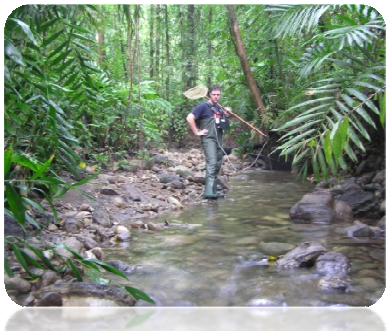
Freshwater fish survey methodology for streams & rivers on tropical oceanic islands





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A brief guide based on the freshwater survey work conducted as part of the Fiji Ecosystem Based Management Project

EBM FIJI ISLANDS

Background

Streams and rivers on islands are considerably different to continental systems in that oceanic island systems are often subject to recurrent flash flooding and many fauna in continental systems are only distantly related to island fauna, rarely having an obligate marine larval phase of their life cycles. Several methods used in surveying continental systems are therefore only of limited applicability in island systems. The field procedures described herein are loosely adapted from those described in Parham, 2005 and Fitzsimons *et. al*., 2007 and refined from the field experiences of the author. These methods are designed to provide the most comprehensive documentation of fishes present in a variety of stream and river habitats in tropical oceanic island ecosystems while also providing an accurate snapshot of the habitable space. In our study for the Fiji Ecosystem Based Management (EBM) project we used fishes, particularly gobioid (Families Gobiidae & Eleotridae) as primary indicators of environmental quality as fishes are taxonomically most well studied and most observable inhabitants of these systems, represent critical components of the food web from primary consumers (herbivores) to predators and because of their recognized value as a food source for inland communities.

Site selection and standardization

Our study was designed to determine the abundance, diversity and biomass of fishes within different reaches of the system during the wet and dry seasons. We surveyed a range of habitats from small creeks to large turbid rivers, torrential mountain streams, mangrove swamps and an upland lake. Specific collecting/survey methods were selected depending on the habitat type and are discussed in the following sections. Rivers and streams were selected on the windward and leeward sides of the island and sampled once during the wet season and once during the dry. Each river or stream was divided into three sections coinciding with known distribution of adult animals. We designated these three sections as lower reach, middle reach and upper reaches. Lower reach sites are generally from the river mouth to the first major obstacle to fish passage (ie. waterfall, culvert, weir). Estuaries, with a free connection to the open ocean and mixing of salt and freshwater, were included in the investigation of lower reach sites. A second lower site was usually conducted just beyond the tidal reach of salty water. Middle reach sections were in pure freshwater at a moderate to low incline with riffle, run and pool development. Again, usually two sections of middle reaches were sampled. One was just above the first major obstacle and another sample was taken 100 - 200 meters further upstream. Upper reach sites are generally characterized by steep gradient headwater areas with waterfalls and plunge pools. Our approach was generally to sample once above the largest headwater waterfall and once below, although variation in length, grade and waterfall formation meant this was not always possible. Overall, we generally sampled twice in each reach in each season. As many techniques were used as possible at each site to gain the most comprehensive understanding of species

presence or absence. Standardization of sampling included apparatus used, length of reach sampled, sampling time and number of surveyers. In general, 50 meter sections of streams and rivers were sampled with a combination of a single electrofishing apparatus, several seine and hand nets by 4 – 6 surveyers, from the downstream section of the site working upstream and were sampled for approximately one hour per site.

Figure 1. Characteristic upper reach (Dreketi River), middle reach (Suetabu River) and lower reach (Kilaka River) sampling sites.

Description of sampling site

Upon selection of the sampling site we embarked on a systematic description of the site to provide a record of the habitat conditions and allow study of conditions suitable for fish habitation. Firstly, two digital photographs were taken facing upstream and then downstream of the sampling site. The site data sheet (Appendix 1) was filled out with field station number, water body name, date, start time, GPS point, collectors, weather condition and a rough site map was drawn. At each sampling site a GPS position and altitude were taken using a Garmin GPS *map* 76Cx. Water quality characteristics were taken before entering the water to minimize disturbance to natural water characteristics. Temperature, pH, conductivity, salinity and dissolved oxygen were taken using a hand-held YSI multi-meter. Turbidity was taken using a turbidity tube calibrated to Nepthalometric Turbidity Units. Flow rate was taken by floating a plastic lid over a marked ten meter section and timing with a stopwatch. Brief notes were also taken on riparian vegetation and instream condition with particular emphasis on substrate type, flow type, instream cover, aquatic vegetation, riparian vegetation, land use type and major disturbance type. Water body maximum width and depth were also taken using a waterproof fibreglass tape.

