

PROTOCOLS FOR BIWEEKLY FAUNA CRUISE

***MACRO & MEIOFAUNA SAMPLES ARE COLLECTED ON A QUARTERLY BASIS ONLY (BEGINNING APRIL 2002).** The last biweekly macro/meio samples were collected on 3/26/02). From now on macro/meio cores will only be collected on the first fauna cruise of January, April, July and October).

Fauna cruise begins (leave lab) 3 hours before scheduled low tide for that day.

Location of Fauna Cruise collections:

<u>Epibenthos</u>	Latitude	Longitude
start	N 33°19'50.5"	W 79°11'18.5"
end	N 33°20'00.1"	W 79°11'14.7"

<u>Zooplankton</u>		
	N 33°20'02.0"	W 79°11'09.8"

<u>Meiofauna</u>		
<u>BB</u>	N 33°19'49.8"	W 79°11'03.5"
<u>DD</u>	N 33°20'01.0"	W 79°10'04.5"

<u>Macrofauna</u>		
	N 33°19'46.9"	W 79°11'01.4"

Lab preparation

Formalin preparation

10% formalin

This formalin mixture is for the smaller meiofauna samples. In a 2 liter polypropylene bottle, mix 1 part 100% formalin with 9 parts 63-micron filtered seawater. Add 1/8 teaspoon of Rose Bengal. Cap tightly and shake to mix. Formalin is stored in the chemical cabinet in the sorting lab. Rose Bengal is stored in the cabinet under the hood in the sorting lab.

100% buffered formalin

This formalin mixture is for all other samples collected. In a new 4 liter bottle of 100% formalin, add 80g of Borax and ¼ tsp of rose Bengal. Cap tightly and shake to mix. Fill centrifuge tubes with this mixture and store in cups in chemical cabinet. Borax is stored in the cabinet under the hood in the sorting lab.

100% ethanol

Ethanol is stored in the chemical cabinet in the microscope/sorting lab. The ethanol is not diluted in any way.

Jar preparation

For each sampling you will need the following:

Zooplankton: 2 32 oz. jars w/lids

Epibenthos: 3 32 oz. jars w/lids

Meibenthos: 4 small 8 oz. jars w/lids (2 each: BB, DD)

Macrobenthos: 8 32 oz. jars w/lids

On the lid of each jar, you will need to write the following information:
NERR, tow #, sample date, sample, rep.

For example:

NERR
545
02/13/03
EPI A

Codes for each sample are:

Zooplankton: ZPK (rep A or B)
Epibenthos: EPI (rep A, B, or C)
Meiofauna: DD or BB (rep 1, 2)
Macrofauna: Macro (reps 1 –8)

Jar label preparation

There is a label template that should be used to print labels for samples. Sample labels should be printed on Nalgene™ PolyPaper® Plastic Paper (coated) Fisher catalog #6304-0811. Labels can be printed using the laser printer if only one sheet is fed in at the time. Labels should look like:

Date _____ Station _____	
Time _____ Tow# _____ Rep: A B C	
Flowmeter# _____	
Flowmeter start _____	
Flowmeter finish _____	
Collected by _____	
Surf T°C _____	Bott T°C _____
Surf S‰ _____	Bott S‰ _____
Surf DO mg/l _____	Bott DO mg/l _____

Example of a blank ZPK, EPI label.

Date <u>01/23/02</u> Station <u>TC</u> <u>EPI</u>	
Time <u>1123</u> Tow# <u>544</u> Rep: A <u>B</u> C	
Flowmeter# <u>18693</u>	
Flowmeter start <u>566879</u>	
Flowmeter finish <u>574952</u>	
Collected by <u>SF, PK, TB</u>	
Surf T°C <u>8.2</u>	Bott T°C <u>8.5</u>
Surf S‰ <u>32.5</u>	Bott S‰ <u>33.1</u>
Surf DO mg/l <u>8.25</u>	Bott DO mg/l <u>8.73</u>

Example of completed EPI label.

Zooplankton: Prepare 1 small (blank) and 1 large (pretyped) label for each sample jar. Small labels should read the same as the jar lid. Large labels can be filled out as information is gathered (lab or field).

Epibenthos: Same as zooplankton

Meiofauna: Meiofauna samples only get a small label that matches the lid. No large labels.

Macrofauna: Same as Meiofauna.

