# Interactions Working Group

***A joint circumpolar project to measure and predict the cascading impacts of "Indirect Trophic Interactions" in arctic terrestrial vertebrate communities***

⚠ **VERY IMPORTANT 2018:** Pay special attention to the highlights and updates fully described in the last page of each protocol (or highlighted in yellow)

**Version 3, April 2018**

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Important progress has been made in recent decades to describe and understand how arctic terrestrial vertebrate interact, especially concerning predator-prey interactions.

Indirect interactions between different prey species (Fig. 1) modulated by shared predators (e.g. Arctic fox) are believed to have important impacts on the structure and/or dynamics of some communities. Yet, our understanding of these types of interactions is still fragmentary.

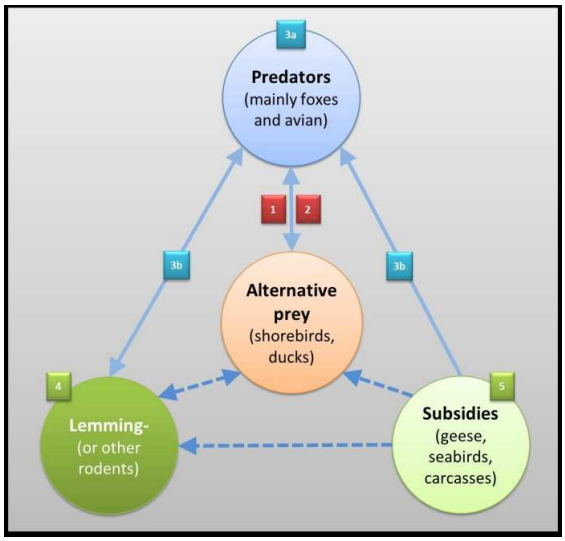



Figure 1: Main trophic interactions found in arctic terrestrial communities
(indirect interactions: dashed lines)

To fill that gap, we will build on ongoing projects exploring related questions in Canada (Marie-Andrée Giroux, Nicolas Lecomte, Joël Bêty) and Greenland (Olivier Gilg, Niels M. Schmidt), while taking advantage of existing networks (ASDN in North America and "Interactions" program in Greenland and Eurasia). The aim of the project is to promote the implementation of several common protocols (numbers on Fig. 1) that will (1) improve each collaborator's knowledge at the site level and, more importantly, that will (2) be merged across sites and years to improve our understanding of the functioning and the influence of indirect interactions on arctic vertebrate communities in general.

Five types of data have been identified as being mandatories to answer questions related to this topic. These data sets will be collected using 5 specific protocols (numbered in Fig. 1) described in the following chapters:



Additional chapters will address:

### *Study sites*

On most sites joining this initiative (Table 1), one or several of these 5 protocols are already implemented. Running all 5 protocols will allow collecting data in a standardized way to answer the various research questions listed provisionally in Table 2. Although some research questions (already identified or pending) can be addressed without implementing all protocols, doing so will nevertheless result in a larger number of scientific contributions.

***Table 1**. List of the 12 study sites (in 5 Arctic countries) collaborating to the project in 2016.*

| Sites from N to S (and PIs) | Lemmings/ Rodents | Main Terr. predators | Ungulates | Geese | Marine resources | Calidris spp |
|---|---|---|---|---|---|---|
| **Hochstetter**, Gree. 75.15N 19.70W (Olivier Gilg & Loïc Bollache) | Collared lemming, high amplitude cycles, fading since 2000 | Arctic Fox<br>Long-tailed Skua<br>Snowy Owl<br>Stoat | Muskoxen: low | High abundance of moulting Pink-Footed G. in July | Limited | Sanderling<br>Dunlin |
| **Zackenberg**, Gree. 74.47N 20.57W (Niels M. Schmidt & Jeroen Reneerkens) | Collared lemming, low amplitude cycles, fading since 2000 | Arctic Fox<br>Long-tailed Skua<br>Snowy Owl<br>Stoat | Muskoxen: high (with many carcasses almost every year) | High densities of Several species | Limited | Little (Dunlin) |
| **Belyi Island**, Russia 73.32N 70.09E (Aleksander Sokolov, Natasha Sokolova & Dorothée Ehrich) | Siberian Lemming, high amplitude cycles | Arctic Fox<br>Skuas<br>Snowy Owl | Reindeer: high | Few moulting (Pink-Footed G.) | Limited | Sanderling<br>Dunlin |
| **Bylot**, Canada 73.15N 80.00W (Joël Bêty, Dominique Berteaux, Gilles Gauthier) | Collared and brown lemming, high amplitude cycles | Arctic Fox<br>Skuas<br>Rough-legged Buz.<br>Snowy Owl<br>Stoat | Absent | Largest colony of greater snow geese in Canadian Arctic | Limited | Baird's (White-rumped) |
| **Karupelv**, Greenl. 72.50N 24W (Benoît Sittler & Johannes Lang) | Collared lemming, high amplitude cycles, fading since 2000 | Arctic Fox<br>Long-tailed Skua<br>Snowy Owl<br>Stoat | Muskoxen: medium | Few breeding (Barnacle) and moulting (Pink-Footed) | Limited | Sanderling |
| **Sabetta**, Russia 71.24N 71.80E (Aleksander Sokolov, Natasha Sokolova & Dorothée Ehrich) | Various species | Arctic Fox,<br>Long-tailed Skua | Reindeer | Medium densities of Several species | None (inland) | Dunlin<br>Little Stint<br>Temminck's |
| **Barrow**, Alaska USA 71.23N 156.75W (Richard Lanctot) | Collared and brown lemming, high amplitude cycles | Arctic Fox<br>Long-tailed Skua<br>Pomarine Skua<br>Snowy Owl<br>Stoat/Weasel | Reindeer: in winter in some years, rare in summer | low density of White-fronted Geese | None (inland) | Dunlin<br>Semipalmated |
| **Ammarnas**, Sweden 69.96N 16.29E (Rob van Bemmelen & Anders Angerbjörn) | Various species | Long-tailed Skua<br>Red Fox | Reindeer: low | Absent | None (inland) | Dunlin (Temminck's) |
| **Igloolik**, Canada 69.40N 81.60W (M.-A. Giroux and Nicolas Lecomte) | Collared and brown lemming, low amplitude cycles | Arctic Fox<br>Long-tailed Skua | Absent | Very low densities (Lesser snow goose, Brant goose and Cackling goose) | Abundant in winter (polynya and seal carcasses) | White-rumped (Semipalmated) |
| **Erkuta**, Russia 68.22N 69.15E (Aleksander Sokolov, Natasha Sokolova & Dorothée Ehrich) | Various species | Arctic Fox<br>Rough-legged Buzzard<br>(Red Fox) | Reindeer: high | Medium densities of Several species | None (inland) | Temminck's |
| **East Bay**, Canada 63.98N 81.67W (Paul Smith) | Various species | Arctic Fox<br>Long-tailed Skua | Reindeer: high | High densities of Several species | High (costal) | White-rumped (Semipalmated) |
| **Churchill**, Canada 58.70N 94.08W (Laura McKinnon) | Various species | Arctic Fox<br>Red Fox<br>Long-tailed Skua<br>Stoat | Absent | High densities of Several species | High (costal) | White-rumped |

### *Research questions*

The various research questions aim at better understanding indirect interactions in tundra ecosystems. Detailed paper outlines will be prepared within a few months and circulated among all collaborators. For the time being, we listed a provisional list of research questions that have been prepared be the initiators of the project (Table 2).

***Table 2**. Preliminary list of research questions identified as likely products of this initiative*

| Research questions | | | Data to be used | | | | |
|---|---|---|---|---|---|---|---|
| **Short title** (and PI) | **Objectives** | **P1 Artificial Nests** | **P2 Real nests Tiny Tags** | **P3 Pred/Lem. Incidental Observations** | **P3Pred/Lem. Collection of fox scats** | **P4 Lemming abundance** | **P5 Herbivore relative abundance** |
| The role of food subsidies in shaping predator-prey interactions in the Arctic: implications for shorebird conservation (MA Giroux and A. Bédard, MSc Student) | Investigate the impacts of subsidies and lemmings on the predation pressure on ground-nesting birds like shorebirds using artificial nests | ● | | ● | | ● | ● |
| Subsidies-Modelling (O.Gilg) | Similar question but answered through (parameterized) modelling work | ● | ● | ● | ● | ● | ● |
| Are predator-prey interactions explaining the distribution of some Arctic birds? (O. Gilg) | A test of Gilg and Yoccoz (2010) hypothesis:<br>• Following McKinnon et al. (2010): Is breeding success of real nests also positively correlated with latitude (or an index of productivity+subsidies)<br>• By measuring the difference between predation pressure (artificial nests / predator counts) and predation on natural nests (Tiny Tags), test if high-Arctic species (and/or populations breeding in areas with high/low levels of subsidies) are more sensitive to predation | ● | ● | ● | (●) | (●) | (●) |
| Shorebirds' nest attendance related to predation (O. Gilg & N. Meyer, PhD Student) | Extend Smith et al. (2012) work on more species and sites (comparing recess time/duration with predation rate and pressure)<br>First analyses have started and will be circulated in late 2018 | (●) | ● | ● | (●) | ● | |
| Predation rate between (covered) artificial and real nests (J. Bêty) | In many studies, predation rates on artificial nests are different than on real nests. We assume that this is partly due to different predators predating unattended (uncovered) and attended (covered) nests. | ● | ● | ● | | | |
| … | | | | | | | |

# Protocol #1:
# Monitoring of predation pressure using artificial nests

**(COORDINATOR: MARIE-ANDRÉE GIROUX & OLIVIER GILG)**

## MAIN OBJECTIVE

Estimate spatial and temporal variations in predation pressure on ground-nesting birds like shorebirds using artificial nests. The protocol will allow estimating separately the predation pressure associated with (1) terrestrial and (2) avian and terrestrial predators.

