MarineGEO
THE TENNENBAUM
MARINE OBSERVATORIES NETWORK

Smithsonian Institution

Protocol: Seagrass community sampling

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1. Summary

Seagrasses provide critical habitat and a range of ecosystem services in coastal regions around the world. For this reason seagrass extent and composition are considered Essential Ocean Variables (Miloslavich et al. 2018). This protocol provides a standardized set of measurements for characterizing the changing structure and health of seagrass communities, including the cover and physical structure of seagrass canopy, fouling load, and community structure of the epifaunal communities that use seagrasses as habitat and are important conduits of productivity to fishes. The latest version of this and other MarineGEO protocols, along with field data sheets, data templates, instructional videos, and standardized analysis scripts, are hosted and updated at the MarineGEO GitHub site[1].

2. Rationale

Seagrass habitats and the services they provide are threatened by a wide range of human activities, notably coastal development and declining water quality (Waycott et al. 2009). Effective conservation and management requires reliable data on the nature and mechanisms of change in seagrass extent and ecological functioning. This protocol is designed to track changes in the health of plant and animal components of seagrass ecosystems (Douglass et al. 2010, Duffy et al. 2015), producing data that are meaningful and comparable to those of other major seagrass monitoring programs, Seagrass Watch (McKenzie et al. 2000) and SeagrassNet (Short et al. 2006), to facilitate analysis of combined data across regions and programs. Because seagrasses are diverse and widespread, no general protocol will be applicable in all cases; the MarineGEO team is happy to work with partners to adapt methods to specific local needs. Components of these protocols can be applied to validate remotely sensed imagery of coastal vegetation to describe the regional extent and quality of seagrass vegetation (McCarthy et al. 2018). MarineGEO sites complete the protocol at least annually during the peak season of biological activity, and preferably seasonally, to monitor seagrass community health and changes over time.

3. Requirements

Personnel: at least 3

Time: Preparation: Field: 3 people x 0.5 day. Lab: 3 people x 8 hours (seagrass sample processing), 30 person-hours (epifaunal sample sorting). Data curation: 1 person x 4 hours.

[1] MarineGEO GitHub: https://marinegeo.github.io/

Frequency: Annual (minimally)

Sampling design: Seagrass community sampling has two main components: (1) non-destructive measurements of plant species composition (seagrass and macroalgae), percent cover, and shoot density; and (2) collection and processing of samples for seagrass structure, fouling load, and associated epifauna. The assessment is structured around three replicate 50-m transects, permanently marked where possible, at each site. Macrophyte percent cover, species composition, and seagrass shoot density (in a portion of the quadrat if shoots are very dense) are recorded within each of five 1 m^2 quadrats located randomly along each transect (N = 15 quadrats total at the site). Three types of samples are then collected (N = 15 each) and returned to the lab for processing: (1) a bag sample of seagrass shoots to measure seagrass leaf length and width, sheath length, and mass of fouling material; and (2) a bag sample of seagrass shoots to characterize the associated mobile epifauna community.

4. Measured Parameters

- Density (cover) of seagrasses, macroalgae, and sessile macroinvertebrates
- Seagrass shoot density
- Seagrass leaf length and width, sheath length
- Fouling mass on seagrass leaves
- Mobile epifauna: species composition and abundance

5. Methods: Preparations prior to field work

Goal: Ensure reliable field and lab operations and safety of staff and data

1) Review this protocol and existing laws and customs related to working in marine habitats in your area. Communicate with local stakeholders and obtain permits if required.
2) Designate a team leader responsible for coordinating field and lab activities related to sampling and data management.
3) Notify all participants in field and lab activities who the team leader is. Exchange cell phone and email information among all participants.
4) Designate a secure physical location for storage of samples (e.g., mobile epifauna vials) and completed data sheets.
5) Designate a secure location with informative folder structure for electronic files (scanned field data sheets, Completed data entry sheet, field photos, etc.). Make sure all participants know these locations and that samples and data sheets are placed there safely at the end of each work day.
6) Download a current field data sheet from the MarineGEO GitHub website, print one copy on waterproof paper for each field worker and location to be sampled, and bring them to the field on a clipboard with pencil.