Rationale and generalized methods for measuring key habitat characters:

Temperature (^{o}C)

The distribution and abundance of aquatic plants and fishes changes is partly shaped by water temperature. Changes in temperature will alter the amount of oxygen dissolved in the water with high temperatures decreasing the amount of dissolved oxygen available and also affecting the rate of photosynthesis by algae and other plants. Increases in water temperature will also cause an increase in the metabolic rate of organisms in the water. Increased metabolism increases the oxygen demand of fishes, insects and bacteria. A short period of high temperatures each year can make the stream unsuitable for sensitive organisms even though the temperature during the rest of the year is suitable. Some species have different temperature requirements at different stages of life. Generally fish larvae tolerate a narrower range of temperature than do adult fish. Fishes can generally tolerate slow changes in temperature. Thermal stress can occur where the temperature changes more than 1 to 2 degrees in 24 hours. Temperature rises are often caused by the discharge of heated water into waterways, reduced flow of water due to siltation or damming, reduction in shading along rivers due to deforestation, as well as through increased turbidity from agricultural runoff or algal blooms. Temperature was taken 15 cm below water surface using the YSI handheld probe submerged until the value stabilized.

pH

pH is a measure of how acidic or basic the water is. On a scale of 0 (extremely acidic) to 14 (extremely basic), water usually has a pH of between 6.5 and 8.5 and is the preferable range for most aquatic organisms. Variation of pH naturally occurs due to the geology of the watershed (eg. limestone produces more basic water) and salinity (salt water is more basic than freshwater with normal seawater having a pH level of around 8) and organisms are adapted to live within the naturally occurring pH level in their ecosystem. A change of pH, even slight changes for some organisms can cause death, with immature (larval) stages of insects, amphibians and fish being very sensitive to pH levels below 5. Human activities can change the pH levels of water. Air pollution from motor vehicle emissions containing sulphur dioxides and nitrous oxides can increase the acidity of the water by forming sulphuric and nitric acid. Industrial water, agricultural runoff or drainage from improperly run mines all affects pH levels. Rapidly growing algae and submerged vegetation caused by elevated phosphate and/or nitrate levels can remove carbon dioxide from the water during photosynthesis. This can result in a significant increase in pH levels in a waterway. pH was taken using theYSI hand held meter submerged 15 cm below the water surface until the value stabilized.

Dissolved Oxygen (mg/L)

Dissolved Oxygen can range between 0 and 14 mg/L and is affected by temperature and salinity. As water temperature changes, the highest potential dissolved oxygen level changes. Lower temperatures result in higher potential dissolved oxygen levels and higher temperatures result in lower potential dissolved oxygen levels. The tropical climate of Fiji and the subsequent warm temperature of the water cause the natural levels of dissolved oxygen to be quite low. This temperature effect is compounded by the fact that living organisms increase their activity in warm water, requiring more oxygen to support their metabolism. Critically low oxygen levels often occur during the warmer months when decreased capacity and increased oxygen demand, caused by respiring algae or decaying organic material, coincide. Naturally occurring salts found in estuarine and marine waters also lowers the levels of dissolved oxygen. An increase in water temperature due to high turbidity levels, discharge of heated water into waterways, reduced flow of water due to siltation/damming or a reduction in shading along rivers due to deforestation can also decrease dissolved oxygen levels. An increase in aquatic plants/algae through fertiliser runoff or sewage contamination can also decrease the available oxygen in the water. Damming waterways or removing riffles (rocky shallow areas) can decrease the oxygenation of the water through a reduction in the speed of flow or churning of the water. Dissolved oxygen levels below 3 mg/L are stressful to most aquatic organisms. Levels below 2 mg/L are not enough to support life whilst levels between 2 – 4 mg/L are only acceptable to a few forms of organisms that are adapted to low oxygen levels. 4-7 mg/L of dissolved oxygen is acceptable for warm water fish as they are adapted to the low levels of dissolved oxygen found in warm waters. Dissolved oxygen levels can also be expressed as a percentage of the maximum possible dissolved oxygen levels at a given temperature. Percentage saturations of over 90% are excellent, 71-90% good, 50-70% fair and below 50% are poor. Saturation levels of between 60-79% are acceptable for most aquatic organisms. Dissolved Oxygen was measured using the YSI hand held probe submerged 15 cm below the water surface until the value stabilized.