## METHODS

### *Artificial nest manipulation*

Two distinct sampling designs can be chosen according to the number and overlap of typical shorebird habitats (Figure 1).

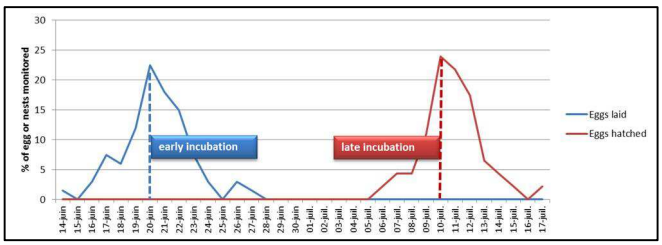
Option A (preferable): the artificial nest will be placed randomly in a single plot of 5-10km² if habitat is relatively homogeneous or if different shorebird habitats (e.g., Dunlin in wet and Sanderling in mesic Tundra) strongly intricate (e.g., a mosaic of small, <1ha, contiguous patches of different habitats).

Option B: if different shorebird habitats are found within the 5-10km² area but cover large distinct patches (>1ha), then sampling should consist of 10-15 nests randomly placed in each of the two major habitats.

In all cases, if an habitat is dominant, it can hold a few more nests, but always ≤15. In addition, the habitat (to be described) in which the nest is deployed will be recorded and treated as a covariate.

The experiment must be conducted twice per summer: once during "early incubation" and again during "late incubation" (1-2 weeks between repetitions). Exact dates can change depending on the year- and site-specific breeding phenology.

For example: If most nest hatch ca. 10/7 (hence considered "peak hatching time"), then "peak laying time" is around June 20 (i.e., 3wks earlier for most Calidris species).
In this case we would define "early incubation" as the week following the peak laying date and "late incubation" the week preceding the peak hatching date. Experiments should hence respectively occur during the following periods 20-27/6 (early) and 3-10/7 (late).




To insure independence between artificial nests, all nests will be randomly distributed within the study plot(s) and located at ≥150-200m away from each other. Avoid deploying nests when a predator is close by and watching you!

Once deployed, nests should be visited 2 times, i.e. after 48 and 96 hours of exposure.

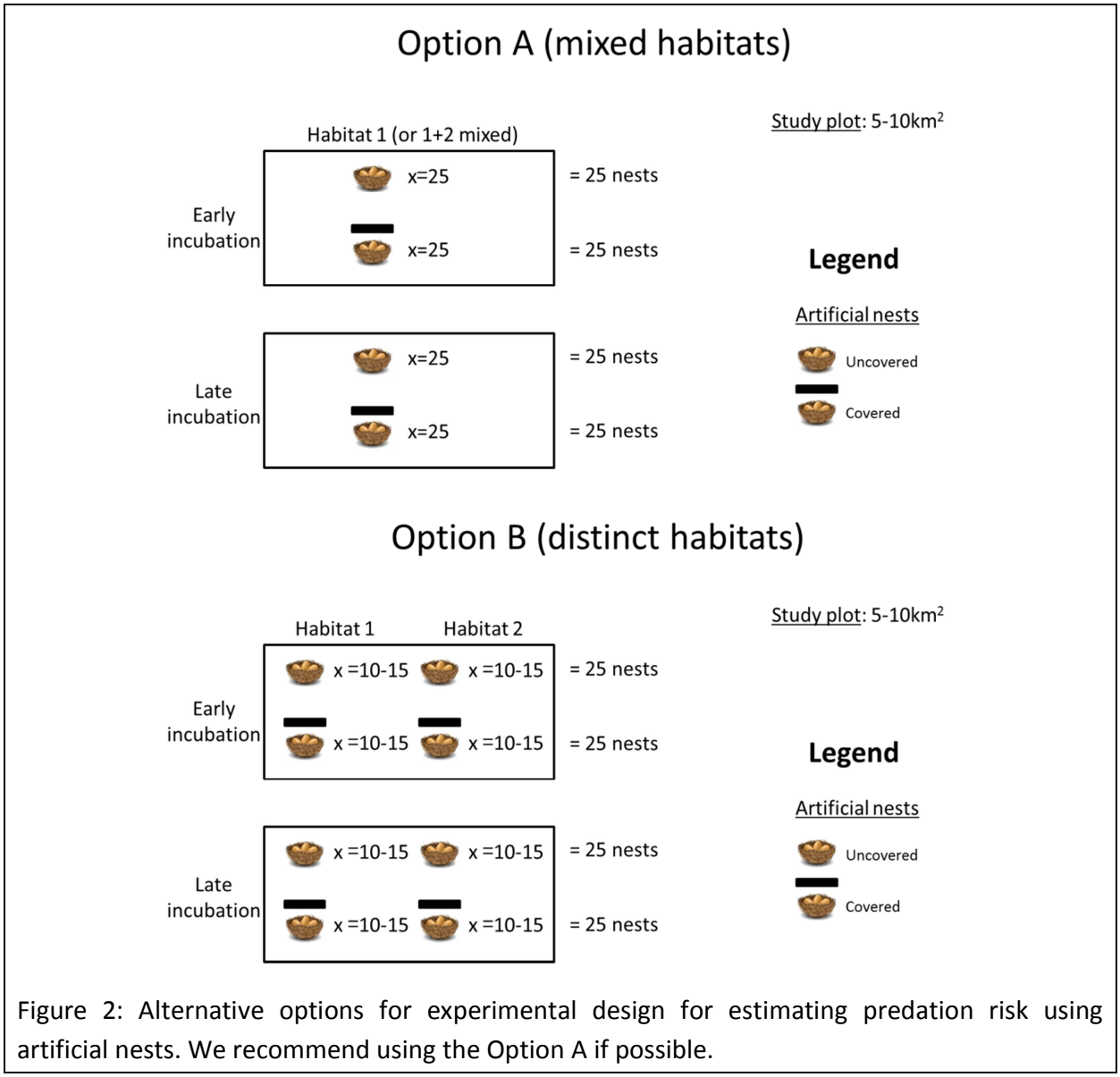



Figure 2: Alternative options for experimental design for estimating predation risk using artificial nests. We recommend using the Option A if possible.

The nests will be deployed in study plots of 5-10 km$^2$ (Options A and B). Those numbers must be similar for ALL SITES, and correspond to a nest density of 5-10 nests/km$^2$, which, in most cases should be below the density of natural shorebird nests (all species combined). Some evidence suggests that individual avian predator can learn and recognise when observers are deploying artificial nests. Hence, if you deploy artificial nests near a nesting avian predator (ex. gulls or jaegers), consider moving the artificial nests away from the avian predator nest (>1 km) after 2-3 years.

To estimate the predation risk associated with “mammalian” vs. “mammalian+avian” predators, half of the artificial nests per study plot will be covered with compacted plant material (see below) and the other half will remain uncovered. The use of cover has been tested for three

years on Bylot Island and camera trap data have shown that only arctic foxes predated covered nests while uncovered nests were predated by arctic foxes and avian predators.

Artificial nests

Each artificial nest will consist of 4 fresh European or Japanese quail eggs (not Chinese quail eggs) placed in a small depression (nest cup) on the tundra. The depression (approximately 7 cm in diameter) can be made using the sole of your boot. Place a marker in the middle of the nest cup so that the top of the marker is flush with the bottom of the cup and place the 4 eggs in the nest cup, fully covering the marker. The marker is only used to find the nests once they have been depredated. We suggest using ½ barbecue wooden skewer as marker. Alternatively, coloured nail, or nail with flagging tape wrapped around the tip can be used. Because markers can be contaminated by predator's scent, the MARKERS MUST BE DISCARDED IF A NEST IS DEPREDATED (and should never be in close contact with "clean" markers).

Every second nest will be a "covered nest". For covered nests, use a popsicle stick instead of a nail. In that case, the stick can stick out 1-2 cm above the eggs to help maintain the lichen cover (or any other dominant plant material found nearby) in place. To reduce odours in the vicinity of the artificial nest, it is important (1) to use nitrile gloves when handling the eggs, feathers, popsicle stick, nail, etc., (2) to avoid kneeling down (or drop your bag!) by the nest at any time, and (3) to rinse all eggs with clean water prior to setting up the nests (with a special attention for eggs that are looking dirty or were in the same boxes that contained broken eggs).

All nests should be deployed within the same time period, avoiding periods of peak activity of the expected main predator (usually arctic fox). If the field site is not monitored via an automatic weather station, make some general notes on the weather (i.e. precipitation, wind speed, cloud cover). Record geographical coordinates of each artificial nest using a GPS and place popsicles sticks at 5 & 7 meters from the nest (both sticks aligned to easily find the nest, especially when they are empty). Record the bearing from the popsicle stick to the nest or always use the same bearing for all nests. Another natural object (e.g. a stone) placed approximately at 7 meters from the nest can be used to replace the second popsicle stick.