Sample notebook preparation:

There are two notebooks used for fauna cruise; one for ZPK/EPI, and another for MACRO/MEIO. Notebooks should be waterproof. Notebook templates should look as follows:

ZPK/EPI

NERR # _____ Date _____
 Pred. Low _____ EST
 Time _____ EST

Physicals

	PAR	‰	°C	DO mg/l
Air				
0				
50				
100				
150				
200				
250				
300				
350				

bottom depth _____m

wind
 water
 weather

NERR # _____ Date _____
 Pred. Low _____ EST
 Time _____

ZPK A B

FM# _____

Start _____

Finish _____

EPI

FM# _____ Time _____

B1		
B2		
B3		
End		

comments

MACRO/MEIO

NERR # _____ Date _____
 Pred low _____ EST

	DD	BB
Time		
AirTemp		
SedTemp		
H ₂ Otemp		
‰		
redox(cm)		
DO(mg/l)		
Depth(m)		

Tides
 Water
 Weather
 Wind
 Comments

The following gear is needed for fauna cruise: *Most gear is stored in the Archive Room except for the nets, which are stored below the building.*

- Sample jars w/ lids, labeled. Take *at least* 1 extra 32oz. jar out in case one breaks.
- 2 cod-ends for zooplankton nets (153 µm mesh)
- 1 cod-end for epibenthic sled
- 2 zooplankton nets with weights (1 each) and flowmeters attached
- 1 epibenthic sled/net w/ flowmeter attached

- YSI w/650 display
- LICOR light meter with LI1400 datalogger and weights
- 2 common (flat head) screwdrivers
- 5 centrifuge tubes full of 100% formalin (13 if quarterly)
- full bottle of ethanol
- sample notebook for ZPK/EPI
- #1 pencils (this # lead works best with waterproof paper)
- cell phone (*very important*)
- sample notebook for macro/meio (if quarterly sampling)
- full bottle of 10% formalin (if quarterly sampling)
- small hand corer w/ stopper (if quarterly sampling)
- large hand corer w/ stopper (if quarterly sampling)
- long corer w/ attached syringe and stopper (if quarterly sampling)
- thermometer (if quarterly sampling)
- cable ties
- pliers/cutters
- Kestrel handheld weather station (handy but not necessary)
- compass (handy but not necessary)

Before loading the nets, you must check to make sure that each of the flowmeters are filled with water. If not, carefully remove the small screw at the base of the propeller and add *tap* water using a syringe or squirt bottle. **DO NOT** over tighten the screw or it will crack the flowmeter body and prevent the propeller from turning correctly.

Boat preparation

For fauna cruise we usually use one of the 17' whalers since we have so much equipment to load. A 15' will do if only 2 people go out.

- Check plugs (one in rear, one in anchor well)
- Check gas
- Make sure the engine will start (batteries tend to die during cold winter months)
- Make sure there is at least 1 life jacket per person, as well as 1 throw device in the boat. Float coats only count as life preservers IF you're wearing them.
- Make sure you have an oar
- Make sure the engine is trimmed up and resting on support arm
- Make sure the boat is latched and chained securely
- Make sure the boat is tied down to the trailer front and back

Make sure that all equipment is loaded onto the boat and that a float plan has been filed. Float plan should include: date, time left, time expected back, people going, boat taken, where going. *Remember:* you should leave the lab 3 hours before the predicted low tide.

The boat is put in at Clambank. The first sampling station is at the mouth of a small creek off of Town Creek. This small creek is on the east side of Town Creek between the large sand bar and Bread and Butter Creek. The boat should be anchored such that the boat is ~30-50 meters off of the creek mouth. Tide and wind conditions will dictate how the boat will be oriented when anchored.

If there are 3 people on the cruise, one person can do physicals while the other two collect zooplankton. If only 2 people are on the cruise, the zooplankton collections must be done simultaneously, so physicals will be done after that collection.

Sampling physicals (water quality data)

- Using the YSI, sample and record the following parameters at both the water surface and bottom: *depth* (m), *temperature* (C), *DO* (mg/l), *salinity* (ppt).
- The old method of measuring PAR using the LI185-B was discontinued in December of 2005. The new method involves using the new LI-1400 datalogger with the same quantum sensor (LI-193SB). Attach the cable from the spherical quantum sensor (tag marked “round” on connector) to the connector on the LI-1400 labeled “sphere air” OR “I1”. This connector port has been programmed with the proper multiplier that is specific to the spherical quantum sensor for air readings. (This multiplier is obtained from LI-COR everytime the sensor is calibrated.) Turn the datalogger on. If “View New Data” doesn’t automatically appear at the top of the screen, press “View”. When “View New Data” appears, press “Enter”. Use the left & right arrows to page between connectors until you see I1A. This is the “sphere in air” channel. The datalogger is programmed to average readings over a 15 second period, so you have to wait ***at least*** 15 seconds before recording the reading. We no longer use the white tub for these readings, but just allow the sensor to sit vertical in the gray tub such that no shadows fall on the sensor. If clouds are passing over, the readings will be jumpy. You should usually wait longer to allow for the old readings from the last depth to clear the dataloggers memory such that they don’t get averaged into the new reading. (i.e. you want the datalogger to average only those readings that were taken at the depth you’re interested in OR in the case of air, only those readings that were taken once you had the bulb situated in the correct position)
- For measuring “light in water”, remove the cable from the Channel I1 connector and reconnect to the “Sphere Water” connector Channel I3. Make sure the weight is attached and lower the sensor into the water for the first reading at the surface. The “surface” is considered 1 bulb diameter below the water’s surface. Using the left or right arrow keys, tab over to the I3A screen. Just as for “light in air”, allow longer than 15s to elapse before recording the reading since the first readings will have been from before you got the bulb in the water. Record your reading and move on to the next depth (0.5m increments). Simply disconnect the sensor, turn the datalogger off, and recover the sensor when you’re done.
- Lastly fill in the information about water, weather, wind, etc. Water would include water color, clarity, etc. Include in the comments anything unusual such as unusually high or low tide, previous storm events that might affect samples, etc.
- ***NOTE:*** Collection of water velocity data was discontinued in February 2007.
- ***NOTE:*** *It is important to be as detailed as possible when giving weather & water conditions as it’s essential to troubleshooting any errors in data recording that are found later during data entry. (i.e. if light readings seem unusually low, it’s important to know how cloudy it was, if water was really turbid, etc., rather than just “gray day” or “brown water”)*

Sampling zooplankton

Zooplankton collections were initiated by Dr. Steve Stancyk in 1981 during the LTER program, and have been collected every two weeks ever since. This study's purpose is the long-term characterization of zooplankton larger than or equal to 153 microns, and to obtain some basic information on each of the taxa encountered.

The zooplankton collections are the first samples collected during the fauna cruise. These collections are made from the same location as are the physical water quality measurements; 30-50 meters off of the mouth of the small tributary to Town Creek. The zooplankton nets consist of 3 metal bars, two of which hold the mouth of the net open, and the other that holds the weight. The net is comprised of 153 micron mesh and ends in a cod end on which is attached a PVC "cup" with 153 micron mesh window. A flowmeter is located in the mouth of the net to measure water flow as the net is deployed. A spring operated closure is attached to the top of the net, to which the heavy-duty nylon line on the bottom half of the net mouth is looped. This closes the mouth of the net until the "messenger" (metal pipe on rope) is released and hits the spring to open the net mouth. The rope is marked by ½ meter increments, which will periodically need retouching.

To get the nets setup for sampling, each person should first tie the end of the rope to the boat railing to prevent loss of the nets. Record the flowmeter number and beginning reading in the notebook. Zooplankton samples are designated **B** for the creekbank side or side facing the mouth of the small creek, and **A** for the side facing the main body of Town Creek. Attach the PVC cod-end to the bottom of the nets using the hose clamps provided. Attach the long slender weights provided to the bottom link in the chain on the "weight" bar of the net. Attach the nylon loop from the net mouth bottom to spring release mechanism at the net mouth top. Make sure the mouth of the net is closed by holding the net up by the top rope and allowing the weight to pull the mouth tight. Adjust if needed. When both nets are set-up, each person should simultaneously lower their nets over the side of the boat (one on the "B" side, one of the "A" side) until they feel the weight hit the bottom of the creek, counting hash marks as it's lowered. Mean depth at mid-tide here is ~3.5 meters. Making sure that the line is taught and that the weight is pulling on the net (don't let the net "rest" on the bottom or else the mouth won't open correctly), note the depth of the net by the number of hash marks that are submerged. Position the top rope such that you can easily release the messenger without obstruction. **At the same time**, each person should release the messenger, keeping the rope taught, to open the net mouth. Collect at the bottom for 1 minute. After one minute, pull the nets up to the ½ way mark (mid water column) and let them sample for 1 minute. Finally, pull the nets up such that the brass ring above the spring is at the water's surface, and sample here for 1 minute.

At the end of the last minute, pull the nets in and wash any material stuck in the nets down into the cod end of the net. Record the flowmeter reading in the notebook. At least 2000 revolutions should have occurred for the sample to be good. With the net and cod-end still attached, carefully pour out excess water through the netting in the PVC. Remove the cod-end and pour the sample into the designated sample jar. There will still be sample remaining in the cod-end, stuck to the sides and netting. Carefully submerge the bottom half of the cod-end into the water (bottom down) and allow water to pour through the netting (**not over the top edge**) and swirl this water around to dislodge the remaining sample. Pour this into the sample jar. The jar should be about 90% full of water/sample. Using a syringe and the 100% formalin mixture, add 40 cc of formalin to each sample. Make sure that if the large tag is not already filled out, that it is removed before adding the sample. The tags can be filled out later at the lab. Cap the sample tightly and shake to mix the formalin with the sample. This is the end of the zooplankton sampling.

Sampling Epibenthic Fauna

Epibenthos collections were initiated in 1981 by Dr. Dennis Allen under the auspices of the LTER monitoring program, and have been conducted biweekly ever since. The study was initiated to determine seasonal and inter-annual changes in the taxonomic/life stage composition and abundance of small motile epibenthic invertebrates and fishes (1-20 mm in length) in the major subtidal habitats of North Inlet estuary.

The epibenthic samples are collected using the epibenthic sled which is a 365 micron net attached to a steel frame on skis. The sled is pulled three times along a 200 m predesignated route along Town Creek. Each pull results in a single epibenthic sample (reps A, B, C). The boat is positioned ~5 meters off of the creekbank from the starting point which is marked with PVC. This point is ~20 m south of the mouth to Clambank Creek on the west side of Town Creek. Each sample is made towing south to north. The stopping point is also marked with PVC. This point is 200 meters north of the starting point, along the western bank of Town Creek.

First, tie the end of the rope attached to the sled to the left rear end of the boat. Second, record the flowmeter number and initial reading. Screw the sample collection cod-end to the net. Approach the starting point (south to north) at the slowest forward speed possible. As the stern (rear) of the boat passes parallel to the PVC marker, throw the sample collection cod-end over and allow the net to straighten out, then slowly lower the sled into the water over the side of the boat, holding the sled by the upper steel bar (left hand) and the ring/rope attachment (right hand). Slowly pay out the line to the sled, constantly keeping tension on the line as it's going out. This will assure that the sled does not tangle with the net and minimizes the change of the sled spinning as it goes down. Record the time that the sled is deployed.

At the slowest forward speed possible, slowly pull the sled along Town Creek until the stopping point is reached (pvc marker). Avoiding the sand/oyster bar off the mouth of Clambank Creek, the boat should run a pretty straight course along the creek and should end up about 10 meters off the bank from the ending point. During this time, one person can be filling out the labels with the water quality information and flowmeter information. Once the stopping point is reached, the driver should turn the boat hard left toward the bank/pvc marker to allow the sled to be pulled in easily. The boat is then put in neutral and two people can help to pull the sled in and prepare the sample.

Pull the sled up into the boat and note the flowmeter reading. At least 7000 revolutions must occur for the sample to count. During the winter months, algae can make up the bulk of the sled sample and severely clog the net and wrap around the flowmeter and sled mouth. If you feel that algae prevented the flowmeter from turning correctly during the tow, the tow MUST BE REDONE. The rpm's are used to calculate the amount of water that flows through the net and thus the amount of organisms per unit area in the creek.

With the sample bottle over the water, fold the net over the end and invert the bottle to pour excess water out but keeping the bulk of the sample in the bottle. Then holding the bottle up out of the water by the net, use your other hand to splash water over the net immediately above the sample bottle to wash down any remaining sample that is stuck on the net or the top of the bottle. Remove the bottle and pour the sample into the designated jar. You might have to dip the bottle again to insure you get the entire sample out of the bottle. The sample jar should be ~90% full of sample. Add a centrifuge tube full of 100% formalin to the sample, cap tightly and shake sample well to distribute formalin. Again make sure that the tag is already filled out or removed to be filled out later at the lab.

Repeat this process for rep B.

For rep C, collect the sample as normal, but make sure all of the water is removed. Use a squirt bottle full of ethanol to rinse the sample from the net into the cod end, and then preserve the sample in 100% ethanol rather than formalin.

Sampling Meiobenthic Fauna

This study was initiated by Dr. Bruce Coull in 1973, and represents one of the longest ecological data sets of its kind in the world. Its original purpose was to determine if natural meiobenthic assemblages exhibited continuity over time, and to monitor several physical variables to determine if these influenced long-term temporal patterns.