Conductivity (µS) and Salinity (ppt)

Measurements of conductivity and salinity assess amounts of dissolved ions such as Calcium, Potassium, Chlorides and Bicarbonates present in water. As such, salinity and conductivity are related. In fact we can roughly estimate salinity by multiplying conductivity by 0.64. Problems in aquatic systems often occur when deep rooted vegetation is removed and water infiltrates soil bringing deep salts to surface. The water then evaporates leaving high salt concentrations to wash into the water body. Many species can only survive in certain salinity ranges so changes in salinity can change the variety of species present. These measures also give us an idea of which species prefer particular salinity or conductivity regimes. Salinity values are also important for determining if electrofishing can be undertaken. Electrofishing is ineffective and can damage the electrofisher in salinities around 5 ppt. We did not use electrofishing in low reach sites with higher salinities.

Conductivity and salinity were measured using a YSI hand held probe submerged 15 cm below the water surface until values stabilized.

Current speed (m/s):

Current speed is a limiting factor for fish communities, preventing certain species from living in an area. Also, fast moving streams tend to have higher levels of dissolved oxygen. Our field method of estimating current speed was by floating a plastic lid over a marked ten meter section and timing with a stopwatch. A tip for this method is to start a few meters before the measured area and place floater in mid-stream in estimated "mean" flow conditions for the site. A commercially available flow probe, however, would yield more precise results.

Turbidity (NTU)

High turbidity decreases the amount of light passing through the water column which limits plant growth also affecting the fish communities which feed on and live in the plants. High levels of turbidity reduce the ability of the water to support a large variety and number of aquatic organisms. Where there is less light penetrating the water, there will be less photosynthesis occurring and this reduces the levels of oxygen in the water. Also, the water becomes warmer because suspended material absorbs heat from the sun. This also decreases the amount of oxygen dissolved in the water. As many fishes are visual predators, high turbidity levels will also reduce their ability to forage effectively and to avoid predation. To measure turbidity we used a turbidity tube calibrated to Nephthalometric Turbidity Units. Water is added to the tube until the black indicator lines at the base of the tube are no longer distinguishable and a measurement is taken when this point is reached. If the value was greater than 200 NTU's then we filled with 2 x the water and multiply the NTU value by 2.

Water body width, depth (m)

Measuring the maximum water body width and depth of a sampling site gives a rough approximation of the volume of water accessible as habitat for fishes. If these values are used in combination with the current speed this can also give an approximation of flow rate (m^3/s). Seasonal variation in stream width and depth can significantly change not only the amount of habitable space but also the degree of fish community interaction with bank vegetation and the riparian zone, altering the physicochemical characteristics of the water body. We used a waterproof fibreglass measuring tape to measure both maximum width and depth. Often maximum depth was gained by using a stick or length of bamboo to probe for the deepest area and then the probe was measured to where the water height reached.

Riparian zone

The riparian zone pertains to the banks and other adjacent terrestrial environs of the water body. It is the terrestrial/aquatic interface and very important in determining the structure and function of stream

ecosystems. Riparian vegetation has critical function in prevention of erosion, moderating water temperature, providing input of plant detritus and insects for biological energy, providing woody debris for habitat and pool creation, slowing flood velocities, maintaining base flows and filtering pollutants. While we did not spend a great deal of time characterizing the riparian zone during our study we did photograph the stream banks for later characterization. We also used a modified data sheet from Queensland Government stream condition assessment (Appendix **) which included assessment of adjacent land use type, % cover by riparian vegetation type, % canopy cover and major disturbance type and rating. This was not done systematically but will be the subject of intensive and directed study in following stream assessment work to follow.