Covers

Covers should be made of a material that is commonly encountered on the ground at the field site and which is likely to remain effective on the nest during the 4 days of the experiment, regardless of wind or rain conditions. At Bylot Island, lichens of the genius Bryoria or Gowardia have been used. To make sure the cover holds on the popsicle stick while hiding the eggs completely, lichens may have to be compacted. On Bylot Island, the compacted lichens were the only material used to make up the cover (no plastic lid was used). Vegetal covers should be 10-12 cm in diameter, round, and 2-3 cm thick (Fig. 2). Depending on site-specific plant communities, compacted lichens may not correspond to the best material to hide nests. In this case, it would be advised to plan a preliminary 4-day test (with no eggs) in order to test the

different biological material that can be used at your site to cover nests. At every site (including sites where lichens represent a good option for concealment), we recommend testing how the type of cover chosen holds on the popsicle stick (see below) during a 4-day period. Differences in the type of material chosen to cover nest could generate biases between sites. To better understand whether such biases could affect our results, we must document the type of cover used on each nest (e.g. by taking notes and a picture, if different types used on the site) and we will estimate indexes of predator activity at every site (see section Indexes of predator activity below). Other options for vegetal material that can be used to cover eggs are disks of Dryas or of dried mosses. If the plant material chosen has a risk of flying in the wind, it may be advised to use fine metal wires (just 2 sections) that can stick out of the tundra and bend over the cover to help keep it in place under strong winds. Obviously, too many of those wires are not advised, as we don't want to create a cage around the egg.

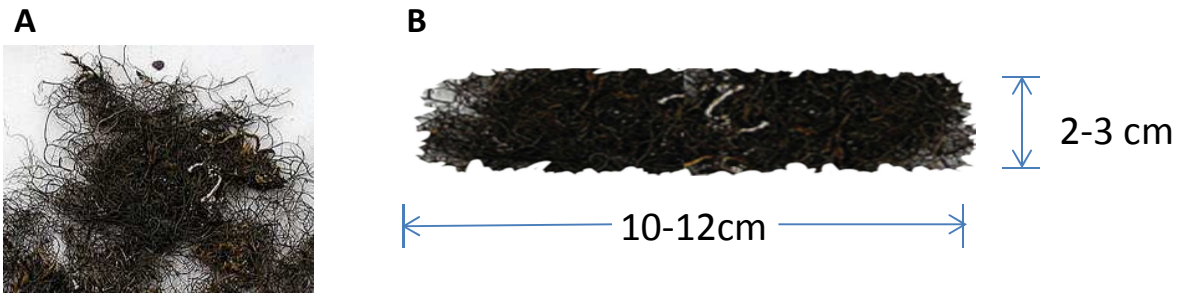



Figure 3: Characteristics of the lichen covers. (A) Covers should be made of a material that is commonly encountered on the ground at the field site and that won't suffer from rain or wind. At Bylot Island, lichens of the genius *Bryoria spp.* or *Gowardia spp.* have been used. To make sure the cover holds on the popsicle stick while hiding the eggs completely, lichens may have to be compacted. On Bylot Island, the compacted lichens were the only material used to make up the cover (no plastic lid was used). (B) Vegetal covers should be 10-12 cm in diameter, round, and 2-3 cm thick.

Visits

Once deployed, nests must be visited 2 times, i.e. after 48 and 96 hours of exposure. At each nest visit, the following information should be recorded:

- Predation
  - 0 – nest intact
  - 1 – partial predation (record the nb of eggs remaining)
  - 2 – total predation
- Signs of predation (predator faeces, egg shells) that could help identify predators:
  - Fox: presence of faeces or smell of fox urine in the nest cup
  - Avian predator: eggs are pierced but found within the vicinity of the nest
  - Unknown: eggs gone, no predator specific signs

All eggs that were not depredated at the end of the first session (early incubation period) should be collected. Nest markers of nests THAT WERE NOT DEPREDATED may be left in place in order to help find the nest cups when setting up the experiment again during the second session (late incubation period). REMINDER: nest marker of depredated nest should always be DISCARDED and put away from clean markers!

Potential confounding effects such as the presence of predators during nest deployment or visits and/or odour of eggs deployed should be noted in the notes section of the datasheet. While visiting artificial nests, it is really important to avoid visiting natural nests in the same run.

Artificial nest data will be entered in the file named
**"P1_Artificial nest_SITE_YEAR.xlsx"**

## Highlights and Updates on "P1_Artificial Nests":

**Smells**: To reduce possible bias (especially between-sites differences in detectability related to egg smell; eggs from different providers likely have slight differences in smell…), we advise to:

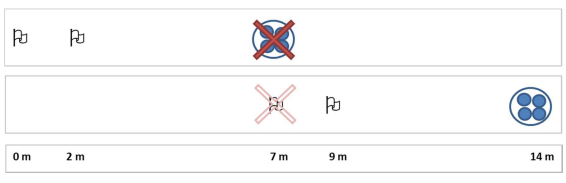
- Marker of depredated nest should always be DISCARDED and put away from clean markers!
- Use same egg provider at regional scale at least: only one provider in Russia (3 sites; coordinator A. Sokolov), 1 in Canada and possibly Alaska (4-5 sites; coordinators Giroux/Lecomte/Bêty) and 1 for Greenland and Sweden (4 sites; coordinator J. Lang)
- Rinse all eggs in water and remove or at least wash very carefully any dirty eggs (e.g. in boxes were some eggs were broken), or eggs obviously smelling strong (this can also happen…).
- Wear nitrile gloves when handling eggs & don't put your knees/leave your bags on the ground close to the nest

**Sticks**: we agreed that we should all place the sticks at 5 & 7m from artificial nests (both sticks aligned in order to find the direction of the nest).

**Change of nest locations between the 2 experiments**: the same location can be used for unpredated nests for the 2 experiments. For predated nests (because foxes might have urinated in the nest), a new nest cup must be prepared at least 7m from old/predated one. We use 7m so that you can just use the old nest cup as the new position of the farthest (7m) stick. By doing so, you don't have to record a new GPS position and change your "waypoints/route". The only difference during the 2nd experiment will be that for positions were nests got predated during the first experiment, your GPS route will bring you to the stick instead of to the nest.

1st experiment (predated):

2nd experiment:




**Additional info**: If you have evidence of heavy predation by a given predator species and for a given session (e.g., raven 2nd session Igloolik), please note such details.

# Protocol #2:
# Monitor real predation pressure on *Calidris* nests using Tiny Tags

**(COORDINATOR: OLIVIER GILG)**

*VERY IMPORTANT: (1) as for artificial nests (and even more important for real nests), don't put your knees and don't leave anything on the ground (except tiny tags), even for short periods, within 20m of the nest, (2) nests are marked with a stick or stone pile 10m from the nest (e.g. opposite an obvious landscape feature or always in the same geographic direction)*

Our intention is to find at least 20 nests of each target species at each site.

Give each nest a unique nest number including: species ID, site initials, year and nest number: DUNL_ERK_2016_01, DUNL_ERK_2016_02, etc.

Record the exact coordinates for each nest (in decimal lat/long).

Make sure that you can find the nest again: coordinates alone in your GPS may not be sufficient! Especially when the clutch is taken by a predator, the nest cup can be very difficult to find. In Greenland (where we seldom have fog!), we place a marker (white stick or small but obvious pile of stones) at ten meters (or large footsteps) away from an obvious landscape feature (station mast, mountain summit, etc). That implies that during a next visit one should take ten steps from the marker in the direction of the station (or another obvious element in the landscape) to find the nest cup. Especially predated nests or nests on which the incubating bird sits tightly are sometimes very difficult to relocate. Avoid markers that might easily lead predators to the nests! In Canada a second stick is placed a few meters away from the first, to give the direction of the nest.

Place the probe of a Tiny Tag in the nest cup (Fig. 4). The probes should be fixed (with a small band of duct tape) to a small (ca. 10-20cm long according to the softness of the terrain) wooden peck (toothpick is definitely too short) that can be stuck in the middle of the nest cup. The probe with thermistor should be in the middle of the 4 (3) eggs so that it does not hamper the incubating bird, but that the thermistor touches the brood patch until the end of the incubation period. Take the eggs out of the nest cup when you place the probe to avoid any possible damage to the eggs! Hide the wire of the probe and the Tiny tag itself as good as possible to avoid it attracting avian predators. The data-logger itself can usually be hidden under a (few) flat stone(s) or dug (using a knife) superficially into the ground (especially in the wet Dunlin habitat), in a crevice in the nearby ground.

The Tiny Tags should be programed to record temperature every minute. Recording time will – automatically- be the same as on the computer used to start the tiny tags; SO PLEASE SET YOUR PC on LOCAL TIME before starting the tiny tags or advise us if you didn't). This must, of course, be done before leaving the base camp with Tiny Tags ready to install each day. To prevent using too much battery power (and too much of the memory of the logger), stop the logging upon return if not used that day. Keep a record of which Tiny Tag is placed in which nest and when (each Tiny Tag has a unique ID written on it).

Remember that some old Tiny Tags have only a memory of 14 days, so they need to be replaced with a new Tiny Tag before the memory is full. New ones will record for ca. 22 days which covers the entire (remaining) incubation period of most nests we will find and monitor.

If you don't have enough Tiny Tags: for accurate estimation of daily nest survival, return to each clutch once every 3 days (and every 2 day before expected hatching date): this is to make sure eggs have hatched or suffered predation just before expected hatching date (floating eggs is then mandatory).

Figure 4: Placing the Tiny Tag: a summary. The probe (red) is attached to a small (ca. 10-20 cm) wooden stick (blue) with a small stripe of duct tape. The wooden stick must then be pushed/fixed through the entire nest lining (e.g. made of dry Salix leaves) in order to find more solid support (in wet mossy Dunlin habitat: use longer sticks). If not, the probe will slowly drop down during incubation and recordings will become useless (to be effective the probe must touch the brood patch of the incubating adult). So very important: the top of the probe must be at the same level (green dashed line) than the top of the eggs once fixed. However, you must remove the eggs before pushing the stick down to prevent damaging the eggs (you may have to place and remove the eggs 2-3 times until you get the right fit).

Once the probe/stick are adjusted, (1) hide the data logger (blue) at ca 50cm of the nest, under a stone or a piece of ground/moss and then (2) hide the probe's cable (red) in the vegetation or under 1-2cm of soil so that it cannot be seen by potential predators (an easy way to do that is to cut the soil with an old knife between the nest and the data logger, to push the cable in this small crack and then gently push both sides together again… don't forget to wear nitrile gloves when doing that!).

