Civic Laboratory for Environmental Action Research (CLEAR)
Lab Book
a living manual of our values, guidelines, and protocols

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Protocols (cont.)

CLEAR Value Framework

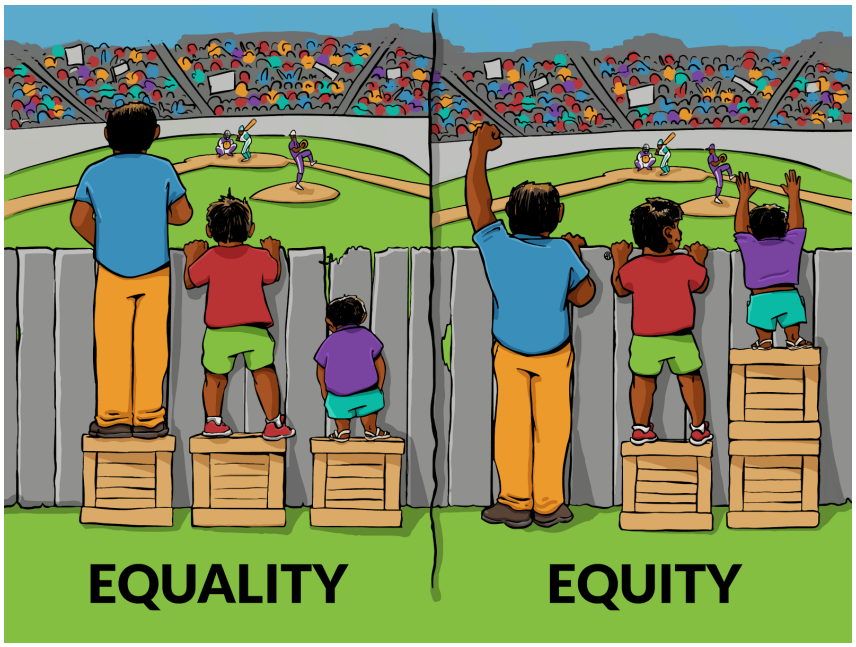
Introduction

This document emerged from a creative, collaborative process where members brainstormed what we value as a lab. It serves to guide our actions, decisions, and work, as well as to help realize our goal to be a *feminist* marine science laboratory! It is a living document, meaning it is something we interact with and update frequently as members come and go, and as we evolve as a lab. While this guide is meant for CLEAR members, it may also be useful for partners to understand how we work, for other labs as inspiration for their own statements, and for those interested in how all knowledge production, including science, is based on values.

CLEAR's main values are Equity, Humility & Solidarity, Supportive Openness, and an orientation to Process.
Note: terms used in this framework are also included in a glossary at the end.

1. Equity

CLEAR's work is fundamentally informed by the feminist value of *equity*, whether we are ordering supplies or building scientific instruments (see Liboiron, 2016). Equity is different than equality. Equality involves treating everyone exactly the same and as a result has no impact on the uneven positions from which different people start. Equity, in contrast, is attuned to the different social, economic, cultural, and political positions of participants and aims to address and transform power relations that lead to this unevenness.



To make these powers explicit we acknowledge our social locations by talking about them openly—something seldom done within the academy. CLEAR has, at time of this *Lab Book* iteration, a mix of faculty (tenured and untenured); PhD, masters and undergraduate students; men, women, and non-binary trans folk; people that are descendents of settlers of European descent, people Indigenous to Turtle Island (aka North America), and people from other countries outside of Europe or Turtle Island. We believe that simply bringing more diversity into science--equality--while maintaining the status quo only perpetuates the violence experienced by peoples who are systematically unacknowledged, underrecognized, and oppressed in and through science. In order for diversity to be meaningful, intersectionality must be embraced. Intersectionality is a feminist concept, from race/ethnicity scholars, that emphasizes the interconnectedness of identities (e.g.: ethnicity, socioeconomic class, gender, sexuality, etc.) and their complex interactions; a straight, cis, upper-middle class man's experiences are different from that of a woman and different again from a black woman's at both a personal and systemic level.

This is called intersectionality. Intersectionality recognizes the interconnectedness of identities such as race, class, and gender of a community or individual, and the intersections of various ways of being: a person is not just a woman and a scientist, but also white, settler, from a rural frontier town, living below the poverty line, a daughter, a sister, an auntie, an academic-in-training, and disabled, among many other social locations. Thus, equity work involves many levels, systems, and ways of knowing and being.

In practice, CLEAR works towards an equitable environment by:

- using "one diva, one mic" during lab meetings: only one person speaks at a time. Usually, white, male, senior people interrupt others, but we prefer that no one interrupt anyone.
- using "round-robins" with a "pass" option during group meetings, where everyone has a turn to speak, which encourages contribution and discourages over-contributing (with exceptions and flexibility)
- through facilitation training every year, we learn to *step up* to advocate for another person's work or ideas if they are quiet, modest, or absent, and to *step back* if we have taken up more space than others during a conversation
- we aim to ask questions as much as we make statements
- we have employment/recruitment equity practices for the lab when hiring or soliciting new members (see *Joining the Lab* in *Guidelines*).
- at the start of every meeting, we have a recurring and/or routinized pronoun check ins
- we use consensus-based decision-making as described by Harnett (2011) that involves open discussion that identifies key concerns, creating proposals that address them, then amending the proposal until everyone agrees to move forward. This can take a few minutes or a few weeks. The aim of consensus is to redistribute power and advocacy. Tenured faculty do not have more say than undergraduate students, though we

acknowledge that faculty still have greater power of persuasion and that unconscious biases are always at work.
- we have an equity in author order process that includes social position as a factor in determining credit in articles (see *Protocols*).
- payment rates are set by the university and/or granting activities, so people are not paid the same in the lab. We have a transparent list of who is being funded by what in the lab. We aim to have no unpaid volunteers.
- anyone is able to work on any project if they like (assuming successful collaboration styles).

There is no "outside" of power relations, and our efforts in the lab are unable to overcome inequity completely. Yet these practices serve (at least) three functions: they are an ethic-in-practice that confirms our solidarity with one another in the lab; they make the politics that are always at work explicit and able to be addressed rather than implicit and unacknowledged; and they are a form of "prefigurative politics" where we work to model the world we want, rather than merely critique the world as it is.

2. Humility & Solidarity

To work with and among others, humility and solidarity are key values to ensure we are respecting others. In science, values of individualism, heroism, machismo, rescue, paternalism, and exceptionalism are dominant. Humility and solidarity counteract this.

Humility is enacting the understanding that our world is interconnected and that there are bigger things than ourselves. It is about recognizing that one still has much to learn regardless of age, education, or lived experience and about remaining teachable no matter how much we already know. Being humble means that we—as members of larger groups of humans and others[1] —recognize that we are not singular nor superior in our knowledge, perspective, experience, or social position, and that we are connected to others whether we want to be or not. We can be humble by being ready to change our minds and actions, being responsive to context, and being mindful of our surroundings so we might adapt to it rather than force it to adapt to us. Humility acknowledges feminist standpoint theory[2] and understands that there are many ways to know things, many different forms of knowledge, and recognizes the limits of a single way to know things (e.g.: strictly via the scientific method rather than lived experience).

Humility is not modesty; modesty usually means not talking about or celebrating your achievements. If you are modest, then you are not acknowledging or celebrating the network you are part of which is also part of those achievements; you do an injustice to yourself and that

[1] "Humans and others" is a less human-centric way to say the more common "humans and non-humans."
[2] Harding, Sandra. 1991. *Whose Science/ Whose Knowledge?* Milton Keynes: Open University Press.

network practicing modesty (erasure of connections) rather than humility (being beholden to connections).

Solidarity is the act of working together towards a shared goal. In this context, it is a collective response to situations of injustice and oppression, deploying supportive actions to transform those situations. Humility is particularly important for solidarity, as different groups might share a goal with others and work to reach that goal together, despite difference. For example, we as marine plastic researchers and the Nunatsiavut Government both wish to avoid polluting life-giving water, so working in solidarity on issues around methylmercury at Muskrat Falls makes sense.[3] Crucially, our acts of solidarity should be in alignment with the actions the Nunatsiavut Government (or others we are in solidarity with) advocate for--this is part of why humility is central to equitable solidarity work.[4] Humility and solidarity act in concert, and operate as synergistic values. Being humble in solidarity means being ready to change oneself, being responsive to and mindful of one's context (i.e.: our social environment) so we might adapt to it rather than force it to yield to as we are. To violate these principles would not be acting from a place of solidarity or humility. Solidarity requires accountability to each other, to other beings, to place, and to the things we produce (i.e.: our lab space, our knowledge/research) because we are always already connected to these things (the basis of humility).

Solidarity is a verb, not a noun. It is not just discursive work (saying things in support, clicking things in support), but involves concrete actions that make material change on the ground ("being in the trenches" rather than "armchair activism"). Like equity, enacting solidarity will be different for everybody as each person has differing relationships to power and privilege and each body has different abilities, needs, knowledges, resources, etc.

CLEAR attempts to create an environment where humility and solidarity are fostered by:

- We have a mandatory community peer review process when we study contamination, where we report our findings back to communities we sample from *before* we publish to ensure we are representing them in a way that is in alignment with their needs and goals (see *Protocol* section for details)
- We work on the species brought to us by fishermen/women and hunters
- We report the plastic ingestion results of fishermen/women's and hunters specific catches back to them so they know if their own food was contaminated
- We make all our data public upon publication
- We work on research questions and topics based on feedback from community meeting surveys-- currently, people have been asking for more biomonitoring surveys and

[3] See Liboiron, Max. (2015). "On Solidarity and Molecules," *Discard Studies.* https://discardstudies.com/2016/10/20/on-solidarity-and-molecules-makemuskratright/
For an example of the place of science and scientists in solidarity movements with Indigenous groups.

[4] Also see Land, Clare. (2015). *Decolonizing solidarity: Dilemmas and directions for supporters of indigenous struggles*. Zed Books Ltd.

education resources, and ask for shoreline and water studies less, so we have adjusted our research focus accordingly (see *Protocols*)

- we do not use the terms “community outreach” or “public education” in our work because these terms assume something called the “deficit model,” where publics/communities need to be educated by academics. This is not solidarity with humility. Instead, we “collaborate with,” “work for,” “work with,” pay, “learn from,” and “partner with” other groups, all of which allows value to flow in two directions, and on shared terms.
- We encourage reflexive/reflective processes, such as when we talk about what we have learned over a season, or what we’ve found hard, or what we’ve valued in others. Sometimes we need to help other people be reflexive.
- We ask questions as much as we make statements
- We listen to people who aren’t scientists with as much focus as when we listen to our professors and they are saying something will be on the exam.
- we only collect data in places where we have been invited. For example, our director went to different communities in Labrador to present our work to see if there was general interest and explicit invitations for doing work in/with different groups and Lands.
- all publications and presentations have a Land acknowledgement. We are always on Indigenous Land.
- not interrupting or capitalizing a discussion by stepping back, which leaves space (physically, mentally, or verbally) for others; this recognizes their worth/contribution is as valued as our own, and that we are learning
- working in groups (or at minimum revising in groups) and/or pairing new lab members with returning member-mentors to encourage the exchange and expanding of knowledges; fresh perspectives always enhance and add value to the collective
- acknowledging each other, both in greeting and for our achievements; whenever a member of CLEAR has an achievement, we congratulate them via the listserv and in person!
- when we have a new member, we all welcome them to the lab when they are added to our email group.
- no projects are done solo: they are either done in groups or returned to the group for review, comments, and sent back for refining

A reading list for Humility & Solidarity:

Indigenous Action Network. Accomplices, not Allies.
http://www.indigenousaction.org/accomplices-not-allies-abolishing-the-ally-industrial-complex/
Land, C. (2015). *Decolonizing solidarity: Dilemmas and directions for supporters of indigenous struggles*. Zed Books Ltd..
Tuck, E. (2009). Suspending damage: A letter to communities. *Harvard Educational Review*, *79*(3), 409-428.

Conflict Resolution: On Remaining in Solidarity during Conflict

Disputes and conflicts will arise whenever we are acting as part of a collective or working with others in a group. It is key in times of conflict to be especially mindful of our value of solidarity

and to that end, a collectively authored *Conflict Resolution Protocol* (see section on Protocols) has been established to guide our lab through disputes.

3. Supportive Openness

Supportive openness is caring for each other so that we can share the important things with each other. When we collectively described what we value about CLEAR and what values CLEAR exercises, the terms open and supportive came up often. This comes from the lived experience of lab members feeling welcome, safe, and at home in the lab. bell hooks does a great discussion of “radical openness”, which approximates our position (https://youtu.be/9oMmZIJijgY?t=59m10s) .

SUPPORT= (Community care + Self-care) x Accountability

Support is the care we show for each other and the contexts we are in. Support is manifested within the lab and extends beyond to all we interact with: to the communities we work with, the technologies that we build, the beings that we are in relation with. We support each other through actions, not just words, which is also in line with our value of solidarity. However, support does not mean always agreeing with each other, but rather moving through difference and disagreement while holding each other. Support is both collective care work and holding each other accountable. Support without accountability is paternalism. CLEAR is dedicated to building the structures that allow us to care for each other and help us be accountable to our contexts.

Support also includes self-care. Plastic pollution and/or environmental research—and activist work in general—are heavy and depleting activities that require significant emotional and physical labour. This is why this lab values each person’s self-care needs and encourages self-awareness and reflexivity in our members so they are attuned to their needs (as opposed to encouraging them to manage or ignore them). An important caveat of support is that self-care on its own is not support, but survival. Self-care requires enabling community structures and processes. When thinking of self-care we ask, what am I going to do for myself that allows me to thrive in the lab?

In this way, CLEAR also values mutual caring and support, otherwise known as an “ethics of care”, believing that caring is not just work we must do for ourselves (self-care) but for each other as well. Support and care are forms of political and ethical practice that “holds things together” (de la Bellacasa, 2011, p.90; Martin et al., 2015). We also acknowledge that care work can disportionately fall upon or be needed by certain groups more than others depending on gender, ethnicity, class, sexuality, and perceived abilities (Upping the Anti, 2016). With this in mind, the lab aims to distribute and acknowledge care work in its various forms. By doing caring

work for ourselves and for each other we assure the longevity of our lab and the wellbeing of its members.

OPENNESS= Sharing what's important with each other

Openness speaks to our value of sharing things that are important to us, especially what can't be shared outside of the lab, with each other. This includes our emotions and states of being: exhaustion, frustration, pain, joy. Openness allows us to celebrate our differences in identity and experience and provides us with context to move through the complexities of difference when it gets tricky.

A supportive environment allows openness. However, openness cannot disable support. For example, someone being open about their racism or sexism within the lab is in conflict with both our values of equity and support and therefore is not acceptable. A supportive, open lab will foster trust between lab members.

CLEAR attempts to create a caring and supportive environment by:

- one of our only rules is that if you are heartbroken, sick, or exhausted, you go home and take care. Your lab work/school work/job is not more important than your life
- when you do take a break, we let each other know, since we are part of a collective and existing collaborations
- having flexible work arrangements (e.g. each member's unquestioned need for time-off, early arrival/leave, etc.)
- accommodating and/or changing to be more accessible wherever possible; providing opportunity at each onboarding of new and returning members for needs to be communicated (e.g.: pronoun checks but also for disabilities/abilities) and once communicated those standards are applied to every lab member not just those who need "accommodating" (i.e.: everyone does a pronoun check, we position ourselves to be lip-read by every member); it is also holding meetings in accessible locations for those with limited mobility
- "feeding" each other, not just taking (figuratively and literally, with potlucks and emotional support; not *hand-feeding*, unless consent is given)
- creating safer spaces in the lab and in the field (e.g.: asking for and avoiding member's triggers, pronoun checks, having conflict protocols ready, etc.)
- asking for and accounting for each other's unique needs (e.g.: considering and honouring dietary restrictions, making members aware of allergies, accessibility concerns and barriers to participation, etc.)
- celebrating each other, including thank-yous for work done
- acknowledging and working with emotionality

- equitably distribute tasks like maintaining equipment, cleaning up, booking rooms, scheduling, and other mundane tasks
- keeping humour, fun, and personality alive and part of the lab work
- trusting each other until given reason not to, and even then allow for growth
- Round-robins when speaking at lab meetings for the opportunity to share
- Showing up to meetings and other events with lab members
- Seeing each other as real people, not confined to their roles, skills, or tasks assigned to them, including asking about people's lives outside the lab

As new members join CLEAR and as returning members evolve as people, new ways to care for each other and keep each other accountable will emerge. Providing a space and/or mechanism for members to share their needs and to enshrine them in this document is vital to upholding this value.

4. Orientation towards process

CLEAR is an activist lab; we strive to make concrete changes in the world through our scientific work. But rather than use scientific data to make arguments for change (though we do that, too), our focus is on making social change through our methods. How we choose new members, how we order supplies, how we gather samples and with whom… every step of our scientific processes aim to achieve equity, humility, solidarity, and supportive openness. This means two things: first, *we are focused on processes and methods rather than outcomes and findings* as processes and methods are an opportunity to insert feminist politics/practices; and secondly, that we are devoted to *change*, and to flexible processes instead of fixed and rigid structures or rules for doing things. There are important differences between a rule bound structure and a system of processes and practices. The former is authoritative and resists humility, and the later is situated-- responsive to what is happening, when, and with whom.

We do this by:

- accepting change as part of our *modus operandi*; the need for change may be discovered through careful reflection, reflexivity, and continuous learning
- this lab book is *living*, meaning it can and will change as membership changes and our focus changes
- our author order protocols are an approach to establishing equity in author order, but they are not rules.
- our weekly lab meetings are essential to constantly re-calibrating what we're up to, what we stand for, and how we exist as a community. We are not established when people are hired or accepted to programs, but are constantly reforming via weekly meetings.
- the protocols listed in this lab book are iterative and adapted that changes and is updated every time they are used, sometimes in small ways, sometimes in large ways,

but always in ways based on the experiences of the users and the sites in which the protocols are used.
- process and project change can be driven by communities, researchers, or findings. This can determine whether finding are published,
- the process and integration of the lab's values
- allowing certain projects to run their course and progress without setting a cap, fluidity
- Presenting findings to lab members and colleagues prior to presenting at conferences and in public venues, feedback can provide the presenter with support, direction, speaking skills
- Project success not necessarily defined by its outputs, but by its ethics,

Glossary of Terms

intersectionality: the interconnected nature of social categorizations such as race, class, and gender as they apply to a given individual or group, regarded as creating overlapping and interdependent systems of discrimination or disadvantage as well as privaledge. i.e.: a white woman and a black woman may have being women in common but race and class may complicate or add further levels for analysis

feminism: bell hooks defines feminism in "Feminism is for Everybody" as "feminism is a movement to end sexism, sexist exploitation, and oppression. (...). Practically, it is a definition which implies that all sexist thinking and action is the problem (...). It is also broad enough to include an understanding of systemic institutionalized sexism. As a definition it is open-ended. To understand feminism it implies one has to necessarily understand sexism." Because feminism's bane is oppression, solidarity with other forms of oppression (race, sexuality, class, etc.) via intersectional feminism is absolutely crucial to feminism. This is why in our lab, we talk about gender, but we also talk about rural living, class, education, and other markers of difference.
(bell hooks participates in an exchange on the nature of feminism as something concrete, not completely subjective: https://youtu.be/9oMmZIJijgY?t=57m38s)

systemic (level of analysis): systemic analysis, as opposed to individual analysis, recognizes that oppression is often structural and not just arising in interaction between individuals; institutions, economics, legislation, and other societal influences can (re)create oppression by limiting, biasing, or compromising our agency/choices and other means (like policing or gatekeeping, etc.); https://youtu.be/5SBFdtqW0GM

discursive: relating to discourse or modes of discussion; analyzing on the level of discussion

reflexivity: like reflecting, but fancy. Commonly refers to someone's capacity to recognize forces of socialization and alter their place in the social structure; loosely speaking this means

being as skeptical of one's *own* views, biases, and position, as we are of other's as they may be informed by our social environment (e.g.: patriarchy, colonialism, white supremacy, etc.); this happens through a process of deep reflection (https://youtu.be/xCiKogar570)

living document: a document that is dynamic and continually updated by members; i.e.: this Book is not an artefact of the past but grows and changes with the lab

decolonial: Following Eve Tuck and other Indigenous thinkers, Indigenous decolonization and decolonial acts means: returning sovereignty and Land to Indigenous peoples. In Franz Fanon's terms, it is replacing settlers with the colonized. It is important not to conflate decolonization with anti-colonization, or other forms of doing good in the face of empire, imperialism, and colonialism. See Tuck, E., & Yang, K. W. (2012). Decolonization is not a metaphor. *Decolonization: Indigeneity, education & society*, *1*(1).

Anti-colonial: Fighting against the effects, logics, and structures of colonialism, including relationships with Land that are based on resource extraction or human interests, ways of knowing that exclude non-scientific thought, and/or discourses of discovery and ownership.

