



NAFC Marine Centre
University of the
Highlands and Islands

SEAWEED CULTIVATION MANUAL

Shetland Seaweed Growers Project 2014-16



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Acknowledgements:

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Chapter 1- Introduction

1.1 Current status of seaweed aquaculture in the UK

Seaweed is currently harvested in relatively small volumes across the United Kingdom. However, increasing demand for seaweeds and their products has fuelled further research into the UK's wild and farmed seaweed resources over the past decade. Wild seaweed is harvested across the UK by small businesses, typically for production of food and alginates. Kelp forests are important natural habitats and, as demand for seaweed grows, seaweed farming can help create a sustainable supply and minimise environmental impacts from potential overharvesting of wild seaweeds. Since the 1980s, British and Irish researchers have been experimenting with methods for seaweed aquaculture - in particular on the Isle of Man, the west coasts of Scotland and Eire, Shetland and at Portaferry in Northern Ireland (for example: Holt, 1984; Kain *et al.*, 1990; Sanderson, 2006; Edwards & Watson, 2011; Rolin *et al.*, (2017)). By applying methods originally developed in Japan and China, seaweed production can be enhanced without depleting wild stocks to the detriment of the local ecosystem.

Seaweed and its derivatives have many uses. Historically, seaweed was commonly used as fertiliser and food for animals or people, particularly during food shortages. In modern times, it has many applications in the cosmetic, food processing, biomedical, pharmaceutical and printing industries and as a feedstock for biofuels and anaerobic digesters.

Production of seaweed has the potential to generate jobs and income for coastal communities. For example, in Ireland the annual turnover from seaweed products is around €5 million (Walsh & Watson, 2011), half of which comes from seaweed sold for cosmetic uses. Along with benefits to the economy, farming seaweed on a commercial scale could have significant environmental benefits by providing a habitat for marine organisms and help mitigate impacts of climate change by fixing carbon (Muraoka, 2004).

1.2. Environmental benefits from seaweed

Kelps are large brown seaweeds that create vast sub-tidal 'forests' along the UK's coast. These provide nursery habitats for fisheries and reduce coastal erosion. Kelp forests increase biodiversity by creating a rich habitat for a diverse range of marine life. For example, they play a vital role in the marine food web for animals feeding on kelp detritus and prey species that live among the kelp. Kelp also benefits the environment by acting as a carbon sink.

As the world's energy demand increases and fuel sources decrease, biofuels are being investigated as an alternative to more traditional/non-renewable fuels such as coal and other fossil fuels.

Seaweeds are a potential source of biofuel and exhibit the following advantages over terrestrial plants currently being used for biofuel production:

- Seaweeds use inorganic nutrients and carbon dioxide to rapidly synthesize biomass. Many species can do so more efficiently than land-based crops, so generating larger equivalent yields and biomass than the latter.
- Because they are already supported by seawater and have holdfasts to anchor themselves in place, seaweeds do not need to waste energy developing complicated support structures, leaving them with more energy to invest in growth.
- Seaweeds do not compete with food crops for land, freshwater or other resources.
- Seaweeds create cleaner seas by absorbing inorganic wastes from seawater such as ammonia and nitrates.
- Once deployed at sea, seaweeds need relatively low maintenance and require little manual labour compared with many terrestrial crops. Nor do they require artificial fertilizers or pesticides.

1.3. The *Shetland Seaweed Growers Project*

1.3.1. Project Funding and Objectives

The *Shetland Seaweed Growers* project was funded by the Coastal Communities Fund and undertaken at the NAFC Marine Centre in Scalloway, Shetland from June 2014 to December 2016. The project explored whether growing seaweed on a commercial scale is a feasible option within Shetland, in order to create jobs, generate income for the community and present new business opportunities for Shetland companies. Other aims of Shetland Seaweed Growers included supporting local businesses to incorporate seaweed in their products and to increase public awareness of seaweeds, their ecology and their uses.

1.3.2 Partners and Industry Associates

Scottish Sea Farms Ltd. was a commercial partner in the project and generously provided a six hectare licensed sea-site for growing seaweed at Sandsound South in Shetland. They also provided use of a work-boat, skipper and crew to help set up the necessary longlines; sea-deploy the hatchery-grown seedlings, and harvest and sample the resulting seaweed crop.

East Voe Shellfish Ltd. were industry associates of the project who were contracted to carry out the same tasks listed above on a second seaweed sea-site at Lea of Trondra, Shetland that is owned by the NAFC Marine Centre.

Grieg Seafoods Ltd. kindly donated some drying/smoking racks to the project to allow the NAFC Marine Centre to dry bulk quantities of seaweed.

1.4. Seaweed Uses

Seaweed is used by many maritime communities in a variety of ways from simple food sources and fertilisers to more complex products such as gels, medicines, cosmetics and alginates.

- Seaweed has been a food source for thousands of years, forming a staple part of the diet in countries like Japan, Korea and China. Particularly important seaweeds to the Japanese market belong to the *Porphyra*, *Laminaria*, *Saccharina* and *Undaria* genera. In European countries, seaweed has often been used as an alternative food source during hard times while Welsh 'laver' (*Porphyra umbilicalis*) and 'dulse' (*Palmaria palmata*), are considered delicacies by many.
- Historically, kelp was burned on a massive scale (including in Orkney and Shetland) to produce 'potash' (potassium salts) for fertilizer. This practice has now been replaced by world-wide mining of potash ores.
- Alginates are extracted from brown seaweed and used to create gels for foams, stabilisers, emulsifiers and industrial gums. They are also used by the cosmetic and health industries for body wraps, facial masks, soaps, shampoo and conditioners, make-up gels/creams, indigestion remedies, encapsulating particles and absorbent wound-dressings, along with a range of other products.
- Liquid seaweed extracts from brown seaweeds are used as mineral and vitamin supplements in agriculture, horticulture, animal husbandry and, more recently, in human health products.
- Agar is made from certain red seaweeds. In addition to being used in food production, it is universally used in laboratories as a substrate for bacteria cultures. There is currently no satisfactory substitute for laboratory agar.
- Carrageenans are also extracted from red seaweed and are widely used in the food industry, for their gelling, thickening, and stabilizing properties. Their main application is in dairy and meat products, due to their strong binding to food proteins.
- More recently the anti-bacterial and anti-cancer properties of seaweed derivatives such as fucoidan and laminarin are being investigated by the pharmaceutical and medical professions.
- There are numerous edible species that grow around the British Isles, including Shetland. Brown seaweeds are the most abundant, with kelps and fucoids such as *Laminaria digitata*, *Alaria esculenta*, *Fucus spiralis*, *Fucus vesiculosus* and *Ascophyllum nodosum*. Dulse and Irish moss are edible red seaweeds that are widely harvested in Ireland.

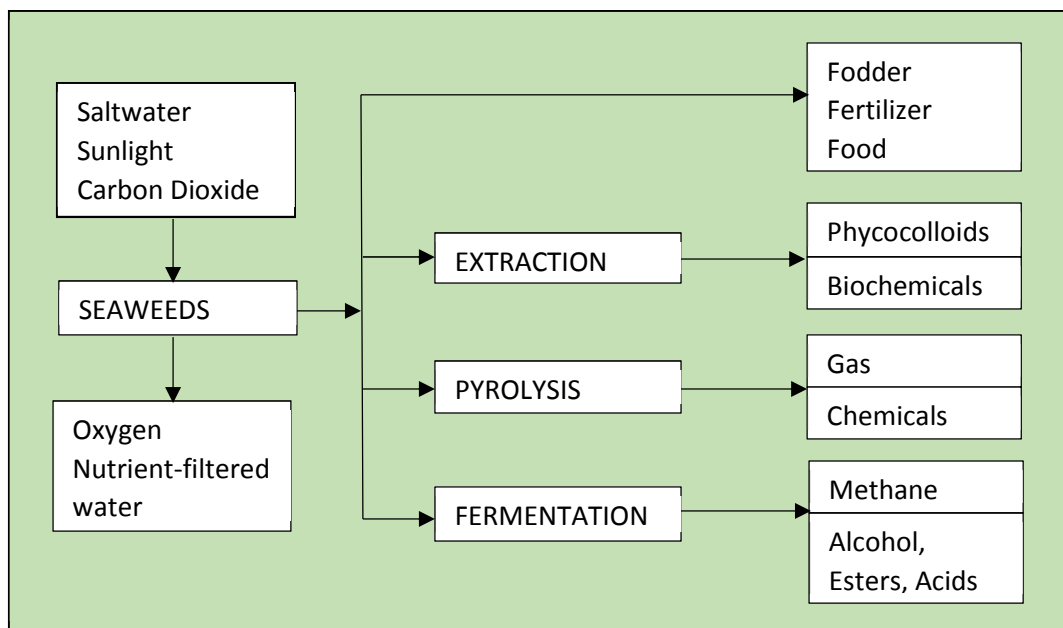


Figure 1: Examples of seaweed processing and subsequent products

1.4.1 Shetland Businesses Currently Making Use of Seaweed:

The *Shetland Seaweed Growers* project encouraged local businesses to diversify by incorporating seaweed grown by the project team into some of their products and/or undergoing training in sustainable harvesting of wild seaweed. Examples of participating companies include:

Artisan Island Cheese, Hoofields, Lerwick. (Caroline Henderson).

<https://food.list.co.uk/place/57672-artisan-island-cheese/>

Mirrie Dancers Chocolatier, Lerwick (David Williams)

<http://www.shetland.org/60n/blogs/posts/mirrie-dancers-chocolatier-launch>).

Orkney Soap, Kirkwall. <http://www.cope.ltd.uk/enterprises/orkney-soap>

Saxa Vord Restaurant, Unst. (Steve Carter) <http://www.saxavord.com/food-and-drink.php>

Shetlandeli, Bixter (Jill Franklin) <http://www.shetlandeli.com/>

Shetland Distillery Company, Unst (Stuart Nickersen) <http://www.shetlandreel.com/>

Shetland Fudge Company, Lerwick. <https://www.shetlandfudge.co.uk/>

Shetland Garden Co., Lerwick (Ingrid Webb) <http://www.cope.ltd.uk/enterprises/shetland-garden-co>.