We also posted a small "field" video at this link in case it can help: https://youtu.be/28IUxmL2NCI)

Once the Tiny Tag is in place and unless incubation is expected to last longer than 22 days (see "flotation of the eggs" below) or you want to trap/ring adults or young, it is - in theory - not necessary to return to the nest until the expected end of incubation to collect the Tiny Tag (keeping the number of nest visits as low as possible will reduce disturbance and anthropogenic predation risk).

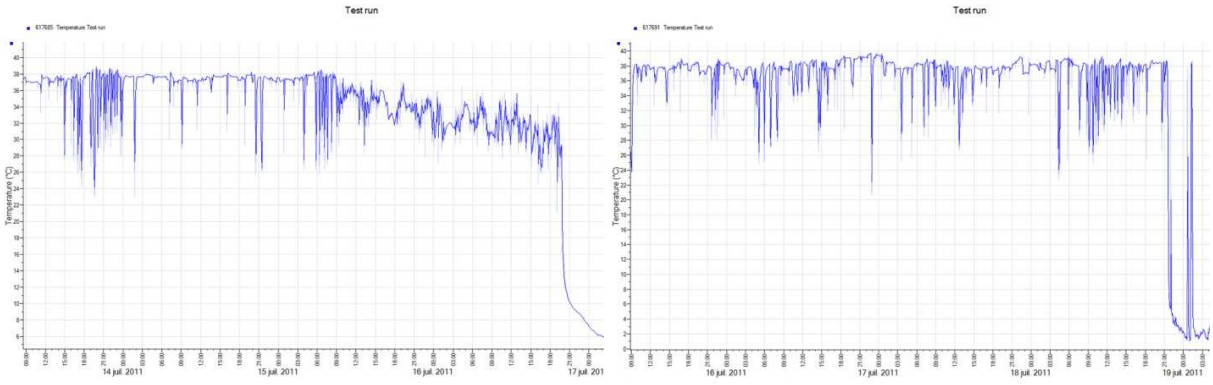
However, we advise that nests are revisited once after 1-2 days to check if the probe is still properly positioned (aligned with the eggs' top as shown by the green dashed line Fig. 4).

Subsequent visits are not mandatory but checking for the absence of incubating birds from the distance (e.g. from the marker at 10m) will allow you to collect Tiny Tags in predated/hatched

nests and to “recycle” them on new nests in case you miss Tiny Tags (in this case make sure to download the data collected on the 1st nest and to reprogram the Tiny Tag before deploying it on the 2nd nest).

We don’t give details here about how to start, download and stop the loggers using the software “TinyTag explorer”, but below are just two figures illustrating a nest that hatched (left) and produced young (evidenced by the slightly declining temperature during the final 36h of incubation), and (right) a nest that got predated (temperature drops dramatically within a minute; later short peaks in temperature just indicate that an adult came back and sat on the empty nest).

Make sure you have it installed on the computer you plan to use in the field and keep a copy of the installation disk with you in case you have to reinstall it!!!




During each nest visit note:

- Date and time of visit (use local time or give details in the form if you don’t)
- How many birds were attending? What was their behaviour (incubating, alarming, etc.)?
- Was/were the bird(s) colour-ringed, and if so, what was the combination? (important for species like Sanderling, Western and Baird’s Sand., that can be either uni- or bi-parental)
- How many eggs were in the nest cup?
- What was the status of the clutch (eggs were warm, cold, with cracks, with a small hole, hatching or hatched (how many chicks?) or likely suffered predation).

If predated, try to find out the possible predator (foxes often urinate or defecate in the nest cup, avian predators might leave egg shell remains with an obvious hole, etc.). If unsure, note any signs that might help solve the mystery: shape and position of the hole, egg shell bits hanging into the hole (in successful nest tiny egg shells can often be found in the bottom of the nest cup, etc). It is often useful to study carefully nests of known fate; e.g. a nest that you knew hatched. Train your eye to see what typical egg shell fragments from a hatched nest are. Similar if you know for certain a nest has been predated (observed predation), what does the nest/egg shell characteristics look like? Does it smell fox? In other words, train! (ADSN manual also gives very interesting and precise information regarding this point: www.manomet.org/sites/default/files/publications_and_tools/ASDN_Protocol_V5_20Apr2014.pdf)

***Flotation of the eggs***

For some analyses it will be important to know the date when incubation started and floating the eggs is how most of us already indirectly collected this information.

Material you need is limited:
1. Float container (e.g. 3 inch Plexiglas cube) with compass angles and millimetre scale written on side (see fig. below)
2. Tepid water (ca. 25 °C) carried in a thermos container (we usually take hot water in the morning and mix it with cold water once in the field)

Methods (see additional info in ASDN protocol if needed):

If a shorebird nest contains a full clutch of eggs (typically 4) when it is discovered, float at least 2 eggs. If the 2 eggs differ in angle significantly, float the 3rd egg (and 4th if necessary).

1. Place eggs on the bottom of the jar before releasing to prevent egg damage from dropping and to ensure they are not held by surface tension. Float each egg separately.
2. With a protractor (or from the scale on the container), measure the angle between the bottom of the cup and the centre axis of the egg to the nearest 5° for each egg (see Fig. below).
3. If the egg floats at the surface, using a ruler (or the scale on the container), record the # of millimetres above the water surface that is exposed to the air and also record the angle of the egg in the water column. Keep in mind the egg may float at the surface but not break the surface. Record these measurements while you are viewing the floating egg at eye level.
4. Carefully place eggs back in the nest (remember to record the date you floated the egg, especially when different from "nest discovery date" or date when you placed the Tiny Tag!)

Note that between the stage of standing on the bottom at 90° and starting to float at the surface, "weightlessness" occurs.
If the temperature is below freezing do not float the eggs. Do it on the next visit.

For our project you only need to float eggs **once preferably on the first (discovery) visit (**because the nest may be depredated on your next visit and age estimates tend to be more accurate when eggs are floated early in incubation).

*Inferring hatching date*

For Canada/Alaska sites/species: preferably use Float Tables in **Appendix J of the ASDN protocol to infer hatching date.**
Note the tables are designed so that the lowest angle that can be considered is 21° (shorebird eggs are oblong and will never lay flat on the bottom).

For Greenland and Sweden (Dunlin and Sanderling incubate for ca. 22 days), you can use the figure below (prepared by J. Reneerkens for Sanderling).

For Russia (Temmink's and Little Stints) or any other species with known incubation duration, you can use the average values inferred from Liebezeit's paper and presented on the next page.

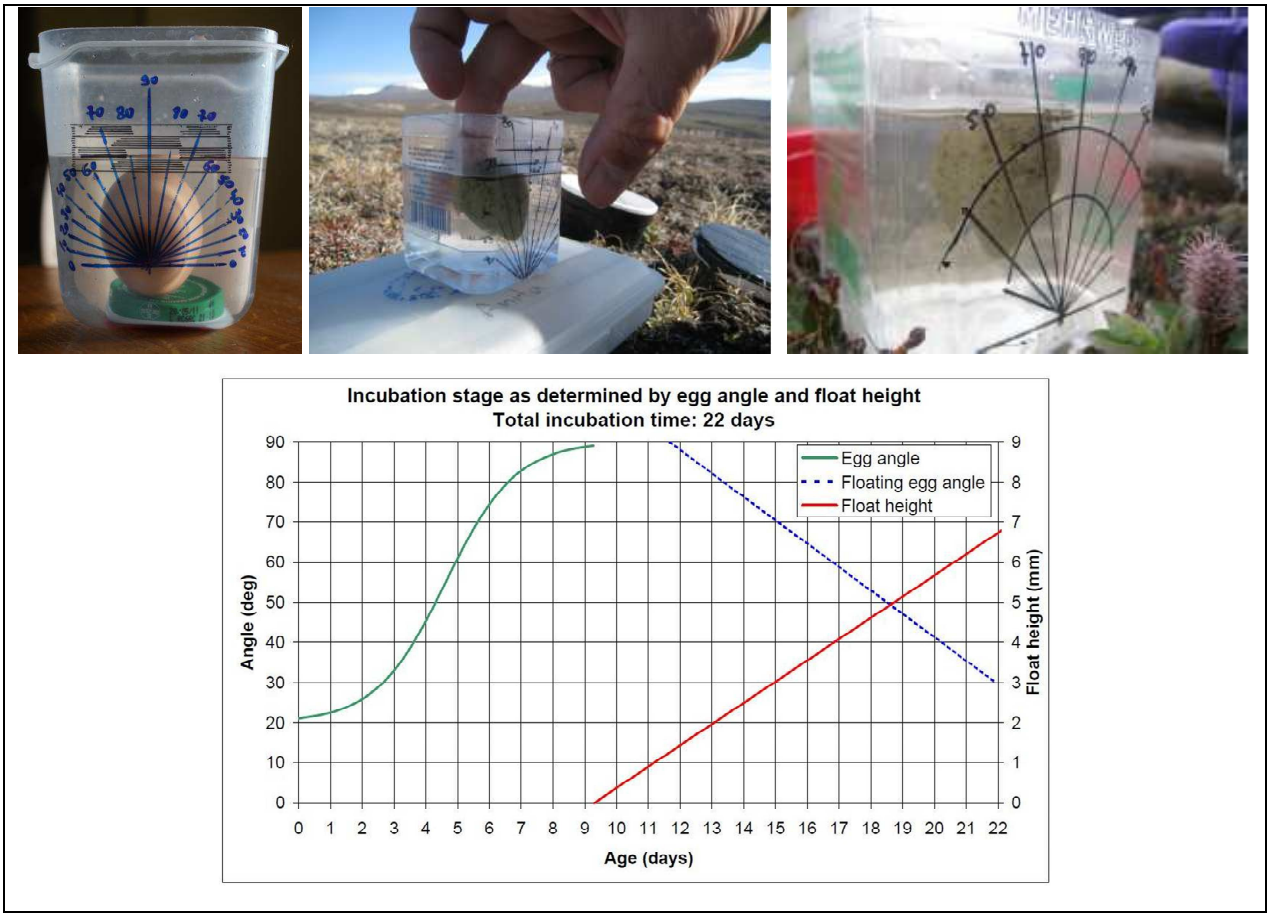



Figure 5: Pictures and graph illustrating the flotation method.