Turtle Island: The land also known as North America according to some Indigenous origin stories. North America was formed when a muskrat got some mud from the bottom of the water to put on the back of a turtle so that Falling Woman (who fell from the sky) could have a place to live. The turtle grew and grew and became this Land. You can tell because North America looks like a turtle.

tenure: the career track typical of universities by which professors get permanent posts as a recognition of scholarly contributions; created as a response to the Cold War-era practice of firing communist sympathizing professors, tenure guarantees job security once a professor has established themselves in the academic community.

cis(gender): having a gender identity that is consistent with the assignment given at one's birth (i.e.: at birth, doctors declared you a boy, then later in life you saw no reason to dispute this and identify as a boy/man)

trans(gender): having a gender identity that is somehow different than the assignment given at one's birth; there are many ways this can be inconsistent and many different subsets of trans identity and gender expression (i.e.: at birth, doctors declared you a boy, then later in life you refuted this and instead identify as a woman or neither/both gender(s), or have gone back and forth)

non-binary/enby (trans): having a gender identity or expression that is neither of the binary--male or female--sexes/genders; non-binary people sometimes use gender neutral pronouns as well (they/them/theirs or ze/zir/zirs, etc.)

CLEAR Rules

There are only three rules:
1. If you are sick, heartbroken, or exhausted, go home. This job is not more important than your well being.

2. No fleece in the lab. It contaminates samples. To reduce contamination, wear a lab coat. Wear a lab coat whenever you wear clothes. Always wear clothes, so always wear a lab coat.

3. Clean up after yourself, and clean up after others. This should mean things are clean.

CLEAR Guidelines

1. **Attend all lab meetings.** Lab meetings are the main way that CLEAR operates as an entire lab, and as a community. Meetings are where we discuss the values, concepts, and ideas that underlie all our work as a lab, and thus are where we work out *how* we work. Without the meetings, we are just some people working on some stuff. If you are not going to make a meeting, email Max, France and/or the lab in advance.

2. **Work autonomously, with others**. We trust and depend on you to get your projects done, and there won't be a lot of oversight to ensure that is happening. Max cares for the lab as a whole, while lab members care for projects, each other, and themselves. When you need help, reach out to other lab members—*we expect that you are able to ask for help when you need it,* and to problem solve. You are *always* welcome to check in with France and Max, but you were hired because you're smart and can figure things out, so try that first. Mistakes will be made. That's cool.Try to make them with as many other people as possible.

3. **Take care of each other, and yourself**. When you first start, work in pairs so other lab members are there to answer questions or lend a hand. If you notice other lab members are struggling, provide whatever support you can. Listen to each other. You can always bring Max or France in to provide concrete support if you or another lab member requires it.

3.5. **Take breaks**! If you work for three hours or more, take a paid 15-minute break every 1.5-2 hours. If you work for six hours, take a paid 30 minute break in addition to the 15 minutes. If you work for 8 hours, take a 1-hour paid lunch break. Don't work more than 8 hours. That's silly.
If you're doing intensive microscope, video, or counting work, rest your eyes every 20-30 minutes. Studies have shown that taking regular breaks makes you more productive. It's also important to care for yourself. If Grandmother is there, he makes great breaks.

4. **Resolve conflicts when they are small**. Conflict resolution is a major part of care. If you're having some trouble with a rule, a process, a person, etc., others may not notice and it's up to

you to articulate the issue so we can address it. You can say things like, “I notice that you’re late sometimes, and then I have to use my time to wait for you when I’d prefer to work at our agreed upon time. How can we work on that?” or “Thank you for volunteering to make the map! I appreciate that. But we want to give everyone a chance to practice map skills, so we’re going to have Sandrine do it this time. How does that sound?” Use the training you have in consensus-based decision making to figure out how to move forward together. See the guidelines for CBDM if you need a primer. If you need support in conflict resolution, let France or Max know.

5. **Take your ideas seriously**. Many of the lab tasks are for ongoing projects that we are beholden to community groups, grantors, or others for. But we are always interested in new projects or new ways of doing ongoing projects. We’re especially interested in ideas for how to make our work more feminist, decolonial, and equitable. If you have an idea for something you want to explore or develop, let us know! For example, a few lab members want to start a queer science reading group within the lab—we can make room in meetings to report what they’re doing, find money in the budget to pay them for their time, and establish ways to bring their new knowledge into lab work.

6. **Be on time**. Because the lab is member-run, if you are meeting others for training or carpooling, it is imperative that you are on time so that others are not waiting for you, and the lab is not paying people to wait on someone. If you’re running late, text ahead and let people know when you expect to arrive. This means you’ll need people’s numbers *before* you’re late.

7. **Acknowledge the shoulders & Land you stand on**. When you write articles or do presentations from the work that has happened in the lab, always include an acknowledgement page as your last slide that thanks people who contributed in various ways by name, as well as the lab (CLEAR) itself. For papers, do the same with author orders (see *protocols*) and citation. All presentations and articles should include an Indigenous Land Acknowledgement (see *Equity Protocol,* Land Acknowledgement). This is because environmental science owes a great debt to those who have cared for the Land before us. This is part of a process of reconciliation, and a way of doing equity, humility and solidarity.

10. **Boost marginalized voices**. For articles, reference as many women, people of colour, and others that are usually left out of science as possible to acknowledge their work and give them credit for it. This is called citational politics: most bibliographies are mainly white men, which perpetuates the idea that science and knowledge are by and for white men. This is one place to change that. We can change how value is accrued in science and academia through our citation practices: our articles should include 51% women authors in our bibliographies. We *cite* knowledge we gain from fishermen and women, Indigenous nations, and others not usually cited in science.

11. **Log your hours**. Keep a word doc or excel sheet that outlines the work you are being paid for: what you are doing, how long you did it for, and when. Every two weeks, submit this to

France (or keep it in a place France can get to it), who then submits it to Max for approval.This is subject to change based on payroll requirements, if applicable. This is important so we can budget for the lab, and catch any issues before they come up, since there is no other regular checking in of work that happens. Sometimes Max will ask you how many hours you have left in a contract, or how many hours you're working a week so she can adjust the budget. Keep your hours document up to date!

12. **If you have to go, go. But tell us**! Sometimes the semester is overwhelming, life happens, and you either need to take a break for a few weeks or leave the lab altogether. That's normal and we will look to support you in that. Just make sure you let Max know so she can ensure your project is cared for in your absence and any outro paperwork is done. We also try to do exit interviews whenever possible to hear how your experience in the lab has been.

Protocols

Protocols are : the “attitudes” or “the manner in which one approaches each and every element in our space” (They are different than pure rules or instructions; they are ways that we establish order and maintain practices across our group and within science-- it defines the way we *ought* to proceed or behave in different situations. As such, they are normative, or premised on values, morals, and an idea of how things ought to be done. They are a manifestation of our values.

Onboarding to the lab

Welcome to the lab!

Intro meeting includes:

- Tour of the lab
- When meetings are (time, place, mandatory)
- Lab expectations- manage your own time, solve problems to the best of your ability, use the entire lab as a resource, give this book
- What are your skills or assets? (video, languages, drawing, fishing, software, webdesign, car, poetry, others?)
- Start a schedule of tasks- what are you going to work on, when?
- Get access to Drive, dropbox, lab key
- Safety stuff

Onboarding

- Write down any questions while you learn about the lab:
- Read website (especially the About page and some of the projects)
- Watch the 20 minute intro: https://civiclaboratory.nl/2017/06/16/tools-practices-ethics-for-monitoring-marine-plastics-from-a-feminist-laboratory-video/
- Read Lab Book: https://docs.google.com/document/d/1ZB9nUo6Y7gYpk9pBA8bQGGZOIZ2rUyyw1z-tguS-a5Q/edit#
- Read “Being a Scientist Means Taking Sides”: http://faculty.washington.edu/skalski/classes/QERM597/papers_xtra/OBrien.pdf

Exiting the Lab

- Communal round robin during a meeting: what we valued about your work with us
- Complete all end of job paperwork
    - Mucep Reflection
- Return lab key!!!!!!
- Exit interview
    - How was your overall experience working in the lab?
    - How did your fellow lab members make you feel / how did you feel in the lab environment (approachable? Comfortable to talk to to? Easy to work with?...)
    - What is the most valuable thing you learned? What else did you learn?
    - How do you think your work/experience in the lab could have been different?

        - What would you like to learn that you didn't get to?
    - Would you like to come back to work in the lab?
        - If so, what would you choose to work on?
    - Is there anything else about your experiences that you would like to share?

Equity Protocol

Below are excerpts from "*Equity in Author Order: A Feminist Laboratory's Approach"* a paper by:

Max Liboiron, Justine Ammendolia, Katharine Winsor, Alex Zahara, Hillary Bradshaw, Jessica Melvin, Charles Mather, Natalya Dawe, Emily Wells, France Liboiron, Bojan Fürst , Coco Coyle, Jacquelyn Saturno, Melissa Novacefski, Sam Westscott, Grandmother Liboiron

Our protocols are informed by equity and are shaped by a shared commitment to consensus, care work and and acknowledgement of social location. We consider each of these in turn below.

Consensus

Consensus allows a group to reach an acceptable and supportable resolution to an issue, even if that acceptance and support is uneven (see Treloar, 2013 on the importance of differentiating between uneven consensus and unanimous agreement). We follow the consensus based decision-making process as described by Hartnett (2011) that involves open discussion that identifies key concerns, creating proposals that address them, then amending the proposal until everyone agrees to move forward. This can take a few minutes or a few hours.

The aim of consensus is to redistribute power and advocacy. Tenured faculty do not have more say than undergraduate students, though we acknowledge that faculty still have greater power of persuasion and that unconscious biases are always at work. We support one another during the conversation by stepping up to advocate for another person's work or ideas if they are quiet, humble, or absent, and stepping back if we have spoken more than others. We aim to ask questions as much as we make statements. The result is that these conversations tend to be fun, interesting, and supportive.

Care Work is Valued Work

Care is a form of political and ethical practice that "holds things together" (de la Bellacasa, 2011, p.90; Martin et al., 2015). We also acknowledge that care work can disportionately affect certain groups more than others depending on gender, ethnicity, class, sexuality, and perceived abilities (Upping the Anti, 2016). With this in mind, the lab aims to distribute and acknowledge care work in its various forms. Different forms of care we consider when discussing author order have included: training new members on protocols; maintaining equipment; cleaning up; contributing to logistical tasks including note taking, scheduling, sending email reminders and booking rooms; caring for members' physical and mental health by listening, sending sick people home, and telling jokes; and thanking each other.

Social Location

CLEAR has eighteen members. Dr. Max Liboiron directs (non-tenured, woman). Dr. Charles Mather facilitates (tenured, man). We have one male PhD student, four female masters students, and eleven undergraduates, ten of which are women. We have one male and one non-binary trans staff member. We are one Indigenous woman, and a whole lot of white people. Our members have had negative experiences in other labs: our members have been mistaken for assistants rather than Primary Investigators (PIs), have had names moved down lists of author order without discussion, are constantly spoken over or interrupted by senior and male colleagues, have experienced data theft by advisors, have been told that they are expected to work long hours without any credit since they do not make intellectual contributions to projects; are expected to prioritize their work over all other personal and professional goals and obligations; and have been represented in media interviews as "girly" or silly instead of intelligent. This is why we believe that simply bringing more women and people of colour into science--equality--while maintaining the status quo only perpetuates the violence experienced.

For decades, feminist STS scholars have articulated how power circulates through science, differentially impacting people depending on their social location (Haraway, 1988; Harding, 2001). Social location--the groups people belong to because of their place or position in history and society, including race, gender, age, sexual orientation, and educational status--influences not just how people encounter science (determining their wages, likelihood of receiving tenure, awards, etc.) but also how science is produced (influencing the values embedded within their research, the questions they choose to ask, methods they use and more) (Tallbear, 2015; Whyte, 2017). For these reasons, we consider social location when awarding author order. Some of the aspects of social position we've considered include:

- Whether the author is an academic; the value of order to a non-academic will be different than an academic. It is important to consider non-academics as full collaborators and value them as such via authorship and involvement in the consensus process.
- Affiliation; which affiliations do we want to highlight, and why? We may want to promote new, unsung, or underfunded organizations and universities.
- Who needs the cultural capital most? Is anyone going on the job market, going up for tenure, or applying for graduate school?
- For whom is authorship a unique opportunity? Publishing opportunities for faculty and graduate students can be numerous, while for others (such as undergraduate students) publishing may be a unique or sole opportunity to be recognized.
- Hierarchical status; often undergraduate and technician work is not valued as much as graduate or faculty work, even where the same labour is performed.
- Payment status; are some members being paid wages or stipends for the work, while others are not? Are they paid the same amounts or the same way?

- Number of publications authors already have; the publication may mean more to someone who has less.
- Direction of member's research; if a paper fits particularly well with a member's research trajectory, it may provide more value than if they are working on a different topic.
- The past struggles of some members; some people have already encountered acute setbacks in their careers due to social location.
- Markers of difference: consider gender, race, Indigeneity, age, disability, and other markers of difference and privilege. How can we address severe underrepresentation of certain demographics in STEM right here, right now?

The Fine Print

These are guidelines for practices, not rules. They can change -- by consensus! While we consider care work and social location to achieve equity in author order, we still heavily weigh the amount of work someone put into a project or paper and whether someone could speak knowledgeably about the paper their name is on. We also usually include a "contributions" section that outlines the roles that all members played in the study for transparency.

A Snapshot of the Process

*Spliced and emphasis added in **bold** to prevent TLDR (too long, didn't read) in readers.*

So, what does this look like, in practice? Here is a snapshot of our discussion to determine the author order for this paper. The discussion occurred after the lab conducted edits of a full draft together on our laptops during a lab meeting:

The meeting was **facilitated by** Dr. Max Liboiron, which is normal for lab meetings. She wrote the names of every participant on a white-board. People called out new names if they thought someone needed to be considered, since this **process has been built up over the past year through many conversations**. Several absent people were added. "Shall we add Coco? She was part of the first conversations about this and organized all those readings for us, plus did all the emailing." **Everyone wiggled their fingers up** in the air, **indicating yes**. Her name was added. **Fingers sideways mean more discussion was needed**, and **fingers down mean no**. We call them twinkle fingers.

Then, Max drew three columns on the board, checking in with everyone to ensure they reflected the type of work we had done to write this paper: 1) involvement in discussions; 2) involvement in editing and discussions; and 3) involvement in writing, editing, and discussions. We then "chunked" people into the **three categories based on how they had contributed**. One person was moved from the editing to the writing section after some discussion. In this way, we reaffirmed that our author order **still privileges the type of labour** that goes into a paper.

How do we order each person within the three sections? This takes the most time and discussion. We started with the lowest ranked section, involvement in discussions, because it

had the lowest stakes, and decisions we made in low stake discussion set precedents for higher stake discussions. It's easiest to choose the first and last author in a section by going down the lists of **care work and social standing** explained above.

For this paper, **Coco rose to the top** of the discussion section **because of her extensive care work for the lab**. **Sam is a white male** pursuing an English degree and has been **with the lab for a single semester**, **so he went to the bottom**. Jacquelyn and Melissa put in similar amounts of work of a similar kind, but **Jackie is trying to get into grad school and is having to come up with her own funding**, while **Melissa** has at least one more semester with the lab and **will be a front author on another project**. We had this kind of discussion for each section. When it was time to order the section which the facilitator was part of, she stepped back and an undergraduate student took her place running the meeting.

For the most part, **people asked to be placed below others**. **Charles, as a white man with tenure, always wants to go last**. We tried to convince him to go higher in author order since he was a significant editor and writer, but he was firm. We decided that we believed self-determination in self representations is a central tenet of equity in science (a concept we discuss often because we work with communities), so he is last in his section. When we were placing an absent member, we had to rely on knowledge we had about her, and used Google Scholar to see how many other papers she had authored. She was mentioned in several published acknowledgments, but had no papers under her name. **She was a victim of data theft. She rose to the top of the list as an acknowledgement of this setback in her career, and as a sign of solidarity.** Even after this, we discussed how generous she was when she found us free equipment. At every instance, when decisions were made, we used our fingers to show consensus.

There was one point **where two people seemed completely even**: Emily and France. There **was a temptation to resort to alphabetical order, but that is not staying with the trouble**[5]. We went down the lists of care work and social location. We thought of other aspects of social location and care we hadn't already discussed, which required us to expand and nuance our definitions of care and position. We recalled that last paper, Emily stepped back so France could have a higher author standing, so this time, France stepped back for Emily.

Results: Why the equity approach?

Throughout the meeting **the atmosphere was unhurried, comfortable, and fun**. The process of determining author order in CLEAR is **performative (i.e.: it enacts and**

[5] i.e.: abandoning the important subtlety of breaking this "tie" and falling on alphabetical convention is dangerous, classifying the trouble as a nuisance to be dispatched rather than teased out; we should stay with the trouble, contemplating and unpacking it further is a more feminist way of doing things. We later discovered that last time a tie between the same two people occurred and was broken by precedent, not convention.

strengthens the values that we hold dear). **Recognizing past care work leads to thanking each other. Talking about what should be included in social standing hones our ideas of equity, and allows us to stand in solidarity with one another.** Crucially, each iteration of determining author order is different in that we are always faced with a new set of issues to consider. **Our approach to authorship order is not, and can never be, systematic. The establishment of systems closes discussion**. Instead, we use a situated process that recognizes diversity and difference while at the same time rewarding the varied contributions that contribute to knowledge production, and most importantly, we become a more reflexive, stronger feminist lab every time we have these discussions.

Land Acknowledgement

We acknowledge that CLEAR operates on the unceded, unsurrendered ancestral Lands of the Mi'kmaq and Beothuk. We would also like to acknowledge the Inuit of Nunatsiavut and NunatuKavut and the Innu of Nitassinan, and their ancestors, as the original peoples of Labrador.

Notes to remember, based on subsequent practices of this protocol:

- Merit, equality, and equity can become conflated. Stop often to redefine and re-hew to the values we are trying to enact
- What you need and equity are not the same thing
- Consensus can come to reflect the dominant pov and subject position in the room. Look out for that.
- Also look out for rising feelings of competitiveness, unfairness, alienation, anger, etc... what is happening? Where is the gap between what is happening and what you want to happen, and how does that relate to the goals of CLEAR? If you have these feelings, feel free to stop the process. Chances are others are having them, too, and they might be addressed
- While this protocol deals with an article as an individual unit of action, keep in mind that the lab produces a body of work and people are involved in this body as much as a paper-- how can the body of work of the lab and equity be addressed as much as equity in a single paper?
- Remember: we aim to do science differently, not to publish. How can this process be part of doing science differently, rather than just adjudicating author order?

The Most Feminist Science Article in the World would include:
(a brain stormed list)

- A land acknowledgement
- A position statement
- A count of all the pretty things
- A map/graph legend with “no contamination” that wouldn’t be used
- A discussion of justice
- Hand drawn graphs/maps
- Local place names
- Multiple languages, including local indigenous languages
    - Multiple understandings (local narratives)
- Section in results of community peer review
- Coloured ink
- Elder review
- Translation between traditional knowledge & scientific knowledge → how they work/ do not work together
- What kind of day you’re having & effects on results
- “Baymen’s terms” & “Baywomen’s terms” abstract → like an abstract, but in less jargony terms
- Dictionary-ish reference at back of paper (like a textbook) for jargony terms
- A romance novel
- Zine or comics
- The social life of contaminants → follow the fish & log all its contaminants, not just the ingestion of plastics
- Sections on hands, brain, heart
- Write it as a discussion or play so that roles & care work is clear
- Explicitly address when we are using scientific categories/names/stuff instead of assuming that it’s normal (ie “cod” vs “Gadus morhua”)
- Always include a “how to”. No good ideas without ideas for implementation
- Say where our thinking has changed (our “mistakes” or changes & learning)
- Write more about fear
- Pay attention to legacies, debts, & origins of practices as politics because science borrows from others all the time (called stealing)
- ‘Personal communication’ should outweigh/out number references to dead white guys
- More references (relations) than text
- Increase our timeline of references to include people erased from history/the record
- Inclusion of gender dynamics (Gender pronouns)
- Be clear about what essential characteristic we care about (ex: in our cod study, we care more about the edible status of the fish rather than if it’s cod)
- Always include interdisciplinary discussions

- Study unimportant, ugly things instead of things useful to humans, keystone species & charismatic species
- What about health of fish rather than humans - decentering the human
- Curse as outcome
- White paper bios (including training)
- More grey literature - efforts to increase our ‘bodies of knowledge’
- Critique our ‘public engagement’ instead of patting on back - be specific, be better
- Do feminist evaluation, did the research do what you said you’d so. Unintended consequences (accountability!)
- Offer the britches
- Power, knowledge, money -global criticism
- Feminism is about ending/mitigating/surviving systems of patriarchy, colonialism, capitalism, so explicitly stating how we are attempting/doing this in methods, topic, results…
- Metrics that matter to the animal
- Use of direct quotes & stories
- Ethics of collection..ethics section in all sections - shooting & vomiting
- Integrational studies (lab + CS)
- Only their words/quotes - not our words
- In contribution section/methods mention our consensus process
- Articulate the value-laden choices (how we chose a research question)
- Having guidelines for what we can & can’t offer to communities
- Include a section on teaching
- How is this activism?
- Really <u>own</u> our feminist epistemology instead of __ it
- Looking sideways on conducting science - covariables, forward thinking, & different perspectives
- Acknowledge languages of lit review
- Don’t let any section start without talking about the body
- Quote Sarah Ahmed
- Let definitions be fluid rather than _/closed
- Killjoy fish?
- Use personal experiences/stories as data
- Citation: White men (300DC-20170
    - White dudes cite as one institution
- Have study [species/being] in authorship
    - Died for the cause
- Local narrative about condition/phenomenon/thing
- Reciprocity
- Check back at every step (people, communities...)