Collection of fishes

As many collection techniques were used as possible at each site to gain the most comprehensive understanding of species presence or absence. The following apparatus and techniques were used:

Electro-fisher (Deka 3000, 600V, 10A; Smith-Root LR-25; 500V, 10A) a primary sampling tool in river and streams. Wearing rubber waders and never venturing deeper than 1.5 meters, the anode (on a meter long rod) was discharged while two people (also wearing rubber waders) held a medium -sized, 1 mm^2 mesh net across the stream several meters upstream from the anode. As the anode reached the net, it was raised and fauna within the net were placed in a water-filled plastic bucket. Care must be taken to not touch water with uninsulated skin while the electro-fisher is being discharged. Stunned fish were also captured by small hand nets in between pulses.

Figure 2. Using the Deka 3000 electrofisher. The surveyer should, however, be wearing insulated rubber gloves as well.

Gill net *(25m x 1.8m, 1 inch mesh)* were used in lower sites and deployed with the floaters along the top edge and the lead weights along the bottom. Gill nets are the most effective way to sample large rivers, but due to their size, expense, and time involved setting and retrieving, are not commonly used for rapid surveys. While we were limited by net availability, it is advisable to use variable mesh sizes to ensure that a range of species are captured**.** Soak time was approximately one hour before the net was removed.

Figure 3. Deploying a gill net in the lower Dreketi river.

Large seine net (2 m x 7 m, 0.4 cm^2 mesh) This net was pulled in a rough circle, with the bottom edge down as close as possible to the substrate and forward of the top floating edge of the net. This technique was executed before anyone could set foot in the water body to minimize the number of fleeing fishes. This was generally used in minor tributaries and slow moving or still waters.

Figure 4. Large seine net being pulled (white) and medium pole seine being used simultaneously in a small slow flowing stream

Medium pole seine net (1.2 m x 0.8 m, 1mm^2 mesh) was used in a variety of ways. Firstly, it was held firmly downstream as people kick and dislodge rubble upstream. This is a useful method for collecting small, bottom dwelling fish. On vegetated banks the net was thrust under submerged vegetation and the vegetation was disturbed on the bank dislodging fishes into the net. Also, this net was used to "scoop" (bottom edge held forward, run along substrate for a few seconds then lifted) from any accessible shallow body of water. This net was particularly useful for narrow streams and the net most commonly used in conjunction with the electro-fisher.

Small hand nets (15cm x 10cm + 10 cm x 8 cm, 1mm^2 mesh) These were used to "scoop" the underside of overhanging rocks and in small crevices in the smaller streams and also to collect fauna when in still water bodies. These were also often used in conjunction with the electrofisher in between pulses to collect stunned fishes.

Observations (mask and snorkel) This method should only be used in clear streams where there is no possibility of bull sharks. It is very effective for obtaining a very quick overview of the local fish population and relative abundance of species. The method essentially consists of making underwater observations with the use of mask and snorkel. In areas that were shallow enough and the water was clear enough, a mask and snorkel were used to observe the benthos and fauna that were not being caught by the nets. Notes were recorded on plastic slates or special waterproof paper.

Figure 5. The author making underwater observations using a mask.

Additional sampling methods not used during this study

Rotenone 8.4% (powder form): Rotenone, a poison derived from the derris plant, is ideal for collecting fishes in small creeks or sections of larger streams where current flow is minimal. However, it requires experience to know how much to use depending on the size of the stream and rate of current flow. It is one of the best methods for obtaining a comprehensive sample as all species can be collected with this method. In general, mix approximately 0.5 to 1.5 kg of rotenone powder with several litres of water. Then disperse this solution over a period of 5 to 10 minutes. After several minutes of exposure, the stunned fishes begin to gasp at the surface and are easily netted. A seine net is generally used to block off the downstream end of the collecting area. Rotenone stations are frequently made in small creeks near their junction with larger streams. A representative yet minimal sample of fishes can be obtained in this manner as the rotenone is quickly rendered inactive when diluted by the flow of the larger stream. The main disadvantages of rotenone is that it is expensive (about AUD $350 per 22 kg bag), cannot be stored for long periods unless kept cool, and it is heavy to transport. As its use is illegal in Fiji and many countries, we did not use it during our surveys.

Spear**:** A small multi-prong spear is effective for collecting elusive fishes such as lutjanids, kuhliids, gobies, etc. It is used while swimming underwater with mask and snorkel equipment. The device is simple to make, consisting of a 1-meter long piece of spring-steel (3 mm diameter) and about 5-6 15-cm long prongs (1 mm diameter) that are glued to the main shaft with epoxy resin. The spear is propelled with a piece a rubber that is attached on one end to a 15 cm long piece of bamboo, aluminium tube, drilled out piece of broom stick, or similar hollow device and a small piece of partially drilled out wood on the other end, which receives the end of the spear after it is threaded through the hollow tube.