Shetland SeaSalt Co., Scalloway (Akshay Borges) info@shetlandseasalt.com

Shetland Soap Co., Lerwick (Ingrid Webb) <http://www.cope.ltd.uk/enterprises/shetland-soap-company>

Chapter 2- Seaweed Life-Cycle and Biology

There are three main types of seaweed: green, brown and red. Kelps belong to the brown seaweeds. They are large plants and form vast kelp 'forests' in the subtidal zone, so providing important habitats for many marine animals and protecting shores by reducing wave action. Kelp are of particular interest for seaweed farming since they grow fast and produce a large biomass. The *Shetland Seaweed Growers* project successfully cultivated the kelp species *Laminaria digitata* and *Alaria esculenta* on longlines at two sea-sites in Shetland. Crops of wild *Saccharina latissima* that naturally settled on unseeded sea-lines were also harvested during the project.

2.1 Species identification:

Alaria esculenta

Alaria esculenta or Dabberlocks (Figs 2 & 3) is a widely-distributed brown kelp that grows well in exposed sites with good water exchange. Its holdfast give rise to a short stipe, which continues into a distinct midrib running down the length of the plant's single, long frond that is olive-green/brown in colour. When fertile, *A. esculenta* develops two rows of branching sporophylls that grow out from the stipe at the base of the blade in autumn/spring. These sporophylls are solely for developing and storing spores.

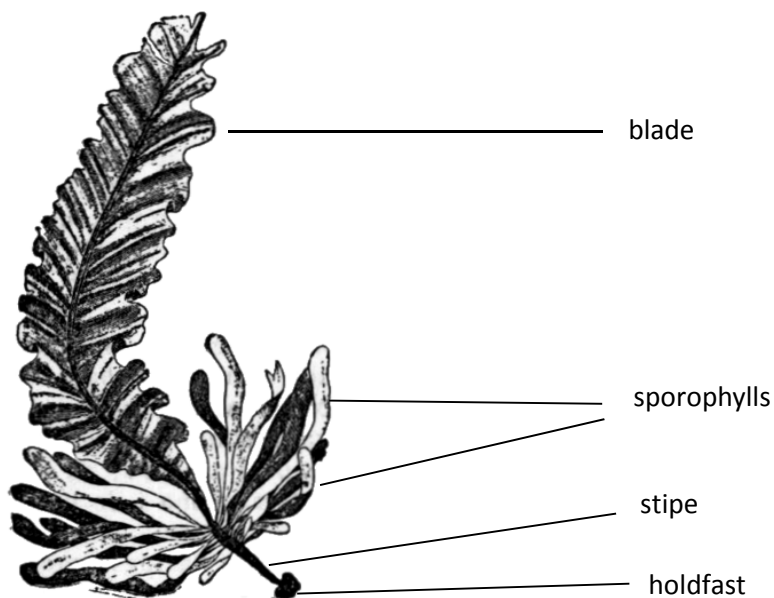


Figure 2: *Alaria esculenta* showing sporophylls at top of stipe, below the blade.
(source: Wikimedia Commons).



Figure 3: *Alaria esculenta* cultivated on a longline at Scottish Sea Farms Ltd's South Sandsound site, Shetland. (Photo: C. Rolin)

Laminaria digitata

Laminaria digitata or Oarweed, is distinguished by its digitated frond that has three to eight finger-like segments when mature (Fig. 4). It is dark brown with a smooth, flexible stipe that is oval in cross-section. A plant can grow up to three metres in total length. In the wild, each plant anchors itself to rocks by its conical holdfast that consists of a number of root-like rhizoids. *L. digitata* has no sporophylls and develops its spores in 'sori' on its frond blades.



Figure 4: *Laminaria digitata*. Photo: K. Gifford

Saccharina latissima

Saccharina latissima, commonly known as Sea Belt or Sugar Kelp, is another large, brown kelp. It is golden-brown in colour and has a single, large, undivided frond with a frilly, undulated edge. It lacks a midrib and its stipe is short with a branching holdfast. Its reproductive sori appear in the centre of the blade, rather than in separate sporophylls on the stipe.



Figure 5: *Saccharina latissima*. Photo: C. Rolin

2.2 Overview of seaweed reproduction

Seaweed life-cycles and reproduction can be complicated. Some seaweeds are perennial, living many years, while others are annuals, living only one year. Sexual and/or asexual reproduction (cloning) are possible. Different species start their life-cycle and reproductive processes at different times of year and use various strategies. Many are heteromorphic, going through many different stages before becoming fully grown.

The kelp life-cycle begins when dark reproductive patches, called 'sori', appear. Depending on the seaweed species, these spore-containing sori are either found on the plant's blades or in specialised structures called sporophylls. For example, sori in *Alaria esculenta* are located in sporophylls that grow out from its stipe (Fig 6), whereas sori in *Laminaria digitata* are found on the blades of the plant (Fig 7).

During kelp sexual reproduction, a particular type of cell division called meiosis produces male and female spores. These haploid spores (containing only a single set of unpaired chromosomes) are then released into the sea. The motile spores settle and develop into either male or female gametophyte plants. The female gametophyte develops eggs which are fertilised by sperm from male gametophyte plants. Fusion of the egg and sperm during fertilisation creates a diploid zygote containing two sets of chromosomes – one from each parent. Each zygote develops into a sporophyte (adult plant), and the process begins again. This lifecycle is summarised in Fig. 8.

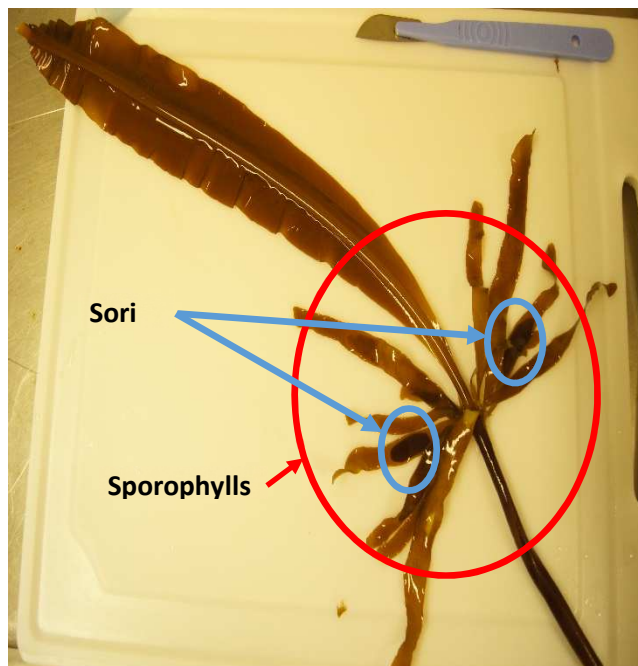


Figure 6: *Alaria esculenta* with sporophylls circled in red and sori in blue. Photo: C. Rolin.



Figure 7: *Laminaria digitata* blade with sori clearly visible as dark patches. Photo: C. Rolin.

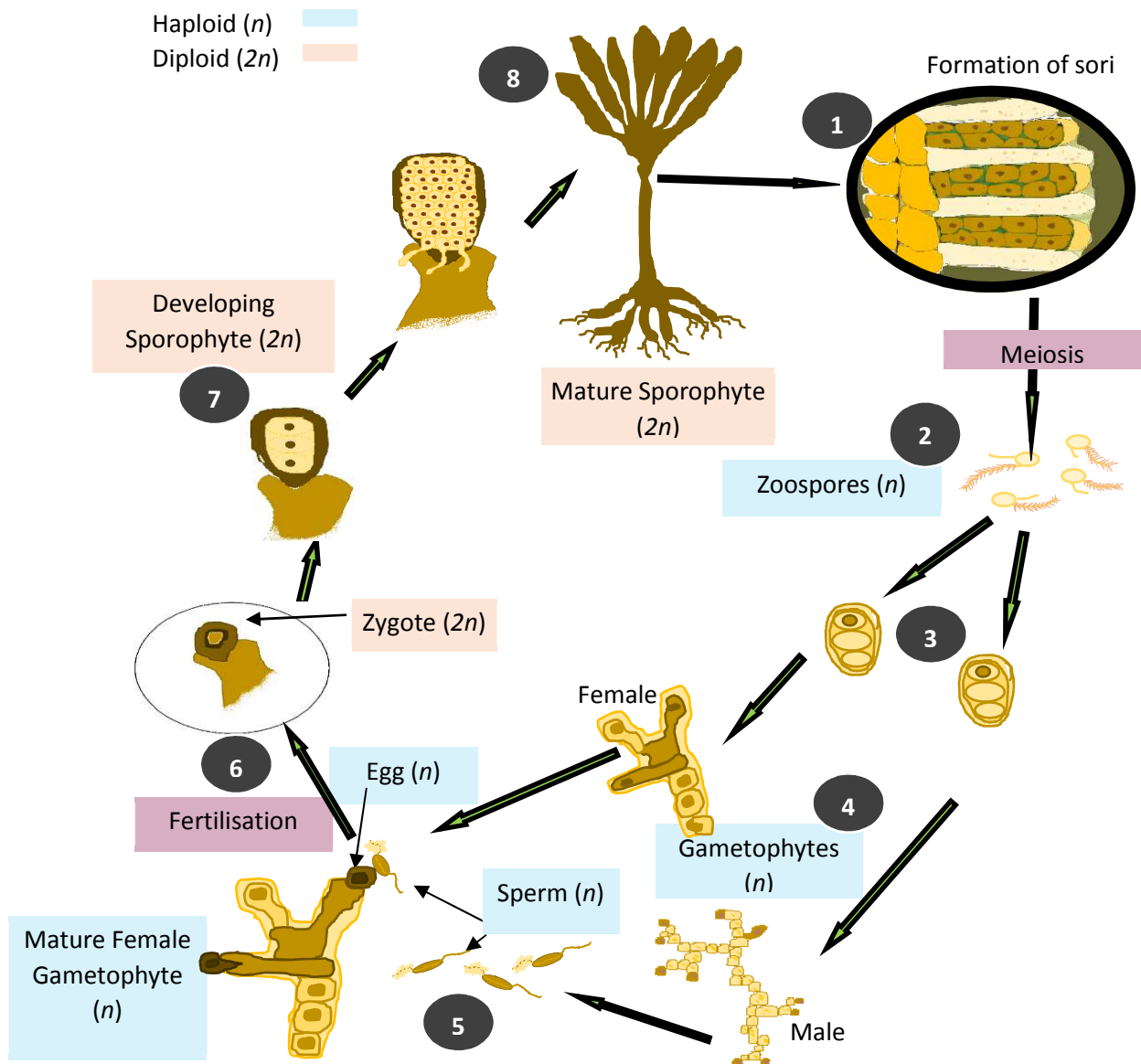


Figure 8: Life cycle of kelp, showing the various stages towards adulthood.

1. Sori, containing thousands of spores, appear on the seaweed's blade or within sporophylls.
2. Motile zoospores with flagellae are released into the seawater to find a suitable place to settle.
3. After 24 hours the flagellae are lost and the spores attach to a surface.
4. The zoospores germinate to form male and female haploid gametophytes (n).
5. The gametophytes become fertile and produce sperm and egg gametes.
6. At fertilisation, a sperm fuses with an egg to form a diploid zygote (2n).
7. The zygote develops into a sporophyte (adult) with holdfast, stipe and frond/blade.
8. The adult sporophyte ripens and the whole process is repeated.

2.3 Farming calendar

Seasons of reproduction will differ between seaweed species and also between different locations. For example: *Laminaria digitata* produces sori between April and October, depending on location, with most sori appearing between July and September. However, sori have also been seen on this species between November and January. By contrast, *Alaria esculenta* produces its spore-filled sori between November and March, with most appearing between November and December. It is important to know the reproductive season of your selected species for cultivation in your particular location so you can organise spore collection and propagation of the young life-stages at the correct times. Example calendars showing annual cultivation cycles for *Laminaria digitata* and *Alaria esculenta* are given below (Tables 1 & 2).

Bear in mind that these schedules are only given as a general guideline and will vary with location. Also note that deploying your young seedlings on sea longlines as early as possible in the winter will yield larger and less biofouled crops since there will be less planktonic larvae competing for settlement space on the longline and fewer fouling organisms.

Table 1: *Laminaria digitata* Farming Calendar

	Jul	Aug	Sep	Oct	Nov	Dec	Jan	Feb	Mar	Apr	May	Jun
Spore Collection	■	■	■	■	■	■	■	■	■	■	■	■
Hatchery Growth	■	■	■	■	■	■	■	■	■	■	■	■
Deployment to Sea Lines	■	■	■	■	■	■	■	■	■	■	■	■
Sea Growth	■	■	■	■	■	■	■	■	■	■	■	■
Harvesting	■	■	■	■	■	■	■	■	■	■	■	■

Table 2: *Alaria esculenta* Farming Calendar

	Jul	Aug	Sep	Oct	Nov	Dec	Jan	Feb	Mar	Apr	May	Jun
Spore Collection	■	■	■	■	■	■	■	■	■	■	■	■
Hatchery Growth	■	■	■	■	■	■	■	■	■	■	■	■
Deployment to Sea Lines	■	■	■	■	■	■	■	■	■	■	■	■
Sea Growth	■	■	■	■	■	■	■	■	■	■	■	■
Harvesting	■	■	■	■	■	■	■	■	■	■	■	■

Chapter 3 – Hatchery Cultivation of Seaweed Seedlings

3.1 Setting up a seaweed hatchery

3.1.1. Overview

Since 1992, the NAFC Marine Centre has operated a marine pump-ashore hatchery equipped with drum, bag and cartridge filtration of seawater down to 1 micron, followed by sterilization by UV light. This facility was used to germinate seaweed spores and to culture the subsequent kelp gametophytes and sporophytes prior to sea-deployment of these kelp seedlings on longlines.

When setting up a seaweed hatchery, consider developing the facility within an existing marine hatchery as this can help avoid some of the most expensive start-up costs.

Below are basic instructions for establishing a seaweed facility similar to the one used within the Coastal Community Fund's *Shetland Seaweed Growers* project at NAFC Marine Centre in Shetland. We adapted and employed a mixture of direct seeding and indirect seeding culture methods that are detailed in Edwards & Watson (2011) and Flavin et al. (2013)

3.1.2. Facility requirements:

Seawater

The seas around Shetland are virtually oceanic in quality with high salinity, clarity and cleanliness. Incoming seawater should be filtered to 1 μ (for example, initially passed through a drum-filter with a 10 -15 μ screen, then through a series of 10 μ , 5 μ and 1 μ cartridge filters) before being sterilised by UV light. All filters and UV lamps need to be cleaned and replaced regularly. Glass culture flasks of seawater with cotton wool bungs need to be sterilised in an autoclave at 121°C, 15psi for 20 minutes and then cooled prior to inoculation with seaweed spores and seedlings.

Temperature control

A temperature controlled room is essential and should be maintained at 10°C for the culture of temperate kelps. At NAFC, this was achieved using a CellarKing CX(E) Standard Cellar Cooler (Heronhill). Air and seawater temperatures should be monitored and recorded. Since incoming seawater at the NAFC hatchery was still at ambient sea temperature, culture tanks were cleaned and filled with seawater the day before their use in order to allow the static seawater tanks to equilibrate to the culture room air temperature (10°C) overnight.

Air supply

A continuous supply of clean air is required to oxygenate and mix seaweed cultures. At NAFC, this was achieved by a mixture of air-blowers and smaller air-pumps with adjustable valves. Air-lines were fitted with 0.2 micron air-filters.

Seaweed spores, gametophytes and initial sporophytes were cultured in cotton-wool stoppered glass flasks containing seawater filtered to 1 μ and fitted with a glass tube for aeration purposes. The flask of seawater plus its inserted foil-capped air tube were sterilized

together within an autoclave (121°C, 15psi for 20mins) prior to use.

Subsequent seaweed seedlings were grown in insulated 660L harvest bins of static, aerated seawater. Airlines to these culture bins were also fitted with air-filters but had air stones (rather than glass tubes) in order to increase oxygen diffusion. These air-stones were weighted to keep them on the bottom of the culture bin. Several airlines were placed at different points in each culture bin to ensure good oxygenation and circulation of seawater and nutrients. Aeration levels were gradually increased as the seedlings increased in size.

Lights

During flask-cultivation at NAFC, 4' waterproof white fluorescent lamps were placed along two laboratory benches allowing flasks to be moved closer or further away to attain the target light intensity of circa $18\mu\text{mol}\cdot\text{m}^{-2}\cdot\text{s}^{-1}$ at the flask surface (Fig 9). Lights were covered with red cellophane during the initial spore/gametophyte stages of cultivation since red light inhibits sexual reproduction in kelps and allows the culture's biomass to increase (see Fig 10 for further explanation).



Fig 9. *Laminaria digitata* spores and gametophytes cultivated in red light at the NAFC Marine Centre, Shetland. Photo: R. Inkster

Water surface lighting levels of circa $50-70\mu\text{mol}\cdot\text{m}^{-2}\cdot\text{s}^{-1}$ were used over NAFC's 660L seaweed culture bins (see sections 3.5 and 3.8 of this report). Banks of three cool-white waterproof fluorescent lamps were covered with blue cellophane since blue light encourages sporophyte growth (Fig 10). These 'blue' lights were fixed to strong marine plywood boards and suspended above each tank by chains to the ceiling. Several of these assemblies were made and were raised or lowered in order to achieve the desired light intensity in the tank directly below them. Light intensity was measured using an Apogee Quantum Meter MQ-200 (Campbell Scientific Ltd).

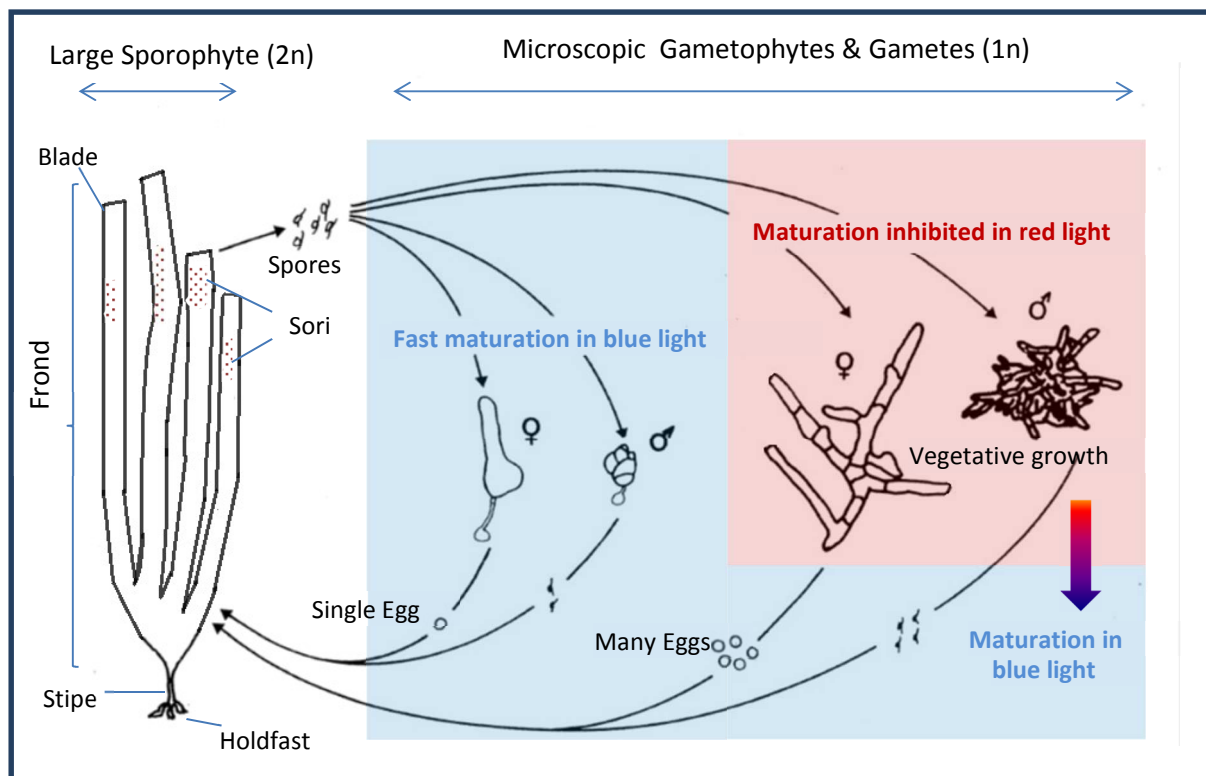


Fig. 10. Life-cycle of the Laminariales illustrating how red light delays gametophyte maturation, allowing further vegetative growth and subsequent increase in biomass and overall fecundity. 2n: diploid; 1n: haploid. (adapted from Kain (1991))

Nutrients

A stock solution of Cell-Hi F/2 'All in One' Algae Nutrient Powder (Varicon Aqua Solutions Ltd) was made up according to the manufacturer's instructions and added at 1.5ml per litre of culture in order to provide nutrients and vitamins to the culture flasks and bins. Fresh stock solution was made up at least monthly and it was stored in a refrigerator and protected from light by a foil covering (in order to prevent deterioration of the vitamins).