How to Run a Feminist Science Lab Meeting

While our protocols, technologies, and experimental designs are all feminist in that they foreground issues of equity and justice, the main place where people notice feminism-at-work when they join our lab is in how we run our weekly lab meetings. Here are some resources to running an anti-oppressive, horizontal, equity-based meeting:

Facilitation

Facilitation is a discussion method that aims to bring collective knowledge together. Rather than styles of discourse characteristic of teaching, leadership, or debate, all of which are more individualistic and based on a single main "knower," facilitation looks to "grease the wheels" of everyone else's knowledge. Facilitation addresses how different people in the room are more or less likely to speak, be heard, or be interrupted, and works to address those disparities. Facilitation is not intuitive. It's a skill, and it has to be trained.

Everyone can work towards facilitation, and there can be designated facilitators. We've done enough training that students now run lab meetings, meaning that the faculty "in charge" speak just as much, or less, than everyone else.

Here are two excellent resources on how to do facilitation:
Aorta Collective Anti-Oppresive Facilitation Guide (Creative Commons license)
USAID Facilitation Skills Training Manual

Round Robins/Talking Circle

One of the simple facilitation techniques we use every meeting is a round robin or talking circle. We go around to everyone at the table, in order, and they have a chance to speak or weigh in on the topic. Anyone can "pass" and choose not to speak, but it also means the junior researchers, introverts, women, people of colour, new recruits, and others that may not otherwise speak have a chance to share their insights. We often do one of these at the end of the meeting to see how everyone is doing/what their main take away was.

Consensus-Based Decision Making

Consensus-Based Decision Making (CBDM) is a process where everyone in a group agrees to move forward on a plan of action. This doesn't mean everyone agrees equally, but that everyone has agreed to move forward regardless of unevenness and differences of opinion. Because it is a method that aims to reach agreement despite difference, it should be carefully and intentionally facilitated.

There is a concrete, step by step process that can help a group research consensus outlined in Tim Hartnet's book, *Consensus-Oriented Decision-Making*. This is the model we use, though there are others. Here is our lab's "cheat sheet" of condensed steps in the consensus process.

Collaboration

Our lab operates as a collective and leverages our shared intelligences, rather than acting as a bunch of individual geniuses. However, collaboration is a skill. We work to develop this skill in a few ways.

The first step is to set ground rules, guidelines, or a code of conduct about how we want to work together. What do we think about lates and absences? How do we want to communicate with one another? How do we make decisions? How do we resolve conflicts? I teach some of this in my classes. Here is a sheet for structuring these ground rules in a rather formal way via a collaboration contract. You can also do it less formally. We've done it by having facilitated conversations about what we value, and then drawn practices & protocols from those values. This took several meetings, but we are damn good at collaboration. One of our most basic ground rules is: "If you are sick, heartbroken, or exhausted, go home. Work is not more important than your health."

Another part of our training is to recognize that people have different preferences for how they maintain their energy, think, make decisions, and communicate. To address and support these differences, we use the *Myers-Briggs Type Indicator (MBTI)* (a free version is here) to help us articulate how we think and work, and then use the "*Types and Teams* framework" to create specific meeting styles and work styles that work for all participants.

A full training manual to *Types and Teams* is here:
https://www.opp.com/en/shop/mbti-step-i/introduction-to-type-and-teams-10-pack/c-24/c-74/p-209

Problem-free? No.

These processes don't mean that we don't have problems. We have them all the time. But we work through our issues equitably, supportively, and consistently. As a result, lab members are collectively able to take on riskier work, act autonomously, stretch their limits and skills, and have fun doing it. CLEAR currently has 18 lab members and 2 faculty members working on close to 20 unique projects, an impossible set up unless everyone is supporting one another consistently, selflessly, and in good humour.

Instructions for Field Collection of Guts

Materials:

- Whirl packs- large and small
- Data sheet-master record sheet
- Information sheet for volunteers (cut small)
- Paper tags to go in with fish
- Sheet for emails (public meeting)
- Binder/clipboard
- Pens/pencils
- Sharpie
- Cooler
- Extra trash/plastic bags
- Aprons or lab coats (optional)
- Fish knife/pocket knife
- Tape measure
- Sample jar of ingested plastic (outreach)
- Box to put it all in ("We want your guts" on outside of box)
- Camera (optional)
- Water/hat/sunscreen/lunch

Roles

Ideally, have three people: one person bagging and tagging, one person recording data on the clipboard, and one person talking to people. If you have two people, one person handles samples and data, and one person talks to people. *Always have one person facing the public, scanning for people to talk to*.

1. **PUBLIC ENGAGEMENT & SOLICITATION**:
    a. **Prepare**: Wear clothing that identifies you as from MUN or CLEAR, or otherwise not-DFO. Although wearing lab coats provide full coverage from gut mucus stains, it may give off the wrong impression (elitist!!) especially if worn during fieldwork in small towns. Wear aprons instead. White lab coats raise suspicion and appear authoritative whereas aprons provide a more grounded appearance.
    b. **The approach:** Approach people when they are on wharfs gutting their fish. What do you say? "We're collecting fish guts! Do you have any?" is a good call out. Follow up: "We're researchers from MUN collecting fish guts to see if fish have been eating plastic in the area. We're trying to see if plastic pollution is a problem here." Identify yourself as not-DFO at some point for sure. You can tell we've found some of the world's lowest ingestion rates in cod caught in

Newfoundland and we're following up. Put a sample of plastics ingested by cod from previous studies in your kit to show people.

i. If someone else is also there and wants to guts for something else, back off and give them the guts. = humility. We can get them elsewhere. We have lots of resources to do so.

c. **Reporting back/reciprocity**: As people to name their fish if they'd like to know if their fish had plastics in it. Give them the website url (flier). If people have troubles coming up with a name, ask what their pet's name is or was. We shouldn't be collecting people's real names. Allowing people to see whether their fish had plastic in it is part of environmental justice; this is not optional unless the person opts out.

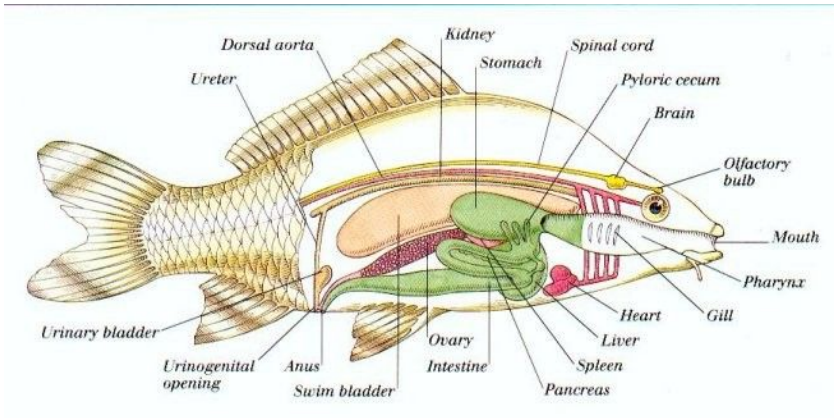
2. **GETTING GUTS** (bagging & tagging):

a. Make sure the bag has a number on it *before* you put guts in and get it wet. Also mark a piece of paper with sharpie and drop it in the bag. Naming includes location and year: Petty Harbour team, add PH16 to your number. St. Phillip's team, add SP16 to your number (ex. SP16-1 for the first fish, PH16-34 for the thirty-fourth fish). Etc.

i. Data on a sheet IN THE BAG: Using a sharpie, record the bag number, date, location that the fish was caught, type of fish (likely cod), size of fish, and the name to call the fish on the sheet, then put the sheet in the bag. If you get guts without the fish and don't have the length, so be it.

ii. Also mark this data on the data sheet.

b. If you are gutting a fish yourself, get everything from the mouth to the anus (all green in the image below). If the intestines are cut open, do not collect that fish, unless you are collecting from other people. Throwing away guts you just asked for is rude. Bag it and mark it clearly as nicked.



a.

4. SHARING DATA (talking):

a. Give the flier to anyone who volunteers guts. They need this to see if their fish has eaten plastics. Give the flier to anyone who wants it, even if they didn't give guts.
b. If anyone is particularly interested in the public meeting, record their email or phone number on the sheet in the binder, then make sure to give to France so she can contact them/we can add them to our list.
c. If there is someone interested in the process, always talk to them. Always, even if it means you miss some fish. Even if they are not fun to talk to.

Marine Plastics in Fish

Marine Plastics in Fish

A citizen science dissection & analysis protocol

This guide will walk you through how to remove the guts from a fish and analyse them for plastics.

Materials

- Spotter's guide (@ end of this guide)
- Dissecting diagram (@ end of this guide)
- Full sized baking sheet or flat surface that can catch fluids
- Fine mesh strainer
- 1 large or 4 small Coffee/Paper Filters
- Sharp scissors
- Fish knife (or other sharp knife)
- Tweezers
- Water Bottle
- Rubber gloves (optional)
- Garbage bag

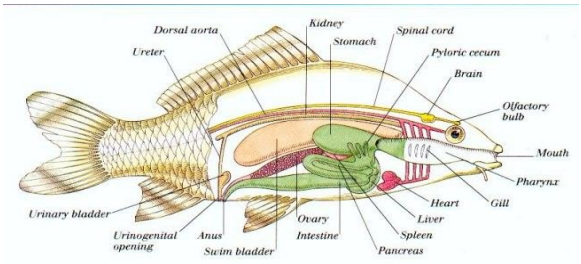
These are items that you can find at home or at local hardware or general stores. Items can be substituted for others that get the job done.
Children should work with an adult on this project because of the use of knives and other sharp tools.

Setting Up Your Workstation

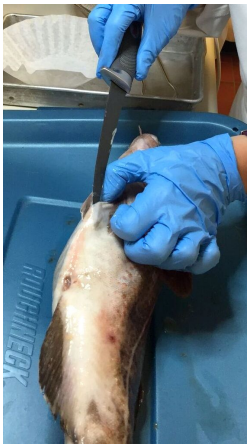
1. Dissecting station should be on a clean and even surface. Prepare the station by placing the cookie sheet on the table or counter.

2. Place the fish on the cookie sheet or other flat surface (we are using the lid of a rubber maid here).

3. Take out the guts: Following the dissecting diagram, use the scissors to remove the entire GI tract (outlined in green).

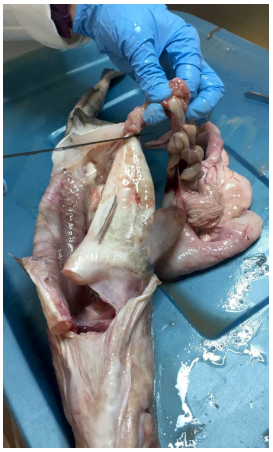


4. a) Cut a straight line of the belly of the fish from the mouth to the anus.



b) Pinch the end of the intestines by the anus with your fingers so contents are not lost. With your other hand, pull up--the entire GI tract should come up. Still pinching the end of the intestines, cut the guts at each end (mouth and anus).

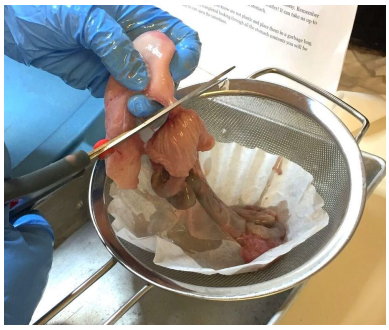
c) Gently place the guts in the paper-lined colander. Try to get it all in one piece with no nicks!



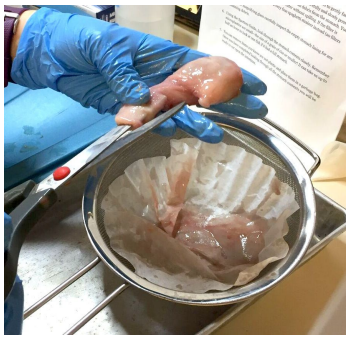
Processing the Stomach

4. Place the coffee filter(s) in the strainer, and then guts in the coffee filter.

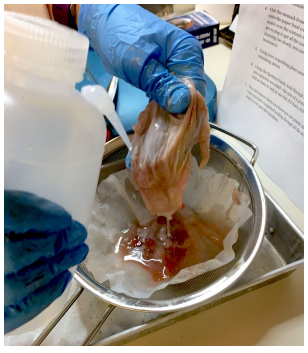
5. Using scissors, cut the intestines from the stomach, being sure to keep anything that spills out into the coffee filter. Move intestines to one side of colander; you will dissect them after the stomach.



6. Cut the stomach from one end to the other allowing the contents to gently fall onto the paper lined colander.



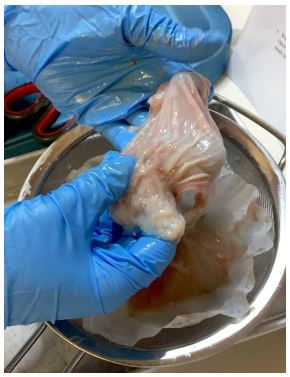
7. Using the water bottle carefully and slowly pour water over the contents to separate and remove all debris from the stomach. You are trying to get all the contents into the filter without spilling. If the filter is draining too slowly, you can use a very fine mesh strainer instead (remove the filter-- the filter just makes it easier to see things).



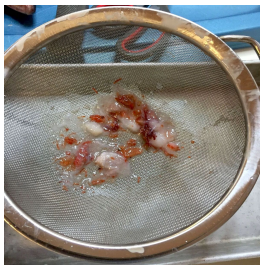
8. Whenever you see a plastic, pick it out and put it aside (in a dish is best, since they can get lost!).

Looking for plastics

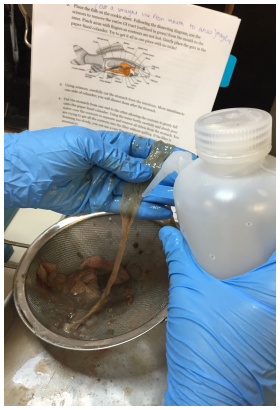
9. Inspect the empty stomach lining for any remaining debris.



10. Put aside the stomach, and look through the stomach contents closely for plastics. Use the Spotters Guide. Most plastics are smaller than a grain of rice, so take your time! I usually takes us half an hour to It can take us up to half an hour to look at one fish if it had a full stomach & intestine.
You can remove items you know are not plastic and place them in a garbage bag.



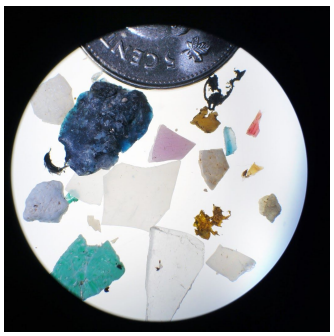
11. Once you have completed looking through all the stomach contents you will be ready to cut open the intestines and repeat cutting, rinsing, and looking. Repeat steps 5-10. The intestines are long and thin, so cutting and rinsing them can take some care & time.

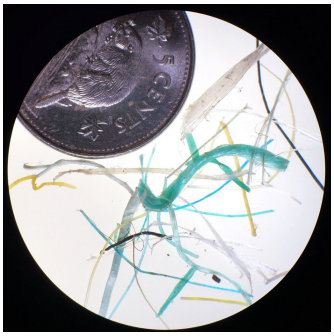


Is it a plastic?

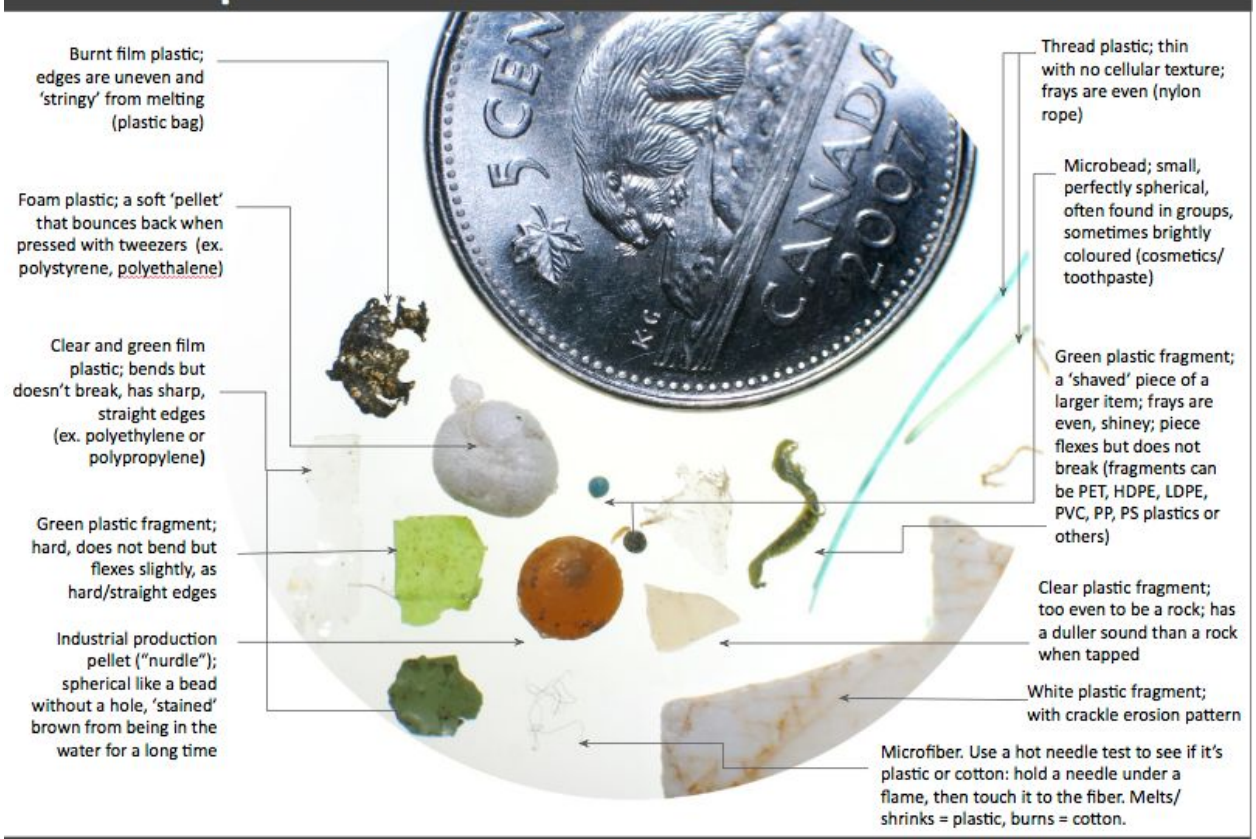
Plastics can be hard to identify because they are so small, and because they can look like other things fish usually eat. Use the Spotter's Guide to help identify plastics from common non-plastics. Other tricks include:

- putting hard-to-identify items into a coffee filter for 48 hours to dry. If it shrinks or twists, it may have been organic and changed form when it lost water.
- Tap it. Does it sound like plastic, or a rock (or something else)?
- If you hold a flame to it, does it melt or flare like plastic, or does it burn like organics (this destroys the sample)
- Use a magnifying glass or microscope to see the underlying structures





Spotter's Guide to Plastic Pollution

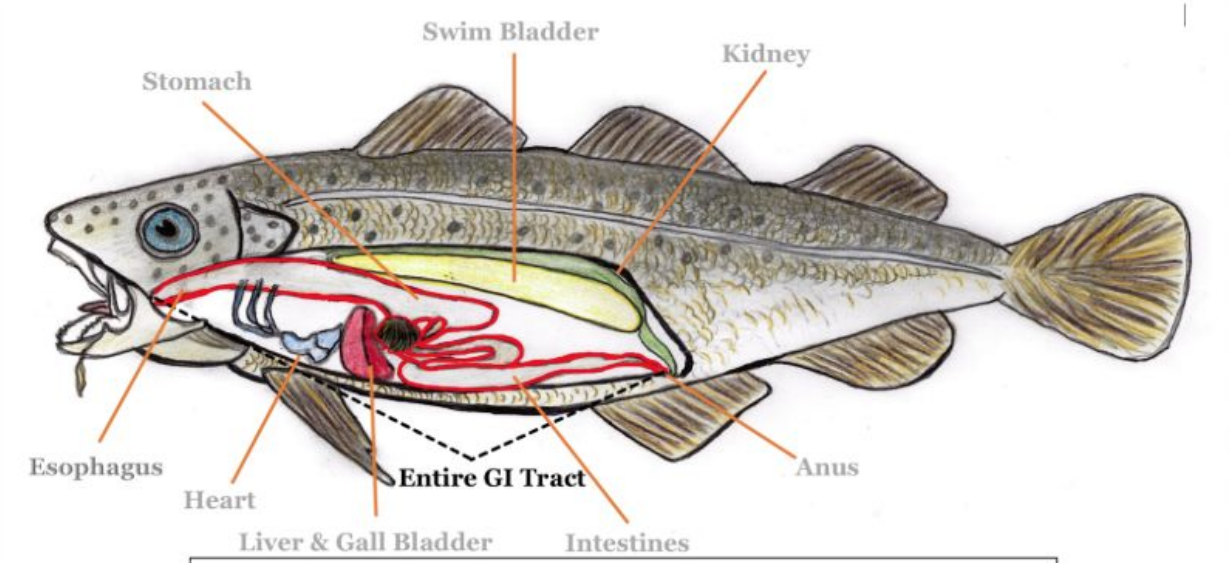


Developed by Civic Laboratory for Environmental Action Research (CLEAR) https://civiclaboratory.nl

Spotter's Guide to Plastic Pollution: non-plastics

Developed by Civic Laboratory for Environmental Action Research (CLEAR) https://civiclaboratory.nl

Dissection Diagram



When removing the **Entire GI Tract** cut from the **Esophagus** to the **Anus**
Discard the Heart, Liver, Gall Bladder, Swim Bladder and Kidney

Image by Created by Natalie Richárd M.Sc. Student Department of Geography and CLEAR

Methods: Plastic Ingestion Lab Processing Protocol

This protocol is for visual processing cod and other fish guts (stomach and intestines) to look for plastics >1mm without the aid of chemical analysis. Anything smaller is not reliably identifiable visually (Song et al 2015).