7) Note that MarineGEO data entry templates are designed to have standard format across protocols, to be compliant with conventions such as Darwin Core[2] for biodiversity information and CMECS[3] for habitat classification, and to be easily imported into R for efficient analysis and visualization. This explains the sometimes cumbersome or non-intuitive names of variables[4], which in the end, will save you more trouble than they cause!

6. Methods: Field activities

6.1. Select and characterize the sites

Goal: Select 3 sites for long-term sampling of seagrass communities

1) Seagrass sites for long-term monitoring should be selected for logistical ease of repeated access and representativeness of the area (Short 2006). *We recommend discussing site selection with MarineGEO team during exploration of potential long-term research sites.*
2) Select three locations[5], ideally separated by at least 0.5 km. Seagrass cover should be as homogeneous as available at the site, and the meadow large enough that seagrass extends through most of each 50-m transect.
3) Once a site has been selected, give the location a name and a locationID, using MarineGEO format and conventions. These location identifiers will be used consistently in all data resulting from this and subsequent MarineGEO research at the site.
4) Record the site's GPS coordinates (decimalLatitude, decimalLongitude).
5) Capture digital photographs, sketches, and/or drone imagery as necessary to characterize site and seagrass characteristics.
6) Print a copy of this protocol and bring it with the assembled gear (see checklist below) to the field site when first sampling and when new team members participate.
7) Repeat for the remaining two sites.

6.2. Measure macrophyte percent cover

Goal: Record macrophyte species composition and areal cover (N=15)

1) Assemble gear (see field checklist below) and the appropriate MarineGEO field data sheet for percent cover. Upon arrival at the field site, fill in metadata clearly and legibly on the data sheet. Write your name (first last) as data recorder. Explain any abbreviations used.

[2] Darwin Core: https://en.wikipedia.org/wiki/Darwin_Core
[3] CMECS: https://www.cmecscatalog.org/cmecs/
[4] MarineGEO data standards and guidelines: https://marinegeo.github.io/standards/
[5] Underlined words refer to names of variables in MarineGEO data entry template associated with this protocol.

2) Lay out the first of three 50-m transects. The three transects should be laid *parallel* to the shoreline, near the upper and lower limits, and the middle, of the meadow. If the meadow is extensive and subtidal, arrange the transects to cover the depth range of the meadow.
3) Mark the beginning and end of the transect with temporary markers such as PVC poles (we use ~1.5 m poles driven into the sediment). Record depth (relative to mean lower low water), GPS coordinates, and state of tide on the field datasheet. Poles or other more permanent markers may be left in the field if it is feasible to sample the same transects repeatedly over the long term and if this does not pose a safety or vandalism hazard.
4) Along each transect, randomly select 5 points for deploying quadrats. All subsequent measurements will be made at or near each of these points.

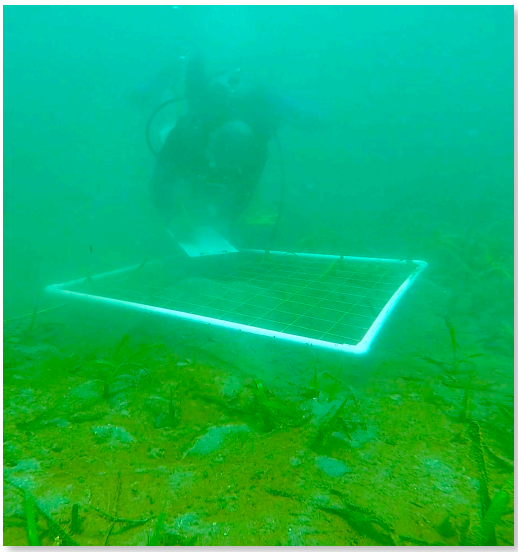

Figure 1. Diver measuring seagrass cover with 1 m^2 quadrat.

5) At each selected point, place a PVC quadrat (1 m^2) adjacent to the transect line. Record a photograph encompassing the whole quadrat from as close to vertical an angle as possible. Use a reliable means to keep track of which photo belongs to each quadrat.
6) Using scuba or snorkel as necessary, identify the macrophyte species directly underneath each point (i.e., intersection of monofilament cross-lines) within the quadrat. Add up the points for a given taxon, and record the total number of points occupied by taxon on the field datasheet. It's often helpful to count one taxon at a time, sequentially (e.g., *Zostera marina* first, then *Gracilaria* sp., then 'red sponge', etc.). Count any point that covers a sessile organism, whether blade, shoot, or other. Be sure to count the number of points over bare substratum as well.
7) Record the total number of grid points (line intersections) within the quadrat you are using, so that the percent cover of each taxon can be calculated later.
8) If possible, record a photograph in situ of any organism on the quadrat you cannot identify.
9) Repeat these procedures for all quadrats on the transect, and then for the other two transects, for a total of three transects and 15 quadrats.

