Florida Microplastic
AWARENESS PROJECT

VOLUNTEER MANUAL

Sea Grant FLORIDA

UF IFAS Extension UNIVERSITY of FLORIDA

www.plasticaware.org

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Contents:

Dear Volunteer,

Thank you for helping with the Florida Microplastic Awareness Project. This project has been funded by a 2015 NOAA Marine Debris Outreach and Education grant to the University of Florida/IFAS Extension. There are 28 regional coordinators located around the state of Florida. If you have not yet made contact with your regional coordinator, please contact Maia McGuire (mpmcg@ufl.edu) to be connected to the appropriate person.

This project has several potential roles for volunteers/citizen scientists. You are welcome to participate in one or more of these:

- Collect water samples (on a flexible schedule—as little as once, or as frequently as monthly; in a single location or multiple locations at the volunteer's convenience)
- Filter water samples (in a location as determined by your regional project coordinator)
- Look at filters under a microscope (in a location as determined by your regional project coordinator)
- Input data online
- Help teach others about microplastics at local outreach events

Procedure for collecting water samples for microplastics project.

1. Talk with your regional coordinator to identify your collection site. In most cases, the collection site will be selected by the volunteer collecting the sample, but we want to make sure that we collect samples from a range of locations.
2. Pick a name for your site. Try and give it something unique (i.e. don't just call it "home" or "beach").
3. Identify the GPS coordinates (approximate) for your collection site. If you have a GPS device, you can use it (please use decimal degrees if possible). If not, Google Maps allows you to find the latitude and longitude of a location. You can use the instructions below to find your coordinates. There are videos available on the "Get Involved" page of www.plasticaware.org that will walk you through this as well as the following procedures.

Using Google Maps to find your latitude and longitude

1. Go to maps.google.com

2. Enter the street address for your collection site, or a general location (e.g. Marineland Beach)

3. Click on the "Satellite" box to get the aerial imagery (this makes it easier to pinpoint your site)

4. Zoom in as needed to be able to find your sampling location. Click on that point with your mouse.

5. Write down the numbers that pop up. The first number is the latitude, the second is the longitude. Note that the negative sign with the longitude is important!

4. Record your site name(s) and latitude/longitude(s) in the table below. Please give your regional coordinator a copy of this information.

Site name	Latitude	Longitude

5. Obtain a 1-L numbered sample container from your regional coordinator for each of your sites. The project is using Nalgene wide-mouth sampling bottles.
6. Ideally, collect your sample on a day/location that is as calm as possible and at a slack tide in areas that are tidally-influenced (this allows plastics to rise to the surface of the water instead of being mixed through the water column). Please try and avoid wearing synthetic clothing (e.g. microfiber, fleece) while collecting your sample.
7. Make a note of the amount of time that you spend collecting your sample(s) (see page 14).
8. At the collection site, rinse the collection bottle and lid three times with water from the site. You do not need to completely fill and empty the bottle—fill it ¼ to 1/3 full with water, swirl the water around the inside of the bottle, and empty the contents. Repeat for a total of three rinses. Rinse the lid by dipping it into and out of the water three times. Screw the lid onto the empty container.
9. Go to a nearby area that was not affected by your rinsing procedure (i.e. upstream slightly).

10. Unscrew the lid of the container. Holding it as horizontally as possible, carefully lower it into the water so you are collecting water from the very surface. Continue to lower the bottle into the water until it is full (you will need to angle it slightly), then IMMEDIATELY screw on the lid (ideally you are screwing the lid on partially underwater).
11. Record the collection bottle number and location/date of collection on your data sheet (page 6.)
12. Keep the data sheet with the sample bottle(s). If you will be dropping the samples off for someone else to analyze, it is essential that the data sheet be with the sample bottle(s).
13. HINT: Samples are easier to filter if they sit for a few days before being filtered. Sample bottles should be kept out of direct sunlight, but no other special storage is needed. Samples can sit for several weeks (or more!) before being filtered.