Equipment:

- 5mm mesh stainless steel sieve
- 1mm (#18) mesh stainless steel sieve
- Watchmakers forceps, scalpel and fine scissors
- Medium non-plastic jars (~125 ml) for drying of microplastics on filter papers (~**30**). *The number used will depend on how many samples are processed in a 5 day window- they are what hold the filter papers while the plastics dry, then they can be reused.* These can be mason jars, mugs, or beakers.
- Small glass scintillation jars (~50 ml) for storage of microplastics (>1mm) (**1 per plastic**)
- Sink
- Wash bottle
- Dissecting microscope
- Fine- tip permanent marker
- Filters (coffee filters will do) (**1-2 per fish with plastic**)
- 2 petri dishes (re-used throughout)
- gloves, lab coat, hair ties

Contaminants (all stages)

- Create a control sample for air deposition of microfibers, which are present in indoor air & dust. Put double sided tape in the bottom of a Petri dish, and leave the dish open for the duration of the study (or week by week if the study is ongoing). Label the dish with the dates inclusive. If anyone enters the lab wearing petrochemical fibers (fleece, nylon, poly blends), take a “pinch” of their lint for the control sample.
- Do not wear petrochemical fibers in the lab (fleece, nylon, poly blends). Wear cotton, wool, etc.
- Wear a cotton lab coat and tie hair back.
- Immediately before opening each gut (comprised of both the stomach & the intestines) rinse your hands, backwash the sieve, and wipe down the tools you will use to open the gut.
- After completing a gut, check your gloves and tools for plastic “hitchhikers.” Always rinse tools, hands, bags, etc INTO the sieve so any lingering plastics end up in the sample.

Processing the stomachs to remove plastics (Stage 1):

The objective is to visually identify and remove >1mm plastics, by separating them from other gut contents (food). It is important to examine stomach for embedded plastics.

Processing the stomach:

1. Do not wear earbuds to listen to music while processing, as this separates you from the fish, which deserves your full attention and respect. You can play music from a speaker, and singing is particularly welcome.
2. Fill in the spreadsheet with the fish code (ex. PH13), today's date, the location the fish was caught, size, sex, and data share name of the fish if it is not already filled in. This will require you to look at the field sheets.
3. Before opening each gut, wash your hands, backwash the sieve, and wipe down the tools, microscope lens & plate, and Petri dishes you will use.
4. Stack the 5mm sieve on top of the 1mm sieve and place both in the sink. The 5mm sieve will catch the larger items and make visual inspection of the 1mm sieve easier.
5. Place the thawed guts in the top of the 5mm sieve. Open the stomach and intestines by cutting it along their length, over a 5.0 mm sieve.
6. Rinse stomach and intestine contents thoroughly with cold water from a wash bottle (or a tap hose on low, if available) to remove mucus and food. Mark on the spreadsheet if there is food present in the gut. Make sure the water doesn't splash, as we can lose plastics that way.
7. Immediately remove any plastics you see and put them in the Petri dish. You will likely have to use tweezers.
8. Once rinsed of any food items or plastic, examine the gut tissue for embedded plastics with your eyes, and under a dissecting microscope if you see any suspicious areas. Then put the guts aside until the end of the process.
    a. Record the number of any microplastics embedded in the wall of the gut (0 if there are none). Embedded = the plastic is stuck in the tissue and requires a light tug to remove it.
8. Using the water bottle or tap, rinse the food in the 5mm sieve to look for plastics. Continue to transfer any plastics to a Petri dish immediately with tweezers (rinse them if they have guts on them). If you are unsure if something is plastic, put it in the Petri dish.
    a. Use a back and forth "grid" approach to cover each part of the sieve visually. Running water, poking with tweezers, and shifting the sieve

will help you see any plastics, since they move differently than other gut contents.

b. Mucus is the most likely element to cause researchers to miss plastics. Inspect any mucus carefully.
c. If there are ingested animals that are intact enough to dissect and look for plastics inside of, do that, and provide a new ID number. If the larger fish was PH13, the smaller food fish would be PH13b. Put this fish aside so its gut contents are not confused with its predator. Mark this new sample on the data sheet and process it separately (start from step 1).
d. Record any non-food items that are not plastic in the stomach, such as rocks, tar balls, etc in the spreadsheet. Record whether there is food or not.

9. Place any non-plastic items (food) back into the whirl bag for disposal.
10. Once the 5mm sieve is empty, repeat step 8 with the 1mm sieve. Once all plastics are removed from the sieves and gut tissue, discard any guts and gut contents that are not plastic into the original whirl pack. Inspect both sieves for rouge plastics sticking to the sides. Clean the sieves. Turn them upside down in the sink so you know they have been cleaned.
11. Move on to the next stage (visual sorting) with any plastics in the Petri dish.
12. Close the whirl pack, which should have the empty gut and non-plastic gut contents, and put it in a larger garbage bag (located under the sink).
13. At the end of your lab shift, make sure you re-freeze all fish guts you've already checked. Make sure they are marked as already dissected. In the spring, we take these back out to the water to dispose of so that we interupt life cycles as little as possible and feed other animals. **Do not throw the guts out in the trash.**

Visual Sorting and Separation of Microplastics (>1mm):

1. Examine the Petri dish systematically with your eyes and then under the dissecting microscope. Careful visual sorting is necessary to separate the plastics from other materials, such as organic debris (shell fragments, animals parts, bones, dried algae, or seagrasses, etc.) and other items (metal paint coatings, tar, glass, etc.). Pieces of microplastics >1 mm can be visually distinguished according to the following criteria: no cellular or organic structures are visible, fibers should be equally thick throughout their entire length, particles often present clear and homogeneous colors. They may also be examined under high magnification.

2. Probable Plastics: If you come across something that may be plastic but will require closer inspection, label an additional filter paper ('DPH13, Oct 11/15, unknown') and collect it. Record the number of these 'Probable plastics in the spreadsheet'. These will be examined further under a compound microscope during the 'Quantification' stage of processing. Fold the filter paper and place it upright in jar so the pieces can dry.
3. Plastics: Collect all plastics from the Petri dish onto the pre-labeled filter paper ('DPH13, Oct 11/15. Plastic'). Record '0' if no plastics are found or '1+' if plastics are found. I recommend not counting microplastics as you go. Saving this until later will save you time in the long run, and you are less likely to lose your place while examining the sample. Plus, if there are quite a few, you may lose count and need to recount.
4. If plastics are found, label a coffee filter paper with the fish ID code (ex. PH13) and today's date (E.g., CAAU#, Feb 11/15). Put all the plastics from the sample in the filter paper and fold it closed, then place the entire thing on a jar on the shelf to dry. Do not put the lid on, as these samples will need to dry for up to 5 days before being sealed.

Quantification of Plastics (Stage 2):

The goal will be to individually assess (and fill out a datasheet), for each piece of plastic. At the end, for each piece of plastic we will have recorded the mass, size, category (E.g., industrial, fragment, thread), colour, and erosion. If the samples will be sent to C-CART for polymer analysis, follow the preparation protocol for that (C-CART protocol sheet).

Equipment:

- Compound microscope (to closely examine probable plastics)
- Digital calipers
- Datasheets
- Scale

Visual sorting of items

1. The first step involves careful visual sorting of items to separate the plastics from other materials, such as organic debris (shell fragments, animals parts, dried algae, or seagrasses, etc.) and other items that can be considered non-plastic anthropogenic debris, or rubbish (metal paint coatings, tar, glass, etc.).

2. This is done by direct examination of the sample by the naked eye or with the aid of a dissecting microscope. To some extent, plastics can be visually distinguished according to the following criteria: no cellular or organic structures are visible, fibers should be equally thick throughout their entire length, particles must present clear and homogeneous colors, and if they are transparent or white, they must be examined under high magnification. Sometimes tapping, pinching, and poking the sample with tweezers will tell you about the material.

Incidence and abundance for each fish/animal

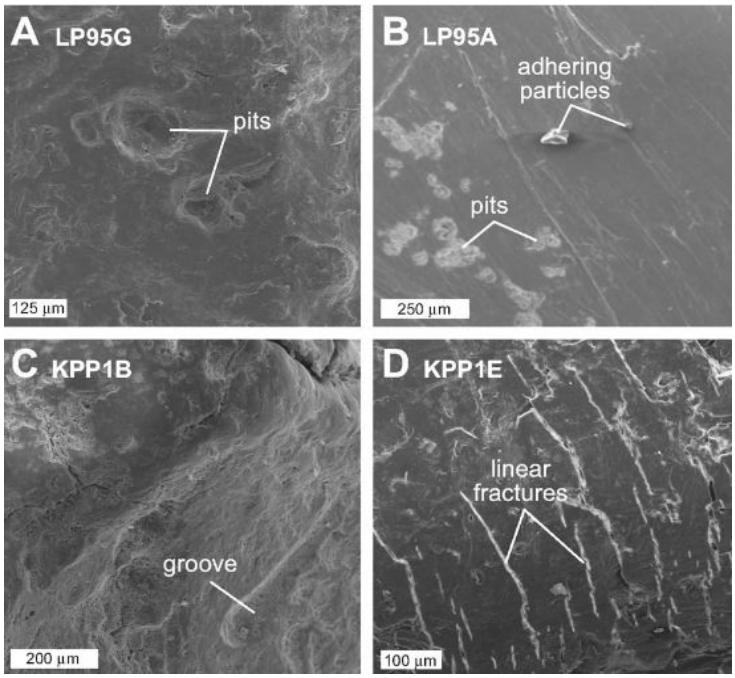
1. For the main categories **plastic** and **rubbish** we record for each individual:
    a. Incidence (Presence or absence) and
    b. Abundance by number (count of Number of items)
    c. Abundance by mass (Weight in grams) using Sartorius electronic weighing scale after a period of air drying at laboratory temperatures. Weights are recorded in grams accurate to the 4th decimal (= tenth of milligram).

Individual assessment of plastics (size, type, colour, shape, erosion)

1. Treat each piece of plastic separately. When filling out each datasheet, taking care to associate the datasheet with the correct Fish ID number, and to number the items being examined. (see sample spreadsheet in dropbox)
    a. Using digital calipers, measure the length (longest dimension), height (second longest dimension), width (shortest dimension) of the plastic. If it is spheroid, measure the diameter.
    b. Categorize the type of plastic:
        i. **Industrial** plastic pellets or “nurdles”. These are small, often cylindrically-shaped granules of ± 4 mm diameter, but also disc and rectangular shapes occur. Various names are used, such as pellets, beads or granules. They can be considered as “raw” plastic or a half-product in the form of which, plastics are usually first produced (mostly from mineral oil). The raw industrial plastics are then usually transported to manufacturers that melt the granules and mix them with a variety of additives (fillers, stabilizers, colorants, anti-oxidants, softeners, biocides, etc.) that depend on the user product to be made. For the time being, included in this category is a relatively small number of very small, usually transparent spherical granules, also considered to be a raw industrial product.

ii. Sheet-like user plastics (**film**), as in plastic bags, foils etc., usually broken up in smaller pieces;
iii. Thread-like user plastics (**thread**) as in (remains of) ropes, nets, nylon line, packaging straps etc. Sometimes ‘balls’ of threads and fibres form in the gizzard;
iv. Foamed user plastics (**foam**), as in foamed polystyrene cups, packaging or aquaculture, or foamed polyurethane in matrasses or construction foams;
v. Small pieces (**fragment**) of more or less hard plastic items as used in a huge number of applications (bottles, boxes, toys, tools, equipment housing, toothbrushes, lighters etc);
vi. Small round beads (**microbeads**), often brightly coloured or white, from personal care products.
vii. Thin fibers (**microfibers**) from clothing. These are thinner and less rigid than threads.
viii. **Other**, items that are ‘plastic-like’ or do not fit into a clear category.

c. **Opacity**: transparent or opaque! It is Opaque if the bottom light from the microscope does not go through it.
d. **Colour:** white, clear, red, orange, blue, black, gray, brown, green, pink, tan, yellow
e. **Erosion**: unweathered, weathered, grooves, irregular surface, jagged fragments, linear fractures, subparallel ridges, burnt/melted, discolouration. See image below (from Corcoran 2009).



Erosin	Description
fresh	
unweathered	
weathered	
grooves	
Irregular surface	
jagged frag	
linear fractures	
subparallel ridge	
burnt/melted*	

References

Corcoran, P. L., Biesinger, M. C., & Grifi, M. (2009). Plastics and beaches: A degrading relationship. *Marine Pollution Bulletin*, *58*(1), 80-84. doi:10.1016/j.marpolbul.2008.08.022

Song, Y. K., Hong, S. H., Jang, M., Han, G. M., Rani, M., Lee, J., Shim, W. J. Comparison of microscopic and spectroscopic identification methods for analysis of microplastics in environmental samples. *Marine Pollution Bulletin* 93: 202-209. doi:10.1016/j.marpolbul.2015.01.015

Protocol Using Premade 10% KOH to Dissolve Fish Guts

Note that we no longer use this protocol as it creates toxic substances. Stored here for reference.

Materials

4L-8L 10% KOH (Instructions & protocol follow)
Incubator Oven
Glass Jars and lids appropriately size for fish guts (guts should only take up ¼ of jar)
250ml Glass Beaker
Plastic Pan (to use as splash guard)
Hood
Protective Gloves
Protective Eyewear
Sink filled with Soapy Water
Paper Towels

Protocol

1. Before beginning protocol make sure you are wearing protective eyewear and gloves. KOH will cause severe throat and eye irritation. Never use without the use of a hood.
2. It is easier if you place the fish guts into the glass jars in a wet lab prior to starting the following steps. This will prevent any fish smell or material entering the chemical or biological lab.
3. Set incubator temperature to 50 Degrees Celsius.
4. Turn on hood
5. Place premade 10% KOH, 250ml beaker, glass jars filled with guts, and plastic pan under hood.
6. Place the jars in the plastic pan.
7. Using the 250ml beaker slowly fill about ¾ full with premade KOH. Do this over the plastic pan to catch any KOH spillage (clean plastic pan in soapy water when finished).
8. Begin to fill the jars with KOH. It is 1 part fish guts and three parts 10% KOH.
9. Immediately seal jar using the lid, wipe any excess KOH off glass jar and place in preheated incubator. Place paper towel in the sink and soak in soapy water. Dispose of in proper container.
10. Repeat steps 7-9 until you have completed all fish guts.
11. The amount of KOH used greatly depends on the size of fish guts. 4L of KOH will complete an estimated 30, medium to small sized fish guts.

12. The fish guts will take up to two weeks in the incubator to fully dissolve all material other than plastic and lipids.

Creating 10% Potassium Hydroxide (KOH) Protocol

Note that we no longer use this protocol as it creates toxic substances. Stored here for reference.

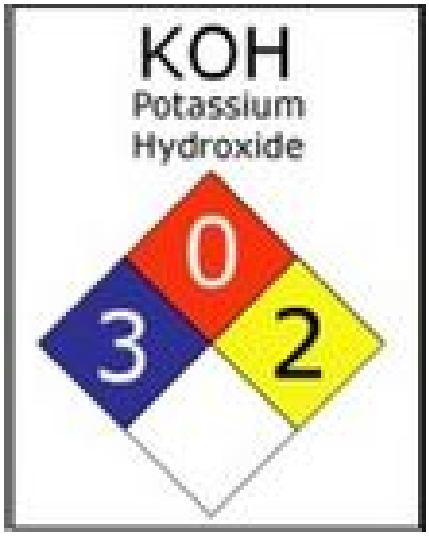
Materials

1000ml Beaker
Purified Water (Nano)
4 Liter Glass Container Properly Labeled with NFPA Label
400g KOH Pellets
Hood (very important not to conduct without the using, fumes will burn eyes and throat)
Protective Gloves
Protective Eyewear
Scale
Scoopula
Measuring plate for KOH

Protocol

1. Before you begin make sure you are wearing gloves and protective eyewear.
2. Using the 1000ml beaker fill the 4L glass container with purified water.
3. Tare the scale with the plate you plan to use to measure out the KOH pellets.
4. To be more accurate measure out 200g of KOH pellets twice to total 400g. You should have to plates of KOH 200g each.
5. One you have measured out 400g of pellets it is important to act quickly but safely. The pellets will begin to soften and melt.
6. Bring your KOH pellets and purified water filled glass container to the hood and turn on.
7. Using the scoopula begin to place the pellets into your 4L glass container. Do this as quickly as possible but try not to spill over. If you do quickly pick up pellets and place into glass jar.
8. Once you have place 400g of KOH pellets into the 4L glass container, gently swirl. You will notice the reaction will cause the glass container to become hot.
9. Your KOH will be ready to use once the reaction is completed. The glass container will cool.
10. Keep stored in the hood at all times.
11. When using KOH always use a hood. The reaction can cause severe throat and eye irritation.
12. If you have any spillage while using KOH clean up with paper towels and soak in water. Dispose paper towels in a proper container.
13. IMPORTANT Never use Prepared KOH with metal for any extended time. It will cause a reaction and begin to melt. Use only plastic or glass.

Proper NFPA Label for KOH



POTASSIUM HYDROXIDE

(CAUSTIC POTASH)

UN1813 **DANGER! POISON!** CAS#: 1310-58-3

TARGET ORGANS: EYES, RESPIRATORY SYSTEM, SKIN

PRECAUTIONARY MEASURE: CAUSES SEVERE BURNS. MAY BE FATAL IF SWALLOWED. Do not breathe dust, mists, or vapors. Use with adequate ventilation. Keep in a tightly closed container in a cool area. Loosen closure carefully. Wash thoroughly after handling. Use appropriate personal protective equipment when handling.

SYMPTOMS OF EXPOSURE: Irritation of the eyes, nose, throat, and respiratory system. Burning of the eyes and skin. Pulmonary edema.

INSTRUCTIONS FOR IMMEDIATE AID IN CASE OF EXPOSURE: Call a Physician. In case of contact, immediately flush affected area with running water for at least 15 minutes while removing contaminated clothing and shoes. If swallowed DO NOT induce vomiting. Medical intervention: Treat symptoms as appropriate. Delayed pulmonary edema may follow acute inhalation incident.

SPILL RESPONSE PROCEDURES: Only appropriately trained personnel should respond to spills. Use proper response procedures and maintain appropriate level of fire protection. Dispose of as required by federal, state, and local regulations.

CONTAINER HANDLING and STORAGE: Keep in a tightly closed container in cool area. Do not store with acids, flammables, or any other incompatible materials. Concentrated solutions may react with water causing spattering and generating heat.

MSDS REFERENCE: Refer to Material Safety Data Sheet for more information about this chemical.

MANUFACTURER: **MSDS LOCATION:**

PHONE NUMBER:

HC305 NMC

Methods:

Seabird Plastic Ingestion (V9)

May 6, 2016

Protocol by Stephanie Avery-Gomm, based on Franeker et al. 2011.

Improved by comments from Max Liboiron, Michelle Valliant, Jocelyn Wood, Florence Poon

Objective: Process gut contents for plastics >1mm and <1mm. If there is no interest in examining stomach contents for plastics <1mm then the instructions in green text may be ignored.

Stage 1: Stomach Processing

Contaminants

Minimizing cross-contamination: Samples can also become contaminated by particles present in the air and dust of the laboratory, on the clothes of workers, in poorly cleaned instruments, or by improperly sealed samples. Control samples should always be used to confirm that there is no procedural sample contamination. The objective is to be able to say: “Our contamination prevention and controls suggest that the fibers found in our study are not a result of contamination during processing.” It will also enable us to characterize plastic contamination in the laboratory.

The following steps must be taken to reduce and account for contamination.

- Wear clothing that would have less of a chance of fibers to come off (i.e. Wear cotton instead of fleece)
- Reduce air circulation by closing windows and blocking fans, or working in a fume hood.
- Wear a lab coat and tie hair back.
- Immediately before opening each stomach (comprised of both the gizzard & proventriculus)wash your hands, the funnel, backwash the sieve, and wipe down the tools you will use to open the stomach.
- Use the recommended set-up (described below) to minimize the surface area open to the air
- Work efficiently and seal the jar containing the solution that passes through the sieve as soon as possible.

- Collect a control before and after each period of stomach processing. These should be recorded alongside the stomach data as 'Control#'.
- Use a set up where samples transfer from the sieve and funnel directly to sample jar, rather than an intermediary space and thus another chance for air and dust contamination.

How to collect a Control for microplastics: After processing a few stomach samples, you will get a feel for how long it takes to wash out a stomach, rinse the funnel and seal the jar. For a control, you must follow all the same steps as you would if you were processing a stomach, for the same approximate length of time. It may help to use a stopwatch when processing your stomachs, and spend the same amount of time collecting a control sample. Theoretically, if there was no risk of airborne microplastics contaminating the samples, then a control sample would be a jar of water, without microplastics.

Processing the stomachs to remove plastics:

Objective here is to focus on removing the >1mm plastics, but retaining stomach contents <1mm for later analysis. It is important to examine stomach for embedded plastics and control for contamination.

Equipment:

- 1mm (#18) mesh stainless steel sieve.
- Watchmakers forceps, scalpel and fine scissors.
- Glass funnel (larger diameter than sieve)
- Large glass jars (~250 ml) for remainder (food, water, microplastics <1mm) (1 per stomach)
- Medium glass jars (~125 ml) for drying of microplastics on filter papers (~30). *The number used will depend on how many samples are processed in a 5 day window- they are what hold the filter papers while the plastics dry, then they can be reused.*
- Small glass jars (~50 ml) for storage of microplastics (>1mm) (1 per stomach)
- Jig to suspend sieve over funnel over jar (see Figure 1)
- Sink
- Wash bottle
- Dissecting microscope
- Fine- tip permanent marker
- Filters (coffee filters will do) (**1-2 per stomach**)

- 2 petri dishes (re-used throughout)
    - Embedded plastics: For gizzard and proventriculus to examine for embedded plastics, and ulcers.
    - Microplastics: Also be sure to examine sieve to make sure you've got the full sample! Drawing parallel lines 1 cm apart on the bottom of the petri dish with fine-tip permanent marker may aid in keeping track while examining the sample through the dissecting microscope.

Processing the stomach:

1. First, label a filter paper, the small jar and the large jar with the species code, the bird ID number and the date (E.g., CAAU2015-1, Feb 11/15). Fill in the spreadsheet with the same information.
2. Before opening each stomach, wash your hands, the funnel, backwash the sieve, and wipe down the tools, microscope lens & plate, and petri dishes you will use.
3. Open the stomach by cutting the proventriculus and gizzard along their length, over a 1.0 mm sieve. The 1.0 mm sieve should be positioned securely over a larger non-plastic funnel and a ~250 ml glass jar, so that all items (including plastics) that passes through the 1.0 mm mesh can be retained for later analysis (**Figure 1**).
4. Stomach contents are carefully and thoroughly rinsed with cold water from a wash bottle to remove mucus from the periventricular walls and digested soft food components. Once rinsed of any food items or plastic, the proventriculus and gizzard tissues are placed in a petri dish to be examined for embedded plastics under a microscope. If sticky substances hamper further processing of the stomach contents, warm or hot running water can be used to clean them. Use water sparingly, but be thorough. This water can all be retained for later analysis of microplastics <1mm.
5. Using a small amount of water, lift the sieve and rinse the funnel to ensure that everything passes into the labeled jar below (we will call this the 'remainder'). Seal the jar immediately for later analysis of microplastics less than <1mm. These jars should be kept in a dark and temperature-controlled environment (stable room temperature) to reduce degradation during storage (i.e., cupboard or box). Ethanol not needed to preserve plastics unless diet is of interest. In that case ethanol would be used and prey items would also be pulled out of the >1mm sample. In this case, we may eventually digest the reminder to look only for plastics, therefore no preserving chemicals are necessary. *If quantifying plastics <1mm is not an objective of this study, the remainder does not need to be retained. The sieve can be washed over a sink rather than a jar, and controls for cross-contamination are not needed.*
6. Replace the sieve, carefully tweeze and then backwash the remaining elements of the stomach contents from the sieve onto a petri dish for sorting under a dissecting

microscope.

7. Over the sink (not the funnel), wash the sieve so it is ready for the next bird and set upside down to dry.

Look for Embedded plastic:

1. Examine both the proventriculus and gizzard first.
2. Record the number of any microplastics embedded in the wall of the proventriculus or gizzard (0 if there are none), and collect it on it's own, additional, filter paper ('Bird ID, Feb 11/15, embedded). If embedded plastics are found, it will be valuable to be able to document what kinds of plastics they are (classification: industry/user, frag, thread, foam etc). This is the rational behind keeping them folded in a separate filter paper.

3. Organ health is recorded in a simple scoring system ranging from zero for extremely poor health to score 3 for good health. Descriptions here cannot be exhaustive. Please use the comment fields to describe situations not properly covered in the table below. Note: Decay of corpses will complicate judgment of organ health: please attempt to give a judgment as if the bird was 'fresh'. Health scores for different organs may assist in defining the cause of death or the duration of the dying process. They are not meant to describe the state of decay of the corpse.

Code	Description
0	severe or multiple wounds, ulcerations or cancers in or around stomach walls
1	affected with wounds, ulcerations or cancers in or around stomach walls
2	slight or minor affected stomach wall
3	unaffected, healthy looking stomach wall

4. Lesions, lacerations or ulcers may be present. A lesion is anything abnormal, while an ulcer can be recognized as an area of complete loss of the mucus layer on the interior of the gizzard or proventriculus. Record the number of ulcers seen (0 if none), but wary of

misidentifying cuts (even circle shaped cuts) due to DNA sampling. Take a picture of ulcers if possible (ensure the photograph can be linked to the bird ID). See **Figure 2 & Figure 3** for example.

5. Retain stomachs with lesions or ulcers for histological analysis. If histological analysis is not part of this study, the stomachs can be disposed of. Note that recent findings indicate that visual identification of lesions/ulcers are often incorrect and therefore the presence or absence of ulcers cannot be verified without histology..

Visual Sorting and Separation of Microplastics (>1mm):

1. Examine the petri dish systematically. Careful visual sorting is necessary to separate the plastics from other materials, such as organic debris (shell fragments, animals parts, dried algae, or seagrasses, etc.) and other items (metal paint coatings, tar, glass, etc.). Pieces of microplastics >1 mm can be visually distinguished according to the following criteria: no cellular or organic structures are visible, fibers should be equally thick throughout their entire length, particles must present clear and homogeneous colors, and if they are transparent or white, they must be examined under high magnification and a fluorescence microscope.
2. Probable Plastics: If you come across something that may be plastic but will require closer inspection, label an additional filter paper (‘Bird ID, Feb 11/15, unknown) and collect it. Record the number of these ‘Probable plastics in the spreadsheet’. These will be examined under a compound microscope during the ‘Quantification’ stage of processing.
3. Plastics: Collect all plastics from the petri dish onto the pre-labeled filter paper (‘Bird ID, Feb 11/15, plastic). Record ’0’ if no plastics are found or ‘1+’ if plastics are found. I recommend not counting microplastics as you go. Saving this until later will save you time in the long run, and you are less likely to lose your place while examining the sample. Plus, if there are quite a few, you may lose count and need to recount.
4. Sand & Food: Sand may indicate ingestion near the beach, and food (i.e., zooplankton) may be important later. For each, record Y or N on the spreadsheet if you see evidence of these in *either* the >1mm sieve fraction or the ‘remainder’.
5. Once all plastics have been collected from the petri dish, wash everything that is left into the ‘remainder’ solution of plastics less than 1mm. Then, carefully fold all of the labeled filter paper(s) (I.e., 1 for embedded plastics, 1 for stomach plastics and 1 for probable plastics) and lower them into the labeled jars. Do not put the lid on, as these samples will need to dry for **up to 5 days** before being sealed. Set a calendar reminder or email someone to let them know when to seal the jars.
6. These should be kept in a dark and temperature-controlled environment (stable room temperature) to reduce degradation during storage.
7. During **Stage 2, Quantifying Plastics**, these plastics will be moved from the filter

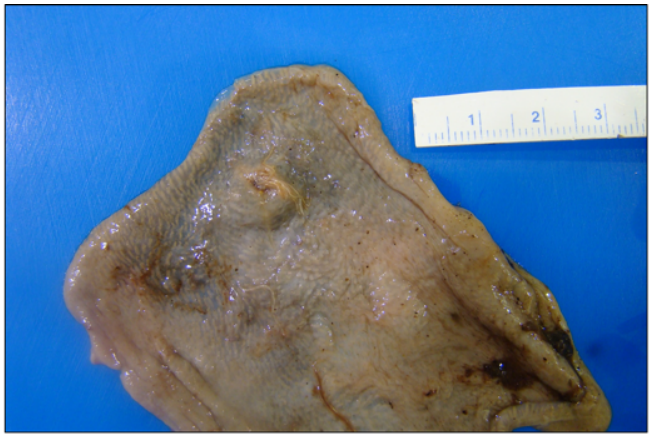
papers **carefully over a Pyrex dish** (as static may cause it to jump) into the smaller jars for longer-term storage. Please do not dispose of these ingested at the end of the study, they have value as props for presentations or educational purposes.

Figure 1. The stomach should be opened by cutting the proventriculus and gizzard along their length over a 1.0 mm sieve. The 1.0 mm sieve should be positioned securely over a larger non-plastic funnel and a ~250 ml glass jar, so that all items (including plastics) that passes through the 1.0 mm mesh can be retained for later analysis.

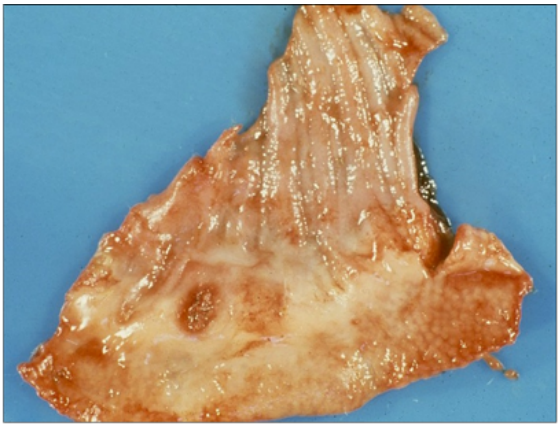
Figure 2. (**A**) It is often reported that plastics can impair gastrointestinal function or cause

physical damage to the bird's gastrointestinal tract, including perforation, blockage or ulceration. However, ulcers may have other causes such as (**B**) nematodes or (**C**) virus infection. It is important to note all ulcers, but not to assume that all ulcers are caused by plastic.

Proventriculus and gizzard. Northern fulmar. A32370-08-2. SI-181. Much plastic ingestion! No evidence of ulceration.

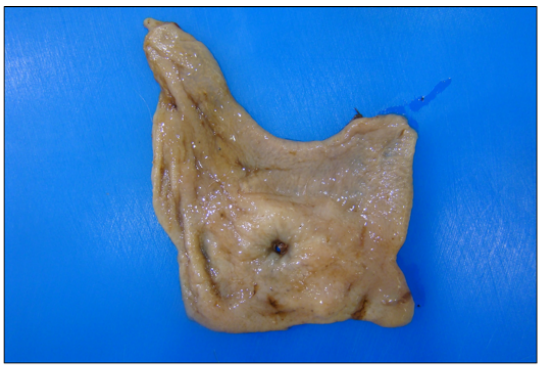


Proventriculus, mucosal surface. Greater Shearwater. A14852-12-1. SI-480. Small nodule in wall with a deep central ulcer (arrow) into which are anchored nematodes (tentatively identified as *Anisakis*). No plastic in proventriculus. Gizzard: one fairly large folded piece of thin plastic.

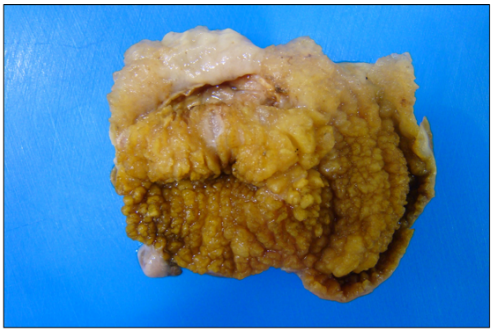


Esophagus (portion) and proventriculus. Canada Goose that died of "duck virus enteritis" (duck plague), a generalized infection by a Herpes virus. Large ulcer (arrow) in proventriculus.

Figure 3. Two more examples of ulcers, and suspected causes.



Proventriculus, mucosal surface. Greater Shearwater. A14852-12-2. SI-481. Nodule in wall with a perforating central ulcer (arrow), causing focal peritonitis. Proventriculus empty. Gizzard: one small bundle of 'strings', a few small pieces of plastic.



Gizzard and small portion of proventriculus, mucosal surface. Sooty Shearwater. A16452-12-2. SI-405. Large deep ulcer (arrow) at junction between proventriculus and gizzard. Two cross sections of nematodes seen on its surface, microscopically. Proventriculus: no plastic. Gizzard: one small thick piece of hard plastic.

Stage 2. Quantification of Plastics:

The goal will be to individually assess (and fill out a datasheet), for each piece of plastic. At the end, for each piece of plastic we will have recorded the mass, size, category (E.g., industrial, fragment, thread), colour, shape, erosion and polymer type.

Data is initially recorded on a paper data sheet, and then entered into a excel file. Separate sheets within the excel file are used to detail information for the birds (e.g., overall number of plastic pieces), and to detail information about the plastics (e.g., dimensions, colour etc).

Equipment Needs:

- Compound microscope (to closely examine probable plastics)
- Digital calipers
- Datasheets
- Excel files
- Analytical scale that measures to 0.001

Visual sorting of items

1. The first step involves careful visual sorting of items to separate the plastics from other materials, such as organic debris (shell fragments, animals parts, dried algae, or seagrasses, etc.) and other items that can be considered non-plastic anthropogenic debris, or rubbish (metal paint coatings, tar, glass, etc.).
2. This is done by direct examination of the sample by the naked eye or with the aid of a dissecting microscope. To some extent, plastics can be visually distinguished according to the following criteria: no cellular or organic structures are visible, fibers should be equally thick throughout their entire length, particles must present clear and homogeneous colors, and if they are transparent or white, they must be examined under high magnification.

Individual assessment of plastics (size, type, colour, shape, erosion)

1. Treat each piece of plastic separately. When filling out each datasheet, taking care to associate the datasheet with a Bird ID number, and to number the items being examined. This information will be entered into the provided excel spreadsheet, with one row per plastic item.
    a. Categorize the **Type** of plastic (van Franeker et al. 2004)
        i. **Industrial** plastic pellets. These are small, often cylindrically-shaped granules of ± 4 mm diameter, but also disc and rectangular shapes occur. Various names are used, such as pellets, beads or granules. They can be considered as “raw” plastic or a half-product in the form of which, plastics are usually first produced (mostly from mineral oil). The raw industrial plastics are then usually transported to manufacturers that melt the granules and mix them with a variety of additives (fillers,

stabilizers, colorants, anti-oxidants, softeners, biocides, etc.) that depend on the user product to be made. For the time being, included in this category is a relatively small number of very small, usually transparent spherical granules, also considered to be a raw industrial product.

ii. **User plastics** (all non-industrial remains of plastic objects) differentiated in the following subcategories:

1. Sheet-like user plastics (**sheet**), as in plastic bags, foils etc., usually broken up in smaller pieces;
2. Thread-like user plastics (**thread**) as in (remains of) ropes, nets, nylon line, packaging straps etc. Sometimes 'balls' of threads and fibres form in the gizzard – if so **do not unbundle the thread** before you measure it with the calipers. Keep it as a bundle and count as one item, measuring the dimensions of the bundle as best you can.
3. Foamed user plastics (**foam**), as in foamed polystyrene cups, packaging or aquaculture, or foamed polyurethane in matrasses or construction foams;
4. Small pieces (**fragment**) of more or less hard plastic items as used in a huge number of applications (bottles, boxes, toys, tools, equipment housing, toothbrushes, lighters etc);
5. **Other**, for example cigarette filters, rubber, elastics etc., so items that are 'plastic-like' or do not fit into a clear category.

b. **Opacity**: transparent Translucent or opaqueOpaque
c. **Colour**: One of white, clear, red, orange, blue, black, gray, brown, green, pink, tan, or yellow. Record only the primary color that would be seen by the bird, not the underlying color under the biofouling. Feel free to record additional colours or make notes in the 'comments' column.
d. **Erosion**: unweathered (new broken plastic, fresh), weathered (been in the ocean for awhile).
e. **Size**: Using digital calipers, measure the length, width and height of the plastic. If it is a sphere, measure the diameter. If it is cylindrical, measure the diameter and the height. If it is a bundle of threads, measure the length, width and height to your best ability.
f. **Mass**: Weigh each plastic and recorded in grams accurate to the 4th decimal (= tenth of milligram). Note: In the event that bundles of threads are encountered, where other debris is tangled within (including organic debris), it is acceptable to detangle the bundle and remove organic materials before weighing. Even if this bundle is observed after de-tangling to be more than one thread, weigh together.

Reporting Incidence and abundance for each bird

1. For the main categories **plastic** and **rubbish** we summarize **for each bird**:

    a. Incidence (Presence or absence)

    b. Number of plastics, rubbish, for each bird

    c. Number of user and industrial plastics for each bird

    d. For each bird, report the total mass for each **type of plastic** (industrial, fragment, foam, thread, sheet, rubbish) from step f above. For example, for each bird, weigh all the fragments together, all the foam pieces together, all the threads together, all the rubbish together, etc. Weights should be recorded in grams accurate to the 4th decimal (= tenth of milligram).

PROCEDURES FOR LABORATORY CHEMICAL WASTE DISPOSAL

(via MUN)

Procedures for disposal of hazardous waste:

Segregate materials according to the categories listed on pages 3 and 4. If possible, also segregate within categories. Unless the materials are used together during the course of an experiment, segregate all waste. Do not mix chemicals together in one container for convenience sake. We can not stress strongly enough that different chemicals have different disposal methods. If you are unsure of which category to use or if the materials can be safely mixed into one dump, call the **safety office (864-3659) or email (health.safety@mun.ca)**. Do not guess and do not assume.

Label all containers with the group name from the chemical waste category and an itemized list of the contents. For example, do not label a container simply `Corrosive Liquids'. List each chemical in the container, including all solvents used. List by full name only. Abbreviations, initials or chemical formulas are not acceptable labels.

Liquid dumps are intended for liquids only. Do not place glass or plastic items, such as tubes or pipettes, into solution dumps. If these items require disposal, package them separately. (Keep plastic and glass waste separate.)
Any waste containing PCB's must not be placed in waste dumps. Special procedures are in place for disposal of PCB's and it is important to keep the volumes small.

Packaging and containers:

All waste must be appropriately packaged for the waste category. For example: corrosive waste should be stored in non-metallic containers.

All liquid waste must be stored in leakproof containers with a screw- top or other

secure lid. Snap caps, mis-sized caps, parafilm and other loose fitting lids are not acceptable.

Solid debris must be placed in plastic bags. Do not place chemical or other non-biohazardous material in a biohazard bag. Biohazard bags are for biohazardous material only. Any waste disposed of in these bags will be treated as such.

For the disposal of vials containing liquid scintillation fluid, place plastic and glass scintillation vials in separate boxes. Plastic vials can be placed loose in a cardboard box lined with a garbage bag. Glass vials should be placed in trays, then placed in a box.. Attach a completed "Waste Scintillation Fluid" label (include all requested information). Please do not "hide" items for disposal in the boxes; the boxes are opened for final disposal and unexpected items can create a safety hazard to personnel.

Sharps (needles) must be well packaged to avoid any possibility of puncturing personnel. Used needles should be disposed of in a commercial sharps container or other suitable heavy plastic container. With the lids secured, place the containers into a cardboard box and seal with tape. Label "Sharps for disposal".

Importance of segregating waste

It is very important that hazardous materials are segregated into the proper categories. Different hazardous waste have different disposal methods. These disposal methods are also reflective in the cost of disposal. For example, waste which has the potential for reuse or recycling, such as non-halogenated organic waste is less expensive to dispose of than waste which is destroyed in a chemical incinerator, such as halogenated organic waste. There is also a tremendous environmental advantage to reusing and recycling chemical waste.

When categories are mixed, the disposal method is always for the "more hazardous" chemical. To use the above examples, when a few litres of a halogenated solvent is mixed with a drum of non-halogenated solvent, the entire volume must be considered halogenated waste. The contents of the drum,

including the recyclable waste, will be destroyed in an incinerator.

Importance of proper labelling

Waste that is picked up from a lab is not sent to the final waste disposal facility in the original container. For example, a 4L bottle of waste lead solution is bulked into a 205L drum with lead solution from other labs. This is either done on-site at our campus transfer station or, in the case of larger volumes, at a waste brokers transfer station. Little on site testing is done before bulking. We depend on the labels you place on the containers. If a container is mis-labelled or incompletely labelled, that is, all the contents are not listed, we may inadvertently place the waste in the wrong bulking drum. With the many hazardous combinations of chemical incompatibility possible, this could have serious implications. The result could be the release of noxious fumes, formation of more hazardous compounds, fire or even explosion.

It is also important when shipping hazardous waste to the disposal companies that the exact contents of the containers are known. Transportation of Dangerous Goods (TDG) regulations require that the transport of hazardous materials include detailed shipping documents. Also, although we do not test the container's contents, the waste disposal companies do extensive testing of all waste to determine the proper waste disposal method. Surprises in the containers will result in a surcharge levied onto the cost of disposal. Besides the unnecessary cost expenditure, this can also result in an embarrassing situation when it appears that we are hiding "more hazardous" waste in with other materials.

Chemical Waste Categories (see flowchart): AVOID MIXING WITHIN, AS WELL AS, BETWEEN CATEGORIES. SEGREGATE WASTE WHEREVER POSSIBLE. CONSULT WITH SAFETY AND ENVIRONMENTAL SERVICES (4320) BEFORE MIXING WASTE.

Organic waste - Phenol

Examples: any waste generated which contains phenol or phenol mixtures, including phenol-acid mixtures and phenol-chloroform mixtures.

Organic waste - Halogenated

Examples: any halogenated organic waste or any mixtures containing halogenated organic waste, except those containing phenol. Including chlorinated oils such as cutting oil. Examples: chloroform, 1,1,1-trichloroethane, methylene chloride

Organic waste - Corrosive

Examples: non-halogenated solvent-acid mixtures, non-halogenated organic acids such as acetic acid, trichloroacetate, acetic anhydride.

Organic waste - Non-halogenated plus water

Examples: non-halogenated solvent-water mixtures or non-halogenated solvents with greater than 20% water such as 80% ethanol.

Organic waste - Non-halogenated

Examples: acetone, toluene, acetonitrile, ethyl acetate, heptane, hexane, alcohol with less than 20% water.

Corrosive waste - Acid

Examples: hydrochloric acid, sulphuric acid, nitric acid, chromic acid, hydrofluoric Acid.

Corrosive waste - Inorganic/acid mixture

Examples: iron III chloride, aluminium trichloride, mercury compounds dissolved in acid, other inorganic compounds dissolved in acid.

Corrosive waste - Alkali

Examples: hydroxides, phosphates, ammonia.

Corrosive waste - Alkali mixture

Examples: compounds dissolved in hydroxides, phosphates, ammonia.

Waste Oil

Examples: used pump oil, crankcase oil, hydraulic oil. Excluding halogenated oils such as cutting oils.

Reactive waste

Examples: air and water sensitive materials such as Grignard reagent, alkaline metals, reactive halides.

Waste oxidizers

Examples: all nitrates, potassium dichromate, metal peroxides such as chromium dioxide.

Inorganic waste

Examples: heavy metal compounds and solutions such as those of mercury,

lead, copper and zinc (except those dissolved in acid), other inorganic compounds not covered by another category.

Hazardous waste - Other

Examples: waste not covered by any other category. All waste in this category must be segregated. No mixtures. Does not include radioactive waste, biohazardous waste, highly hazardous waste, explosive waste or surplus Chemicals.

Materials not covered under these procedures:

Radioactive waste

Follow procedures in place for the disposal of radioactive waste.

Biohazardous waste

Follow procedures in place for the disposal of biohazardous waste.

PCB waste

Includes any waste containing or suspected of containing PCB's. Follow procedures in place for the disposal of PCB's.

Explosive or other highly hazardous materials

Examples: peroxide formers such as aged ether, di and tri -nitro compounds, old flares, azides. These materials require special disposal. Consult the safety office for arrangements.

Surplus chemicals

Examples: any chemical which is no longer used or needed but which is still in good, usable condition. Consult the safety office for an assessment.

To request laboratory waste collection:

Send a completed "Request for Laboratory Waste Collection" form to the Office of Safety and Environmental Services, Facilities Management. If you have any questions or are unsure of any part of these procedures please call **(709)864-3659 or or email (health.safety@mun.ca)**.

Hazardous Materials Waste Request Form

How to do a Land Survey

Land Survey

Plastic pollution monitoring for shorelines, fields, and land

This guide will take you through how to survey an area of land for plastic pollution and other human-made waste. 80% of marine plastics originate on shore, which is why land-based surveys are important for understanding pollution in aquatic environments.. Also, plastics that reach the ocean often wash up on shorelines.

Land surveys give valuable information as to the types, quantities, and even sources of plastics that are polluting an area and may travel to the ocean. This means you can monitor plastics on land, on shorelines, and even in your own backyard!

This guide will teach you how.

If you have any questions, contact Civic Laboratory for Environmental Action Research through our website, or by emailing our director, Dr. Max Liboiron, at mliboiron@mun.ca.

This guide is funded by Memorial University of Newfoundland Public Engagement Accelerator Grant, and by the Social Science and Humanities Research Council IDG # 430-2015-00413

Getting Started

Materials:

- **1 rope** that is the length of your shoreline/area of study. We recommend a rope that is 15 feet long at minimum.
- **Garbage bags** for disposing of any extra plastics you find on the beach.
- **Gloves** for picking up trash.
- **Paper or booklet** to record information.
- **A smart phone or tablet** with the Marine Debris Tracker App (http://www.marinedebris.engr.uga.edu/). This protocol recommends the use of the Marine Debris Tracker to log items so that data goes into a public data set that scientists can use. This is optional, but recommended.

These are items that you can find at home or at local hardware or general stores. You can substitute items that suite your area. For example, using twine instead of rope.

If you do not have access to tablets or smartphones, this guide has a paper version to log items with (appendix).

Download & practice on the Marine Debris Tracker App. All participants should be trained on the logging app if you are going to use it. Note that you cannot input data from pen and paper into the app at a later date because the app geotags items, recording where they are found. If you input data after the fact, the app will think all the plastic is in your office!

Both Apple and Android products can download the Marine Debris Tracker App (http://www.marinedebris.engr.uga.edu/). Once downloaded, practice using the app to make sure you and your students are familiar with it. Just don't submit any of the practice data!

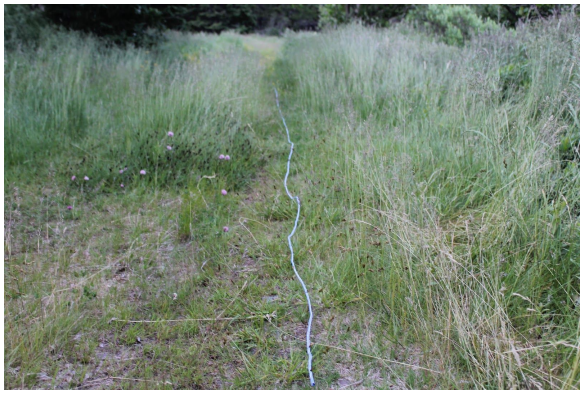
Choosing your site

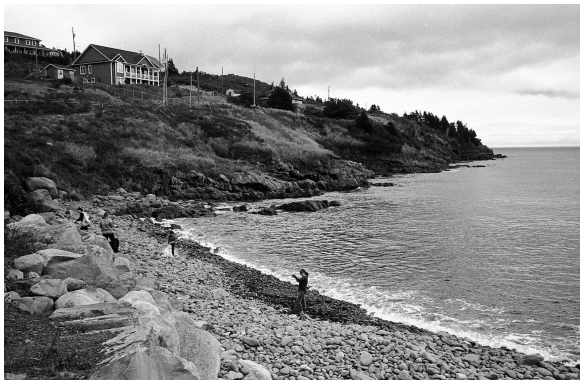
Before you go:

Choose your site.

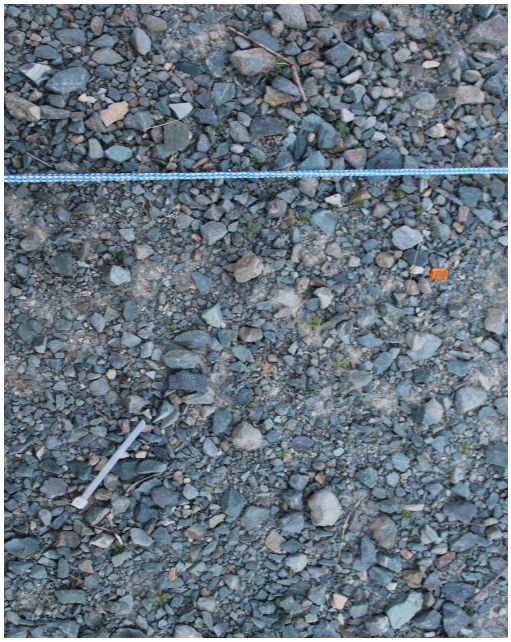
Choose somewhere that is easy and safe to access. School grounds, parking lots, roadsides, and shorelines are all ideal places. Ensure the location has some amount of waste so data will be rich. Since weather and season affect waste patterns, think about how weather will affect your data collection. For example, after rain, shorelines have more waste, and parking lots have less.

If you do this yearly, we recommend going to the same location at the same time of year each year so results are comparable.





Doing the Land Survey



/ˈtransekt/: a straight line or narrow section through an object or natural feature or across the earth's surface, along which observations are made or measurements taken.

1. **Lay down the rope and establish the study zone.** Open fields, parking lots, and shorelines don't have consistent and obvious edges. To define your research area, lay your rope down in a straight line that defines the length of the area you want to study. Then pick up waste that is 1 meter from either side of the rope. If it is difficult to stay within 1 m of the rope, you can take two ropes and pick up all the trash between them. Scientists, especially ecologists, use these lines to delimit study areas. They are called "transects."

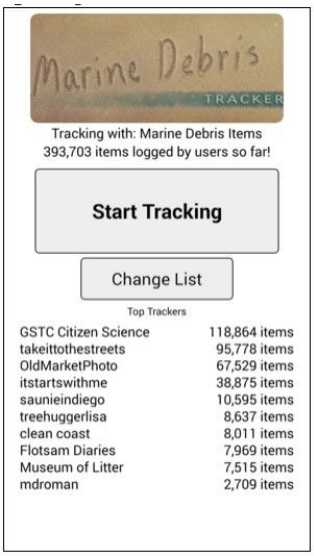
2. **Work in pairs.** One person will be in charge of picking up and calling out item names, and the other will log the data. The picker should have the gloves and bag, and the logger should have the app/paper.

   Note that most people do not often collect trash smaller than a piece of rice. Yet over 93% of plastics in the ocean are this size. Encourage students to look for and pick up tiny pieces of waste—cigarette butts, gum, and fragments of paper, but even smaller plastics are out there as well—tiny fragments that have broken off of larger items.

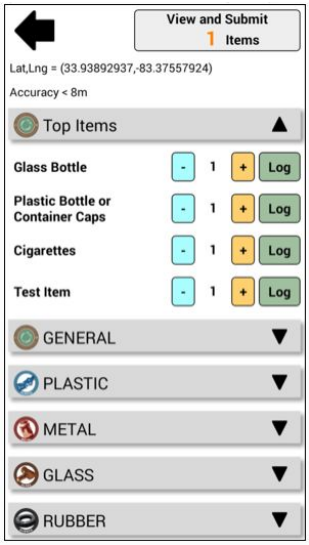
Using the Marine Debris Tracker App

If you are not using the app, skip this section, and just use a paper and pen to record what you pick up.

a) Once you download the app and open the app, you will be taken to the start page. You can use the default account and press “Start tracking” on the first screen.



b) For each item you pick up, find the material category that matches your item and expand it to see the suboptions. Click the green **Log** button to input an item. The item’s GPS coordinates will be automatically taken. The counter at the top that says “View and Submit # Items” will increase by one each time you hit a green **Log** button and will vibrate.



c) Keep logging items one at a time as you find them.

d) To correct any errors, click on view and submit in the top right hand corner. The view and submit screen will let you delete any items by clicking the red X next to the log. If you have been practicing and not logging any real items, delete all items—otherwise they go into the database that scientists use! Then press the black **back arrow** to go back to logging.

Troubleshooting: If you come across an item that is difficult to categorize, use your best judgement. The most important thing is that plastics are in the plastic category, and metal are in the metal category. If the metal was a door knob and there is no door knob category, put it in something as close as possible but still in the metal category.

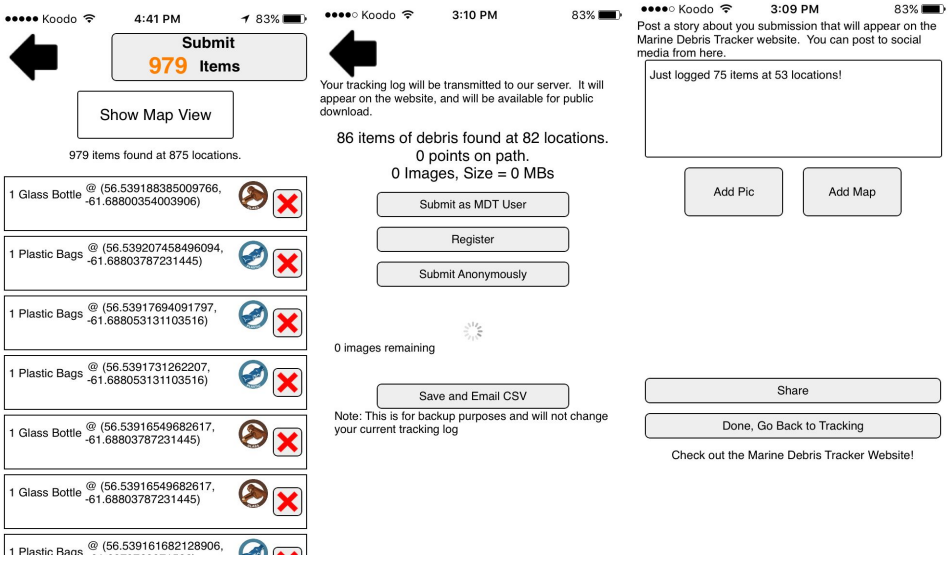
Submitting data on the Marine Debris Tracker App

a) If you are using the MDT, upload the data either in the field or at a location where there is wifi. If you are using paper, collect all sheets and add the types of waste from each sheet together on a master sheet that represents all the data.

b) For the MDT: Go to the **View and Submit** screen. Before you submit your data, you can view all your items on a map by pressing **Show Map View.** (this requires a wifi signal or data and may take a while to generate, depending on how many items you have logged). You can also get this map later via the MDT website (http://www.marinedebris.engr.uga.edu/)

c) Whether you look at the map or not, submit your items by pressing **Submit Items**. This requires wifi or data. If you do not have a connection, you can close the app and open it again later when you have a connection. It will remember your items.

d) Close the app, and open it again. The start page will have **Submit Queued** button. If your list is there, click on it to submit. If not, then it was already sent!



a) The next screen will ask whether you want to submit as a MDT user, register, or submit anonymously. We usually choose “submit anonymously” since you can always find your data later by location and date on the MDT website, but you can register as a unique MDT user. On this screen, you can also save and email yourself a CSV file with all your data. We recommend this.

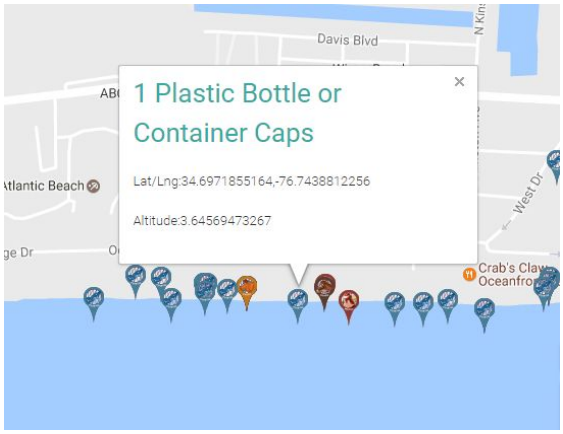
b) The data will appear on the MDT website if you upload it! Note that you can’t upload paper data to the MDT because it will geotag incorrectly.

Finishing Up

Reuse, recycle, or dispose of your collected items. We find that sorting items into recyclable, reusable, and waste is too hard to do while we are also tracking and collecting data about type of item, so we recommend sorting trash at the end. If you want, you can record the amount of recyclable, reusable, and disposable items at the end as extra data to discuss with your class (the app does not have settings for this kind of data).

After data gathering, Analyze your data! What was the most prevalent material? Within that material, what was the most prevalent item? Least? Why? Using the MDT website, you compare your data to other data in other areas-- how is your area similar to or different from others? If you generate a map via the MDT website, you can also analyze your data spatially. Does it clump, or is it spread out evenly? Why? Do you think this would change with different weather patterns like rain or wind? What about tides (if there is one)?

Scientists can use your data, too! Our laboratory at Memorial University of Newfoundland often uses the data on the MDT as raw data in our research to see trends in waste over time, between locations, and in specific regions. Thank you for contributing to our data!



Paper Form of MDT

TEAM NAME: DATE:

PLASTIC	
Item	**Number Logged**
Plastic Bags	
Balloons and/or strong	
Plastic Bottle	
Plastic Bottle or Container caps	
Cigarette lighters/tobacco packaging	
Cigarettes	
Foam or Plastic Cups	
Plastic Food Wrappers	
Rubber Gloves	
Other plastic jugs or containers	
Personal Care products	
Plastic or foam fragments	
Plastic utensils	
Six-Pack rings	
Straws	

FISHING GEAR	
Item	**Number Logged**
Buoys and floats	
Fishing lures and lines	
Plastic rope/Small net pieces	
Rope or net pieces (non-nylon)	

METAL	
Item	**Number Logged**
Aerosol cans	
Aluminum or tin cans	
Metal Bottle caps	

GLASS	
Item	**Number Logged**
Glass bottle	
Glass jars	

RUBBER	
Item	**Number Logged**
Flip-flops	
Tires	

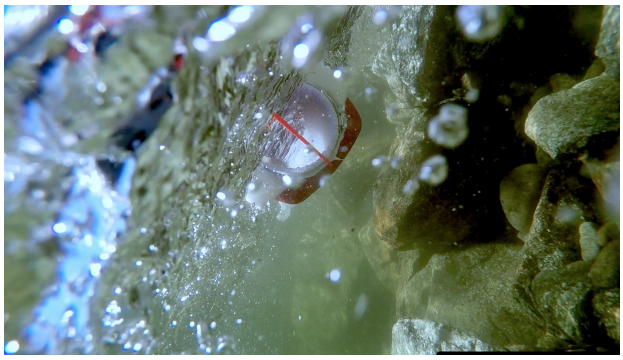
CLOTH	
Item	**Number Logged**
Clothing and shoes	
Fabric pieces	
Gloves (non-rubber)	
Towels or rags	

OTHER ITEMS	

How to Build & Use BabyLegs

Baby Legs

A citizen science research trawl for studying marine plastics



Created with baby's tights, soda pop bottles, and other inexpensive and easy to find materials, Babylegs can be used to skim the surface of water in rivers, streams, and oceans for floating marine microplastics by hand or from a boat. Plastics in the water pass through the mouth of BabyLegs, and accumulate in the toes. Once sampling is done, you turn the leggings inside out to see what they have caught!

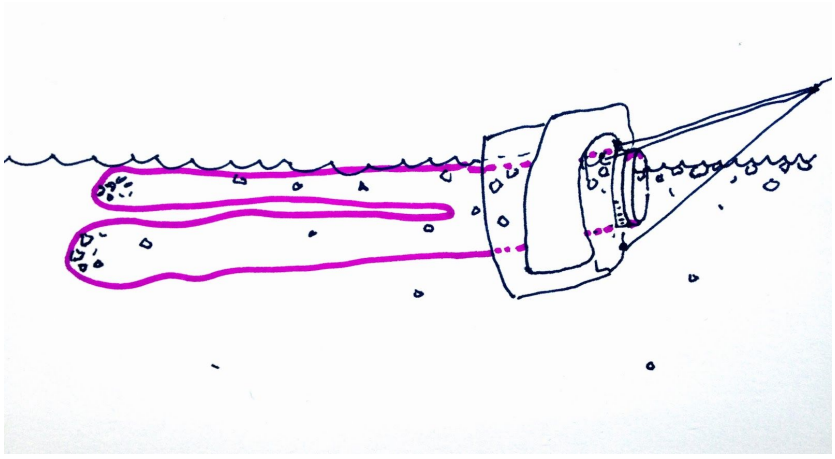
This guide will walk you through how to build and use BabyLegs.

If you have any questions, contact Civic Laboratory for Environmental Action Research through our website, or by emailing our director, Dr. Max Liboiron, at mliboiron@mun.ca.
This guide is funded by Memorial University of Newfoundland Public Engagement Accelerator Grant, and by the Social Science and Humanities Research Council IDG # 430-2015-00413.