Results of the monitoring of real nests (one file per species) will be entered in the file named
**"P2_Real Nests Monitoring_SITE_YEAR_SPECIES.xlsx"**

| Species | | Duration of incubation | | | | | |
|---|---|---|---|---|---|---|---|
| | | 18 | 19 | 20 | 21 | 22 | 23 |
| ***Calidris alpina*** | Dunlin | | | | | | |
| ***Calidris alba*** | Sanderling | | | | | | |
| ***Calidris minuta*** | Little Stint | | | | | | |
| ***Calidris temminckii*** | Temminck's Stint | | | | | | |
| ***Calidris bairdii*** | Baird's Sandpiper | | | | | | |
| ***Calidris fuscicollis*** | White-rumped Sandp. | | | | | | |
| ***Calidris pusila*** | Semipalmated Sandp. | | | | | | |
| ***Phalaropus fulicarius*** | Red Phalarope | | | | | | |
| ***Phalaropus lobatus*** | Red-necked Phalarope | | | | | | |

| Position | Egg angle | mm above | % completed | Nb of days of incubation left | | | | | |
|---|---|---|---|---|---|---|---|---|---|
| bottom | 21 | 0 | 0,02 | 17,7 | 18,7 | 19,7 | 20,7 | 21,6 | 22,6 |
| | 25 | | 0,08 | 16,7 | 17,6 | 18,5 | 19,4 | 20,4 | 21,3 |
| | 30 | | 0,12 | 15,9 | 16,8 | 17,6 | 18,5 | 19,4 | 20,3 |
| | 35 | | 0,15 | 15,4 | 16,2 | 17,1 | 18,0 | 18,8 | 19,7 |
| | 40 | | 0,17 | 15,0 | 15,8 | 16,7 | 17,5 | 18,3 | 19,2 |
| | 45 | | 0,18 | 14,7 | 15,5 | 16,3 | 17,1 | 18,0 | 18,8 |
| | 50 | | 0,20 | 14,4 | 15,2 | 16,0 | 16,8 | 17,6 | 18,4 |
| | 55 | | 0,22 | 14,1 | 14,9 | 15,7 | 16,5 | 17,2 | 18,0 |
| | 60 | | 0,23 | 13,8 | 14,6 | 15,4 | 16,1 | 16,9 | 17,7 |
| | 65 | | 0,25 | 13,5 | 14,3 | 15,0 | 15,8 | 16,5 | 17,3 |
| | 70 | | 0,27 | 13,2 | 13,9 | 14,7 | 15,4 | 16,1 | 16,9 |
| | 75 | | 0,29 | 12,8 | 13,5 | 14,3 | 15,0 | 15,7 | 16,4 |
| | 80 | | 0,31 | 12,3 | 13,0 | 13,7 | 14,4 | 15,1 | 15,8 |
| | 85 | | 0,36 | 11,6 | 12,2 | 12,9 | 13,5 | 14,2 | 14,8 |
| | 89 | | 0,45 | 9,9 | 10,5 | 11,0 | 11,6 | 12,1 | 12,7 |
| weightless | | | | | | | | | |
| top | 90 | 0 | 0,42 | 10,4 | 11,0 | 11,6 | 12,2 | 12,8 | 13,3 |
| | | 1 | 0,48 | 9,4 | 9,9 | 10,4 | 10,9 | 11,4 | 12,0 |
| | | 2 | 0,55 | 8,1 | 8,6 | 9,0 | 9,5 | 9,9 | 10,4 |
| | | 3 | 0,62 | 6,8 | 7,2 | 7,6 | 8,0 | 8,4 | 8,7 |
| | | 4 | 0,68 | 5,8 | 6,1 | 6,4 | 6,7 | 7,0 | 7,4 |
| | 80 | 0 | 0,46 | 9,7 | 10,3 | 10,8 | 11,3 | 11,9 | 12,4 |
| | | 1 | 0,53 | 8,5 | 8,9 | 9,4 | 9,9 | 10,3 | 10,8 |
| | | 2 | 0,59 | 7,4 | 7,8 | 8,2 | 8,6 | 9,0 | 9,4 |
| | | 3 | 0,66 | 6,1 | 6,5 | 6,8 | 7,1 | 7,5 | 7,8 |
| | | 4 | 0,73 | 4,9 | 5,1 | 5,4 | 5,7 | 5,9 | 6,2 |
| | | 5 | 0,79 | 3,8 | 4,0 | 4,2 | 4,4 | 4,6 | 4,8 |
| | 70 | 0 | 0,50 | 9,0 | 9,5 | 10,0 | 10,5 | 11,0 | 11,5 |
| | | 1 | 0,57 | 7,7 | 8,2 | 8,6 | 9,0 | 9,5 | 9,9 |
| | | 2 | 0,63 | 6,7 | 7,0 | 7,4 | 7,8 | 8,1 | 8,5 |
| | | 3 | 0,70 | 5,4 | 5,7 | 6,0 | 6,3 | 6,6 | 6,9 |
| | | 4 | 0,77 | 4,1 | 4,4 | 4,6 | 4,8 | 5,1 | 5,3 |
| | | 5 | 0,84 | 2,9 | 3,0 | 3,2 | 3,4 | 3,5 | 3,7 |
| | | 6 | 0,90 | 1,8 | 1,9 | 2,0 | 2,1 | 2,2 | 2,3 |
| | 60 | 1 | 0,61 | 7,0 | 7,4 | 7,8 | 8,2 | 8,6 | 9,0 |
| | | 2 | 0,68 | 5,8 | 6,1 | 6,5 | 6,8 | 7,1 | 7,4 |
| | | 3 | 0,74 | 4,6 | 4,9 | 5,1 | 5,4 | 5,6 | 5,9 |
| | | 4 | 0,81 | 3,4 | 3,6 | 3,8 | 4,0 | 4,2 | 4,4 |
| | | 5 | 0,88 | 2,2 | 2,3 | 2,4 | 2,6 | 2,7 | 2,8 |
| | | 6 | 0,94 | 1,0 | 1,1 | 1,1 | 1,2 | 1,2 | 1,3 |
| | 50 | 1 | 0,65 | 6,3 | 6,6 | 7,0 | 7,3 | 7,7 | 8,0 |
| | | 2 | 0,72 | 5,1 | 5,3 | 5,6 | 5,9 | 6,2 | 6,5 |
| | | 3 | 0,79 | 3,9 | 4,1 | 4,3 | 4,5 | 4,7 | 4,9 |
| | | 4 | 0,85 | 2,7 | 2,8 | 2,9 | 3,1 | 3,2 | 3,4 |
| | | 5 | 0,92 | 1,4 | 1,5 | 1,6 | 1,7 | 1,8 | 1,8 |
| | | 6 | 0,99 | 0,2 | 0,3 | 0,3 | 0,3 | 0,3 | 0,3 |

# Highlights & Updates "P2_Real Nests Monitoring":

**Sample sizes** were small on some sites or for some species so we need to increase our effort in 2017 or at least maintain it. Remember that a sample size of 20 nests (per year/species/site) is the target but, of course, duration of the monitoring also matters (better 15 nests monitored for 20 days than 20 monitored for only one week; total monitoring days*species*site*year= 300 days in the first case but only 140 the second…).

**On several sites, the recording of nest temperature was not optimal** (no clear "plateau" seen in the time series around 38-40°C). Hopefully these were sites where nests were also regularly visited and hence when the temperature recording was not good enough to discriminate between hatching or predation, we could also use data from nest visits (and egg flotation) to infer the fate of these nests.

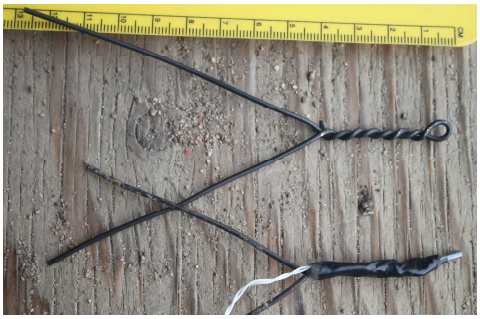
To improve this in 2017 will be easy since in most if not all cases, this problem is only due to probes positioned too low in the nest.

1. If the probe was intentionally positioned too low and did not touch the brood patch of the incubating bird(s), then you should just put it higher in the nest (see figure 4 in manual).

2. If the probe was "sinking" in the nest during the incubation period, then using longer sticks will in most cases solve the problem. In dry habitats (Sanderling, Baird…), sinking happens if the stick is too short or the soil too sandy (Temminck). But in wet/mossy habitat (Dunlin), this happens almost all the time unless you use a very long (up to 20cm or more in very mossy areas) stick. Also, in some case (e.g. early Dunlin/Phalarope nests on flooded hummocks), the stick sometimes touches the concrete-hard permafrost and later sinks with melting permafrost (in this later case there is not much you can do except revisit the nest after a few days to place a longer stick).