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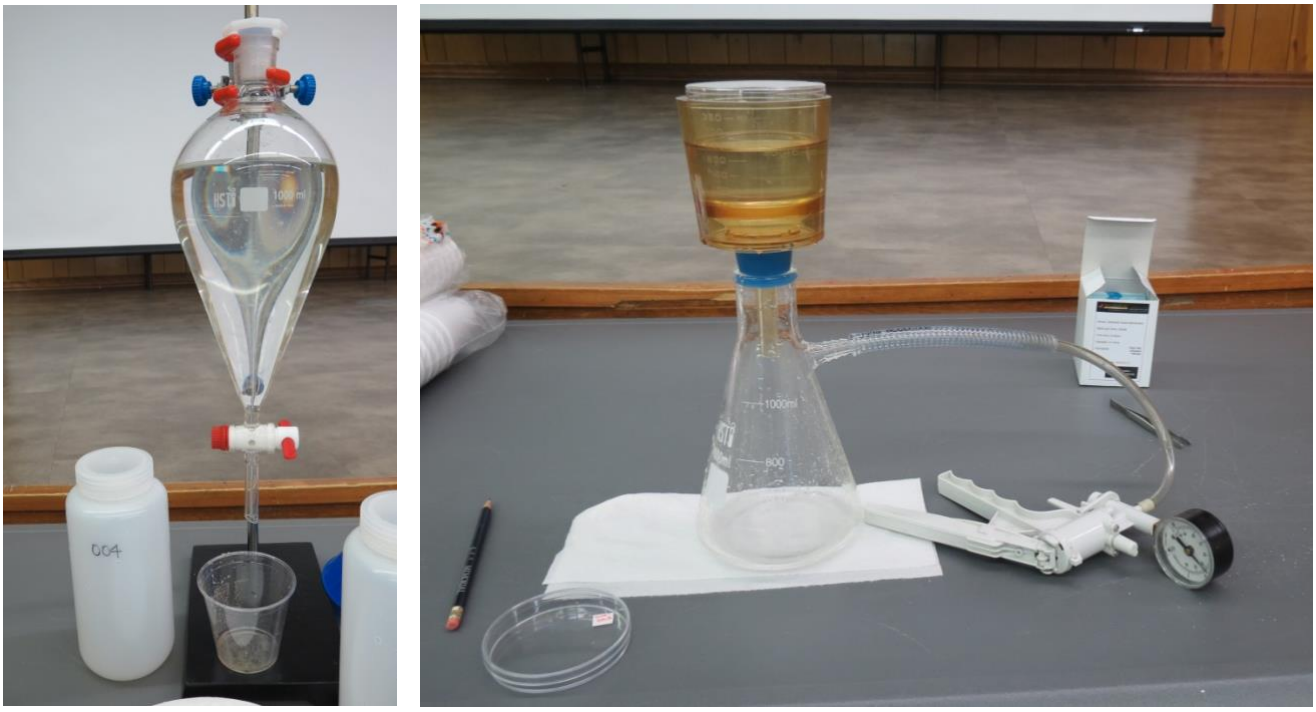
DATA LOG SHEET

Sample bottle #	Sample collected by	Sample collection date	Sample collection location (name and GPS coordinates)	Sample filtered by	Sample filtration date	Filter observed by	# of FIBERS seen			# of plastic FRAGMENTS seen	# of MICROBEADS seen	# of pieces of FILM seen	Date data entered online
							Unknown (not tested with flame)	Natural	Synthetic				

Analyzing Water Samples for Microplastics

Materials needed:

- Vacuum filter holder that can take 47-mm filters
- 1-liter side-arm Erlenmeyer flask
- Vacuum pump and hose that fits outlet of flask
- 0.45 micron gridded filters
- Filter forceps
- Squirt bottle
- Tap water
- 1-liter separatory funnel and stand/clamp/4" ring (optional—useful for samples that contain a lot of sediment)
- Dissecting microscope (20-30 or 20-40 X).



