and can often out-compete native species and spread very quickly throughout the ecosystem. The green crab (*Carcinus maenas*), common periwinkle (*Littorina littorae*), and Japanese crab (*Hemigrapsus sanguineus*) are three common introduced species. High numbers of these species might not indicate other environmental problems associated with the salt marsh, but should be cause for concern. In some cases, careful monitoring or **mitigation** measures might be appropriate.

Percent Insects, Spiders, and Mites

The presence of aquatic freshwater insects (such as chironomid midges, beetles, true bugs, mosquitoes, and dragonfly **larvae**) in salt marshes might indicate low salinity regimes possibly caused by excessive stormwater runoff from the surrounding landscape, the influence of groundwater springs, or the lack of tidal flushing due to a tidal restriction. The presence of terrestrial organisms such as spiders, mites, flies, aphids, and grasshoppers very close to the water edge can indicate a reduction in flooding by tidal salt water. For example, if you found 28% of Insects, Spiders, and Mites in the study site and only 4% in the reference site, then it is likely that the two sites have a different salinity and/or hydrological regime(s). You would probably want to measure salinity or tidal hydrology to support these findings.

ADVANCED ANALYSIS

You can now see how to use metrics to make comparisons that may suggest health problems for different aspects of the invertebrate community at a study site. The information you have obtained to this point is very important. It forms the basis for the final step in the invertebrate community health analysis, the calculation of the **Invertebrate Community Index (ICI)**. Volunteers are not asked to take this step because it requires special expertise. Volunteers should complete the **Habitat Assessment Score (HAS)**, and by reading the section “Summary of ICI and HAS” volunteers can see how the invertebrate data and habitat evaluation can be graphically displayed and presented to decision makers and managers.

Invertebrate Community Index (ICI)

Project leaders use invertebrate data to calculate an ICI with assistance from a professional biomonitoring scientist. The ICI is a summary of the multiple metrics and **indices** that have been selected for each site. The ICI summarizes the degree of impact to the invertebrate community at the study site as compared to a reference site(s) by comparing all of the invertebrate metrics that were calculated. The study site is given a score between 0% and 100%. A score of 100% means that the invertebrate community at the study site is the same as that of the reference site(s). A score of 0% means there is no resemblance at all with the reference site. Scores will nearly always fall in between 20% and 90%. For example, a score of 72% means that the invertebrate community from the study site differs by 28% from that sampled at the reference marsh.

Habitat Assessment Score (HAS)

Invertebrate community health is reliant on both habitat quality and water quality. It is important to document habitat quality and water quality variables in order to put invertebrate results into context. Form 1 and Form 5



provide a means to express habitat and water quality in a way that is comparable to the invertebrate community metrics and the ICI. This manual uses 10 important **variables** of habitat condition to compute an overall score, called the HAS. The HAS is expressed as a percentage of a theoretical optimal condition. Follow the procedure below to compute the HAS:

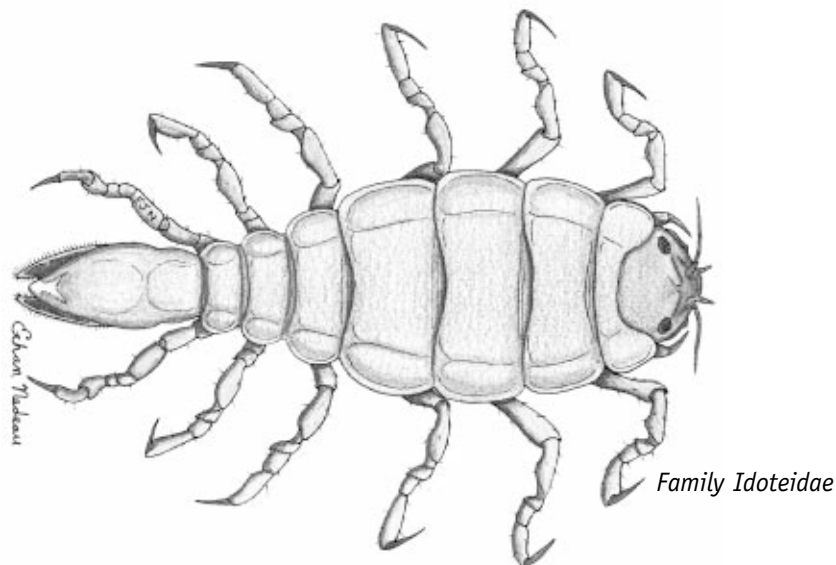
1. Use the information recorded on Form 1 and your best judgment to determine a score for each of the variables on Form 5. Scores range from zero to five, with zero = poor and five = excellent. You may use partial numbers (i.e. 3.5). Record the score in the appropriate column on Form 5.
2. Sum the scores for each variable and convert the total to a percentage. Conversion to % = total score for attributes/50 x 100