3.1.3. Making Seaweed Spools

The NAFC Marine Centre employed the same types of seaweed spools used by Edwards & Watson (2011), constructed as follows:

1. Square drain pipe was cut into lengths of about 40cm.
2. Large holes (5cm diameter) were cut along the length of each of the four sides of the drain pipe to allow circulation of seawater through the structure.
3. Two small holes were drilled in the top and bottom of the pipe and threaded with thin nylon string to allow the spool to be suspended from rods spanning across the top of the tank. (Usually, after several weeks, there was greater seaweed growth at

the top of the spool nearest the overhead lights. Drilling additional string-holes in the bottom of the spool allowed them to be inverted in the tank, moving the lower, smaller seedlings closer to the lights to improve their growth-rate).

4. Cross braided white nylon string (1.3mm thickness) was wound around a square plastic-coated wire basket (such as a freezer basket). The basket was then placed in the sink and covered with hot freshwater. That was emptied and refilled several times a day for at least 3 days in order to leach the string of any contaminants that might badly affect seaweed settlement. The leached culture string was then allowed to dry naturally at room temperature, still wound on the basket.
5. Dry, leached culture string was secured on the drain pipe using an elastic band and wound around the pipe, leaving a 'tail' of string hanging from the wound spool for sampling and observation purposes. About 35-40 metres of culture string can be wound around each 40cm piece of drain-pipe.
6. The wound spools were then covered in foil to keep them clean and stored in plastic bags prior to use: either for direct settlement of spores or being sprayed with a blended seaweed culture.

3.2 Collecting Fertile Seaweed

The methods described below are adapted from Edwards and Watson (2011) and Flavin *et al.*, (2013).

1. Kelps are most easily collected during low tide, so plan ahead by checking your local tide tables for a suitably low tide during daylight hours and check the weather forecast for safe weather conditions. Do a careful, dynamic risk assessment for the activity and stop if conditions worsen and threaten to become unsafe. Always wear appropriate waterproof clothing and footwear and a compact life-jacket. Never go sampling alone. Always inform others where and when you are sampling and give them an estimated time of return. Sheath sampling knives when not in use.
2. Refer back to Chapter 2 of this manual for information regarding seaweed biology, suitable sampling habitats and how to identify reproductive tissue (*sori*) in different species.
3. When a *sori* patch has been found, it should be cut away from the seaweed, leaving behind as much of the seaweed undamaged as possible. Remember to take only what you need.
4. When selecting seaweeds to harvest take only the ones that are 'clean', and unfouled by other organisms and plants.
5. Immediately place collected *sori* into an insulated cool-box filled with fresh, cool seawater.

6. When collecting *Alaria esculenta* DO NOT put the sporophylls directly into seawater in a cool-box, instead transport the sporophylls in seawater-dampened tissue paper wrapped in seaweed inside the cool box.

3.3. Processing Fertile Seaweed back at the Hatchery

Processing the collected seaweed *sori* should be carried out in a temperature-controlled environment where the seaweed is not exposed to temperatures below 10°C or above 15°C.

1. Fill three plastic trays with sterilised seawater and place a thermometer in the tray to closely monitor the water temperature throughout.
2. Remove the collected, un-fouled seaweed *sori* from the cool-box and rinse in Tray 1.
3. After rinsing, place the seaweed on a chopping board and gently dry with white tissue paper, rubbing in one direction only, to remove the mucus covering. Once dry, gently scrape the seaweed with a blunted razor blade.
4. Repeat this rinsing and drying process twice more (rinsing the seaweed in Tray 2 and then in Tray 3), however do not re-scrape the seaweed using the blunt razor.
5. When processing *Alaria esculenta* sporophylls, do not use the blunted razor blade at any point during the cleaning process.
6. Once the seaweed has been through the full cleaning and drying process, lay it flat between a few sheets of dry tissue paper and place inside a closed zip-lock plastic bag.
7. Leave the seaweed in a dark incubator at 10°C for between 18-24 hours.

3.4. Inducing Spore Release

1. Remove seaweed from the incubator. Staining of the tissue by the seaweed is a good sign because it indicates that the *sori* were ripe and some spores have been released onto the tissue.
2. Cut the seaweed into pieces about 5cm x 5cm.
3. Add around 15 pieces of the cut-up *sori* into a glass beaker along with a thermometer and 800ml of sterilised seawater.
4. Once the seawater has been added, stir the seaweed and observe at 5 minute intervals, for 30 minutes, occasionally stirring and recording changes observed in the beakers. During this time a release of spores from the seaweed may or may not be obvious.
5. After 30 minutes, the contents of the beaker should be strained through a mesh to remove the larger pieces of seaweed.

6. If this process has been carried out using several different spore-release beakers, combine the strained contents of all the beakers into one large receptacle.
7. Stir this combined spore-release seawater and place a drop of it onto a haemocytometer to allow the spores to be counted under the microscope.
8. Count all the spores contained within the large central counting square of the haemocytometer, recording the number using a manual counter. Multiply this count number by 10,000, to give the number of spores per millilitre.
9. Optimal spore stocking densities for cultivation are generally between 5,000-10,000 spores/ml (Flavin et al. 2013). NAFC aimed for approximately 8,000 spores per ml in their flask cultures and in their direct spore settlement inoculation tanks.
10. **Apply the following formula from Flavin et al. (2013) to calculate what volume of spore-release seawater to add to your culture flask or to your inoculation tank in order to generate the desired final stocking density of spores for cultivation:**

$$\text{Desired spore density in flask or tank} / \left(\frac{\text{Counted spore density in spore-release water (spore per ml)}}{\text{Volume of seawater in culture vessel (ml)}} \right)$$

= ml of spore-release water to be added to flask or tank to achieve desired final spore density.

Worked Example:

If you count 20 spores in the central haematocyte square, this equates to a spore density of $20 \times 10,000 = 200,000$ spores /ml in your spore-release water.

Applying the equation above, and assuming you wish to generate a final spore density of 8,000 spores/ml within an inoculation tank of 20 litres of seawater, you need to add 800ml of your spore release water to the tank, as calculated below:

$$8,000 \text{ spore per ml} / \left(\frac{200,000 \text{ spores per ml}}{20,000 \text{ ml culture water}} \right) = 800 \text{ ml of spore release water required.}$$

3.5. Flask Cultivation of Spores & Gametophytes:

1. Add 800ml of filtered, sterilised seawater to 1.5L glass conical flasks, add a cotton wool stopper and cover flask neck and cotton wool with aluminium foil before autoclaving at 121°C, 15psi for 20 minutes. Then cool the flask and its contents to 10°C.
2. Briefly flame the neck of the flask and carefully pour the calculated volume of spore suspension into the flask (see section 3.4), avoiding it from touching the inside of the warmed neck.
3. Add 0.6ml of germanium dioxide (0.1%) and 1.25ml of F/2 Cell-Hi stock solution (Varicon Aqua Solutions Ltd.) to the conical.
4. Add a sterilised filtered airline to the conical (i.e. 6mm glass tube connected to silicone

- tubing fitted with an inline air-filter), re-flame the neck and replace the cotton bung.
5. Connect the airline to an aeration pump or air-blower and place the flask in front of warm white fluorescent tubes covered in red cellophane so that the light intensity at the flask surface is $17 - 20\mu\text{mol.m}^{-2}\text{s}^{-1}$
 6. Flasks should be re-cultured on average every 10 days as follows:
 - Stop the aeration to the flask and allow the seaweed gametophytes to settle for several minutes. Carefully pour away the seawater above them without losing the gametophytes. When you are left with a concentrated gametophyte suspension at the bottom of the flask, pour this into a fresh flask of seawater that has been previously autoclaved, cooled and has had nutrients and germanium dioxide added, as above. Add a new sterile filtered air-line and connect to an air-supply.
 7. Aeration should be increased gradually over time, see Table 2 for further details.
 8. As the gametophytes grow and their biomass increases it will be necessary to transfer them to larger flasks of autoclaved seawater.
 9. Cultures should be grown at 10°C either in a temperature controlled environment.
 10. Cultures cannot be held in the gametophyte stage indefinitely on red light, and reproductive structures start to develop after a few months. However, red light can delay reproductive onset sufficiently to allow gametophyte biomass to be increased tenfold.
 11. Examine the maturity and health of the gametophytes under the microscope at each flask-change. Healthy cultures are green/brown in colour. Bubbles from the aeration are normal but the cultures should not have froth accumulating on the top of the culture.

3.6. Spraying Cultures onto Spools of Culture String.

At the NAFC Marine Centre, seaweed gametophyte cultures were held under red light for up to 20 weeks through the summer, however some reproductive structures started to develop after this time. Using the methods of Edwards and Watson (2011), they can be sprayed onto spools of culture string from about seven weeks of flask cultivation onwards, as follows:

1. Remove the airline from the culture flask and allow the gametophytes to settle on the bottom of the flask, then drain off excess water leaving a concentrated suspension.
2. Using a hand-held blender, blend the seaweed for short bursts for about 40 seconds, then decant into a garden-spray bottle.
3. Set a large rectangular plastic container on its side against a wall (to act as a shield to 'catch' any excess gametophyte suspension during spraying – see Fig. 11) and stand the spools of culture string upright inside it.

4. Using the mist-spray setting, spray the blended seaweed suspension evenly onto all four sides of the stringed spools by turning each of the spools around during spraying. Several can be sprayed at the same time, side by side.
5. Once sprayed, allow the spools to air-dry for 25 minutes, then suspend the spools of sprayed string in filtered, UV sterilised seawater at 10°C in 660L insulated polyethylene harvest bins.
6. Although time-consuming and costly, flask culture of gametophytes allows a ready supply of gametophyte cultures to be held in the hatchery until required, so reducing the risk of 'missing' the period of sori maturity and it allows the available biomass of gametophytes to be increased greatly in the laboratory.