Separatory funnel (L) and filtration apparatus (R)

Procedure:

1. You can let samples sit for weeks before processing (they do not need to be refrigerated, although they should be kept in the dark to prevent algal growth in the bottles). Make sure that you have the sample's data sheet before you start filtering it. **It will be easier/faster to filter samples if they have been sitting for at least a week**.
2. Plastic fibers from clothing or in the air can contaminate your sample. Please avoid wearing synthetic clothing while filtering your sample, and make sure to follow directions carefully.
3. **Your coordinator may have filtered water available for you. If so, you can skip this step.** Run about 100 ml of tap water through a 0.45 micron filter (vacuum filter it). Use this filtered water to rinse the inside of the side-arm flask (the one you've used to collect it in) and discard. Repeat 2 more times. (Essentially you are triple-rinsing the flask with filtered water). Similarly, triple rinse a squirt bottle with filtered tap water. Collect the next 500 ml of filtered water and use it to stock the squirt bottle. You will use this filtered water for rinsing the funnel, etc.
4. *This part is optional, but recommended for samples that contain a lot of sediment (it will make those samples much easier to filter). When ready to process, triple rinse a 1-L separatory funnel. Pour the sample into the funnel (supported by a clamp on a heavy-duty stand). Let sample stand for at least a few minutes—if the sample contains fine sediments, it is better to let it sit for a longer time period—even several hours. Drain off the sand/silt from the bottom of the sample into a cup (this will be discarded).*
5. With no filter inserted, rinse the inside of the filter apparatus three times with pre-filtered water. Use a petri dish or other flat object as a cover for the filter apparatus (only remove when adding more sample). This will help reduce environmental contamination of the sample (e.g. by lint in the air). You will need to leave a small air gap between the petri dish and the top edge of the filter apparatus.
6. Peel open the plastic covering for the filter paper. Insert the filter (gridded side up) into the apparatus, being certain to remove the round paper cover from

the filter. Add sample to fill the filter funnel. Keep the separatory funnel or sample bottle stoppered when not adding sample to the filter apparatus. *Drain sediment from the separatory funnel as needed.*

7. With the petri dish over the filter funnel, vacuum filter the sample. Remember to leave a slight gap between the petri dish and edge of the filter apparatus. Add more sample until it has all been run through the filter, but empty the flask before it becomes so full that water gets drawn into the vacuum pump. Once your sample has been entirely filtered, rinse the sides of the filter funnel with a small amount of filtered water, and vacuum this rinse water through the filter.
8. Release the vacuum pressure. Remove the top part of the filter funnel. Use filter forceps to remove the filter and place into a clean petri dish. Cover with the petri dish lid. Remember to label the sample with the bottle number (either on the petri dish lid, or with a small strip of paper placed inside the petri dish, but not on the filter).
9. Your sample should take between 5 minutes and 45 minutes to filter completely.) If your filter seems to be clogged (e.g. sample will only drip very slowly through the filter regardless of the vacuum pressure), you have some options:
    a. Pour your sample back into the collection bottle and allow it to sit for several days before filtering (even if the sample appears clear).
    b. If your sample will not filter through at all, check to be sure that you removed the paper cover from the filter paper (you can pour the sample back into the sample bottle in order to do this).
    c. Use the separatory filter to remove sediment from the sample.
    d. You may use multiple filters on a single sample—be sure to label them all as being from the same sample, and combine data from all filters before reporting.
    e. Contact your regional coordinator (or the state coordinator) for additional support.
10. You can use the filtered water in the flask to fill up the squirt bottle.
11. If possible, let the filter dry at least overnight before viewing under a