Getting Started

Materials:

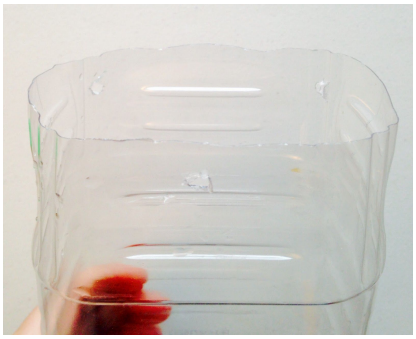
- 1 waterproof container with a mouth at least 6 inches wide. Square or rectangular works best.
- 1 plumber's clamp that will fit around the container (you can always merge two together if they are too short)
- 1 pair of nylon baby or toddler's tights- we recommend pink or red in case fibres contaminate your sample, as they will be easy to pick out. Cotton tights will get too heavy when wet, so ensure tights are nylon.
- 1 rope (~10 meters)- we recommend a bright colour in case threads contaminate your sample
- 1 pair of scissors or utility knife
- 1 nut driver or flathead screwdriver to open and close the plumber's clamp
- 1 drill or grommet punch (optional, but useful)
- 1 file or sandpaper (optional, but useful)



Creating the Body

(1) 

Step 1: Cut the bottom (and top if not already open, ie: soda bottle) off a waterproof container to use as Baby Leg's body. We recommend containers with openings 6 inches and larger to get adequate samples. Square-mouthed containers will give you a better idea of how much water passes through if you want to calculate the volume of water sampled, but if you are just sampling for the presence of plastics, a round-mouthed container will be fine. Any container will work. We've used bulk ketchup containers, kitty litter containers, small buckets, juice containers, and even a cardboard milk carton. Use what works best in your environment based on what you have available.

(2) 

Step 2: Carefully cut, punch, or drill three holes around the edges of the mouth of BabyLegs. These will be for the rope to attach. The mouth should be the side with the smallest opening (otherwise it creates drag). Put holes above where you are going to put the tights. There should be two holes at the top and one at the bottom, as this configuration gives the trawler the most control over maneuvering BabyLegs.

Step 3: Trim, file, melt, or sand the holes so they don't have sharp edges that might snag the tights. Do this for the cut edges at the top and bottom of the container as well. We don't want runs in our tights–it's bad for science because the snags might let samples escape.

Adding the Legs

Step 4a: Open a plumber's clamp wide enough to put over the body. If your clamp isn't big enough, you can screw two clamps together. Put the tights between the container and the clamp. The strongest part of the tights—the waist band—should be where the clamp attaches.
4b: You may have to crimp the round clamp around a square body if your container is square; bend the edges with your hands so the clamp and the body are the same shape.

4c: If possible, tuck the tights around the clamp for added strength, or double up the tights under the clamp if the clamp is too loose. If you clamp the thinner material, it may create holes.

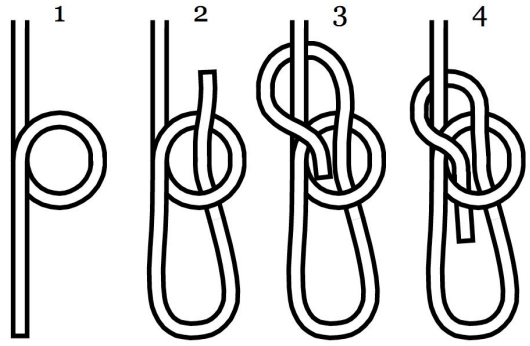
4a  4b  4c 

Step 5: Close the clamp securely, but not too tightly that it stresses the container. If your container is square, you may have to keep crimping the corners so the clamp tightens evenly. Test by tugging on the tights. They should stay firm.

Attaching the Rope

Step 6: Cut 3 equal sections of rope, and leave a 4th, much longer piece for trawling Baby Legs. Attach each of the 3 pieces of rope to the three holes you've created around the mouth of the trawl. Tie them so that they evenly attach at one central point. If it is uneven, BabyLegs can spin in the water and snag the lines. Melt* or otherwise tie off ends of the rope so threads do not contaminate your sample. If you can't melt them or are concerned that they might contaminate your sample, tie them so the ends are on the outside of the trawl rather than the inside.
*Adults may need to assist with this step, if children are participating in building BabyLegs.

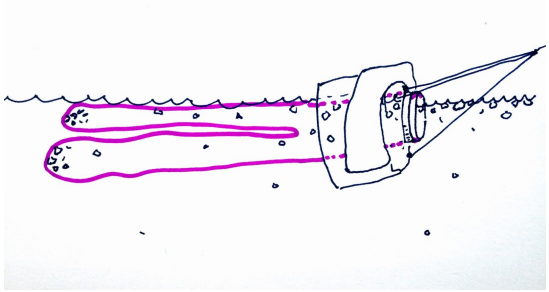
You can tie any kind of knot, but a bowline is a strong knot and is illustrated here.



Amendments and hacks

Since these are do-it-yourself tools, often people will use them in ways and conditions that we don't cover here. That's great, and is what advancing knowledge is all about! There are other additions to can add to BabyLegs depending on the type of water and vessel you are using. You can add "water wings," soda pop bottle pontoons, manta wings, and other stabilization or flotation devices for very calm and slow water, or choppy water.

A. BabyLegs with a cardboard body and black tights, used by hand instead of pulled by a rope.



B. BabyLegs design with a "water wing," or floatation device to keep the mouth of the container at the water's surface in rivers.

C. BabyLegs with pontoons to stabilize her in rough and choppy waters.

Using BabyLegs

Research trawls are essentially fancy butterfly nets for catching surface plastics. You can use BabyLegs anywhere where there is water and you can get the water to flow through the device. Here are some of the ways people have deployed BabyLegs:

A. Behind a human-powered boat like a canoe or skiff.

B. From a motorized boat, which requires an arm keeping the device out of the wake.

C. By hand in flowing water.

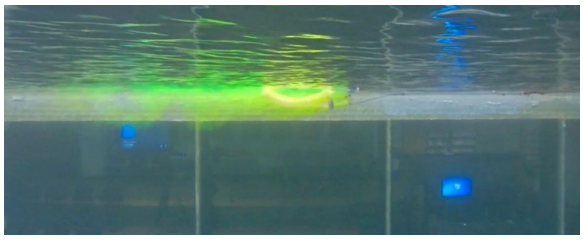
D. Tied off to a wharf with fast flowing water.

E. Tied off from a bridge with fast flowing water beneath.

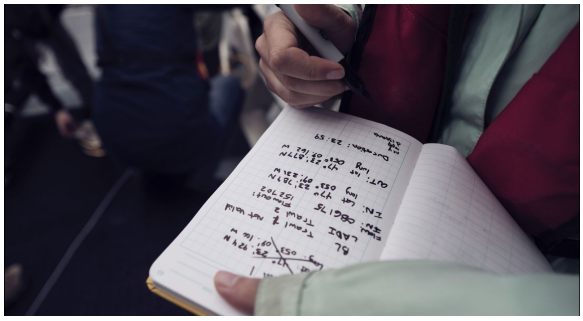
Proper compartment

No matter where you use BabyLegs, the main issue is to make sure the mouth of the container stays at the surface of the water at all times, so most of the mouth is underwater, and a small amount is above water. This ensures the surface of the water, where most buoyant plastics are, is always entering the container.

Keep BabyLegs out of wake, turbulence, or disrupted water flow. Turbulence “down wells” plastic, pushing them deeper in the water column. If you’re tying BabyLegs to a wharf or bridge, ensure she doesn’t bump against anything or end up in an eddy. From a boat, keep BabyLegs out of the wake. We recommend using an extending arm to pull her alongside a vessel rather than behind it. This shows a make-shift arm we made for a research trawl.



Water should always be running through BabyLegs, not “choking up” or flowing out again. Otherwise you’ll lose plastics. This is especially important when you are taking BabyLegs out of the water or changing direction (if you are in a boat or doing things by hand). This image shows dye moving through BabyLegs without flowing back out of the mouth.



Timing will vary, though we do not recommend trawling for less than half an hour if you want to get a good sample. If the water is particularly swampy or full of debris, we do not recommend sampling with a trawl at all, as it will be extremely time consuming to separate plastics from organics.

Removing the Sample

When you have trawled for a set amount of time, pull BabyLegs up and let the water drain out of her toes. There are two methods for removing the accumulated sample:

a) If you have a hose: Leaving the clamped to the body, slowly turn the tights inside out, one leg at a time, and rinse everything captured into a sieve (if you don't have a scientific sieve, you can use a fine mesh spaghetti strainer used in kitchens–the smaller the holes, the better). Repeat with the other leg. You can then transfer the items from the sieve into a jar. Add a capful of hydrogen peroxide (from a drug store) to the jar to keep the sample from smelling.

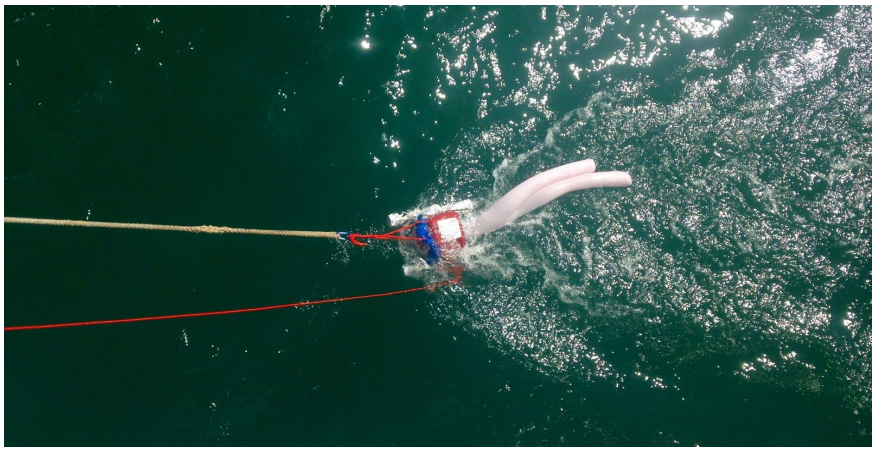
b) If you do not have a hose or a sieve, or if you are in a rush: Unclamp the tights, squeeze the water out, bundle the tights up, and put them in a sample bag or jar, and put on a new pair of tights if you want to sample again. You will process the tights afterwards in a sink. You will need many spare pairs of tights for this method. Add a capful of hydrogen peroxide to the jar/bag to keep the sample from smelling.

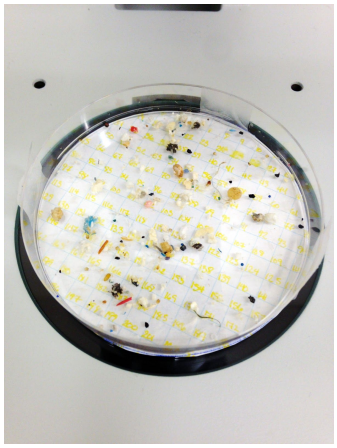
Capacities

Speed: We have found that BabyLegs (Ketchup Container version) can easily handle up to 5 knots of speed in semi-choppy waves for over an hour. In high speeds, attach a “leash” to BabyLegs-- an extra line that does not take any pressure and is tied to the boat directly (red line in the image). If your pole breaks or the main line malfunctions, you can use the leash to keep the device from being lost at sea, or being sucked under the boat.

Type of data collected: BabyLegs can collect samples of marine plastics so that you can see the types and sizes of plastics in an areas. You can also see the ratios of different kinds of plastics: are there more fishing line threads than foam? Or is it mostly foam and fragments? This can tell you about the type and sometimes even the sources of plastics in your area. You can also tell if there are “hot spots” or areas of high density of plastics, such as at sewage outfalls or in certain bays.

However, the data collected by BabyLegs will not hold up in scientific publications because of two reasons: 1) the faster BabyLegs goes and the older she gets, the more the tights stretch and the holes change size slightly. This means that you cannot be sure of the minimum size collected consistently. 2) At different speeds and in different amounts of chop, BabyLegs can bob above or below the surface of the water, especially with different designs, meaning that the behaviour isn’t exactly consistent across or between uses. In rigorous science, you need exactness and consistency to make robust claims about precisely how many plastics are in an area. If you need this kind of data, we recommend using our LADI trawl (see our website). But if you don’t need this kind of precision, BabyLegs is just fine. The image to the left is a sample from BabyLegs in the Hudson River.





How to Build & Use the Ice Cream Scoop

Ice Cream Scoop

A citizen science research trawl for kids

This guide will walk you through how to build your own research trawl for monitoring plastics in aquatic environments using simple, affordable, and readily available materials.

The Ice Cream Scoop is made using a modified ice cream or other container which lets water pass through it. A rope handle such as a skipping rope allows the user to pull the scoop through water, and a fine screen mesh is attached on one end of the container to collect samples from the water's surface.

The scoop can be easily assembled and materials can be substituted if needed. Children should assemble the Ice Cream Scoop with an adult.

If you have any questions, contact Civic Laboratory for Environmental Action Research through our website, or by emailing our director, Dr. Max Liboiron, at mliboiron@mun.ca.

This guide is funded by Memorial University of Newfoundland Public Engagement Accelerator Grant, and by the Social Science and Humanities Research Council IDG # 430-2015-00413.

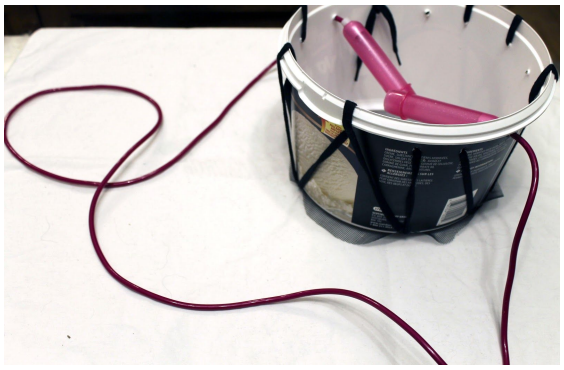
Getting Started

Materials:

- Empty Ice Cream container or other bucket (lid is not needed) - 4L tub is ideal
- Rope – we used a skipping rope
- Shoelaces (2)
- Nail
- Screen mesh
- Scissors
- Utility Knife
- Hockey or Duct tape

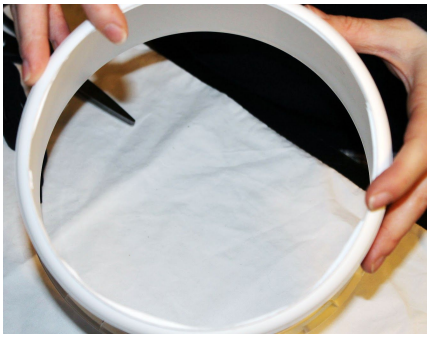
These are items that you can find at home or at local hardware or general stores. You can substitute items that suite your area. For example, using a salt beef bucket or kitty litter container instead of an ice cream container, or using twine instead of a skipping rope.
Note that the skipping rope will not be cut and can be used again!

Children should work with an adult on this project because of the use of knives and other sharp tools.

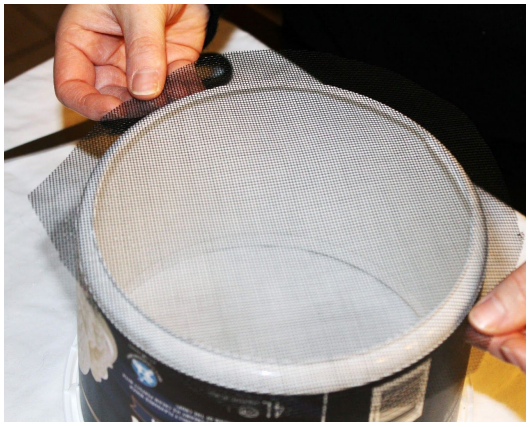


Building the body of the scoop

1. Carefully cut out bottom of ice cream container with scissors or utility knife. This will let water pass through the device. Make sure there are no sharp or jagged edges left when you cut; this might snag you or things in the water! Adults may need to help with this step.

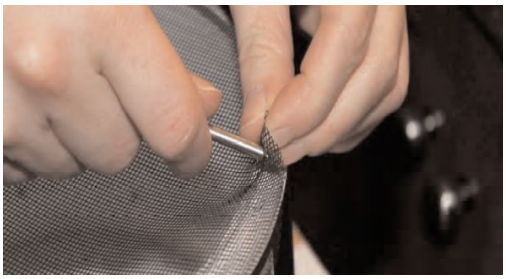
2. Lay a piece of screen mesh over bottom of ice cream scoop to measure the size you need and cut with scissors to fit, leaving approximately 1 inch of screen around the perimeter of the bucket. This will become your net!




Threading the net onto the scoop

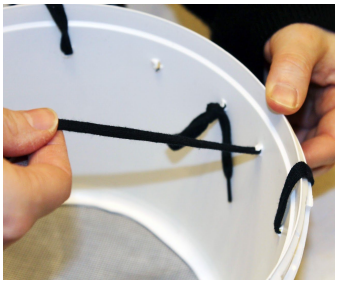
4. Punch holes around the top rim of the bucket with a nail, screw, or the end of scissors, about 2 inches apart, being careful not to tear the plastic. These holes will be used to thread the shoelaces and skipping rope in later steps.

5. Once holes have been punched through the bucket, place the screen mesh over the bottom of the bucket and puncture holes through the screen with a nail. These holes should match up with the ones you put in the bucket.



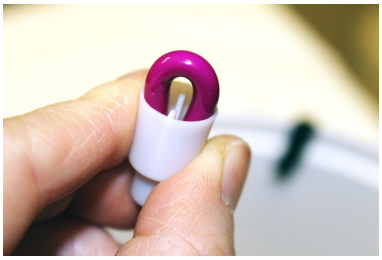
6. Thread the shoelace through bucket and tie the shoelace on the inside of the bucket, preventing it from slipping out.

7. Thread the shoelace through the hole you've punched in the screen, then back through the bucket. You're "sewing" the net and bucket together (see page 2 & 5). Continue this process until the shoelace has been threaded fully around the bucket and screen. Put tape around the edge of the screen, sealing it to the bucket. **Ensure you leave two punched holes (on opposite sides of the bucket) empty, as the skipping rope will be threaded through these.**

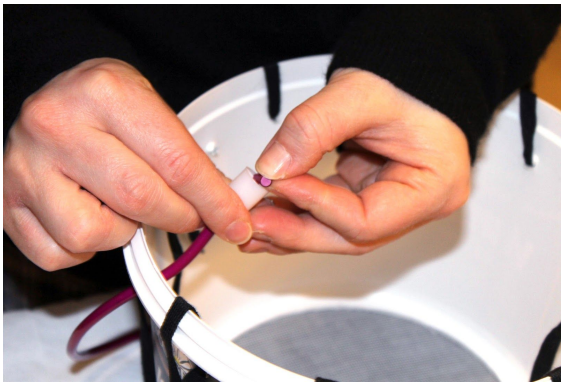


Attaching the final rope

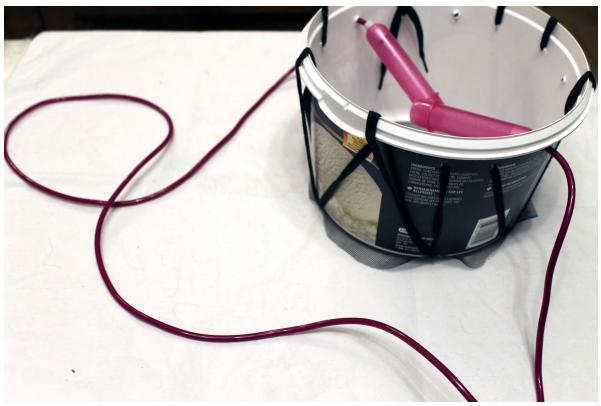
8. Remove the handle of the skipping rope. You can secure the rope by tucking it into the plastic part to make it easier to thread.



9. Thread the skipping rope through an empty hole in the bucket. Repeat this step on the opposite side of the bucket.
Re-attach the skipping rope handles once the rope itself is threaded inside the bucket.

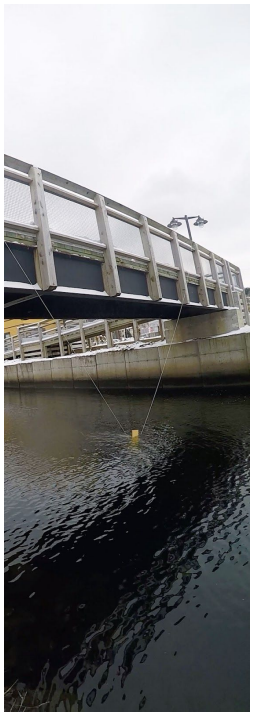


Congratulations! You've created your very own do-it-yourself trawl to monitor microplastics! You're now ready to use it.



Using the Ice Cream Scoop

Research trawls like the Ice Cream Scoop are essentially fancy butterfly nets for catching plastics that float on the surface of the water. You can use the Ice Cream Scoop anywhere where there is water and you can get the water to flow through the scoop.



You can:

- Put on boots and wade in the water with it,
- Drag it through the water from the side of a dock or wharf
- Dangle it off a bridge and let the water run through it
- Tie it to a stick and use it like a fishing rod

The ways you can use the Ice Cream Scoop is only limited by your imagination!

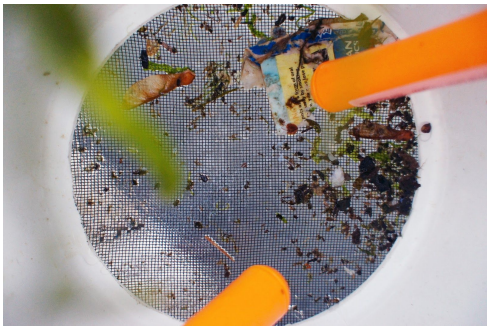
The only things to keep in mind is that water has to be flowing through the bucket, and that water can’t flow backwards otherwise your sample will flow out! And of course, be safe!

Using the Ice Cream Scoop

1. Whether you are pulling the Ice Cream Scoop through the water on a wharf or by hand, make sure water is always flowing *through* the container and not “choking up” and flowing back out. You can make it go back and forth in a zigzag, or do lots of straight lines.
   We recommend letting water flow through for at least 15-20 minutes to get a good sample.