***Hook and line*:** (8 lb test line, 3.5 cm hook) These hooks are baited predominantly with large local insects or larvae and are thrown from the bank into the larger, faster moving, water bodies that cannot be fished effectively by any of the other methods. A small weight attached to the line to aid in the casting process. This method can be used in the major tributaries and fast moving rivers in conjunction with traps and other local methods.

Cone trap: This is generally a conical woven bamboo basket with a baited, spring-loaded circular door (1.2 m length with 20 cm diameter opening at one end). These traps are also baited with insect and placed in crevices underwater along the banks of the larger, faster water bodies. There are also other variations to these traps.

Interviews (Fish markets/ on fishing grounds): This is a good way to see if your collections are fairly complete and can gives you an idea of the important local food fishes and relative abundance. If there are interesting fishes that you have not managed to collect you can usually buy them and preserve them on the spot. Important questions to ask are: by what methods they used to catch the fishes, where they caught the fishes, what time of day they caught the fishes, how long it took to catch the fishes, are they seasonal and do they prefer a certain type of habitat. It is also useful to have a colour field guide to use in combination with your questions about certain fishes. It is very useful to pay attention to and even use any other traditional methods of catching fishes that you see. The locals are generally more knowledgeable than you at catching their local fishes and know where the best spots are. This local knowledge will also provide important insights into the biology of the local ichthyofauna.

Preservation of specimens

We collected fishes for positive identification by museum authorities and to create a reference collection for USP and regional museums. The recommended method of preservation is that used by museum biologists. The main ingredient is full-strength formalin, which can be obtained from a pharmacy, university laboratory, or fisheries station. It is a dangerous chemical and care must be taken to avoid breathing the fumes (where possible use a fume hood). This chemical should always be handled with gloves and, obviously, it must be kept out of reach of children. The preserving solution is made by diluting one part of formalin with nine parts of water. The fish should be fully immersed in the solution. If larger than 12 to 15 cm a slit along the side of the belly will facilitate preservation of the internal organs. For long-term storage it is best to transfer the specimen to a 70 per cent solution of ethyl alcohol (7 parts ethanol, 3 parts water) after the fish is fixed in formalin (that is, after several weeks). If formalin or ethanol are not available, any alcohol based solution such as methylated spirits can be used. Anyone sending specimens to a museum or other institution should first contact

appropriate staff for detailed instructions on packing and shipping. Freezing is another option, which avoids messy chemicals and is good for short-term storage, particularly if the specimen can be hand-delivered to local authorities. If specimens are needed for DNA work a clip of the caudal fin can be placed directly in a small vial of 80% plus ethanol.

For our study, voucher specimens were collected, fixed in a 10% formalin solution and transferred to 70% ethanol solution after 1-2 weeks of fixation. Some specimens were stored directly in 80% ethanol for DNA analysis. Voucher specimens were deposited at the University of the South Pacific, Suva collections. As color loss is rapid, accurate preservation of color patterns was recorded by photography. Fresh specimens were placed in a portable aquarium with some local aquatic vegetation and benthos to enhance the photography. Some tips for good photos are to remove small bubbles on glass with paint brush and to photograph with sun behind and/ or with a flash angled to avoid reflection from the glass.

Figure 6. The author using a small portable, photography aquarium to capture live colours of a specimen.

For larger fishes that cannot fit into the aquarium, the following steps ensure the photos are of good diagnostic quality:

- The specimen should be photographed when fresh to avoid rapid fading.
- Fins should be spread to show their true shape.
- Wet fish should be blotted dry to prevent harsh glare.
- The fish should be placed on a contrasting background.
- The length of the fish should be recorded for future reference.

Fins of small fishes can be held erect with sewing pins on a piece of flat Styrofoam or cardboard. If full-strength formalin is then applied to the fins with a small paintbrush or eye-dropper and allowed to set for a few minutes, they will remain stiff when the pins are removed.