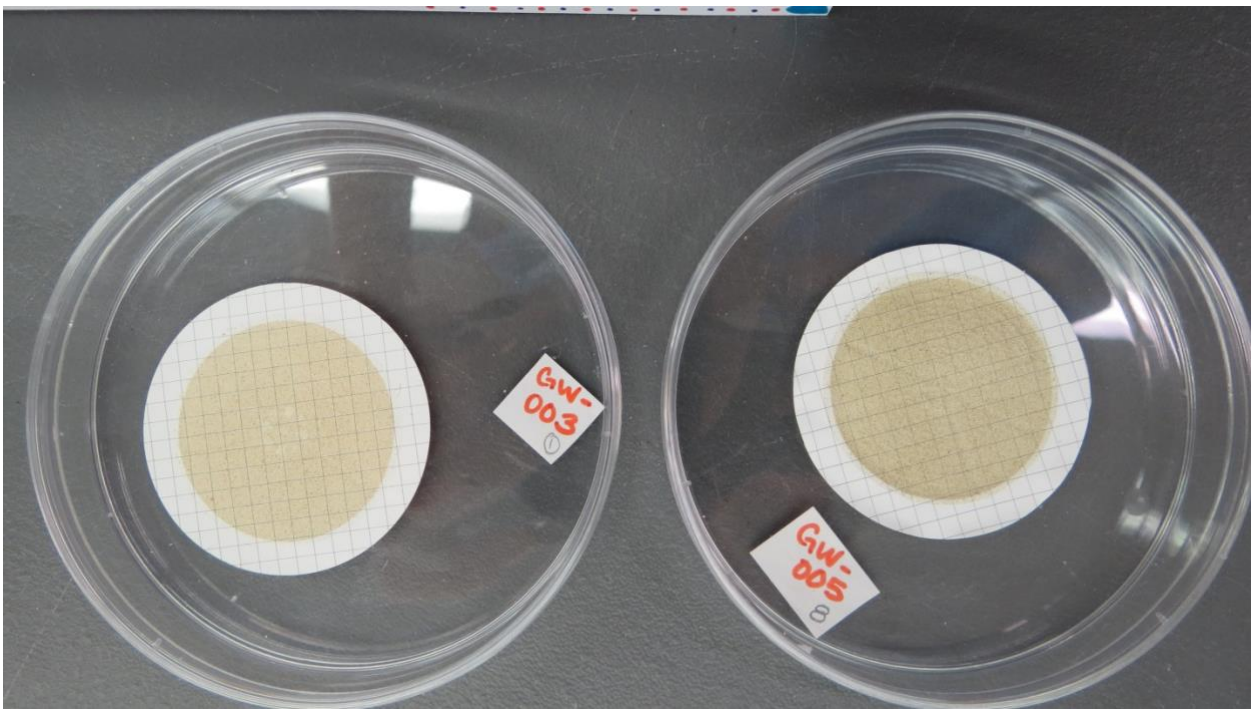
microscope (this is not required, but it's easier to differentiate plastics from plankton once the plankton have dried out somewhat. It's also easier to scan without the reflection from the wet filter).

12. If processing several samples collected in the same general location one right after the other, you do not need to rinse the separatory funnel or filter funnel in between...but should do so before switching sample locations. When rinsing the funnels, use filtered water. You do not need to have filter paper in the filter funnel when rinsing between samples.
13. Observe the filter papers under a microscope at at least 20X magnification. Scan the filters systematically, moving row by row to prevent double-counting or missing plastics. Plastic will generally be milky/white or colored (not clear). Sand grains are easily mistaken for plastics.
14. Many of the fibers seen on the filters will be extremely small.
15. Refer to the Marine and Environmental Research Institute's *Guide to Microplastic Identification* (available at www.plasticaware.org) and the videos on the website for help in determining what is and is not plastic.
    a. The MERI guide explains how to use a hot sewing needle to determine whether or not a fiber is plastic. We have found that cotton fibers respond to the hot needle in the same way as plastic fibers, therefore we no longer recommend using the hot needle technique.
    b. Instead, we suggest using fine forceps to pick up an individual fiber and hold it about 1 cm above a candle flame for about one second. DO NOT HOLD IT IN PLACE FOR TOO LONG, or it may be completely incinerated. We recommend practicing this technique using cotton fibers from 100% cotton balls/swabs, or 100% polyester (e.g. pillow stuffing).
    c. Move the heated fiber back under the microscope's field of view. Natural fibers will be partially/mostly singed (black), but will still be about the same length as they were before heating. Synthetic fibers will melt and generally form blobs on the end(s) of the fiber.
16. For the Florida Microplastic Awareness Project, we are dividing the plastics

into four types:

a. Fibers: These are likely to be the most common type of plastic seen. They will look like thin threads, and are often clear or colored (blue and red seem to be the most common colors). Look carefully at any green "threads" that you see—they might be filamentous algae rather than plastic. If in doubt, check with your regional coordinator.
b. Fragments: These are pieces of plastic that seem to have come from larger plastic items (but are not fibers). Pieces of algae can be mistaken for fragments, as can sand grains.
c. Microbeads: These will be completely spherical and could be up to 1 mm in diameter. They will probably be colored. Diatoms (a type of plankton) are commonly mistaken for microbeads. Diatoms are easily crushed with the forceps, while microbeads are not. Diatoms are also disc-shaped when viewed from their sides.
d. Film: Pieces of thin plastic (like grocery bags, plastic wrap etc.) should be recorded separately from fragments.