6.3. Measure seagrass shoot density

Goal: Record seagrass shoot density (N=15)

1) *For dense seagrass (>100 shoots/m^2)*: Within each quadrat where cover was measured, randomly place a ring (6 inch diameter) of PVC or other material on the sediment surface.

2) Count and identify the number and species of seagrass shoots within the ring in each quadrat. Count only shoots rooted within the ring.
3) *For sparse seagrass (<100 shoots/m2):* If seagrass shoots are too sparse to be counted accurately in a ring (say, <20 m-2), count all shoots within the quadrat used for cover. You should still measure the cover of seagrass under every point in the 1 m^2 quadrat as described above, including seagrass leaves.

6.4. Collect samples for shoot characteristics

Goal: Collect samples for seagrass shoot dimensions and fouling load (N=15)

1) At each quadrat position where cover was measured, select a point ~1 m from the transect (to avoid destructive sampling within the transect) to collect a sample for seagrass shoot dimensions and fouling load: at each point, carefully cut 3 shoots at the sediment surface and place all three gently into a single pre-labeled ziploc bag (one bag per quadrat position, N=15 total), being careful to disturb attached epiphytes and detritus as little as possible.
2) Place an internal label in the bag, written in pencil on waterproof paper. Use the MarineGEO conventions for labeling seagrass samples: (see reference above):
   a) Convention: [*eventCode*]*SeagrassShoots*[*date*]
   b) Example: PBT001SeagrassShoots2018-08-11 (where PBT = Panama, Bocas del Toro)
3) Place the sample bags in the cooler for transport to the lab.

6.5. Collect samples for mobile epifauna

Goal: Collect mobile epifauna samples (N = 15)

1) At each quadrat position where cover was measured, select a point near, but >1 m away from, the transect (to avoid destructive sampling within the transect) to collect epifaunal samples: at each point, use a single fine mesh (or ziploc) bag to gently but quickly enclose a clump of seagrass shoots. Close the open bottom of the bag around the base of the seagrass shoots as best you can, cut the seagrass blades from underneath the mesh bag, and quickly close and secure the bag (zip it closed if ziploc, or use rope or cable tie for mesh bag) to capture detached seagrass blades and associated animals within. (Note: mesh bags allow water to escape for more compact sample transport, but some workers prefer ziplocs because animals can cling to mesh and are difficult to remove.)
2) Place an internal label in the bag, written in pencil on waterproof paper. Use the MarineGEO convention for labeling epifauna sample bags:
   a) Convention: *[eventCode]SeagrassEpifauna[date]*
   b) Example: *PBT001SeagrassEpifauna2018-08-11*
3) Place the sample bags in the cooler for transport to the lab.

Note: Epifaunal samples are best processed within 24 hours of collection, and can be stored overnight (but not much longer) in a refrigerator. Otherwise the samples can be frozen, although specimens deteriorate and can be more difficult to identify after freezing.

6.6. Review and curate data sheets

Goal: Ensure security and accuracy of data

1) Before leaving the field, review the field datasheets. Check that all fields are filled in, that numbers have appropriate units specified, that all entries are clear and legible, and that both the field team and data recorder's names are entered in case questions arise.
2) If any changes or omissions to the protocol were necessary, note these clearly in the notes field on the data sheet.
3) Upon returning to the lab, assign a unique locationID code to each location if not done already. Scan the field datasheet and, immediately if possible, store both the paper copy and the scanned electronic (PDF) copy in a secure place designated for this purpose. Use the MarineGEO label convention for the electronic file names (see above):
   a) Convention: [*eventCode]SeagrassCover[date]*
   b) Example: *PBT001SeagrassCover2018-08-11*

7. Methods: Lab activities

7.1. Process samples to measure mass of fouling material

Goal: Measure fouling on seagrass leaves, including epiphytes, detritus, and sediment.

At the end of this procedure you should have 15 fouling mass samples and 15 associated seagrass leaf samples.

1) Download the current MarineGEO lab data sheet for this activity, and print one copy for each location sampled. Fill in metadata clearly and legibly. Write your full name on the sheet as data recorder. Explain any abbreviations used.
2) In the lab, carefully transfer contents of the ziploc bag containing the shoot sample into a large, shallow sorting tray (white is best) *without* water.
3) Remove any loose unattached macroalgae and sessile macroinvertebrates, if present, from the seagrass.
4) Separate seagrass above-ground biomass from any below-ground biomass by cutting the shoot where color changes from green (above-ground) to white/clear (below-ground). Discard any below-ground material.
5) Remove all fouling material from the blades using whatever combination of methods is effective. When fouling material is primarily fluffy diatoms and loose detritus, this can often be removed by rubbing gently with the fingers or a loose paintbrush or toothbrush. Material

that adheres to the blades more tightly may need to be scraped off with a plastic knife blade. Set aside the scraped seagrass tissue for measurements of shoot dimensions (see below).

6) Prepare a labeled aluminum foil pack or tray for the fouling material for oven drying. Write the label clearly, using a dark sharpie or equivalent, on the foil pack and use a unique label that will not be confused with other samples that may be around (i.e., more informative than "sample 2").
7) Record the sample number and tare (empty) mass of the foil container on the lab data sheet.
8) Place the scraped fouling material in its labeled aluminum container.
9) Place the labeled foil containers with fouling mass samples in a drying oven at ~60°C and dry to constant weight (usually 1-3 days, depending on the volume of material).
10) When the samples have dried thoroughly, remove the foil container from the oven, weigh to nearest mg, and record the sample (including foil) dry mass on the lab data sheet.

7.2. Process samples to measure shoot dimensions and biomass

Goal: Measure seagrass leaf length and width, sheath length, and dry mass (N = 45)

1) Return to the seagrass shoots scraped of fouling material in the previous activity. You should have 3 shoots in each sample. From each shoot, measure the length and width of the single *longest* leaf and record on the lab data sheet for that sample. Record the sheath length from the meristem (visible as a constriction at base of shoot) to the top of the sheath surrounding the leaf bundle (Figure 2).

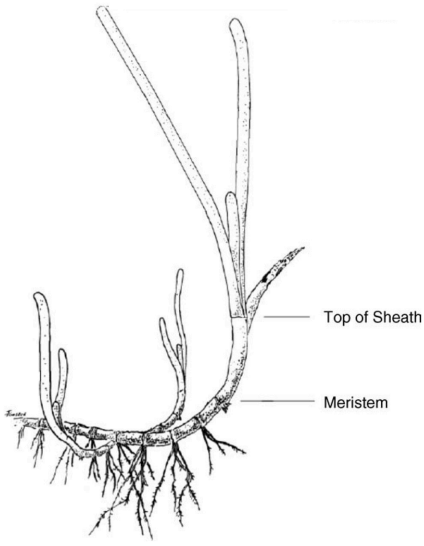

Figure 2. Morphology of an eelgrass (*Zostera marina*) shoot, showing leaves and sheath. Adapted from Gaeckle et al. (2006).

1) Prepare a labeled aluminum foil pack or tray for the scraped seagrass leaves for oven drying. Write the label clearly, using a dark sharpie or equivalent, on the aluminum container and use a unique label that will not be confused with other samples that may be around
2) Record the sample number and tare (empty) mass of the aluminum container on the lab data sheet.
3) Place the scraped leaves in the labeled aluminum container.
4) Place the labeled aluminum containers containing the scraped leaves in the drying oven at ~60°C and dry to constant weight.
5) When the samples have dried, remove the aluminum container from the oven, weigh to nearest mg, and record the sample (including container) dry mass on the lab data sheet.

6) Periodically review the lab data sheet for completeness*:* check that all fields are filled in, units are specified for all measurements, all entries are clear and legible, and the data recorder's full name is entered.
7) If any changes or omissions to the protocol were necessary, be sure to note these clearly in the notes field on the data sheet.
4) Before leaving the lab for the day, scan the completed lab data sheets and store both the paper copy and the scanned electronic copy in a secure place designated for this purpose. Use the MarineGEO label convention for the electronic file name (see above) using the date of *sample collection*, if different than date of data entry:
   a) Convention: [*eventCode]SeagrassShootMeasurements[date]*
   b) Example: *PBT001SeagrassCover2018-08-11*

7.3. Process and preserve mobile epifauna

Goal: Preserve a sample of the mobile epifaunal community with contextual data

At the end of this procedure you should have 15 mobile epifauna samples and 15 associated seagrass samples from which the epifauna were collected.

1) Download the current MarineGEO lab data sheet for this activity, and print one copy for each location sampled. Fill in metadata clearly and legibly. Write your full name on the sheet as data recorder. Explain any abbreviations used.
2) In the lab, carefully transfer contents of the sample bag containing the mobile epifauna and associated seagrass into a large, shallow sorting tray (white is best) filled with water. Gently rinse the sample bag into the tray, repeatedly if necessary, to ensure that all material from the sample is in the tray.
3) Assemble a nested pair of sieves, with the 1 mm sieve on top and the 0.5 mm sieve below. This will be used in following steps to concentrate the epifauna.
4) Agitate the seagrass material in the tray of water for a minute or so to dislodge associated animals. Then pour the water and dislodged animals into the nested pair of sieves.
5) Carefully refill the tray with tap water, repeat the agitation of the seagrass sample, and pour the water with any remaining animals into the paired set of sieves. Repeat this step until few or no animals are left in the agitated water.
6) Carefully remove all seagrass, macroalgae, and sessile macroinvertebrates (hereafter 'epifaunal-seagrass sample') from the tray, checking to ensure that no mobile epifauna are clinging to the removed material from the tray. Set aside to be weighed after processing the mobile epifauna. .

7) Locate two 20-ml scintillation vials (preferably plastic) for storage of the mobile epifauna from the sieves.
8) Carefully label each vial with both (a) an internal label, written firmly with pencil on water-proof paper, and (b) an external label, written with sharpie or, better yet, ethanol-resistant felt-tip pen. Check that the two labels for the vial are identical and follow the MarineGEO convention for labeling mobile epifauna samples above, but with addition of the sieve size:

a) Convention: *Sitecode date (yyyy-mm-dd) "fauna" sample number, sieve*
b) Example: *FP 2016-05-10 fauna 6, 0.5 mm*

9) Rinse the contents of the 1.0 mm sieve under a gentle stream of tap water to flush through as much sediment and debris as possible.
10) Carefully transfer the contents (mobile animals and associated debris) from the 1.0-mm sieve to its appropriate labeled vial. This can be done by picking up the mass of small animals with a spatula or spoon, and transferring to the vial. Then use a squirt bottle of water to gently concentrate remaining animals in the sieve, repeating as necessary. The final residue of animals can be washed carefully into the vial with a squirt bottle.
11) Remove as much water from the vial as possible, being careful to avoid removing any animals, then fill the vial about ¾ full with 95% EtOH.
12) Check to make sure that both internal and external labels are in place and identical.
13) If the epifaunal sample will not fit in a single 20-ml vial, divide the collected animals into as many vials is necessary. Assign each vial a unique subsample number:
    a) Convention: *Sitecode date (yyyy-mm-dd) "fauna" sample number-subsample number, sieve*
    b) Example: *FP 2016-05-10 fauna 6-1, 0.5 mm*
14) Repeat the last four steps for the contents of the 0.5-mm sieve.
15) Record the unique label information of each vial on the lab data sheet.
16) Store the epifaunal samples in a secure, designated place for later identification and enumeration.

17) Return to the 'epifaunal-seagrass sample' from which epifauna were removed. Prepare these samples for weighing, using a separate container for each seagrass, macroalga, or sessile macroinvertebrate taxon in each replicate sample.
18) Prepare a labeled aluminum foil pack or tray for each epifaunal-seagrass sample for oven drying. Write the label clearly, using a dark sharpie or equivalent, on the aluminum container and use a unique label that will not be confused with other samples that may be around
19) Record the epifaunal-seagrass sample number and tare (empty) mass of the aluminum container on the lab data sheet.
20) Place the epifaunal-seagrass sample in the labeled aluminum container.
21) Place the labeled aluminum containers containing the epifaunal-seagrass sample in the drying oven at ~60°C and dry to constant weight.
22) When the epifaunal-seagrass samples have dried, remove the aluminum container from the oven, weigh to nearest mg, and record the sample (including container) dry mass on the lab data sheet.