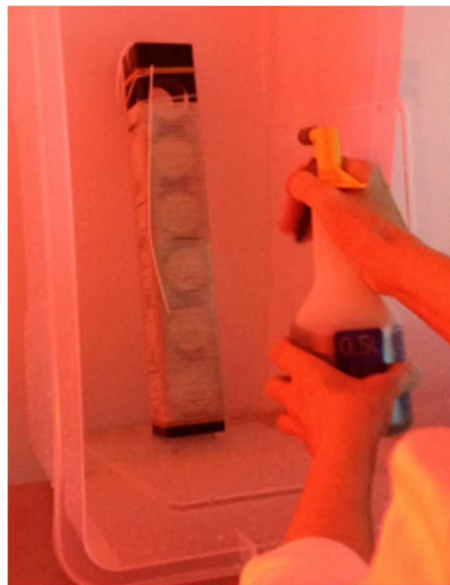


Fig 11. Spraying blended *Laminaria* cultures onto spools of culture string.

3.7. Direct Settlement of Spores onto Culture String Spools.

If preferred, seaweed spores do not need to be cultured in flasks before being transferred onto spools. Instead they can be allowed to settle directly onto spools of culture string, so eliminating the time consuming and expensive step of flask culture. Indeed, in the Shetland Seaweed Growers Project, we noticed a more even settlement of sporelings and higher yields at sea when we switched to using the direct settlement method. It is carried out as follows:

1. Add 15L of filtered, UV sterilised seawater to a large rectangular container along with 10ml of germanium dioxide stock solution (0.1%) and 10ml of F/2 nutrient stock solution (Varicon Aqua Solutions). Finally, to the container add the calculated volume of spore suspension required to generate the desired spore density usually about 8,000 per ml – see section 3.4 of this report for details of how to count spores and calculate spore densities).
2. To the container add two glass rods, one either end of the container. Place four spools on top of these rods to raise them off the bottom of the container (to allow spore settlement on the string on the lower side of the spool too).

3. Replace the lid of the container and leave in a dark room at 10°C for 24 hours.

3.8. Transfer of Seeded Strings to Cultivation Tanks

1. Fill 660L insulated polyethylene bins with filtered and UV sterilised seawater at a temperature of 10°C. The tank may need to be filled the day before use to allow the seawater temperature to stabilise to the temperature of 10°C within the controlled temperature room.
2. Add 35g of F/2 Cell-Hi All-in-One powdered nutrients (Varicon Aqua Solutions Ltd) dissolved in a small amount of freshwater to the tanks and mix well.
3. Place poles across the tank and fix in place (for example, in Fig. 11, polystyrene collars have been fitted at the ends of the poles to prevent them slipping into the tank).
4. Take the seeded seaweed spools and lower them very gently and slowly into the seawater to prevent the spores from detaching from the string. Suspend them from the poles by thin nylon string threaded through the small holes pre-drilled at the top of the spool. Ensure the spools are fully immersed in the seawater.
5. Hang up to 4 spools per pole and about 16 spools per harvest bin.

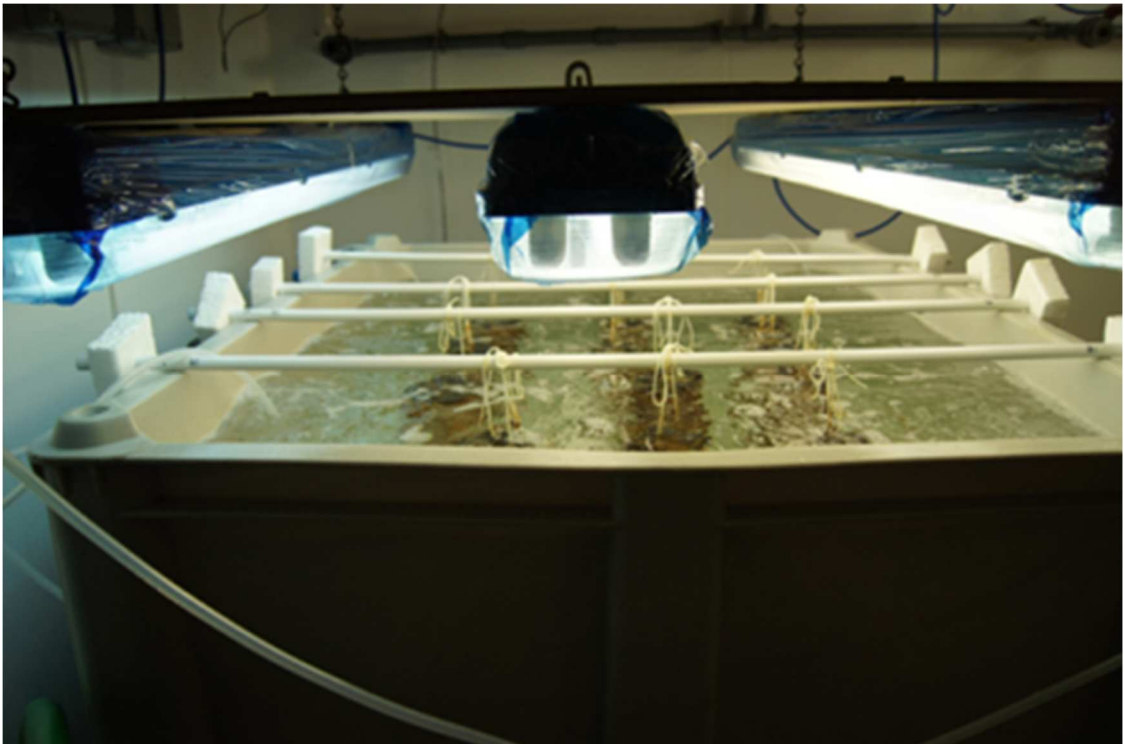


Fig 11. Spools of seeded culture string suspend in 660L seawater tank under blue lights.

Do not add airlines to the tank for the first three days. After this time, airlines are placed in each of the corners of the tank to ensure good circulation of seawater, oxygen and nutrients.

Over the next four weeks aeration and light levels should be increased gradually from low to high using up to four airlines per tank (see Table 2).

Table 2: Pattern of increasing aeration and light intensity during hatchery-cultivation of culture string spools seeded with either *Laminaria digitata* or *Alaria esculenta*.

No. days seeded spools suspended in 660L tanks.	Aeration level	Light level $\mu\text{mol}^{-2} \text{s}^{-1}$ (blue)	
		<i>L. digitata</i>	<i>A. esculenta</i>
0-3	None	14	21
4-11	Add 2 airlines Low level of aeration Slowly increase aeration	Increase up to 45	Slowly increase up to 35
12-19	Increase aeration of 2 airlines to moderate	Slowly increase up to 55	Slowly increase up to 52
20-27	Add 2 more airlines to the tank Slowly increase aeration	Slowly increase up to 65	Keep at 52
28+	High aeration	Slowly increase up to 70	Keep at 52

3.9. Cleaning Spool Tanks:

Twice weekly, the seaweed spool bins need to be emptied, cleaned and refilled with filtered UV-sterilised seawater at 10°C, with fresh nutrients added as in points (1) and (2) in this section. To allow this to happen, there needs to be a spare 660L harvest bin set up the day before cleaning occurs and its seawater allowed to acclimate to 10°C before the seaweed spools are gently and slowly removed from the dirty tank, still suspended from the pole and transferred to the clean tank. Before slowly lowering the suspended seaweed spools into the new clean tank, it helps tank-hygiene greatly if the spools are first gently ‘dipped’ into an additional smaller tank of sterilised seawater at 10°C before being lowered into the clean 660L tank. The small ‘dipping’ tank also needs to be filled with sterilised seawater the day before cleaning to allow its temperature to stabilize to 10°C.

The quality and growth of the small seaweed sporophytes can be assessed by snipping a small sample from the ‘tail’ of culture string deliberately left hanging from each spool (see section 3.1.1) and evaluating it under a binocular microscope (Fig. 12).



Fig 12. Sample of culture string removed from a spool, showing *Laminaria digitate* sporophytes growing along its length.

Chapter 4 – Sea-Site Selection and Permissions

4.1 Sea-Site Selection

Selecting a suitable on-growing site for your seaweed is very important as this will strongly influence how well it grows. Firstly, learn about your chosen species' habitat preferences in terms of wave exposure, temperature range, water depth and flow, type of sediment and light intensity. Websites such as **MarLIN** (<http://www.marlin.ac.uk/>) and **AlgaeBase** (<http://www.algaebase.org/>) are particularly helpful in supplying this type of information.

Select a site that meets as many of your species requirements as possible in order to achieve optimal growth conditions

- Most seaweeds can tolerate a range of temperatures. They are highly tolerant to winter lows but sensitive to summer highs. For the best growth, the optimal temperature of the target species should be considered.
- Light availability is important to photosynthesis, a lack of light can lead to poor growth. Select a location that is exposed to sunlight for most of the day, so avoid selecting areas with lots of shade, look for an area with clear water and low sedimentation.
- Avoid setting up in shallow areas with high levels of sediment. Sediment kicked up from the bottom can smother seaweed, preventing it from photosynthesising and taking up nutrients. This will inhibit growth and could damage crops
- Flow must be sufficient to bring in a fresh supply of nutrients and carbon dioxide for the seaweed to absorb. Good flow will also reduce the settlement of encrusting organisms and sediment. Some species are particularly well adapted to more exposed sites, for example *Alaria esculenta*.

Other important considerations to take into account during site selection include:

- Are there other sea-farms in the area such as fish or shellfish farms or other seaweed ventures? This can effect planning permissions and licences.
- Is it close to any sewage outlets, industrial waste outlets or fresh water run off?
- What happens in the area? Is the area a well-used area for tourist, fishermen and boat traffic? Are there boating clubs using the area? If so, will any of these activities negatively impact on your seaweed farm operation, or *vice versa*?
- Is anyone likely to protest against a seaweed farm in that location?

4.2 Regulations and Licensing

When setting up a commercial venture it is important to know the relevant local and national regulations with which you must comply and the necessary permissions and licences you need to obtain. For example, most of the UK foreshore and seabed up to 12 nautical miles from the shore is owned by the Crown and is managed by the Crown Estate - the small remainder being a mixture of public and private ownership. So from the outset, you need to identify the landlord, seek their permission and determine if a lease and rent payment are required. You also need to apply for the necessary site licence(s) to comply with local and national planning regulations before installing any equipment, moorings, lines or buoys. In Shetland, marine farmers need to apply for licences from the Shetland Islands Council as well as from Marine Scotland. In other Scottish areas, only a licence from Marine Scotland is required.