17. Use the videos on the "Get Involved" page at www.plasticaware.org to help you with the identification of fibers and microbeads.
18. Record the number of each type of plastic seen on the data sheet, along with your initials. If the sample does not contain any plastic, please enter zeros in the boxes.
19. Check with your coordinator to see what they want you to do with the filter. They may wish to keep the filter (e.g. so they can photograph some of the plastics) or they may have you discard it.
20. See page 14 for information about how to enter the data online so it gets put into the project database.



Filters in petri dishes

Plastics on filters (grid size of filters is 3 mm x 3 mm)

Photo credits: Maia McGuire



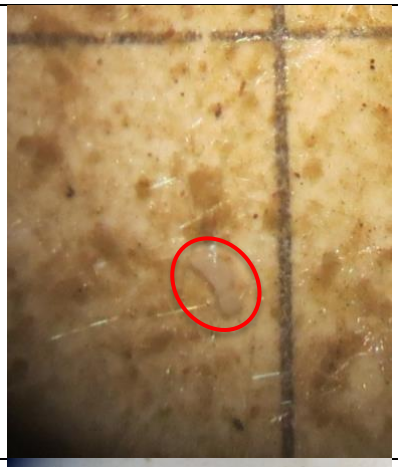
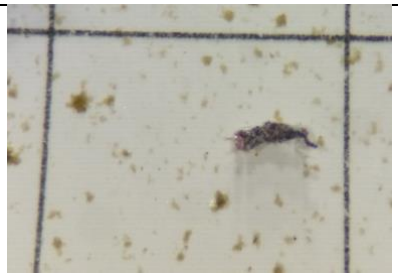
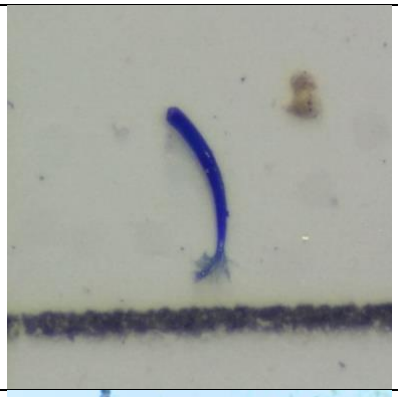
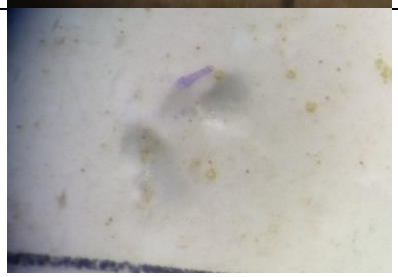
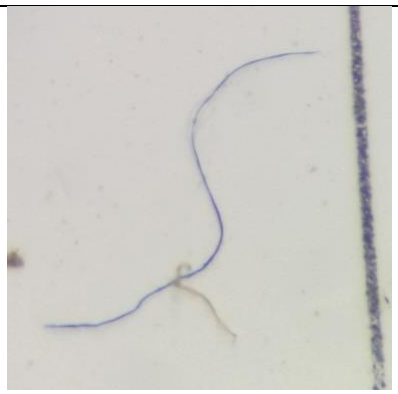
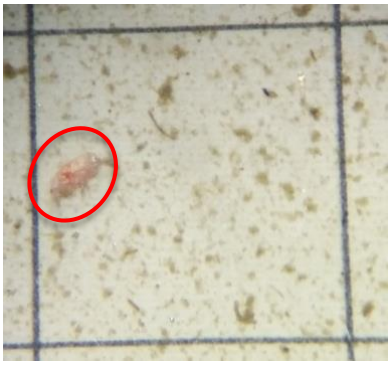





Photo credit: Cheryl McCormick



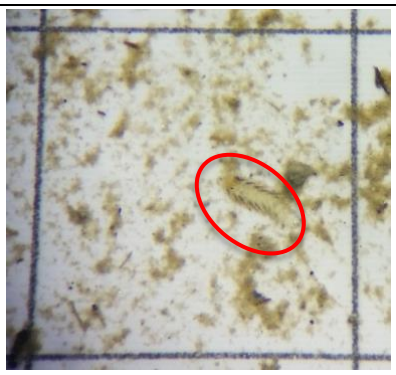
Plankton on filters (from saltwater samples)

Photo credits: Maia McGuire



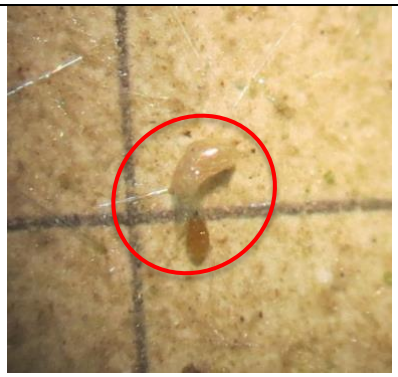
Copepod

Ostracod

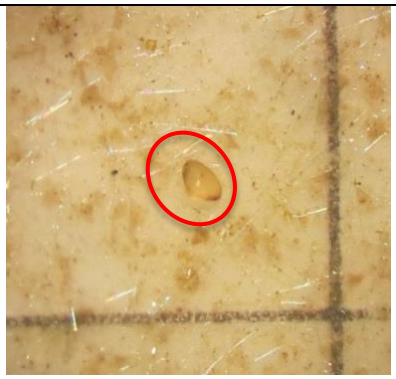


Polychaete worm

Zoea larva (crustacean)

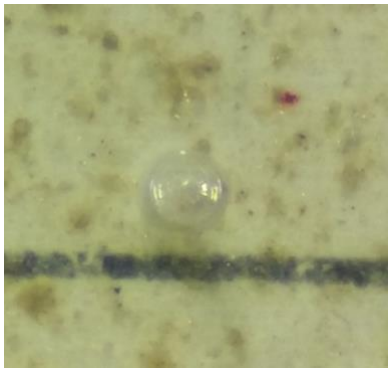


Copepod with egg sacs

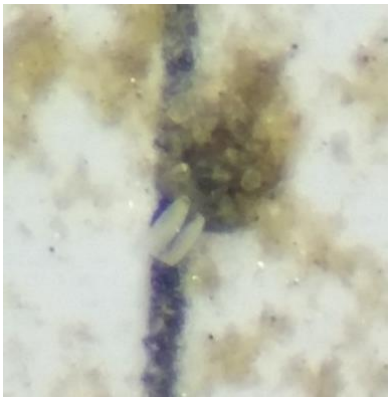


Bivalve larva

Copepod (top); snail larva (bottom)



Diatom (top view) (often confused with plastic, but diatoms are fragile and will crush when "poked")



Diatom (side view)

Sand grains (will sink when placed in a drop of water)

Reporting your Time and Data

It is very important that you track and report the time that you spend collecting and analyzing samples, or at outreach events. This time includes your travel (if appropriate) and time spent setting up/breaking down at events. The time you spend getting trained also counts towards your time spent on this project.

To report your time:

1. Go to www.plasticaware.org
2. Click on "Get Involved"
3. Click on "Report Volunteer Time" and enter the appropriate information in the online form and submit.
4. You may choose to enter your time each time you volunteer, or may want to do it periodically (e.g. once a month).

To report data:

1. Go to www.plasticaware.org
2. Click on "Get Involved"
3. Click on "Report Data" and enter the appropriate information. If you have data for multiple samples from a single location, you can do so on one form (enter the average for all samples, then in the comments box, provide ranges or individual sample data). If you are reporting data for several different sites, please submit one at a time (i.e. once you have submitted data for one site, go ahead and repeat the process for the next sample site.)

What will happen to your data?

Once you submit your data in the online form, it will be sent to the overall project coordinator (Maia McGuire). Maia will periodically be taking submitted data and entering it into a Google Map (you can see the map by clicking on the link at www.plasticaware.org).