Identification of fishes

In general, the fish fauna of oceanic Pacific islands is quite poorly known, particularly at the species level within the highly speciose gobioid fishes. Based on previous work by the first author and students in Fijian rivers and streams, we constructed a waterproof set of index cards called the Fiji Freshest Fishdex, with the known freshwater/estuarine fish fauna on one side and the name on the other. These were used for both field identification and training of field identification (available from Wetlands International – Oceania). We also commonly used the field guides of Allen, 1991 and Kieth *et al.,* 2002 from Papua New Guinea and New Caledonia respectively. Other useful identification sources are listed in references. Unidentifiable fishes were noted according to definable characteristics in the field, photographed and preserved. All fishes that were not clearly identifiable were taken back to the USP Marine lab and available keys from the literature were used to key out all specimens. Particularly difficult species were sent to museum specialists in those particular taxonomic groups for confirmation.

General fieldwork plan for each site

- Photograph sampling site
- Start filling out data sheet – field station #, River name, Date, Start time, GPS, Collectors, Weather, Site Map
- Get pH, Temp and Turbidity before getting in the water
- If clear water then inspect by mask and snorkel and record species seen (can do a transect)
- Start sampling, work from downstream to upstream to avoid upstream water quality disturbance
- Record species caught, record number returned and size of specimens returned
- Record interesting remarks on behaviour, community structure, habitat…..
- Record stop time, length sampled, width and depth
- Keep species alive in plastic bags or bucket for photography on site if possible (separate photo voucher specimens)
- Kill fish in saturated bicarbonate soda solution then preserve (1 specimen in alcohol and rest in formaldehyde)

- Move to next site

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APPENDIX 1 - Data Sheet

Code _____ Date _____ Northing _____ Easting _____ Zone _____

Recorder Initials _____ Time _____ Air Temp. _____ Site Length _____

Water Temp. _____ % Oxygen Sat. _____ Conduct (uS/cm) _____ Turbidity (NTU) _____ pH _____

Site Location _____ Overall Rating _____ Canopy Cover (%) _____

% SUBSTRATE

Flow Type	% of Stream	Av. Width (m)	Av. Depth (m)	Max. Depth (m)	Flow (cm/s)	Rock	Boulder	Cobble/Pebble	Gravel	Sand	Silt

LAND USE

- Rainforest ☐
- Bananas ☐
- Sugar Cane ☐
- Orchards ☐
- Tea ☐
- Urban ☐
- Pastoral ☐

INSTREAM COVER

Flow Type	Cover Type	% Cover

AQUATIC VEGETATION

Flow Type	Species	% Cover

RIPARIAN VEGETATION (%)

- Native Trees (>30m width) ☐
- Native Trees (<30m) ☐
- Exotic Trees ☐
- Native Shrubs ☐
- Exotic Shrubs ☐
- Native Grass ☐
- Exotic Grass ☐
- Sedge/Rushes ☐
- Mangroves ☐
- No Vegetation ☐
- Other ☐

DISTURBANCE TYPE & RATING

- Siltation ☐
- Bank Erosion ☐
- Cattle Access ☐
- Bridge ☐
- Road ☐
- Ford ☐
- Sand/Gravel Extraction ☐
- Urban Rubbish ☐
- Feral Animals ☐
- Pump Inlet ☐
- Drain/Waste Outlet ☐
- Picnic Site ☐
- Camping Site ☐
- Other ☐

Comments:

APPENDIX 2. Fish and habitat sampling data sheet.

Field station #: _____ River/Creek:_____

Date:___/ ___/ ___ Time: ___:___ to ___:___

GPS: _ _°_ _.__ S' _ _°_ _.__ E' to _ _°_ _.__ S' _ _°_ _.__ E'

Collectors:_____

Weather:

_____

Site map:

Flow rate: _____ Turbidity:_____ Depth (min-max)_ _ _m - _ _ _m

Width (min – max): _ _ _ m - _ _ _ m Length of reach sampled: _____m

pH: _ _ . _ _ Temp: _ _. _ _ °C Conductivity _____ µS

Collection method(s):

Electrofisher __V: ___/____
Pole seine
Purse seine
Fish traps
Gill net
Throw net
Hook and line
Rotenone (Derris)
Other (specify)...eg. Observation

Appendix 2b. Reverse side of fish and habitat sampling datasheet.

Field station #: _____

Species	# kept	Size of specimens kept	# returned	Size of specimens returned	Total no.	Estimate of total No.	REMARKS

Additional notes: