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REVIEWS IN Aquaculture

REVIEW

Quantifying habitat provisioning at macroalgal cultivation sites

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Abstract

Macroalgal cultivation is expanding rapidly, and promises to contribute significantly towards future food and energy security, sustainable livelihoods, ecosystem services and habitat provisioning for a range of associated organisms globally. Habitat provisioning underpins biodiversity and ecosystem structure and functioning, supports many ecosystem services and has possible benefits to other marine industries, including enhancement of commercial fish stocks. In macroalgal cultivation, however, only recently has habitat provisioning started to be assessed at a local scale (within a farm's footprint) and with a range of different approaches. This review evaluates techniques used to quantify habitat provisioning in and around macroalgal cultivation sites, for species ranging from microorganisms to megafauna, and outlines recommendations to enable a more comprehensive ecological valuation of macroalgal cultivation in the future. The majority of information on biodiversity associated with macroalgal cultivation is associated with quantifying biofouling or pest organisms, rather than the contribution of colonising species to healthy ecosystem functioning. We suggest how better monitoring of macroalgal cultivation could enable an ecosystem approach to aquaculture (EAA) in the future. To achieve this, we highlight the need for standardised and robust methods for quantifying habitat provisioning that will enable assessment and monitoring of macroalgal cultivation sites of varying scales and within different regions and environmental settings. Increased evidence for the potential habitat value of macroalgal cultivation sites will help inform and shape marine legislation, licencing and certification for macroalgal farmers and potentially reduce marine user conflicts, helping the industry to continue to grow sustainably using EAA.

KEYWORDS
census techniques, ecosystem services, environmental monitoring, marine ecology, seaweed aquaculture

1 | INTRODUCTION

Aquaculture is the fastest growing marine sector globally, and it promises to contribute significantly to future food and energy security.[1] Algae, including seaweeds (macroalgae), form >20% of total aquaculture production biomass, and algal cultivation is growing rapidly compared with other farmed species (approximately 8% yr^{-1}) following the growth and diversification in global markets for algal or macroalgal products.[1–4] Beyond the direct commercial value of macroalgal products, the responsible expansion of macroalgal cultivation

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could provide environmental benefits, which have been detailed alongside potential negative impacts in several recent reviews[5–13] (summarised in Figure 1). Many of these environmental effects relate to key ecosystem services (ESs) including climate regulation, storm protection, biogeochemical cycling and provisioning of food and habitat, or refugia to support secondary production for wild capture fisheries[6,14–17] (Figure 1). The proposed ESs enhanced by macroalgal cultivation would support several UN sustainable development goals including: contributing to global health and well-being; providing economic growth and resilience in coastal communities; enabling responsible consumption and production; facilitating climate action and benefiting marine ecosystems.[18]

Habitat provisioning is a hugely important ecological process that underpins biodiversity, ecosystem structure and function, and