To know if you stick/probe won't sink, push it down carefully with your little finger (as a 50g bird would with its brood patch…) once it is in place (but before replacing the eggs!) If it does not easily sink further, then it should be fine. I never tested it but you can also imagine carefully placing a 200g weight (as max. pressure from an incubating Calidris?) on the top of the probe to check if it is stable…

On Bylot was also tested a "two legged" metal sticks instead of our simple "one legged" wooden sticks (Fig. below). This might help if you have only no wooden sticks, frozen terrain just below the nest cup, or very soft terrain. But as discussed above, simply using longer sticks will solve most problems and is much easier to place in the nest cup. So we don't currently advise the use of such metal sticks unless you have very good reasons to do so. Indeed, placing such "double sticks" could (if poorly placed) damage the nest material. Also, because the thermal conductivity of metal is much higher than for wood, the recorded T°C can be impacted by the ground temperature (which might make inter-nests or inter-sites comparisons associated with temperature-related questions difficult or even impossible in future analyses). So if you have no other choice than to use this design, make sure to "insulate" the probe from the 1.25mm metallic wire with electric tape (ca. 3-4 layers). This will also prevent the eggs to be cooled down or damaged by direct contact with the metal (to avoid!!!).



# Protocol #3: Observations of predators and lemmings

**(COORDINATOR: MARIE-ANDRÉE GIROUX & OLIVIER GILG; MSC STUDENT: AUDREY BÉDARD)**

Data on predators will be central in the planned analyses, because one cannot understand how indirect interaction (between lemmings, subsidies and alternative prey; dashed lines Fig. 1) work without knowing first how these prey type directly interact with predators (lines Fig. 1).

Ideally, we should collect all data needed to describe the numerical and functional response of predators to available resources (see e.g. Gilg et al. 2006 Oikos, Therrien et al 2014 Ecology). This would, however, require important field work that most teams can't afford…

Hence, we ask you to collect and report 3 types of "simplified" information on predators:

1. Record incidental observation of predators AND lemmings (mandatory to answer most research questions included in this project);

2. Collect 20-50 fox scats per year on every site (to be analysed later using DNA barcoding) to assess the functional response of this major nest predator (very important to answer some of our research question and not very time consuming for field teams)

3. Forward any other available information on predators' numerical (or functional) responses that will help us to understand and discuss the specific patterns observed on each site.

## 1. Incidental observations of predators **and lemmings**

*Protocol modified from the ASDN (Brown et al. 2014) and INTERACTIONS (2014) protocols*

Incidental observations of all vertebrate predators and lemmings will be used to create an index of predator and of lemming relative abundance at each study site, which will allow investigating the influence of predator and lemming abundance estimates on predation risk between years and sites. Incidental observations of lemmings will provide a comparable index of lemming abundance between sites, complementary to lemming density estimates collected through traditional methods (snap-trap, live trapping; see section "Indexes of lemming abundance" below).

### Count method

Counts will be conducted daily by one or several observers. Designated counter(s) will record observations of all predators and lemmings in their study area throughout the day regardless of their primary activity when the observer is alert (e.g. during nest searching, and/or environmental monitoring but NOT during captures).

Observers will count independently **(never pool results from several observers)** and make efforts to minimize double-counting (see below). If two observers work together in the field or are in the same area, only consider the observations for one of them. If multiple observers count independently during the same time period, it is critical to record these observations on different lines in the data form and file.

Predator and lemming counts will only occur when people are within the study area and should not include observations gathered during transportation to and from the study area (i.e., if you stay outside the study area) nor incidental observations at camp (some predators are attracted by camps).

In 2018, we are requesting additional information to better assess the possible differences in detection ranges of predators between sites. This will be done differently for foxes and avian predators, as described here. **FOX:** For each week we request you to record the approximate distance and time at which 10 foxes were first seen.

**AVIAN PREDATORS:** For each week we request you to record the approximate distance and time at which 10 avian predators (all species combined) were first seen.

If you see a group of the same species (e.g. 10 parasitic jaegers together), please enter only one detection distance and time. This additional information should be reported on the form **"P3_Incidental observations_Fieldform_SITE_2018_OBSERVER"** where new columns have been added (see recap Figure at the very end of this P3 chapter).

Other guidelines:

- The total observation time at each site during the whole field season should sum up to a minimum of 200 hours. For lemmings, this has been estimated as the required effort to obtain a good correlation between live trapping and incidental observations data.
- In addition to counting the predators that you see, also count the predators that you hear.
- Count an individual twice if you see it a second time after not having seen it for at least 20 minutes. However, do not count this individual twice if you see it continuously for 20 minutes or more.
    - E.g. If you see a fox at 15:10 and again a fox at 15:35 (but you did not follow it between 15:10 and 15:35), count 2 foxes even if it's possible that this was the same individual. Recording the intensity of predator activity is more important than abundance for this protocol aiming at estimating the predation risk.
- For birds, do not count incubating avian predators
- For foxes, only count the adult size individuals (i.e. foxes capable of hunting), there are the one responsible for predation (but mention smaller sized pups and weaned young in the comments)
- For lemmings, you do not have to identify the species but, of course, record the exact species if you can identify it.
- Don't count the individuals that are obviously not using the "terrestrial" habitats (e.g. Arctic skua following the coastline, Glaucous gulls resting in a delta or flying up and down large rivers) or are not really using it (e.g. gulls on a landfill).
- For aggregation or group of species, record the number of individuals which is the most realistic rather than recording a range of values (we prefer "ca. 75" than "50-100" or ">50").

Incidental observation and detection data will be entered in the file named **"P3_Incidental observations_Fieldform_SITE_2018_OBSERVER"** (some modifications have been made since the 2017 version)

## 2. Sampling of fox scats for fox diet and response (to be assessed through DNA barcoding)

Foxes (Arctic but also Red) are likely to be the most important predators to explain variation in predation pressure and rates between sites and years. Documenting the direct interaction between them and prey (box 3b on Fig. 1) is very important… but also quite difficult! Indeed, traditional methods to describe the functional response of predators/foxes are both time consuming and (often) imprecise. To make it both easy and quick for us to document a rough estimate of the functional response of foxes across sites and years, **we only ask you to collect 20 (minimum) to 50 (maximum) fresh fox scats per site and per year (of known species, Arctic and/or Red).** The plan is then (if funded) to describe the summer diet of each fox population using DNA barcoding (method currently developed with scats from the 3 Greenland sites from 2011-2015).

*Guidelines for sampling:*

1. Collect only fresh scats (according to bleaching/moulding)
2. Collect from any place within your study area (fox den, prey carcass, predated nest, erratic rock…), the more collection places the best!
3. Dry the scats if necessary before storing them (avoid collecting e.g. after or during rain)

## 3. Any available information on the "Numerical responses of breeding predators"

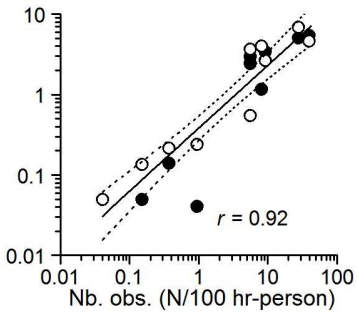
If the monitoring of (some) breeding predators is already part of your field work, or if you think it would not require much extra work (at least for some predator species), then please provide us with standard information on breeding densities and breeding success as presented in the figure below (for more details see Gilg et al. 2006 Oikos).

| Demographic variable | 1988 | 1989 | 1990 | 1991 |
|---|---|---|---|---|
| Snowy owl (75 km²) | | | | |
| No. of breeding pairs | 0 | 6 | 12 | 0 |
| Mean clutch size | - | ≥ 4 | 6,7 | - |
| Breeding success (mean no. of fledglings produced per ne | - | 3,5 | 5 | - |
| Hatching date of first egg (median date) | - | 30 Jun | 16 Jun | - |
| Long-tailed skua (15 km²) | | | | |
| No. of territorial pairs | n.d. | n.d. | ≥ 13 | n.d. |
| No. of breeding pairs | 0 | 8 | 12[b] | 0 |
| Mean clutch size | - | 1,75 | 1,77 | - |
| Breeding success (% of eggs producing fledglings) | - | ≤ 57 | ≤ 73 | - |
| Hatching date of first egg (median date) | - | 19 Jul | 12 Jul | - |
| Arctic fox (75 km²) | | | | |
| No. of dens with reproduction (no. of dens checked) | 1 (4) | 1 (4) | 6 (7) | 0 (6) |
| Mean no. of weaned young per litter[d] | 0 | 6 | 3,5 | 0 |

# Highlights & Updates "P3_Incidental observations"

For most of us, recording incidental observation sounds like something scientifically very weak. However, for some of the most important species in our project (i.e., foxes and Lemmings…) these standardized data will likely be the best available proxy we have at hand at the end of this project for all sites and all years.

For many sites for ex., we already know that fox numbers recorded on breeding dens don't fully inform us about inter-annual changes



in abundance. In years following lemming peaks, many non-breeding foxes are recorded by "incidental observation" protocols but not by the monitoring of the dens…
For lemmings also, incidental observations are highly correlated (e.g. at Bylot) with the most expensive and time-consuming trapping methods (see unpublished figure to the right: nb of observations (X axis) versus number trapped (Y axis), black=mesic, open=wet; Fauteux et al. 2018).

It is hence very important that these data are collected by all of us, and also in a standardized way.

While cleaning and synthetize all data collected for this protocol in 2016 and 2017, we identified several important problems that have to be fixed. For future analyses, we will definitely need these unique data (on predators AND lemmings). If they are not following the protocol described above, or not available for some sites or years, then unfortunately we might not be able to include this/these sites in the study/papers.

Among the improvements needed:

1. Every day spent on the field by the dedicated trained observers must be reported in the "field form", even if you did not see a given species (then report "0" value). Please also report days when the dedicated observers were not on the field (report "."), but indicate that no observations were conducted (see examples in the "**P3_Incidental observations_Fieldform_SITE_2018_OBSERVER"**), otherwise, it will artificially bias estimates downward. In most data tables received in 2016 and 2017, we have missing days and don't know if none of the target species was recorded that day or if no observation occurred (rainy day with no field work?);

2. Daily counts should be independently recorded by 1 or (ideally) 2 dedicated "trained" observers (i.e., not by students who are not experienced with local species). The field form must be filled separately by each person following the protocol and, in most cases, this should not take more than a few minutes per day (except if some species are so abundant locally). To avoid bias and loss of critical information, we strongly recommend avoiding relying on your memory and filling in the data only back at the camp every evening). You should either note observations and hours directly on the field form while in the field or transfer them (from your notebook, voice recorder, etc) on the field form in the evening when back at the camp;

3. Never pool counts from distinct observers! Our protocol has nothing to do with e.g. a "daily species list" for the study site. So if you just ask everybody at dinner what they have seen and report this number in our file, it will be completely useless!

4. In the 2016 version of the protocol we mentioned that counting of common species should stop when >20. This has been deleted. Please give the best possible estimate without considering any threshold value; it's easier to consider an estimate like "approx. 150" than just "more than 20".

5. To help you clean your data before sending them to us in August, we created a data-cleaning guide for this protocol. Thanks to use this guide that you will find in the "data-cleaning-guide" sheet of the file "**P3_Incidental observations_Fieldform_SITE_2018_OBSERVER"**.

Additions to the 2017 and 2018 versions of the protocol:

6. In 2018, we are requesting additional information to better assess the difference in detection of predators between sites (different approaches for foxes and avian predators). Please consult the addition to the protocol (above) and record the information requested in the new columns of the file **"P3_Incidental observations_Fieldform_SITE_2018_OBSERVER "**.

7. The total observation time at each site during the whole field season should sum up to a minimum of 200 hours. For lemmings, this has been estimated as the required effort to obtain a good correlation between live trapping and incidental observations data.

8. In addition to counting the predators that you see, also count the predators that you hear.

9. We also request you to record the time when the observer is not alert between start.time and end.time (e.g. lunch time). A new column "break.time" has also been added to the file **"P3_Incidental observations_Fieldform_SITE_2018_OBSERVER "**.

**PLEASE USE THE NEW VERSION OF THE FIELD FORM THIS YEAR**

**"P3_Incidental observations_Fieldform_SITE_YEAR_OBSERVER.xlsx"**

**REQUIRED FOR EVERY SITE**

NEW IN 2018

Predator detection data for:

- 10 foxes or max number observed/week
- 10 avian predators or max number observed/week

| ak.time | location | fox.time | fox.distance | avian.time | avian.distance | glgu (Glaucous Gull) | poja (LPomarine Jaeg |
|---|---|---|---|---|---|---|---|
| :30 | sola | 16:18 | 30 | 11:50-14:30-15:23-16:45 | 200-150-40-400 | (30) | 2 |
| . | . | . | . | . | . | . | . |
| :00 | sola | . | . | 11:30-14:55 | 100-150 | 0 | 0 |
| :00 | wpw | . | . | . | . | 0 | 0 |

**FACULTATIVE FORMS THAT CAN BE USEFUL**

**"P3_DETECTION-summary_2018.docx"**

Will help you follow the number of foxes and avian predators for which detection data have been collected per year

**"P3_Incidental observations_Small-Fieldform_2018.docx"**

If you wish to have a smaller version of the fieldform to put in notebooks

# Protocol #4: Index of lemming abundance

**(COORDINATORS: MARIE-ANDRÉE GIROUX & NICOLAS MEYER)**

The majority of collaborators, who will participate to this project, already estimate lemming abundance during summer through a variety of methods (snap-traps and/or live trapping, winter nest counts, etc). The current study does not require standardizing those methods that have been used for many years at many sites. We will rather use lemming density estimates that will be obtained through live-trapping and will derive a density estimate from snap trapping data using the conversion equation given in Krebs et al. 2002 and Gauthier et al. 2005. Sites that are not conducting lemming sampling are encouraged to do so, either by implementing live-trapping grids or snap trap transects (see "Technical manual for sampling small mammals in the Arctic": http://www.cen.ulaval.ca/bylot/files/Small_Mammal_Sampling_v1.pdf).

Lemming data will be compiled differently at different sites and the density estimates (live trapping) or the snap trap index (snap trap) will be entered in the file named
**"P4_Lemming index_SITE_YEAR.xlsx"**

## 2017 Updates on "P4_Lemming Index":

Collecting specific lemming data (from live-, snap-trapping or any other method; P4) does not mean you should not record incidental observations in the previous (P3) protocol. Observation from P3 will constitute the only fully comparable (standardized) rodent data set and it is hence of great importance (also because it is highly correlated with trapping data as shown by the figure on the previous page).

# Protocol #5: Index of herbivore abundance

**(COORDINATOR: OLIVIER GILG)**

**VERY IMPORTANT in 2018:** After a thorough evaluation of this protocol, based on data collected on our 12 sites in 2016-2017 and expertise from elsewhere, we concluded that:
* this protocol wasn't suitable for monitoring inter-annual changes in herbivores' dynamics;
* 10 transects were not enough to get a representative coverage of each study area;
* but 30 meters was apparently more than what was needed to get a representative sample of pellets at the transect position.

**We hence decided to sample ≥30 x 10m-long transects (instead of ≥10x30m-long transects)**

This won't change the sampling time for new sites. For the 12 initial sites where 10 transects of 30 meters have already been sampled, we are hence asking to sample ≥20 additional transects of 10 meter long (or less if >10 transects have been sampled in the past years, as long as we end up with 30x10m-long transects in total!)

**Why?**

The protocol is intended to serve as a tool in obtaining an *estimate of herbivore relative abundance comparable between tundra areas and research and monitoring sites*. This protocol aims at giving an *area representative* estimate of herbivore abundance, independent of site-specific habitat classifications.

**What?**

Faecal standing crop gives amount of accumulated faeces (smaller "pellet removal plots" can give year-to-year or seasonal variation but are no longer considered and required by IWG).

The study takes place in a set of tundra regions and includes 30 replicates per site assumed to cover and be representative of main habitats. Defecation rates are assumed to be similar within herbivore species between regions, while decay rates is probably "climate/site specific" (we won't investigate this on each site but hope to get some methodological insights on this question from studies conducted elsewhere).

**How?**

- Locate 30 points (or more) that will serve as random transect starting points:
    - Either spread points totally even in the area you aim at covering (i.e. your study area or, if too large, the same 5-10km² area where you implement the artificial nest protocol), or allocate random in blocks
    - leave out areas that would not be part of the herbivore abundance estimate such as glaciers or water bodies and, in order to get area representative estimates, we want to avoid stratification using ecological criteria in this study

- Your ≥30 transects (the more the best but 30 is a strict minimum) will be 10 m long, and should be at minimum 100 m from each other, and preferably further, depending on the size of the study area. If some starting points need to be discarded (see below), just move it 50m to the E or W or move to the next position.
    - The first transect start is at any logistically feasible point. Discard the point if it is in area that you do not want to include in your area estimate. This can vary between sites, and can be e.g. standing water, human disturbance or other cause you consider a reason for exclusion from the final area.
    - If the starting point is suitable for sampling, walk a 10 m transect towards the north. Record start and end point in GPS in the field.
    - Strip transect: Each transect is 1 m wide and divided into $1m^2$ segments. The 1 m strip can be delineated using a rope with marks or with measuring tape, and walked with a 1 m measuring stick hold perpendicular to the transect. Pellets are counted for each 1 m segment along the transects.
    - Photo: take 2 photos on each transect on the first and last 1x1m segment, the photo covering 1x1m, and including the 1m measuring stick on it to give the scale.
    - For each $1m^2$ segment of the transect, count the pellets belonging to each of these 5 species group: geese, hares, ptarmigan/grouse, reindeer, muskox (and ground squirrels if present on some sites?). Don't give "estimated numbers" or categories (e.g. "100-200", or "a few hundreds");
    - For reindeer and muskox we suggest separating between winter pellets and summer dung. For summer faeces it is often possible to see what came out as a "dung" or "pile" even if they are somewhat disintegrated. Note this as "clump/pile" (can later be coded to e.g. value 30 following ongoing methodological adjustments by Herbivory Network).
    - Covariates to be recorded for each transect:
        - *slope* along transect in degrees (If even: once; if slope changes: note a new slope as necessary and note for which meters each slope value is for), use a compass with clinometer if possible;
        - *GPS coordinates in decimal latitude and longitude*;
        - *Height of vegetation at start and end of the transect* (measure to nearest cm the maximum height).

**Time use and precision:** At low-Arctic sites, if you are time constrained, count what is visible without using time to search for pellets in dense vegetation (clearly state in the data form what is the % of the transect that had to be overlooked; and if possible extend the transect in order to compensate these "losses" and end up with a "10m-long equivalent sampled transect").

Each transect is estimated to take max. 0.5h in high-Arctic sites but up to 1h in the low-Arctic (see §6).

Except for a GPS, the only equipment you will need is a 10m-long rope (with 1m marks) and a 1m long stick to control for the width of the transect (see §7).