Begin this licence application procedure as soon as possible because it can take a long time to complete. This is because the planning authorities first need to seek written advice from other key stakeholders and regulators (for example, the Scottish Environment Protection Agency, Marine Scotland Science, the Northern Lighthouse Board, Scottish Natural Heritage, the relevant District Salmon Fishery Board, Historic Scotland and the Royal Society for the Protection of Birds). Public notices must also be advertised for a sufficient length of time to allow other commercial stakeholders and members of the general public to object to the development if they wish. If a license is refused, the would-be developer is allowed to appeal or to amend their plans and restart the application process.

Traditionally, marine farming regulations have focused on fish and shellfish farming but, following a recent public consultation exercise, the Scottish government is working to introduce regulations specifically aimed at seaweed farming - so it is important to stay up to date with current requirements.

Regulations alter between UK countries and the districts within them. So it is important to know the specific requirements in your location and the various permissions you need to seek.

Please note: This manual does not state or give legal advice - you must seek this yourself.

Chapter 5. Farming Seaweed at Sea

5.1. Project Sea-Sites

The two seaweed on-growing sites used in *the Shetland Seaweed Growers* project were located in sheltered areas with good water exchange. Both sites were suitable for fish farming and located near mussel farms.

The site at Lea of Trondra ($60^{\circ}06.833'N$, $001^{\circ}16.967'W$) (Figs. 13 & 14) had a depth range of 9 – 16 m with longline moorings at 9 – 12 m depth. The two 80 m longlines at this site were set at 2 m below the water surface and each consisted of a continuous length of 24 mm Euroflex[®] sinking rope (W & J Knox Ltd., UK). A mixture of 100 kg concrete and fluke anchors were employed while 32 mm SEASTEEL 3 strand rope (GaelForce Marine Ltd.) and 32 mm open link chain (GaelForce Marine Ltd., UK) were used for mooring attachment.

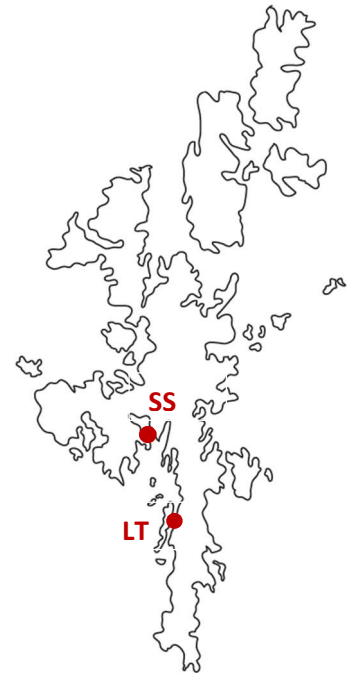


Fig 13. Project sites in Shetland
LT: Lea of Trondra.
SS: Sandsound

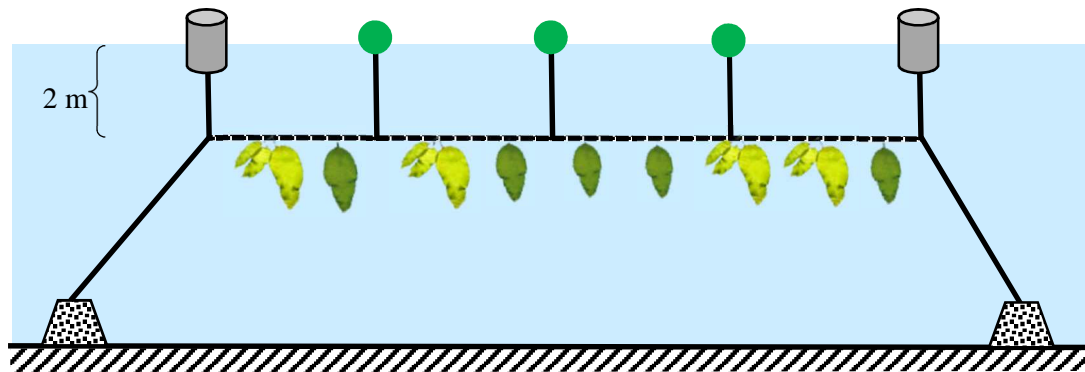


Fig. 14. NAFC Marine Centre's seaweed site (2 x 80m longlines) at Lea of Trondra, Shetland.

The seaweed site at Sandsound (60° 06.833'N, 001° 16.967'W) was kindly offered to the Shetland Seaweed Growers project by Scottish Sea Farms Ltd. (Figs. 13 & 15)

It had a depth range of 8 – 22 m with a longline mooring grid 5 m below the water surface. The two 245 m longlines at this site were also set at 2 m below the water surface and each line consisted of 20 mm "SEASPUN" STAPLE SPUN floating, 'hairy' polypropylene rope (Gael Force Marine Ltd., UK) in seven 35 m segments, connected together by steel rings, as indicated in Fig 14. Each longline was held in place by a rope grid system consisting of 20mm SEASTEEL rope and fluke anchors.

A1 Polyform Buoys (GaelForce Marine Ltd., UK) were attached at regular intervals to the longlines at both sites.

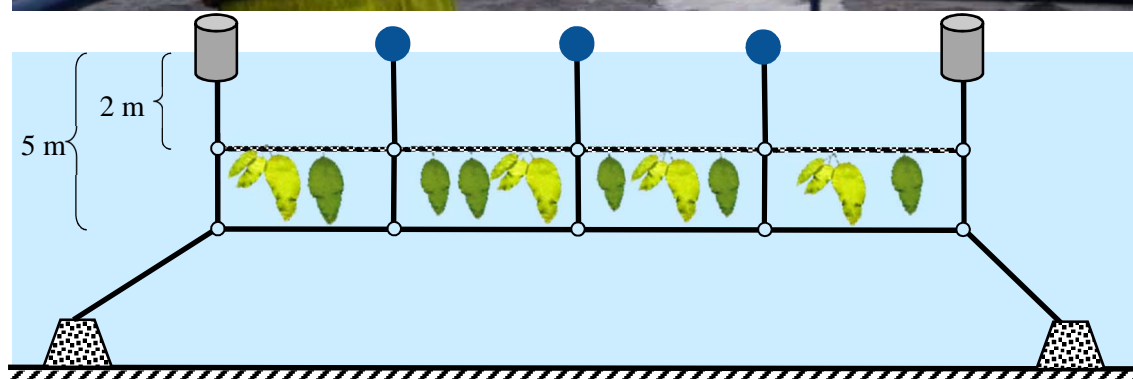


Fig. 15. Scottish Sea Farms Ltd's seaweed site (2 x 80m longlines) Sandsound South, Shetland.

A third design was also tested at Lea of Trondra using the existing structure of NAFC's replicated trial-site, consisting of 12 replicated cages, each 6 m x 6 m x 5m depth. This structure was already anchored to the seabed so instead of using buoys to hold up the ropes, they were weighed down from the cage-platform by a 2 m rope with diving weights. Using a zig-zag design, 30 m of continuous 24 mm sinking rope was deployed by hand inside each cage (see Fig 16).

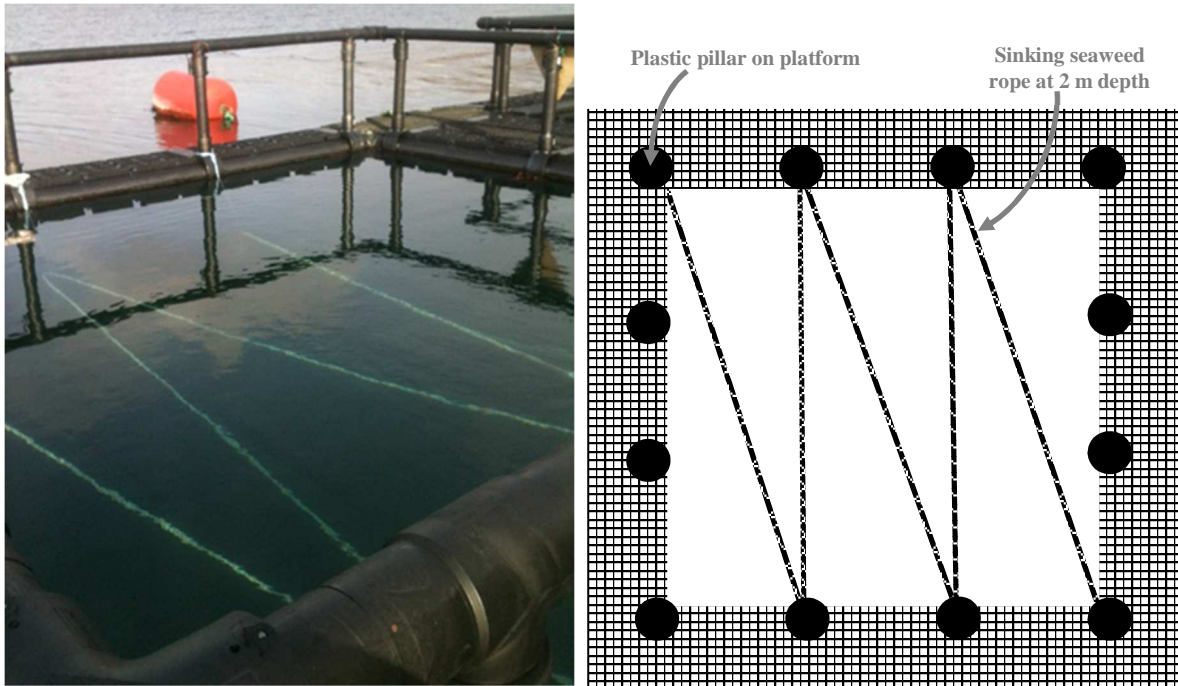


Fig. 16. Deployment seaweed culture strings on ‘zig-zag’ header lines stretched across NAFC Marine Centre’s fish trial cages at Lea of Trondra.

5.2. Deployment of cultivated kelp

Following one to two months of cultivation on spools in the hatchery (see Chapter 3), kelp seedlings are generally ready for sea deployment at a minimum length of 2mm. The optimal deployment time is between October and January for the kelps described in this manual. In 2014 and 2015, *L. digitata* was deployed in November – December at the two trial sites in Shetland. Early deployment may allow for a longer growth period but if done *too* early when there are still plenty of plankton around, fouling may result. Conversely, deploying too late may not allow the seedlings to establish themselves on the rope before fouling organisms start to settle in the spring. Optimal deployment times may vary between sites and regions.



Fig. 17. Sealable plastic containers used for transporting seaweed spools well-wrapped in muslin sheets soaked in sterile seawater. (Photo: C. Rolin).

In order to prevent damage to the seedlings during transport and deployment, the seeded string-spools need to be kept very damp with seawater, maintained at a fairly stable temperature and protected from wind-chill. This is done by wrapping the seaweed spools in muslin sheets soaked in UV-sterilised, filtered seawater inside clean, sealable plastic containers (as in Fig 17). This ensures the spools remain damp but prevents them being damaged by any 'slushing' action of seawater during transport by car and boat.

On site, the header rope is threaded through the seaweed spool and re-attached to its anchor point. Then the string of the seaweed spool is tied and spliced into the rope. The spool is then held firmly by hand while the header rope is released and the boat slowly steams along. The boat's movement will cause the string to unwind itself from the spool and around the header rope, as shown in Fig 18. Ensure the string is wound sufficiently tightly around the rope and that the string (and fingers!) do not snag. When each spool is nearly empty, the end of its culture string is spliced into the header rope and a new spool started.

Extreme temperature fluctuations can kill the young sporophytes, so ideally a calm day with modest winter temperatures, low wind and calm seas should be chosen. This will help quicken deployment and minimize air exposure of the seaweed spools and will-chill. When deploying the spools, be mindful of any subsequent abrasion along the newly seeded rope when raising/lowering the header line.



*Fig. 18. Sea-deployment of *L. digitata* at Scottish Sea Farm Ltd's Sandsound South site. Note how the seeded culture string unwinds from the spool and coils around the header rope as the boat moves along the line. (Photo: C. Rolin).*

5.3. Monitoring growth and environment

The Shetland seaweed lines were monitored monthly between March and August allowing the crop to establish themselves on the rope and develop strong holdfasts for 2 - 3 months before lifting the lines. A 15 cm sample was taken at 4 – 5 random points along the lines, cut closely to the rope to ensure the blades were kept intact and then placed in an individual Ziploc plastic bag (see Fig 19). Sampling points can be chosen by random generation or by dividing the line into segments e.g. using the buoys as markers. Immediately upon return to

shore, any excess liquid was drained from the bags and the samples were weighed in the Ziploc bag and the weight of the bag subtracted. These sample weights allow the wet yield (kg m^{-1}) to be estimated.



Fig. 19. Showing how cultivated kelp was sampled monthly from several 15cm lengths of header-rope and put into zip-lock bags for transport back to the laboratory for measurement and analysis (Photo: C. Rolin).

Environmental parameters such as temperature, turbidity and light intensity were also measured during the monthly samplings. Although seawater temperature can be sampled with a simple thermometer, it is best to sample it at 2 m depth next to the header line. A Secchi disc was lowered until it was no longer visible and the depth recorded - the higher turbidity the less light reaches the seaweed and it may also indicate algae blooms or high loads of nutrients/sediment in the water which can settle on the kelp. Light was sampled at 2 m depth by attaching a light meter to the Secchi disc. During the trials further measurements were taken using image analysis to determine growth rates, species composition, numbers of seaweed plants per metre and biofouling. After recording each sample's wet weight, they were stored in the fridge until processing.

To process a sample, all individual blades were laid down onto a white background - preferably a sheet of plastic for easy cleaning. A label was written with the date, site and sample number along with a ruler or other scale. A photograph was then taken directly from above, as close to perpendicular as possible. This was repeated for each sample. After the photographs were uploaded to a computer, each intact individual blade longer than 10 cm was labelled with a number and this ID number was attached to each measurement. Such labelling can be performed in Microsoft Paint or any other image editing programme. Using *ImageJ*, the scale was first measured and set for each sample image. The length and width were then measured using the line tool. Furthermore, the area of each blade and all fouling organisms were measured. This type of image analysis can be time consuming but provides detailed information on differences between species and sites.

After each sample was photographed, they were put in a tin foil boat and dried at 30 – 80 °C until constant weight was achieved (usually 2 -3 days). The sample dry weights were then used to estimate the dried yield per metre of rope and the percentage dry matter. The dried samples could be further analysed - for example: analysis of their chemical composition.

5.4 Harvesting

Optimal harvesting times vary from region to region and across latitudinal gradients with higher latitudes seemingly being able to harvest later into the summer. In Shetland the optimal harvest time seems to be from late May to mid-July but may vary from year to year. Early harvesting tends to generate lower yields because the plants have had less time to grow to a large size. However, late harvesting carries a high risk of severe loss in quality due to erosion and fouling organisms on the kelp. The longlines in Shetland were harvested using workboats belonging to local mussel and salmon farmers that were fitted with hydraulic winches capable of lifting the longlines out of the water (Fig 20). The kelp crop was then harvested by hand, section by section, using short knives and placed into large baskets or one tonne feed bags.



Fig 20: Seaweed longline being raised ready for harvest at Lea of Trondra, Shetland.

5.5 Growth & Yields of Cultivated Kelp

Figs. 21 & 22 show the mean *Laminaria digitata* wet and dry sample weights (cut from random 15cm lengths of longline) for both the Lea of Trondra and Sandsound South sites in 2015 and 2016, respectively.

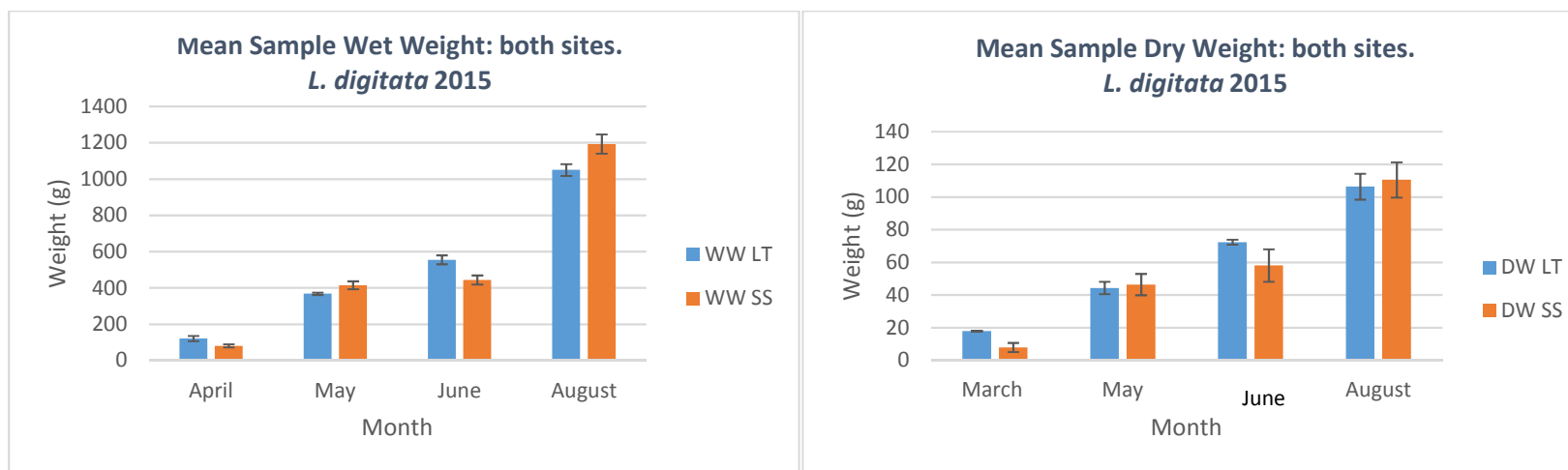


Fig. 21a & b: *Laminaria digitata* mean wet and dry sample weights from Lea of Trondra (LT) and Sandsound (SS) sea-sites in 2015. Each sample corresponded to the seaweed crop from a randomly sampled 15cm length of longline. Error bars indicated (n = 5-7).

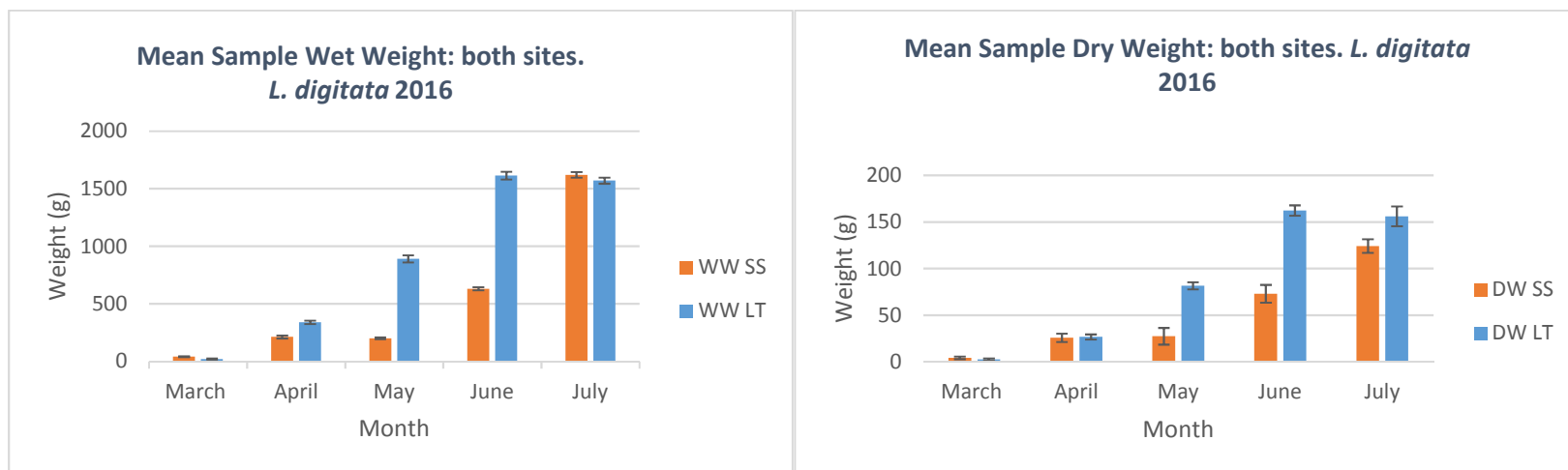


Fig. 22a & b: *Laminaria digitata* mean wet and dry sample weights from Lea of Trondra (LT) and Sandsound (SS) sea-sites in 2016. Each sample corresponded to the seaweed crop from a randomly sampled 15cm length of longline. Error bars indicated (n = 5-7).

Both longline arrangements at Lea of Trondra and Sandsound South worked well: they withstood severe gales through the winter and generated healthy crops of *Laminaria digitata* and (at Sandsound) *Alaria esculenta*. We also observed some natural settlement of *Saccharina latissima* (another useful brown kelp) on the lines at both these sites, particularly at Lea of Trondra.