23) After samples have been weighed, and before leaving the lab for the day, scan the lab data sheet and store both the paper copy and the electronic copy in a secure place designated for this purpose. Use the MarineGEO label convention for the electronic file name:
    a) Convention: *Sitecode date (yyyy-mm-dd) fauna*
    b) Example: *FP 2016-05-10 fauna*

7.4. Count and identify mobile epifauna

Goal: Characterize species composition and abundance of epifauna associated with seagrass.

1) Download the current MarineGEO lab data sheet for this activity and print one copy for each epifaunal sample (N = 15 per site). Fill in metadata clearly and legibly. Write your full name on the sheet as data recorder. Explain any abbreviations used.
2) *Note: Standard MarineGEO protocol is to enumerate only those epifauna retained on the 1.0 mm sieve, but to preserve those retained by the 0.5 mm sieve for comparison with other studies as necessary. We also exclude from the mobile epifauna category all primarily meiofaunal taxa, including nematodes, copepods, ostracods, and mites. If these are retained on the sieves they can be ignored.*
3) Empty the contents of the vial containing the 1-mm sieve epifaunal sample into a 10 cm glass Petri dish or similar vessel for sorting. If the sample is large, begin with a portion of the sample, and repeat until the sample is completed.
4) Prepare the sample for photographic measurement of epifaunal body sizes: remove as much debris as possible and spread the animals out so they are separated on the bottom of the dish. If there are too many animals to separate clearly, return some to the vial to clear space.
5) Place a centimeter rule under or adjacent to the dish to provide a scale for measurement.
6) Take one or more digital photographs to capture all animals in the dish at sufficient resolution that their body sizes can later be measured from the image using the included scale. Photographs may be captured with a camera attached to the microscope or with a cell phone fitted with a macro lens.
7) Inspect the images to make sure they are sharp enough to measure body sizes of animals.
8) Once all animals in the sample have been photographed, it's time to identify and count them. If you don't know the taxonomy well (and even if you do), it's often easiest to separate the animals into groups by morphospecies.
9) Count all individuals of a given (morpho)species. As you count them, remove them to a small vial with 70% ethanol and an internal label with the sample information (as above) and species name. If you cannot reliably identify the taxon to species, identify it to the lowest taxon that you feel confident in, and give it a provisional name, (e.g., Nereid polychaete A).
10) Record the taxon name, provisional or otherwise, and the number of specimens on the appropriate lab data sheet.
11) *Note: Strive to keep separate any taxa that you suspect are different species, even if you don't know their names. For example, if your sample contains two groups of amphipod specimens that you can't identify but you feel pretty confident are different species, you can call them amphipod A and amphipod B. This is better than lumping them together under "Unknown amphipods".*
12) *Note: When in doubt about an identification, record the name only to the finest resolution you are confident in (e.g., Amphipod A or Gammarus sp.). If you think you know the species but are unsure, list this in notes column with a question mark (e.g., Gammarus mucronatus?).*

13) Record photographs of species you can't identify at the highest magnification where you can still achieve a sharp image. Label these images with the sample information and provisional species name you assigned on the data sheet.
14) Review the lab data sheet. Check to ensure that all requested fields are filled in, all entries are clear and legible, and the data recorder's name is entered.
15) If any changes or omissions to the protocol were necessary, be sure to note these clearly in the notes field on the data sheet.
16) After the sample has been sorted, and before leaving the lab for the day, scan the lab data sheet(s) and store both the paper copy and the scanned electronic copy in a secure place designated for this purpose. Use the MarineGEO label convention for the electronic file name but with addition of "counts":
    a) Convention: *Sitecode date (yyyy-mm-dd) "fauna" sample number, sieve, "counts"*
    b) Example: *FP 2016-05-10 fauna 6, 1.0 mm, counts*

8. Methods: Curate the data

Goal: Ensure that all data are accurate, secure, and distributed to the appropriate parties