DD meiobenthos

During the quarterly meio/macro sampling cruise, meiobenthos and macrobenthos cores are collected after the zooplankton and epibenthic samples are collected. After the epibenthic samples have been collected, you should have about 1 hour before low tide. We typically take the boat over to Debidue Beach and anchor off of the backside of the beach to wait for low tide. When low tide is reached, slowly motor the boat into the shallows on the southern side of the large sandflat that forms at the intersection of Town Creek and Debidue Creek. Using the oar as a depth finder, anchor the boat in about 1½ meters of water. Using the pole-mounted corer, collect two meiofauna samples. Using a field thermometer, measure the temperature of the sediment immediately after collection. Using a small ruler, measure the redox depth of one of the cores before emptying the core into the sample vial. Redox is estimated based on the depth at which the sediment turns black from sulfide precipitation due to anoxic conditions. Add just enough 10% formalin to these samples to cover the top of the sediment, cap and shake thoroughly. Using the YSI or Hydrolab, record depth, temp, DO and salinity at the bottom of the water column where the cores were collected. From here you move on to Bread & Butter Creek for the second set of meiobenthos cores as well as the macrobenthos cores.

BB meiobenthos

Bread and Butter Creek is the small creek located directly across Town Creek from the mouth of Clambank Creek. At low tide, the mouth of this creek is fairly shallow due to sand/oyster bars on either side, so be careful when entering this creek with the boat. Slowly motor the boat up Bread and Butter until you reach the large mud flat in the creek bend where the meiobenthos cores are collected (there's no marker and no real way to describe where this mudflat is). **You want to create the minimal amount of wake as possible when you near the mudflat so as not to disturb the sediments.** When you get near the mudflat, turn the nose of the boat directly toward it and then **SLOWLY** run the boat up onto the mud so you can collect the cores.

From the side of the boat, use the small handheld corer to collect 2 sediment cores from an undisturbed area of sediment off the side of the boat. Using the same techniques as with the Debidue cores, measure redox, sediment temperature and water quality parameters from this sampling location. Add the appropriate amount of 10% formalin, cap and shake the cores to mix. Ideally you want only about 3 cm of sample in the collection jars, but you may need to collect more in the corer in order to measure redox depth. Excess core material can be thrown overboard, saving the top 3 cm of the core for the collection.

Sampling Macrobenthic Fauna

Dr. Bob Feller initiated these collections in 1981. The purpose of this study is the long-term characterization of macrobenthic organisms in the North Inlet estuary and to learn some basic data on the taxa found therein.

BB Macrobenthos

Macrobenthic cores are collected after the meiobenthic cores are collected. These cores are collected on a mudflat that's further upstream from the meiobenthic mudflat. This mudflat is marked by a pvc stake along the creekbank above the mudflat. Approach the mudflat as before, with as little wake as possible, and beach the boat on the edge of the flat so you can take cores from the side of the boat.

Using the larger hand-held corer, take eight core samples, being careful not to core in the same spot more than once. Save only the top 5 cm of core sample for preservation. (This can be done by allowing a small gap between the top stopper and the core tube, and letting the unwanted portion of the core slide out) Add 60 cc of 100% formalin to each core, cap, and shake.

Post cruise chores:

- Trailer boat and return to lab
- Offload all of the equipment and samples at the lift. Thoroughly rinse the epi sled and zpk nets with fresh water and hang them back up to dry under the building.
- Clean the boat and park, making sure to run fresh water through the engine.
- The remaining equipment and samples are to be taken into the archive room. Place all of the sample jars in the sink and rinse with fresh water to remove remnants of formalin and dirt. Allow the samples to air dry. Once they're dry, use the electrical tape in the cabinet to seal around the caps of each of the jar to eliminate evaporation while the samples are archived. Make sure that all of the tags are filled out completely and in the appropriate sample jars.
- ZPK and EPI samples are archived in the archive room on their designated shelves.
- Meio and Macro samples are (as of 12/2003) being boxed and stored in the Leonard shed up by the Kimbel Lodge. This may change depending on the availability of someone to start sorting the macro samples. These samples are stored in separate boxes with the range of dates for each written on the outside of the box with black sharpie.
- Fauna physical data collected after the ZPK samples are to be entered into the fauna physical excel spreadsheet (SWMP lab).

Net specifications:

Fauna Cruise Net Ordering Protocol

Nets should be replaced every year. When ordering nets to replace the Fauna Cruise Zooplankton nets and the Epibenthic Sled Net use the following specifications:

2 Zooplankton nets:

- 205cm (total length) double stitched 153 micron mesh* Nitex zooplankton sampling net with 30 cm diameter mouth opening
- the front cylinder portion should be 101.5 cm and the cone cod end should be 73.5cm
- the net should have 3 **Dacron** collars
 - o mouth collar should be 10 cm deep (long) with cord sewn into mouth edge and brass grommets every 9cm
 - o belly band should be 10cm deep (long) which connects the front cylinder to the back cone

- o cod end collar (which terminates net) should be 10cm deep (long) and 9.5 cm in diameter with 3 loops sewn on outside for hose clamp

1 Epibenthic Sled Net:

- 200 cm (total length) double stitched conical 365 micron mesh Nitex zooplankton net with a 50cm diameter mouth opening
- the net should have 2 **canvas** collars
 - o mouth collar should be 10 cm deep (long) with cord sewn into mouth edge and brass grommets every 9cm
 - o cod end collar which terminates the net should be 10cm deep (long) and 10 cm in diameter

*153mm Nitex is an older mesh size. The weave is a Taffeta weave. Whoever makes the net will have to match it to a newer mesh. Therefore, whatever they use should also be Taffeta weave. The trim, collars, etc... should be white (a vender once used black nylon for the collars and trim and the nets had to be sent back (very expensive mistake for the vender)

Our net supplier as of 12/2003 is Sea Gear Corporation of Melbourne, Florida. Their information is below:

Sea Gear Corporation
700-B1 S. John Rhodes Blvd.
Melbourne, FL 32904-1507
ph: 321-728-9116
fx: 321-722-0351
email: SeaGearCorp@aol.com

We had our nets made to order and the designs should be kept on file at Sea Gear. The Sea Gear Model Numbers are:

90-50x4-365-B	½ m diameter 365 micron epi net (specify all white)
9730-153-BML	30 cm. Bongo Style 153 micron zpk net (specify all white)

Equipment calibrations/maintenance:

Datasonde:

YSI

-YSI's should be calibrated as needed for SWMP work and can be used for the fauna cruise in conjunction with the 650 handheld unit.

Light Meter:

Model LI-1400 Datalogger
s/n DLA-3091

LICOR Spherical Quantum Sensor

Model Spherical Bulb
s/n SPQA381
Cable: LICOR underwater cable
2222 UWB-10 10-meter cable

Calibrations done by: LICOR Biosciences
4421 Superior Street
Lincoln, NE 68504
800-447-3576
402-467-0621
FAX: 402-467-2819
www.licor.com

-calibration sheets are stored in the gray filing cabinet in the SWMP lab.

Flowmeters:

General Oceanics Mechanical Flowmeters

Model 2030R

These can be purchased from General Oceanics (www.generaloceanics.com), or may be cheaper from other sources such as Wildco (www.wildco.com), or Forestry Supplier (www.forestry-suppliers.com).

These flowmeters are used on the epi sled and both of the zooplankton nets. The flowmeters are attached to the mouth of each net/sled using heavy-gauge fishing line and double-sleeves. Note that there is a “best” way to attach the epi flowmeter for ease of reading in the field. Flowmeters should be calibrated at least once a year. This is done at Coastal Carolina in their lap pool.

To calibrate, each flowmeter is suspended ~3 feet from a pole and the s/n and flowmeter reading recorded. The flowmeter is then walked along the length of the long side of the pool twice to cover a distance of 50 yards. This is done 3 times (3 50-yard tows) for each flowmeter at 3 different paces (slow, medium, fast). The # of revolutions for each tow is recorded, averaged and used to calculate the average number of revolutions per *meter* for each flowmeter. This number is then used in calculating the number of organisms per m³.

Ordering Jars:

Jars should be ordered in bulk such that you’re never short. Note that some cruises use more jars than others. Note also that during the winter months, large amounts of algae can be collected in the sled tows requiring a larger jar for storage. Oyster Landing fish surveys also require larger 64 oz, and 128 oz. jars for fish storage.

Jars should be clear (flint), glass, straight-sided, with WHITE PLASTIC caps with a liner. It’s best to shop around different vendors to get the best price.

Vendors include:

Oberk Company	www.oberk.com
All-Pak Inc.	www.all-pak.com
Freund Container Inc.	www.freundcontainer.com
Brad Pak Inc.	www.brad-pak.com