Reference marshes may or may not score 100%. Reference marshes are selected because they are minimally disturbed or because they are less disturbed than other salt marshes within a region — not because they are perfect. Most of the best marshes in New England show some signs of historical disturbance such as ditches or tidal restrictions. Similarly, study sites sometimes receive a higher HAS than reference sites. Some types of disturbance may seem important but may have little influence on overall habitat quality in a salt marsh — for example, the odor of sulfur that is natural. Investigators should examine both the overall HAS and the individual variables to fully understand how habitat may affect invertebrate communities.

Summary of ICI and HAS

The Salt Marsh Invertebrate and Habitat Summary Graph (Figure 4) is a graphical representation of the HAS and the ICI. The vertical axis of the graph represents the ICI and the horizontal axis represents the HAS. The graph provides a visual representation of salt marsh invertebrate community condition and provides some indication about the relative importance of habitat quality when marshes are plotted against the two axes. This graph is a valuable **evaluation tool** and has implications for planning and management in light of the rapid rate of development that is threatening many of New England's salt marsh habitats.

Typically, reference sites will be near the upper right hand corner of the graph and impacted sites will be closer to the lower left hand corner of the graph. If the ICI is very low (indicating impairment) yet the HAS is high (indicating good habitat), then it is likely that something other than the habitat variables you measured are causing **biological impairment** (for example, toxic pollution). Any anomalies — such as poor invertebrate communities and excellent habitat, or excellent invertebrate communities and poor habitat — should be followed with a more intensive investigation. This manual provides instructions on measuring tidal influence, salinity, and three other biological parameters — all of this information can complement your invertebrate study. Other types of information that may be helpful include **land use analysis**, a more detailed habitat assessment, measurements of water quality and sediment quality, and **toxicity tests** such as **Microtox** and **bioassays**.



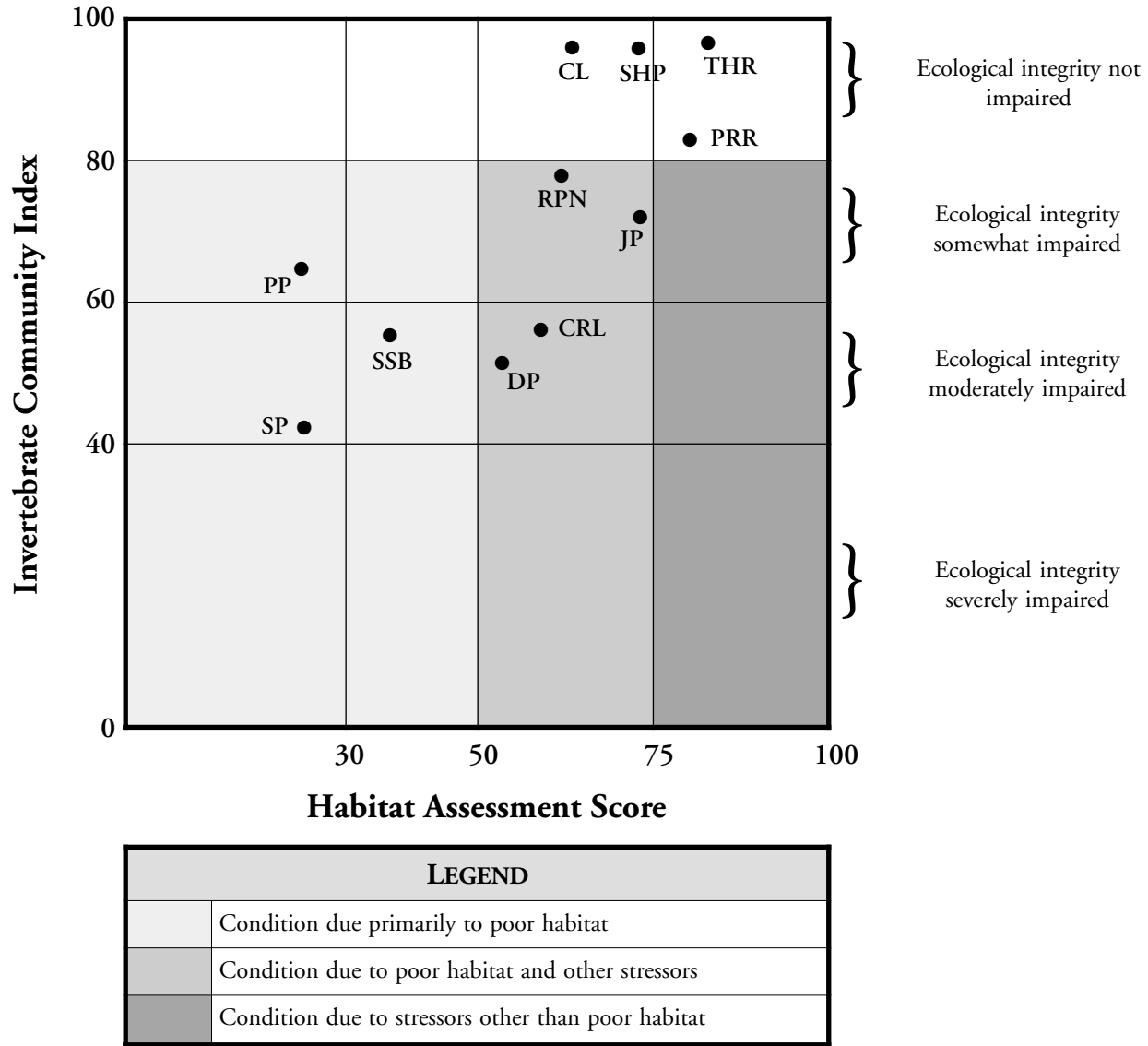
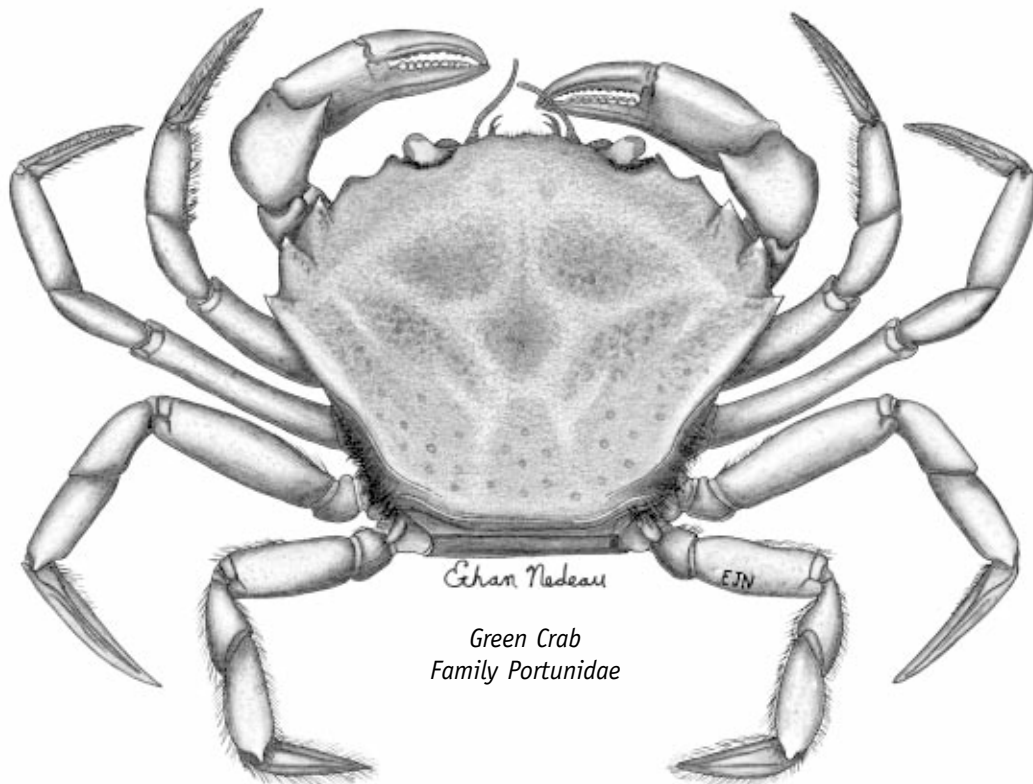


FIGURE 4. ICI & HAS SUMMARY GRAPH

The abbreviations on the graph represent different wetlands and are meant for illustrative purposes.

REFERENCES AND OTHER SUGGESTED READING

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Green Crab
Family Portunidae



NOTES



chapter five
APPENDICES

- APPENDIX 1.** FORM 1: SALT MARSH INVERTEBRATE FIELD SHEET
- APPENDIX 2.** FORM 2: INVERTEBRATE SAMPLES RECORD SHEET
- APPENDIX 3.** FORM 3: INVERTEBRATE LABORATORY BENCH SHEET
- APPENDIX 4.** FORM 4: INVERTEBRATE DATA FORM
- APPENDIX 5.** FORM 5: HABITAT ASSESSMENT SCORE SHEET
- APPENDIX 6.** FORM 6: ICI AND HAS SUMMARY GRAPH



FORM 1: SALT MARSH INVERTEBRATE FIELD SHEET

Monitoring Team: _____

Site #: _____ Date: _____

Site Name: _____ Latitude: _____

Topo Map Series: _____ Longitude: _____

Photographs Taken?: _____

Estimated size of Wetland Evaluation Area (WEA): _____

Approximate distance from site to seacoast: _____

Percent of Wetland Buffer at least 100' Wide (Circle One):

0 - 30% 30 - 50% 50 - 80% 80 - 100%

WEATHER (check appropriate boxes)

WEATHER IN PAST 24 HOURS	
Storm (Heavy Rain)	
Rain (Steady Rain)	
Showers (Intermittent Rain)	
Overcast	
Clear/Sunny	

WEATHER NOW	
Storm (Heavy Rain)	
Rain (Steady Rain)	
Showers (Intermittent Rain)	
Overcast	
Clear/Sunny	

HYDROLOGY

<p>1. Water Sources to the Marsh: (Circle those that apply)</p> <p style="text-align: center;">Precipitation Runoff Freshwater Stream/River Groundwater Tidal Influence</p>
<p>2. Average Tidal Range During Year:</p>
<p>3. Tidal Restrictions (number and describe each one, including dimensions for tidal movement)</p>

VEGETATION Note approximate abundance: N = Absent, R = Rare, C = Common, A = Abundant

VEGETATION - MARSH	
Salt Marsh Grasses	
Non-persistent Salt Marsh Plants	
Phragmites australis	
Scrub/Shrub Vegetation	

VEGETATION - STREAM	
Algae and Seaweed Attached to Banks	
Submerged Algae and Seaweed	
Floating Algae Mats	
Bare Substrate	

Evidence of Disturbance: _____

Evidence of Eutrophication: _____

ABUNDANCE OF FOOD FOR, AND PREDATORS OF, INVERTEBRATES

Note the approximate abundance:

- N = Absent
- R = Rare
- C = Common
- A = Abundant

FOOD / PREDATOR	
Green Slime on Banks	
Unattached Surface Floating Algae	
Attached Filamentous Algae and Other Seaweed	
Organic Detritus	
Soft-Stemmed Macrophytes	
Hard-Stemmed Macrophytes	
Macroinvertebrates	
Fish	

SEDIMENT TYPE AND QUALITY

Note approximate abundance and provide a short explanation if necessary.

N = Absent, R = Rare, C = Common, A = Abundant

SUBSTRATE TYPE	
Bedrock	
Boulders	
Cobble	
Gravel	
Sand	
Mud	
Peat	

SEDIMENT ODOR	
Normal	
Sewage	
Petroleum	
Sulfur	
Other	

Notes:

IMPACTS TO SALT MARSH

Check appropriate boxes and provide a short description.

FEATURE	check	DESCRIPTION
Drainage / Channelization		
Dredging		
Filling		
Bank Modification		
Bank Erosion and Slumping		
Vegetation Removal		
Invasive Species		
Dumping		
Hard Wall Structures		
Tracks through Marsh		
Recreational Activities		
Litter		
Other Impacts		

WATER QUALITY AND ODOR

Check boxes as appropriate, and provide a short explanation if necessary.

SUSPENDED MATERIAL	
None	
Algae	
Silt/Clay	
Fine Particulate Organic Matter	
Other	

WATER ODOR	
Normal	
Sewage	
Petroleum	
Sulfur	
Other	

Evidence of Eutrophication and Other Notes:

WATER CHEMISTRY

VARIABLE	MEASUREMENT		
	STATION 1 at 0'	STATION 2 at 150'	STATION 3 at 300'
Depth			
Temperature			
pH			
Dissolved Oxygen			
Conductivity			
Salinity			
Total Dissolved Solids			
Color			

INVERTEBRATE SAMPLES RECORD CHECK

Preservative Used (type, concentration):

Did you complete labeling and seal bags tightly?

Station	Sample	Sample #
Station 1	D-Net	
	Auger	
Station 2	D-Net	
	Auger	
Station 3	D-Net	
	Auger	

SKETCH OF MARSH

Include: approximate scale, WEA shape and dimensions, location of streams, direction of coastline, ditches, restrictions, surrounding land uses (including roads and storm drains), north direction, and any other relevant information. Include a legend if useful. Indicate the three sampling stations.

QUADRAT SAMPLES RECORD SHEET

SITE #:

FAMILIES / GROUPS	STATION 1 at 0'	STATION 2 at 150'	STATION 3 at 300'
Amphipods			
Talitridae			
Other			
Isopods			
Oniscidae			
Other			
Mussels			
Mytilidae			
Barnacles			
Balanoidae			
Other			
Crabs			
Portunidae (Green)			
Ocypodidae (Fiddler)			
Other			
Snails			
Melampodidae (Marsh)			
Littorinidae (Periwinkle)			
Other			
Orthoptera (Grasshoppers)			
Delphacidae (Planthoppers)			
Collembola (Springtails)			
Aranea (Spiders)			
Acari (Mites)			
Diptera (Flies)			
Homoptera (Aphids, Leafhoppers)			
Other:			
Other:			
Other:			
Other:			
Other:			
Other:			

FORM 3: INVERTEBRATE LABORATORY BENCH SHEET

Site Number: _____ Sample Number: _____
 Technician: _____ Phone Number: _____
 Date: _____
 D-Net Number: _____ Auger Number: _____ Quadrat Number: _____

PHYLUM/CLASS	ORDER	FAMILY	FG	TALLY	TOTAL
Turbellaria	-	-	DF		
Rhynchocoela	-	-	DF		
Nemertea	Heteronemertea	-	DF/PR		
	Other				
Sipunculoidea	Peanut Worms		DF		
Oligochaeta	Haplotaxida	Naididae	DF		
		Tubificidae	DF		
		Other			
Polychaeta	Capitellida	Arenicolidae	PR		
		Capitellidae	DF		
		Maldanidae	DF		
	Cossurida	Cossuridae	DF		
	Ctenodrilida	Parergodrilidae	DF		
	Eunicida	Arabellidae	DF		
		Dorvilleidae	OM		
		Eunicidae	DF/OM		
		Lumbrineridae	DF/SF		
		Other			
	Opheliida	Opheliidae	DF		
		Scalibregmidae	DF		
	Orbiniida	Orbiniidae	DF		
	Phyllodocida	Glyceridae	DF/PR		
		Goniadidae	PR		
		Hesionidae	DF/G		
		Nereidae	OM		
		Nephtyidae	PR		
		Phyllodocidae	DF/OM		
		Pisionidae	PR		
		Sigalionidae	DF		
		Syllidae	OM		
	Sabellida	Sabellidae	SF		
		Spirorbidae	SF		

PHYLUM/CLASS	ORDER	FAMILY	FG	TALLY	TOTAL
	Spionida	Spionidae	DF		
	Cirratulida	Cirratulidae	DF		
		Paraonidae	DF		
	Terebellida	Ampharetidae	DF		
		Terebellidae	DF		
	Unknown				
Cephalopoda		Loliginidae	PR		
		Ommastrephidae	PR		
Polyplacophora	Ischnochitonida	Chitonidae	G		
Gastropoda	Archaeogastropoda	Acmaeidae	G		
	Neogastropoda	Buccinidae	PR/DF/SF		
		Columbellidae	G		
		Nassariidae	G		
	Mesogastropoda	Calyptraeidae	SF/SC		
		Cerithiidae	G		
		Hydrobiidae	G		
		Lacunidae	G		
		Littorinidae	G		
	Basommatophora	Melampodidae	G		
	Nudibranchia	Elysiidae	PR		
		Polyceridae	PR		
Pelecypoda	Veneroidea	Mesodesmatidae	SF		
		Tellinidae	SF/DF		
		Veneridae	SF		
(Bivalvia)	Myoidea	Myidae	SF		
	Myoidea	Pandoridae	SF		
	Mytiloidea	Mytilidae	SF		
	Nuculoidea	Nuculanidae	SF		
	Ostreoidea	Ostreidae	SF		
	Ostreoidea	Pectinidae	SF		
Echinoidea			DF/G		
Stelleroidea	Sea Stars		PR/G		
Holothuroidea	Sea Cucumbers		DF		
Hemichordata	Acorn Worm		SF/DF		
Crustacea	Decapoda	Crangonidae	DF/SC/PR		
		Hippolytidae	DF		
		Palaemonidae	DF/SC/PR		
		Pandalidae	DF		
		Penaeidae	DF		
		Cancriidae	OM		

PHYLUM/CLASS	ORDER	FAMILY	FG	TALLY	TOTAL
		Hippidae	OM		
		Majidae	OM		
		Paguridae	SC		
		Pinnotheridae	OM		
		Portunidae	OM		
		Xanthidae	G		
		Other			
	Cirripedia	Balanoidae	SF		
	Amphipoda	Ampeliscidae	DF/SF		
		Ampithoidae	DF		
		Aoridae	DF		
		Calliopiidae	DF/SF		
		Gammaridae	DF/G		
		Haustoriidae	DF		
		Hyalidae	DF		
		Ischyroceridae	DF		
		Talitridae	DF		
	Isopoda	Idoteidae	DF		
		Janiridae	DF/PR		
		Limnoriidae	DF		
		Oniscidae	DF		
		Other			
	Tanaidacea	Leptocheliidae	DF		
	Cumacea		DF		
Insecta	Collembola		DF		
	Diptera	Chironomidae	DF/SF/PR		
		Culicidae	SF/PR		
		Tabanidae	PR		
	Hemiptera		PR/G		
	Homoptera		G/PR		
	Odonata		PR		
Acachnida	Araneae	Clubionidae	PR		
		Micryphantidae	PR		
		Salticidae	PR		
	Acari (Acarina)	Mites	PR		
Others					
Total Taxonomic Groups:			Total Number of Individuals:		

FORM 4: INVERTEBRATE DATA FORM

Site Number: _____
 Date Sampled: _____
 Date of Lab Work: _____

Salt Marsh: _____
 Name(s): _____

TAXA	FG	D1	D2	D3	\bar{D}	%
NEMERTEA (N)						
Heteronemertea	DF/PR					
Other						
Subtotal N						
CAPITELLIDA (C)						
Arenicolidae	PR					
Capitellidae	DF					
Others						
Subtotal C						
COSSURIDA (CO)						
Cossuridae	DF					
Other						
Subtotal CO						
CTENODRILLA (CT)						
Parergodrillidae	DF					
Others						
Subtotal CT						
EUNICIDA (E)						
Arabellidae	DF					
Dorvilleidae	OM					
Lumbrineridae	DF/SF					
Onuphidae	PR					
Others						
Subtotal E						
ORBINIIDA (O)						
Orbiniidae	DF					
Paraonidae	DF					
Subtotal O						

TAXA	FG	D1	D2	D3	\bar{D}	%
SABELLIDA (S)						
Sabellidae	SF					
Other						
Subtotal S						
SPIONIDA (SP)						
Spionidae	DF					
Other						
Subtotal SP						
OPHELIIDA (OP)						
Opheliidae	DF					
Subtotal OP						
PHYLLODOCIDA (P)						
Glyceridae	PR/DF					
Goniadidae	PR					
Hesionidae	DF/G					
Nereidae	OM					
Nephtyidae	PR					
Phyllodocidae	DF/OM					
Polynoidae	PR/SF					
Sigalionidae	DF					
Syllidae	OM					
Other						
Subtotal P						
TEREBELLIDA (TE)						
Terebellidae	DF					
Ampharetidae	DF					
Subtotal TE						

TAXA	FG	D1	D2	D3	\bar{D}	%
UNKNOWN POLYCHAETA (UP)						
Unknown						
Subtotal UP						
AMPHIPODA (A)						
Ampithoidae	SF					
Caprellidae	SF					
Gammaridae	SF/G					
Hyalidae	SF					
Ingolfiellidae	SF					
Ischyroceridae	SF					
Talitridae	SF					
Other						
Subtotal A						
TANAIDACEA (T)						
Leptocheliidae	DF					
Subtotal T						
GASTROPODA (G)						
Acteonidae						
Cerithiidae	G					
Columbellidae	G					
Hydrobiidae	DF					
Littorinidae	G					
Melampodidae	G					
Nassariidae	G					
Other						
Subtotal G						
DECAPODA - Shrimps (DS)						
Crangonidae	DF/SC/PR					
Hippolytidae	DF					
Palaemonidae	DF/SC/C					
Pandalidae	DF					
Penaeidae	DF					
Other	DF					
Subtotal DS						

TAXA	FG	D1	D2	D3	\bar{D}	%
DECAPODA - Crabs (DC)						
Cancridae	OM					
Majidae	OM					
Xanthidae	SF					
Other						
Subtotal DC						
ISOPODA (I)						
Idoteidae	DF/C					
Janiridae	DF/SC					
Limnoriidae	DF					
Other						
Subtotal I						
PELECYPODA (PE)						
Mesodesmatidae	SF					
Myidae	SF					
Mytilidae	SF					
Nuculanidae	SF					
Tellinidae	SF/DF					
Veneridae	SF					
Other						
Subtotal PE						
OTHER GROUPS (OG)						
Cumacea	DF					
Echinodermata	DF					
Insecta	Mixed					
Acarina	PR					
Aranea	PR					
Merostomata	PR					
Nudibranchia	PR					
Oligochaeta	DF					
Polyplocophora	G					
Turbellaria	SF					
Urochordata	SF					
Other						
Subtotal OG						

PERCENT COMPOSITION OF MAJOR GROUPS

SUBTOTALS	NUMBER	PERCENT
Subtotal N		
Subtotal C		
Subtotal CO		
Subtotal CT		
Subtotal E		
Subtotal O		
Subtotal S		
Subtotal SP		
Subtotal OP		
Subtotal P		
Subtotal TE		
Subtotal UP		
Subtotal A		
Subtotal T		
Subtotal G		
Subtotal DS		
Subtotal DC		
Subtotal I		
Subtotal PE		
Subtotal OG		
TOTAL		

PERCENT COMPOSITION OF MAJOR FEEDING GROUPS

FEEDING GROUP	PERCENT
Predator (PR)	
Deposit Feeder (DF)	
Grazer (G)	
Omnivore (OM)	
Scavenger (SC)	
Suspension Feeder (SF)	
Mixed	
TOTAL	

PERCENT INVASIVE SPECIES

SPECIES	PERCENT
Littorina littorea	
Palaemon macrodoctylus	
Hemigrapsus sanguineus	
Carcinus maenas	
TOTAL	

PERCENT INSECTS, SPIDERS, MITES

GROUP	PERCENT
Insects (Insecta)	
Spiders (Aranea)	
Mites (Acarina)	
TOTAL	

SUMMARY

Average Number of Organisms	
Taxonomic Richness	
% Contribution of Dominant Taxonomic Group	
% Contribution of Dominant Feeding Group	
% Insects, Spiders, and Mites	