1) Develop the habit of checking data sheets regularly to make sure they are complete, legible, and understandable to someone who was not present when they were filled in (e.g., avoid obscure or non-standard abbreviations, numbers without units, dates without years, etc). Specifically, check data sheets before leaving the field, and before leaving the lab for the day.
2) Scan the completed field data sheets to PDF, name the PDF files with informative file names that include site and date, and store both paper and electronic copies in a secure folder designated for this purpose by your site PI. Then back them up.
1) As samples are processed, weighed, and counted, enter the data, along with all metadata, from the lab data sheet(s) into the MarineGEO seagrass data spreadsheet (available at https://marinegeo.github.io). Be sure to enter your full name and date of data entry, and fill in all columns as best you can. Save the file with a unique and sensible suffix to distinguish it from the template. Save the data spreadsheet in the designated folder, and back it up. *Always* retain the original paper and scanned data sheets, even after data have been entered into the spreadsheet.
3) Once field and lab processing of all samples are complete, email the entire package of files (data spreadsheet, scans of all paper data sheets, and any images collected) to MarineGEO HQ at: MarineGeo@si.edu.

9. Selected literature

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Protocol: Seagrass community sampling

10. Materials checklist: Field

☐ *MarineGEO standard field datasheet (waterproof paper)*	1
☐ Hand-held Global Positioning System (GPS)	1
☐ PVC marker poles, 3/4" diameter (Length ~1.5m)	18
☐ Field hammer (for pounding in poles)	1
☐ Bin or buckets for transporting and holding gear in field	1
☐ transect tape, 50 m	1
☐ 1 m^2 quadrat(s) with monofilament lines forming at least 50 points	1-3
☐ Clipboards	1-3
☐ Pencils (usable underwater)	1-3
☐ Scissors (for cutting seagrass shoots)	1-3
☐ PVC ring, 6 inch diameter (for sampling shoot density)	1-3
☐ 500 μM mesh bags (or ziplocs) for seagrass and fouling samples	20
☐ labels: water-proof paper, pre-cut (for all samples)	30
☐ Ties for securing mesh bags (cable ties, ropes, or rubber bands)	~100
☐ 1-gallon Ziploc bags (for epifaunal samples)	10
☐ Cooler (for storing and transporting samples to lab)	1
☐ YSI salinity/temperature meter (or equivalent)	1
☐ LICOR light meter (if available)	1

Protocol: Seagrass community sampling

11. Materials checklist: Lab

☐ *MarineGEO data entry template (electronic)*	1
☐ *MarineGEO seagrass shoot and fouling mass data sheet*	1-3
☐ *MarineGEO seagrass-epifaunal sample data sheet*	1-3
☐ *MarineGEO epifaunal species data sheet*	1-3
☐ Trays for sorting (large, shallow, preferably white)	3
☐ Forceps	3
☐ Scissors (for cutting labels)	1
☐ Pencils (for writing internal labels for ethanol vials)	1-3
☐ Meter stick (for measuring seagrass blade length and width)	1-3
☐ Large 1 mm-mesh sieve (for concentrating mobile epifauna)	1-2
☐ Large 0.5 mm-mesh sieve (for concentrating mobile epifauna)	1-2
☐ Squirt bottle: DI water	1-2
☐ Squirt bottle: 95% ethanol	1L
☐ 20-ml plastic scintillation vials (for preserved epifauna)	>15
☐ labels: water-proof paper, pre-cut (for all samples)	30
☐ Aluminum foil roll (for drying and weighing samples)	1
☐ Sharpies for labeling Aluminum foil	1-3
☐ Drying oven	1
☐ Refrigerator/freezer (for storing samples before processing)	1
☐ Electronic balance, accurate to 0.001 g	1

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12. About the Smithsonian MarineGEO program

The **Marine G**lobal **E**arth **O**bservatory (MarineGEO) is a community of practice, led by the Smithsonian Institution, with the mission of understanding how coastal ecosystems work, and how to keep them working. We use standardized, comparative methods to do so, focusing on nearshore seabed communities because this is where marine biodiversity is concentrated and where humans interact with it most. Our mission is realized through two main components: (1) repeated measurements of key environmental and biological components to produce long-term time series; and (2) experiments leveraging these observations across sites to discover ecological mechanisms in a comparative framework. To place these continuing studies in context and maximize their effectiveness we are assembling a rich database of existing knowledge and new biodiversity collections at each partner site.