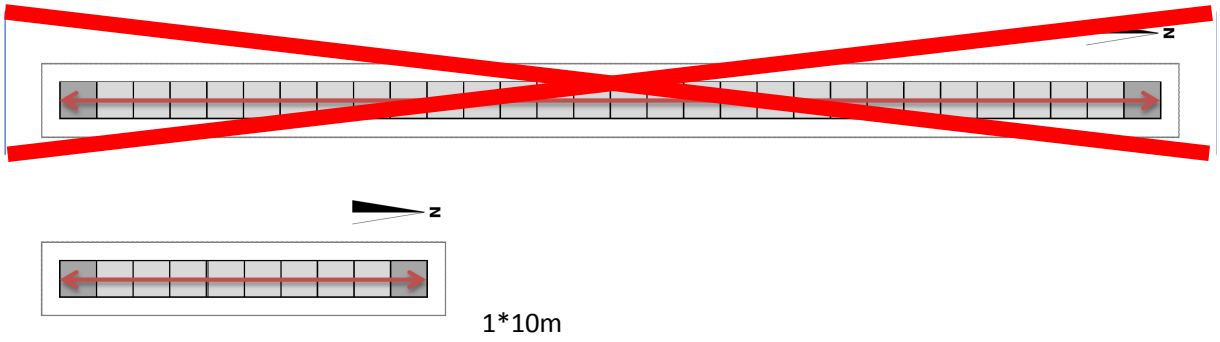



Figure 6: each of the ≥30 herbivore transect is marked with a 10m-long rope (red line) oriented towards the North and made of 10 successive 1x1m segments. On each of the 1st and last segments (dark grey): take a picture or the 1m² segment and measure the max height of the vegetation.

Fecal count data will be entered in the **NEW UPDATED** file named

**"P5_Fecal count_SITE_YEAR_VERSION_AUTHOR_V2.xlsx"**

*Information about other alternate resources (marine, carcasses, human) should also be given in a separate sheet of this file*

# Updates on "P5_Index of herbivory abund.":

**All sites**: Use the new field form (2018) available on dropbox (it was updated for 10m-long transects and has different columns for single faeces or piles).

**New sites:** Implementing this protocol in 2018 (30 x 10m-long transects) is mandatory

**Other sites:** Make sure you end up will data from 30x10m-long transects at the end of your field season. If you already had 30x30m-long transects in previous years, then you are done! If you only had 10x30m-long transects (most sites), then you need to sample an additional 20x10m-long transects this summer (data from different years can only be combined if they were not collected on same transects!)
Following the tests made last year, we acknowledged that monitoring same transects for several years is not suitable to assess dynamics of herbivores. Sites willing to assess such dynamics are encouraged to use different protocols (on smaller grids) but this is no longer an aim for our IWG.

In 2016, most of the sites did not fill the second sheet of the data file (P5_Fecal count_SITE_YEAR_VERSION_AUTHOR.xlsx), except folks working at Karupelv (good job Karupelv's folks ☺). We will mention that specific information in early August in the reminder to send data, but in the meantime, thanks to fill in this second data sheet in the last version of your 2016 data file (Dropbox) and of your new 2017 data file.

## 6. Personnel (indicative: can greatly vary according to study site and personnel skills)

**Protocol #1: Artificial nest manipulation**

The initial set-up (50 artificial nests) will require approximately 3.5 hours of effort for 2 people (7 person-hours).

The first (after 48h) nest visit will require approximately 2.5 hours for 2 people (5 person-hours). The second nest visit (after 96h for eggs removal) will require approximately 3.5 hours for 2 people (7 person-hours). For the two visits, the effort will sum up to 6 hours for 2 people (12 person-hours).

Because the manipulation will be conducted twice during the summer (early and late incubation), the effort for setting up the manipulation will sum up to 19 hours for 2 people or 38 person-hours per year.

**Protocol #2: Monitoring of real nests with Tiny Tags**

On most of the 11 sites listed in Table 1, searching for *Calidris* nests is already part of the basic monitoring and will hence not be estimated here (according to site characteristics, species behaviour and breeding densities, success in finding nest varies a lot, even for a skilled ornithologist: probably from <1 nest per day to >10 nests per day).

If we only consider the additional time needed for each monitored nest to prepare (settings), deploy (place and hide) and recover (at the end of incubation) the Tiny Tags, implementing this protocol probably takes ca. 1 person-hour per monitored nest.

Since our aim is to monitor 20 (if one species) to 40 (if two species) *Calidris* nests per site, total time needed can be estimated at 20-40 person-hour per year.

However, one should also realize that monitoring breeding success (hatched versus predated nests) with Tiny Tags saves a lot of time (not to mention higher accuracy and reliability of results) compared with traditional monitoring (revisiting nests at regular intervals until hatching).

**Protocol #3: Incidental observations of predators and lemmings**

The incidental observation protocol will be done simultaneously to other field work, and will thus add a maximum of 1 person-hour per day to record observations (on data forms directly, notebook or voice recorder) and, eventually, to fill the data form in the evening.

**Protocol #4: Index of lemming abundance**

We assume that this is already part of the work routinely implemented on each study site.

**Protocol #5: Index of herbivore abundance**

Time needed to implement this protocol is also site specific. In the high-Arctic where herbivore faeces will be rare on most transects, counting and removing faeces on a 10*1m transect will usually take less than 1 hour. Hence, after adding a few hours to mark and walk between transects, implementing this protocol should take ≤15 person-hours per year for 30 transects).

In the low-Arctic where vegetation is denser and herbivore faeces more abundant, completing one 10*1m transect could in some cases take 1h, i.e. up to 30 person-hours for 30 transects (if we add a few hours to mark and walk between transects). We advise that in such situations, if time constraints are too strong for the field workers, faeces could be estimated by counting them 10 by 10. Remember that you can work on this protocol at the end of your field season when time constraints are often lower.

Table 3: Summary of the effort (in person-hours) required to implement the different protocols

| Protocols | Total time needed | Distribution of effort | Timing |
|---|---|---|---|
| **P#1 Artificial nests** | 38 person-hours per year | Two sessions of 4 days per year | Early and late incubation periods |
| **P#2 Monit. of real nests** | 20-40 person-hours per year (depending on the number of *Calidris* species monitored: 1 or 2) | Daily search of nests | In priority during first half of the incubation period (to monitor the nests as long as possible) |
| **P#3 Predators/lemmings: Incidental observations** | 1 person-hour per day (for ca. 30-45 days) | Daily | During the shorebird breeding season |
| **P#3 Predators/lemmings: Collection of scats** | (while working on other activities; no specific time) | Continuous | During the shorebird breeding season |
| **P#4 Index of lemming abundance** | (already implemented on most sites) | 2 or more estimates over the summer season | Should cover the entire field season |
| **P#5 Index of herbivore abundance** | 15-30 person-hours according to site | Once | Any time during summer (can be late in the season) |
| **Total per year** | 100 to 150 person-hours according to site and nb of *Calidris* species | | |

# 7. Material

**Protocol #1: Artificial nest manipulation**

- 400 fresh quail eggs (4 per nest x 50 nests x 2 sessions). Extra eggs are recommended in case of breakage during transit. The eggs that were not predated during the first experiment can also be reused, yet the number remaining is unknown.
- A few pairs of nitrile gloves
- 50-60 barbecue wooden skewers to mark the nest and relocate it even if eggs were predated (place a broken half of a skewer in the middle of the nest and push it down so that its top is at the same lever that the eggs); these wooden skewers can replace the 50-60 nails with flagging tape we suggested using in previous years and can also be used (but the you will sometimes need to use an entire skewer) to hold the moss/lichen cover of the 25 covered nests (if your area is exposed to strong winds better use popsicles (below)
- ~30 popsicle sticks (the size of medical tongue depressors) for the covered nests
- 50-100 barbecue wooden skewers for indicating the proximity of every nest (placed at 5 m or 5 and 10m); sticks can be reused for late incubation.
- 25-50 vegetal covers (Fig. 2; 50 if they can be used for both early and late experiments).

**Protocol #2: Monitoring of real nests with Tiny Tags**

- 15-20 Tiny Tags + probes (for one species*site)
- "TinyTag Explorer" and USB connection cable (to program the data loggers and later download the data… so there is also need for one computer!)
- 1 large old knife to cut the ground and place the probe's cable in it (can also be used to remove a piece of ground/moss under which the Tiny Tag data logger will be hidden)
- 1 square, transparent and graduated plastic container to float the eggs and assess laying/hatching dates (see Chapter 2 and further details in ADSN protocols)
- 1 thermos flask to carry warm water in the field (to be used for floating the eggs)
- and 1 GPS, of course, to locate the nests

**Protocol #3: Incidental observations of predators and lemmings**

*No specific equipment needed (assuming every collaborator has a pen, a notebook + field forms, binoculars, and is an experienced arctic naturalist ;-)*

**Protocol #4: Index of lemming abundance**

*Most collaborators already monitor rodent densities on their sites, either by snap trapping, live trapping, or by counting active burrows, winter nests, etc. Hence we won't list specific equipment for this protocol, but if any collaborator is not yet collecting data on rodent abundance and would like to start to do that, then please contact O Gilg to discuss possible (and preferred) options.*

**Protocol #5: Index of herbivore abundance**

- 1 GPS to locate the beginning of the 30 transects
- 1 rope of 10m (marked every 1 m) to materialize the transect, and a 1m long stick marked in its middle to control for the width of the transect

# 8. Data management, data sharing and co-authorship rules

***Data management***

Data should be recorded on hard copy data sheets each day and will then be entered into the 5 excel template files provided. Data files and TinyTag raw files should be put on our shared Dropbox folder as soon as possible, at the latest on August 20$^{th}$ of the year of the experiment (preliminary reporting to the French Polar Institute, which is funding most Tiny Tags, being usually due before Sept. 10$^{th}$).

***Data sharing and co-authorship rules***

These topics are fully addressed in the Collaborative research agreement (files "Collaborative Research Agreement_IWG_april 2017.pdf" and "Research Questions IWG April 2017.pdf" on our shared Dropbox folder).