FORM 5: HABITAT ASSESSMENT SCORE SHEET

Site Number: _____

Site Name: _____

Form Completed by: _____

Phone: _____

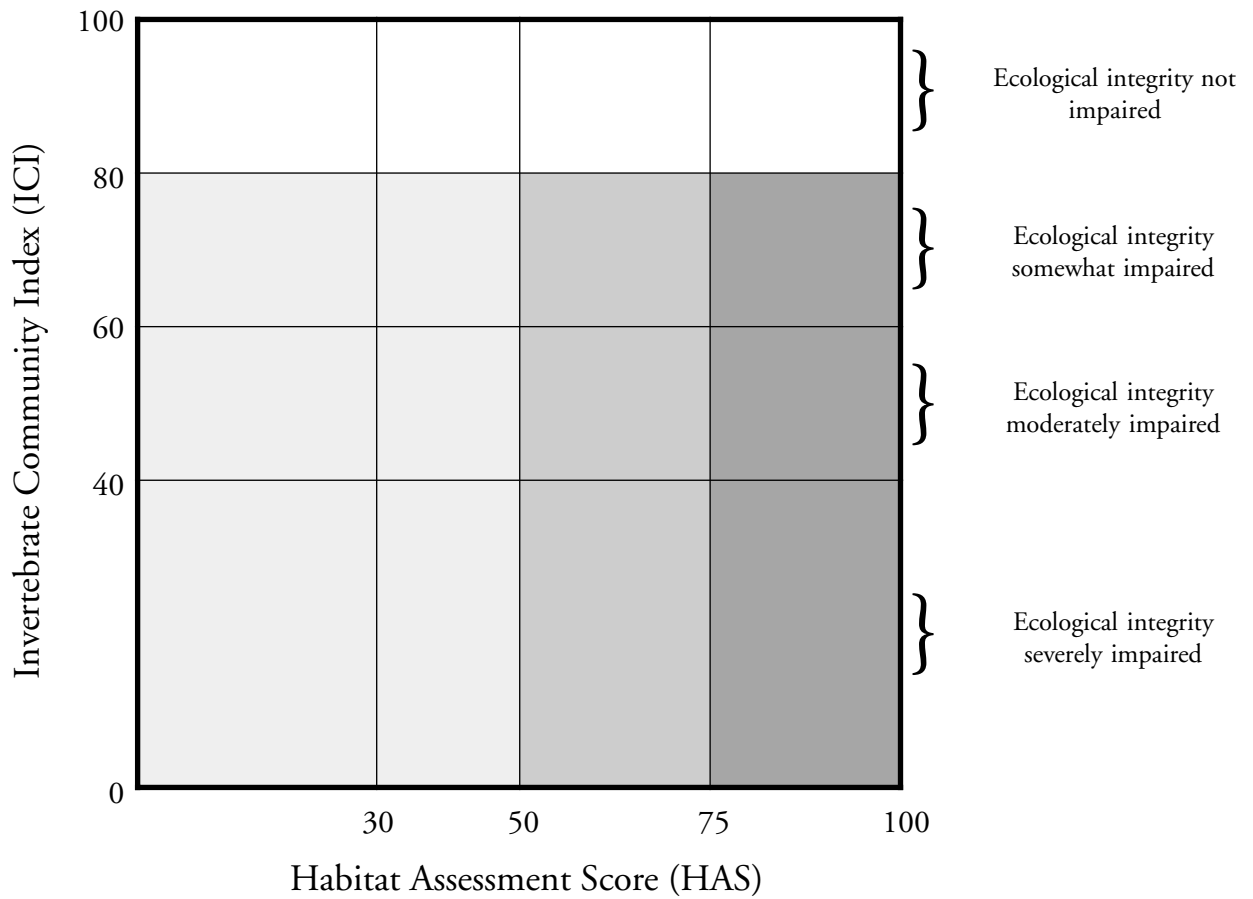
Use the range from 0-5 to score each condition. Half numbers (e.g. 3.5) are permissible).

INDICATOR	4 - 5	2 - 3	0 - 1	SCORE
Size of estuarine salt marsh	Larger than 20 acres (8 hectares)	Between 20 and 4 acres (8 -1.6 hectares)	Less than 4 acres (1.6 hectare)	
Tidal flushing	Natural tidal surges are unimpeded	Some modification to natural fluctuation	Salt marsh cut off from normal tidal fluctuation	
Outlet restriction	No outlet restriction	Outlet restriction between 30' and 5'	Outlet restriction < 5'	
Erosion of banks	Normal estuarine bank erosion with little slumping	Some evidence of accelerated bank erosion; slumping in progress	Severe bank erosion; slumping is common, stream widening occurring	
Vegetation cover	Even and complete vegetation cover of salt marsh	Some patches of exposed ground evident	Large areas of marsh are unvegetated	
100' vegetated wetland buffer	> 80%	80-40%	40%	
Nature of substrate within estuarine stream	Composed of a mixture of substrates: sand, silt, mud, and organic matter present	Mixture of two types of substrate	Predominantly one substrate type	
Evidence of freshwater intrusion	No evidence of fresh-water intrusion (Specific Conductivity > 5,000)	Some evidence of fresh-water intrusion (Specific Conductivity between 5,000 and 800)	Conductivity below 800, or little or no evidence of salt water	
Food sources for invertebrates*	Abundance of aquatic macrophytes, attached macro-algae, periphyton, CPOM and FPOM	Some attached algae and periphyton with CPOM and FPOM	No aquatic macrophytes, attached algae, or periphyton; only some CPOM and FPOM	
Degree of impact from human activities**	No human impact evident	Low to medium level with minimal impact	High level with marsh severely degraded	
TOTAL SCORE				
PERCENT SCORE [(TOTAL SCORE / 50) x 100]				

* CPOM = Coarse particulate organic matter, FPOM = Fine Particulate Organic Matter

** Disturbance from fishing, swimming, boating, trails, roads, vegetation removal, ditching, shoreline modification, solid waste dumping, etc.

FORM 6: ICI AND HAS SUMMARY GRAPH



LEGEND	
	Condition due primarily to poor habitat
	Condition due to poor habitat and other stressors
	Condition due to stressors other than poor habitat