2. The best place for sampling for surface plastics is at the exact surface of the water and a little bit lower. There should always be a bit of air at the top of the scoop to make sure you’re getting the surface of the water.

3. See what you’ve caught! There will be some organic (natural, biotic) material, and some human-made (unnatural, abiotic) materials. There are two ways to process your sample away: 1) put the whole thing in a bag and bring it back to the classroom or kitchen to sort, 2) sort it out, using tweezers, right there on the shore!



LADI and the Trawl

Protocols for Deployment and Sample Collection

By Melissa Novaceski

Before starting your trip:

1. Download a location tracker app that uses GPS, and doesn't require cell service (we used MapMyRun by Under Armor, which is free on iOS and Android).

2. Check that the boat you are using has an arm for trawling which can withstand the drag of LADI, or figure out how to McGiver your own (we have had success tying flagpoles and planks of wood to stick out of the side of the boat, see Figure 47).

3. Check that the boat also has a water pump on board, or bring additional materials for cleaning the samples from the net (listed below).

4. Check the weather and plan your trip, dress, and departure time accordingly.

5. From previous visits or a brief visual inspection, talking with local fishermen or others, Internet research, consideration of recent wind patterns or a brief visual inspection, estimate how much debris will be on the surface of the water. This will help you determine how many and how big the samples will be. Accordingly, plan how long you want to trawl, how many trawls you will do, and how many sample bottles you need.

6. The more trawls you do, the more accurate your results will be. If you just want to see what kinds of plastics are in an area or if there are any at all (a qualitative analysis), shorter or fewer trawls are fine. If you want to model how many plastics are in an area, you need to do enough trawls for lengths of time that correspond to the body of water you are sampling–over the open ocean, in a gyre for example you can trawl for over an hour and do 20+ trawls. In a Bay, trawl times might be 30 minutes and you would need 10.

7. Make sure you have these materials:

   (a) The trawl herself
   (b) Cod End and extra cod end, if you have one
   (c) 3x 3" or 4" hose clamps for the cod end (or whatever size you used)

(d) Extra hose clamps if possible
(e) Nut driver
(f) 1/4” wrench
(g) Extra rope for trawling
(h) Glass or plastic sample jars (the number depends on how many trawls you plan to do, how much ‘junk’ is on the surface of the water, and how long you plan to trawl. Bring more than you think you will need!)
(i) 335 µm sieve
(j) 2x permanent marker
(k) Log book for data (a waterproof notebook will help)
(l) Duct Tape
(m) Rubber bands
(n) Drinking water and food
(o) If your vessel doesn’t have a pump, bring a bucket or two (either pre-filled with fresh water or to use for filling with filtered water)
(p) a few bottles which can be used for squirting water out of the top

After reaching your desired trawl location:

1. Set up the arm and trawling ropes so LADI will be on the side of the boat, not close enough to be bumped and not in the wake (which causes downwelling of plastics, and will ruin your sample).

2. Before each trawl, make sure there are two or three hose clamps attaching the net and the cod end and that they are as tight as you can make them!

3. Do a short ‘practice run’, no data recording or sampling necessary, to flush out any particles in the net, and to make sure LADI is in the location you want at speed.

4. Adjust the ropes or arm if necessary. Wash out the cod end.

5. Record latitude and longitude just before deployment.

6. Start your tracker app just before deployment

7. Lower the trawl into the water on the side of the boat, making sure to drop it upright with the net trailing behind and avoiding the wake.

8. Pull the trawl (try to maintain a consistent speed) for your predtermined length of time, or if you notice the cod end is filling up quickly, try to end the trial before the net gets overfilled.

9. When you have finished your trawl, stop the tracker app, stop the boat, and pull the trawl out of the water.

10. Collect the sample (Protocol in Section 5), and thoroughly wash out the entire net and

cod end.

11. Trawl in both or multiple directions to avoid direction bias, either within or between trawls.

Troubleshooting

If the sampling area contains a large amount of debris on the surface trawl for a shorter period of time (10 minutes or fewer). Aim to have the water level half way between the top and bottom of the mouth of the trawl, this can be adjusted by adding weights such as rocks or fishing line weight to make it sit lower in the water. The mouth of the trawl should be perpendicular to the surface of the water, this can be done by adjusting the length of the chin strap of the trawl to angle it forwards or backwards.

If the trawl dumps, spills, or is caught in the wake, the sample can only be used for qualitative data rather than for statistical modeling of the quantity of plastics in the area.

Transferring trawl sample to jars

By Emily Wells

Sample transfer in the field

Equipment
• LADI Trawl with collected sample
• Nutdriver to unscrew hose clamp(s)
• Spray bottles or hose (with uncontaminated water)
• 335 micron mesh sieves (Figure 48)
• Permanent marker for labelling
• Large sample containers (500 mL); glass or plastic for holding the sample (plastic can freeze and won't break-- make sure it does not contaminate sample)

Transferring sample from trawl to sample container(s).

1. Using running water from a source such as a hose or spraying bottle, rinse the sample from the trawl net into the cod end, starting near the opening and working towards the end.

2. If the sample fits into the cod end, remove the posterior hose clamp and (a) invert net over a sieve and thoroughly rinse from the top to the bottom with uncontaminated water, thereby placing the sample in the sieve, (b) set the cod end aside for later processing, or (c) trawl for less time for future trawls to avoid the cod end overfilling. If the sample is large and does not fit the into cod end, (a) remove the hose clamp from anterior end of the PVC and rinse the sample from the upper netting into the sieve. Process the sample as per (iii) and (iv). Remove the hose clamp from the posterior end of the PVC and rinse the sample from both the PVC and the cod end into the sieve. Process as per steps (iii) and (iv).
   NOTE: Rinse the seams of the netting thoroughly, as small particles - including plastics - tend to accumulate there. To hold the cod end open for easier rinsing, have one person rinse while another pushes the side seams towards each other, thereby creating a cylinder-like shape. Rinse from top to bottom. Ensure that the mouth of the cod end does not touch the sieve while transferring the sample and rinse anything that contacts the sample using uncontaminated water over the sample container.

3. If feasible, remove large organic materials from the sample. Pick up the organic with your hand, rinse it thoroughly over the sieve, visually inspect it to ensure that no plastic debris remains adhered, discard it, and then rinse your hand over the sieve, ensuring no plastic debris remains on your hand.
   NOTE: Mind the water pressure when working with the sample in the sieve, as high blasts of water may splatter the sample and splash it from the sieve.

4. For samples that contain a large number of large organic materials, use your hands to transfer the sample directly into labelled containers and then rinse your hand over the container with uncontaminated water. For the remaining sample, and for smaller samples, spray the sample into a single point on the edge of the sieve. Spraying may be performed from the front or through the back of the sieve. Transfer the sample into a labelled container by placing the container near the rim of the sieve and directing the sample into it with your finger. Thoroughly rinse your finger over the container with uncontaminated water. Cap the containers and set aside for later processing.
   NOTE: The container label indicates: trawl run (example: LADI4); of more than one container, indicate the container identifier (example: LADI4.A); and the location.

5. Add a few mL of hydrogen peroxide to sample jars.

6. Repeat steps 1-4 for each trawl run.

General Laboratory Work Protocol: Trawl Samples

Equipment
• Lab coat and hair ties for long hair
• Petri dishes (amount depends on the number of samples) • 335 micron mesh sieves
• Dissecting microscope
• Tweezers
• Fine point permanent marker for labelling
• Sink or source of running water
• Double sided tape
• Optional: masking tape for labelling

Contamination:

Samples can become contaminated by particles (micro fibers) present in the air, on the clothes of workers, in poorly cleaned instruments, or by improperly sealed samples. Control samples should always be used to confirm that there is no procedural sample contamination. A control ensures that the fibers found in our study are not a result of contamination during processing. It will also enable us to characterize plastic contamination in the laboratory (instructions for how to create a control below). To reduce and account for contamination:

- Wear natural fibers like cotton or wool instead of petrochemical fibers like fleece or nylon
- Wear a lab coat or cotton overcoat and tie hair back
- Immediately before sampling, rinse your hands and wipe down any tools
- Work e ciently and cover the petri dishes and sample bottles whenever they are not in use
- Tie back long hair
- Collect a control sample during sample collection processing (Step 1 below).

Store the samples: You may top up sample jars with hydrogen peroxide and store samples in fridge until analysis. This will alleviate a strong smell as organic matter decays.

Processing sample to remove plastics:

1. To create a control that captures background microfibre and other plastic particle contaminate, put double sided tape in the bottom of a petri dish and leave the dish open next to the work area. Record the date the control was started on the dish. Everyone working on the samples should dust their clothes o↵ into the dish.

2. If samples are large and contain many organic materials, dump the content of the bottle into the 335 micron sieve and remove any remaining large organic materials by rinsing and disposing them (as discussed in (Stage 1-iii)). Depending on the size of the sample, this may take a long time.

3. Place an opened petri dish labelled ‘Control’ in the area where you are analyzing the samples.

4. Place an amount of sample in a petri dish so that it forms a thin layer in the dish: enough so that the contents do not float.

5. Systematically analyze the dish under a dissecting microscope. Move the dish back and forth in the field-of-view and use tweezers to probe through the dish contents. Remove any suspected plastics and place them on a separate petri dish (see below Plastic Analysis Guide).

Tips:

- If the probable plastics are sticking to the tweezers, place a drop of water on the probable plastics dish. The material better adheres to the water than the plastic dish.
- Placing the probable plastics dish on top of a blank piece of paper will make the contents visible.
- You may wish to organize the probable plastics according to suspected type for ease of processing (pellet, film, thread, foam, fragment, microbead, other; see Plastic Analysis Guide).
- Once a first viewer has analyzed the entire dish, have a second viewer repeat the analysis to ensure that all plastics have been detected.

6. Label the dish cover with trawl run identifier (example: LADI4.A), either directly with marker or on a piece of masking tape. Replace the cover on the petri dish and set the samples to dry overnight (until samples are dry) along with the control dish.

Validation and Quantification of Plastics Equipment

- Microsoft Excel
- Digital calipers

- Compound microscope
- Scale
- Small jar

1. An example spreadsheet that we used for analyzing data from Holyrood is included in Appendix C.

2. Assess whether the probable plastics are plastics (see Plastic Analysis Guide).

3. Separately quantify each plastic and log into an excel file: Trawl ID: Corresponds with the trawl run (example: LADI4) Plastic ID: The identifier for an individual plastic (example: LADI4.1 is the first plastic quantified from trawl run LADI4; LADI4.2 is the second plastic; etc.)

4. Plastic type: pellet, sheet, thread, foam, fragment, microbead, or other (see Plastic Analysis Guide) Plastic color: The predominant color of the plastic. Can be white, clear, red, orange, blue, black, gray, brown, green, pink, tan, or yellow

5. Plastic size: Use caliper for all size measurements. If it is a sphere, measure the diameter. If it is cylindrical, measure the diameter and the height. If threads are found in a bundle or are coiled, do not unravel them. Keep them as a bundle or in coiled shape and count as one item, measuring the dimensions as best you can. If the measurement is too small to be detected by the caliper, record that it is less than the smallest measurement the calipers record.

   (a) Plastic length: the longest measurement

   (b) Plastic height: the middle measurement

   (c) Plastic width: the smallest measurement

   (d) Plastic weight: Weigh the plastic on a scale. If no weight is detected, record that it is less than the smallest measurement the scale records.

   (e) Plastic erosion: General comments on the weathering of the plastic (jagged edges, faded, angular; see Plastic Analysis Guide for more terminology)

   (f) Notes: General comments on the process and shape of plastics.

NOTE: If a plastic is lost during the quantification procedure, note it in the Notes column and place and X in the cells with missing data (not a 0, as this would conflict with numerical data).

6. Place plastics from the same trawl run in a small jar labelled with the trawl ID (i.e. LADI4).

Plastic Analysis Guide

Careful visual sorting is necessary to separate plastics from other materials, such as organic debris (shell fragments, animals parts, dried algae, or seagrasses, etc.) and other items (metal paint coatings, tar, glass, etc.). It is recommended that more than one individual verify the probable plastics.

Pieces of microplastics <5mm can be generally identified by the following criteria:

- Lack of cellular detail and organic structures (i.e. venation)
- **Color**: plastics may be distinctly colored, such as: white, clear, red, orange, blue, black, gray, brown, green, pink, tan, or yellow
- **Weathering**: scratches and edge formation can help identify plastics
- Reflectivity
- **Strength and brittleness:** plastics are hard, flexible, or squishy (like foam). Organics tend to stretch when wet and break apart when dry
- Fibres are generally equally thick throughout their length

Types of Plastic

1. **Industrial Pellets (nurdles)**: These are small, often cylindrically-shaped granules about 4mm diameter, but also disc and rectangular shapes occur. Various names are used, such as pellets, beads or nurdles.

2. **Film (Sheet-like plastics)**, as in plastic bags, foils etc., usually broken up in smaller pieces;

3. **Thread,** as in (remains of) ropes, nets, nylon line, packaging straps etc. Thicker and coarser than microfibers.

4. **Foam**, as in foamed polystyrene cups, packaging or aquaculture, or foamed polyurethane in mattresses or construction foams;

5. **Fragments (Small pieces or larger, hard plastics)** broken off from a huge number of sources (bottles, boxes, toys, tools, ships, equipment housing, toothbrushes, lighters, etc);

6. **Microbeads** are used as an exfoliant in personal care products like face wash and toothpaste. They are often brightly coloured, very small, and perfectly round.

7. **Microfibers** are from synthetic clothing like fleece or nylon. They are much thinner than threads.

Equipment

- Dissecting microscope (Olympus SZ61, model SZ2-ILST, with a magnification range of 0.5–12×)
- Compound microscope (Ecoline by Motic, Eco Series, with a magnification range of 4-100×)

Visual sorting of items

1. The first step involves careful visual sorting of items to separate the plastics from other materials, such as organic debris (shell fragments, animals parts, dried algae, or seagrasses, etc.) and other items that can be considered non-plastic anthropogenic debris, or rubbish (metal paint coatings, tar, glass, etc.).
2. This is done by direct examination of the sample by the naked eye or with the aid of a dissecting microscope. To some extent, plastics can be visually distinguished according to the following criteria: no cellular or organic structures are visible, fibers should be equally thick throughout their entire length, particles must present clear and homogeneous colors, and if they are transparent or white, they must be examined under high magnification. Sometimes tapping, pinching, and poking the sample with tweezers will tell you about the material.

Incidence and abundance for each fish/animal

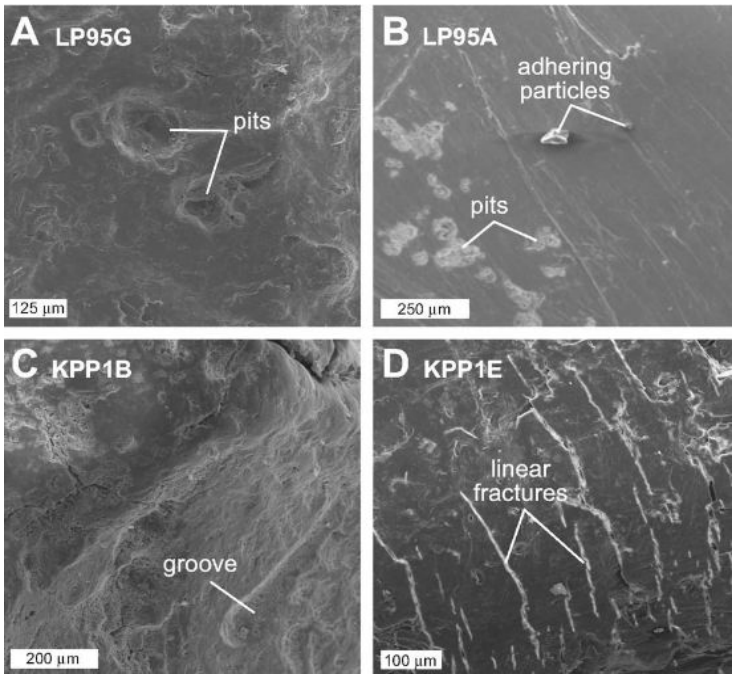
1. For the main categories **plastic** and **rubbish** we record for each individual:
    a. Incidence (Presence or absence) and
    b. Abundance by number (count of Number of items)
    c. Abundance by mass (Weight in grams) using Sartorius electronic weighing scale after a period of air drying at laboratory temperatures. Weights are recorded in grams accurate to the 4th decimal (= tenth of milligram).

Individual assessment of plastics (size, type, colour, shape, erosion)

1. Treat each piece of plastic separately. When filling out each datasheet, taking care to associate the datasheet with the correct Fish ID number, and to number the items being examined. (see sample spreadsheet in dropbox)
    a. Using digital calipers, measure the length (longest dimension), height (second longest dimension), width (shortest dimension) of the plastic. If it is spheroid, measure the diameter.
    b. Categorize the type of plastic:

i. **Industrial** plastic pellets or “nurdles”. These are small, often cylindrically-shaped granules of ± 4 mm diameter, but also disc and rectangular shapes occur. Various names are used, such as pellets, beads or granules. They can be considered as “raw” plastic or a half-product in the form of which, plastics are usually first produced (mostly from mineral oil). The raw industrial plastics are then usually transported to manufacturers that melt the granules and mix them with a variety of additives (fillers, stabilizers, colorants, anti-oxidants, softeners, biocides, etc.) that depend on the user product to be made. For the time being, included in this category is a relatively small number of very small, usually transparent spherical granules, also considered to be a raw industrial product.
ii. Sheet-like user plastics (**film**), as in plastic bags, foils etc., usually broken up in smaller pieces;
iii. Thread-like user plastics (**thread**) as in (remains of) ropes, nets, nylon line, packaging straps etc. Sometimes ‘balls’ of threads and fibres form in the gizzard;
iv. Foamed user plastics (**foam**), as in foamed polystyrene cups, packaging or aquaculture, or foamed polyurethane in matrasses or construction foams;
v. Small pieces (**fragment**) of more or less hard plastic items as used in a huge number of applications (bottles, boxes, toys, tools, equipment housing, toothbrushes, lighters etc);
vi. Small round beads (**microbeads**), often brightly coloured or white, from personal care products.
vii. Thin fibers (**microfibers**) from clothing. These are thinner and less rigid than threads.
viii. **Other**, items that are ‘plastic-like’ or do not fit into a clear category.

c. **Opacity**: transparent or opaque! It is Opaque if the bottom light from the microscope does not go through it.
d. **Colour:** white, clear, red, orange, blue, black, gray, brown, green, pink, tan, yellow
e. **Erosion**: unweathered, weathered, grooves, irregular surface, jagged fragments, linear fractures, subparallel ridges, burnt/melted, discolouration. See image below (from Corcoran 2009).



Erosin	Description
fresh	
unweathered	
weathered	
grooves	
Irregular surface	
jagged frag	
linear fractures	
subparallel ridge	
burnt/melted*	

References

Corcoran, P. L., Biesinger, M. C., & Grifi, M. (2009). Plastics and beaches: A degrading relationship. *Marine Pollution Bulletin*, *58*(1), 80-84. doi:10.1016/j.marpolbul.2008.08.022

Song, Y. K., Hong, S. H., Jang, M., Han, G. M., Rani, M., Lee, J., Shim, W. J. Comparison of microscopic and spectroscopic identification methods for analysis of microplastics in environmental samples. *Marine Pollution Bulletin* 93: 202-209. doi:10.1016/j.marpolbul.2015.01.015

Raman micro-spectrometry

By: Jessica Melvin

"Raman micro-spectrometry is a nondestructive spectroscopic method that can be used to determine a material's molecular structure by revealing vibrational characteristics of the sample down to the µm range (Imhof et al., 2012; Lenz et al., 2015). The method is expensive and technically difficult, factors that make it poorly suited to citizen science methodology in general. It was used here, however, in an attempt to determine the accuracy of visual identification methods for plastics over one millimetre in size . Raman micro-spectrometry can accurately distinguish between plastic and natural particles and can therefore identify (if present) any false-positives taken from the visual identification stage, while having the added benefit of identifying specific plastic polymers (Lenz et al., 2015)."

- Wash particles identified as plastics in the visual identification stage in ethanol and allow to dry prior to analysis.
- Place samples on a silica wafer of known Raman spectrum (520 cm^{-1} peak) and analyse using a Raman micro-spectrometer (Reinshaw InVia with 830 nm excitation) at a 20x Olympus objective.
- The instrument is controlled by WiRE 3.4 software.
- To ensure samples are not burnt, do not allow laser power to exceed 5%, and in cases of high fluorescence (in 4/5 samples), reduced laser power to 1%.
- Compare the Raman spectrum of each particle to reference spectra for common marine plastic polymers.
- Polymers include: polyethylene (PE), polyethylene terephthalate (PET), polycarbonate (PC), polypropylene (PP), polyvinylchloride (PVC), polystyrene (PS), polyamide (PA), polyurethane (PU), poly(methyl methacrylate) (PMMA), acrylonitrile butadiene styrene (ABS), and cellulose acetate (Bråte et al., 2016; Engler, 2012; Lenz et al., 2015; Plastics Europe, 2016)."

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