As might be expected, corresponding monthly wet and dry sample weights showed similar patterns with sample weights peaking in June/July. Dry weights were approximately 10-13% of their corresponding wet weight. Sample yields were similar between both the sites within each year, but increased markedly from *circa* 3kg/m in 2015 to 10kg/m in 2016. Possible reasons for this include:

- Flask culture and spraying of seaweed spools with blended cultures was the predominant method employed in 2015, whereas the method of directly seeding culture strings, which was employed for all trials in 2016. We observed that the latter, direct seeding method seemed to generate a more evenly settled culture string and this probably translated into a more densely populated seaweed header-rope.
- Environmental variables such as weather conditions, sea temperatures and cloud cover varied between the two years.
- The 2015 *Laminaria* spools had to be held longer than recommended in the hatchery because of sea-deployment delays caused by unsuitable weather and site licence renewal. This was not the case in 2016.
- There was relatively more additional, natural settlement of *Saccharina latissima* in 2016 compared to 2015, especially at Lea of Trondra.
- Project staff were more experienced in the necessary cultivation and harvesting methods by 2016.

The 2015 August sample was badly affected by biofouling. Although the fouling organisms were removed as much as possible, the sample weights were probably still affected, so they have not been used to extrapolate yields. Because of this biofouling problem in the late summer, all harvesting and sampling was completed by July in 2016.

In January 2015, the only available site for us to deploy the project's *Alaria esculenta* spools was the NAFC's trial cages at Lea of Trondra. These strong healthy seedlings were therefore deployed on 'zig-zag' ropes attached inside the Lea of Trondra trials cages. However, they did not thrive in this location and the trial was abandoned after a few months. Their poor performance was most likely because this cage-site location was far too sheltered for this particular species, since it prefers more exposed sites. By contrast, in 2016, spools of cultivated *Alaria esculenta* were deployed at the more exposed Sandsound site and they fared much better (see Fig 23). The 2016 *Alaria* was also deployed at sea a month earlier than the 2015 seedlings (December rather than January), so this may also have impacted on the results. This example illustrates the importance of careful selection of site and deployment time.

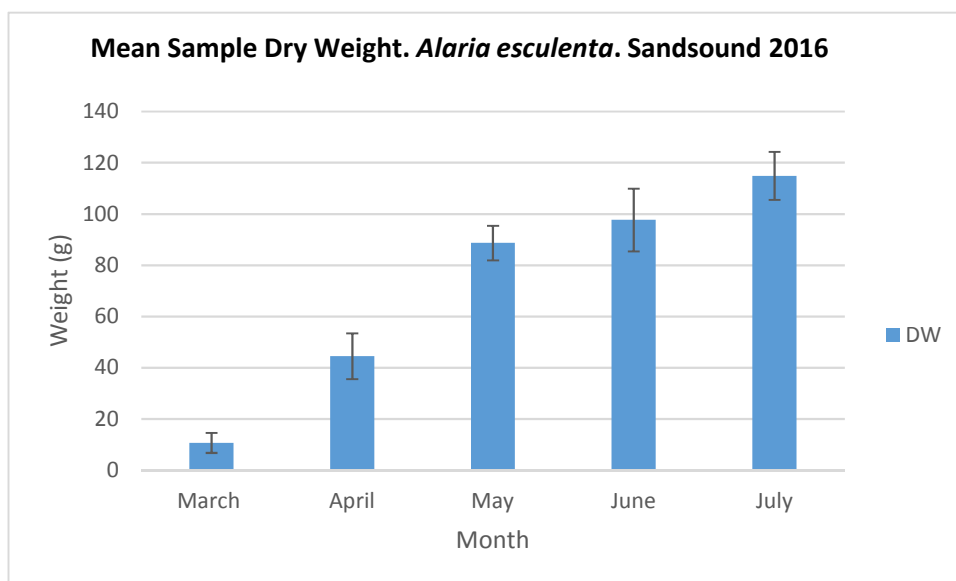


Fig. 23: *Alaria esculenta* mean sample dry weights at Sandsound South in 2016. Each sample corresponded to the crop from 15cm of longline. Error bars indicated (n=5-7).

By extrapolating from the peak (June/July) monthly seaweed sample weights for 15cm of header line, the yield from the longlines in kilograms per metre, were estimated as indicated in Table 3.

Table 3: Estimated total wet and dry yields of *Laminaria digitata* and *Alaria esculenta* (kg kelp per metre of header line) grown at Lea of Trondra and Sandsound sites in 2015 and 2016.

<i>Laminaria digitata</i>	Lea of Trondra Yields	% dry weight of LoT sample	Sandsound South Yields	% dry weight of SS sample
2015 Wet Yield	3.7 kg/m	13%	3.0 kg/m	13%
2015 Dried yield	0.48 kg/m		0.39 kg/m	
2016 Wet Yield	10.8 kg/m	10%	10.5 kg/m	10%
2016 Dried Yield	1.08 kg/m		1.04 kg/m	
<i>Alaria esculenta</i>				
2016 Wet Yield	-----		7.6* kg/m	10%
2016 Dried Yield	-----		0.76 kg/m	

*assuming approx. 10% dry weight

In May/early June of 2015 and 2016, the *Shetland Seaweed Growers Project* donated a significant portion of its seaweed crops to the *MacroBioCrude* EPSRC research project to investigate the feasibility of using ensiled seaweed for producing aviation fuel, including some early-harvests. We also donated some early-harvested seaweed to local SMEs in Shetland to encourage product diversification (see section 1.4.1 of this report for further details). These early harvests took place in April-May before the cultivated seaweed had reached its maximum weight and blade-length. However, by using the peak monthly sample weight (from June/July 2015 and 2016) in Table 3, we can estimate the potential total biomass on the longlines if all the seaweed had been left unharvested until June/July each year. These potential June/July biomasses for each species and each site are given in Table 4

Table 4: Potential Biomass Available if Entire Crop was Harvested in June/July (Max. Growth)

<i>Laminaria digitata</i>	Lea of Trondra 2 x 80m longlines	Sandsound South 2 x 245m longlines
2015 Biomass	0.6 tonnes	1.5 tonnes *
2016 Wet Yield	1.7 tonnes	5.1 tonnes *
<i>Alaria esculenta</i>		
2016 Wet Yield	-----	3.7 tonnes **

*Assuming both longlines lines are used for *Laminaria digitata*.

** Assuming both longlines are used for *Alaria esculenta*.

It is important to point out that Scottish Sea Farms Ltd's six hectare site at Sandsound has the potential to produce significantly greater harvests of seaweed if a 'continuous grid' arrangement of header lines were to be deployed (see page 56 in Edwards & Watson (2011) for further detail), rather than the two simple longlines deployed within the *Shetland Seaweed Growers* project. However, we were advised that it can be very difficult to monitor and harvest crops grown in grid-systems because of the lack of boat-access inside the grid and we therefore made the decision to use longlines which are easier to deploy and access.

The *Shetland Seaweed Growers* Project noticed that the kelp growing on our longlines had relatively short blades compared to reports from other regions. For example, after one year's growth in Shetland, *Saccharina latissima* harvested from our longlines reached an average length of 40cm, with some individuals exceeding 100cm, while our *Laminaria digitata* reached an average length of 30cm and a maximum length of 65cm (Rolin *et al.* 2017). By contrast, Edwards & Watson (2011) reported *L. digitata* lengths ranging from 40cm to 100cm in June during their Irish cultivation trials and *S. latissima* lengths of 80cm to 150cm. However, it is difficult to make direct comparisons between studies because of large natural variations in growth caused by different locations, weather conditions, temperature, light levels, seaweed species, cultivation techniques and sampling methods.

5.6. Seaweed Drying Facilities

Although small 15cm monthly samples can be easily dried in an oven, bulk harvests require dedicated drying facilities. In the *Shetland Seaweed Growers* Project, large seaweed harvests were brought back to the NAFC Marine Centre where the plants were immediately sorted and distributed between stackable, plastic 'bread trays' (as used by many supermarkets) and allowed to drain and partially dry overnight in a well ventilated room. (Being Shetland, it was generally necessary to keep them indoors to avoid risk of wet weather or strong winds, however in other more clement areas, they could probably be left outside to dry). The next day, NAFC's harvested plants were transferred to a disused walk-in

freezer that was converted as a dedicated drying room by adding externally-draining dehumidifiers, fans and heaters. The crop would then be dried in batches by spreading the plants out on metal racks (Fig. 24) that were kindly loaned for use within the project by Grieg Seafoods Ltd., Shetland.



Fig 24. NAFC Marine Centre's dedicated seaweed drying room: a redundant walk-in freezer fitted with drying racks, dehumidifiers, heaters, and fans.

Preliminary Conclusions from the Project Trials:

- Both longline arrangements used within the Shetland Seaweed Growers project worked very well, withstanding severe gales through the winter and generating healthy crops of kelp in the summer.
- Careful selection of sea-sites is vital to ensure the prevailing environmental conditions and level of exposure suit the seaweed species being cultivated so that growth is maximized.
- Direct seeding of culture string saved a lot of time, labour and electricity costs compared with flask cultivation and appeared to generate more evenly-seeded strings than indirectly seeded (sprayed) strings.
- Natural settlement of *Saccharina latissima* on some of the project's *Laminaria digitata* lines may have helped to enhance the overall kelp harvest, but may also have limited the growth of the *Laminaria* plants to some degree.
- There was large natural variation in seaweed growth, both within and between sites.
- Biofouling, particularly by sea squirts, became a serious problem in late summer but varied in intensity between the two project sites.
- Crops need to be monitored regularly (at least monthly during the summer) to ensure harvesting occurs at peak biomass and quality, before biofouling and/or blade breakage and erosion cause severe degradation of the crop.
- Seaweed harvested in Shetland should be used within the isles as far as possible because of high freight costs to the mainland.
- Bulk drying of seaweed requires dedicated facilities and is costly in cold/wet regions such as Shetland. Other methods of preservation may be more suitable, such as ensiling which is currently being trialled with seaweeds within the EPSRC-funded *MacroBioCrude* project.
- In addition to allowing two Shetland aquaculture companies to work alongside the NAFC Marine Centre to trial the various techniques and methodologies required for seaweed farming, the *Shetland Seaweed Growers* project succeeded in encouraging nine other local Shetland businesses to diversify by incorporating seaweed (both cultivated and wild harvested) into their products. These seaweed products have been very positively received and are planned to continue after the life of the project.

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