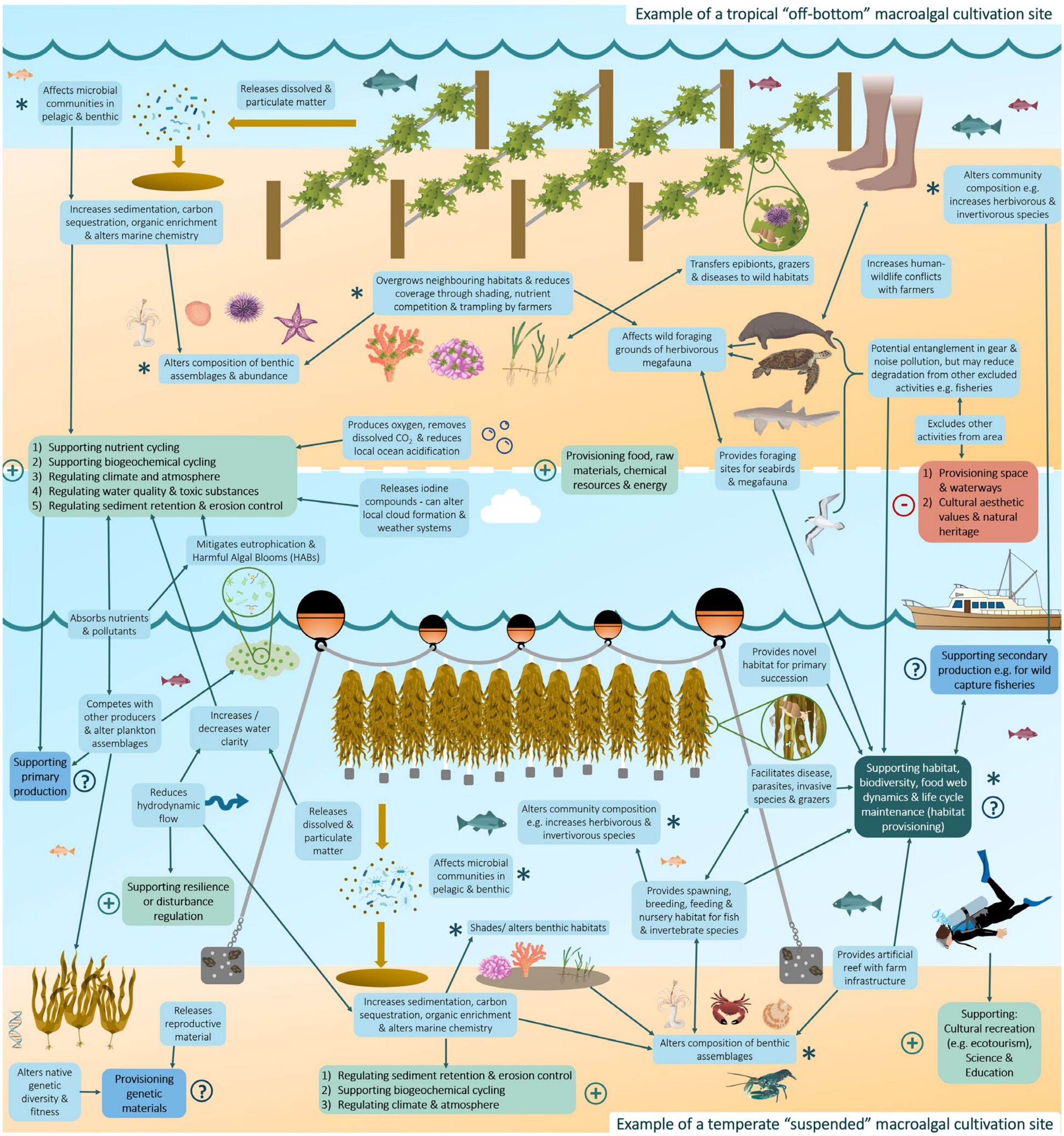

FIGURE 1 Summary of the proposed environmental impacts (pale blue) and potential effects on ecosystem services of a tropical 'off-bottom' macroalgal cultivation site (above) and a temperate 'suspended' macroalgal cultivation site (below) as derived from recent reviews.[5–13] Potential effects on ecosystem services detailed as positive (green (+)), negative (red (−)) and neutral or undetermined (blue (?)) and with habitat provisioning highlighted (dark teal). Some effects linked to habitat provisioning are indicated with an asterisk (*) to aid with clarity. *Graphics are from the Integration and Application Network, University of Maryland Centre for Environmental Science* (http://ian.umces.edu/imagelibrary/) *and BioRender* (*Biorender.com*)

supports many ESs such as food provisioning, water quality, maintenance of pest and disease control and recreation and ecotourism.[19] Habitat provisioning is included differently under prominent ES classification systems, and they are (1) maintaining habitats and nursery populations[20]; (2) refugia, or nursery and migration habitat[21]; (3) habitat heterogeneity[22]; and (4) life cycle maintenance – nursery service.[23,24] This broad range of terminology makes defining and quantifying the value of habitat provisioning difficult, with vague metrics in place to do so.[25] Furthermore, as shown in Figure 1, there may be complex interactions between species, which are difficult to separate and monitor. In the broadest sense, habitat provisioning will vary based on abiotic conditions, farm location or farm type. For example, an offshore kelp farm in Europe will provide a very different potential habitat to a shallow-bottom eucheumatoid farm in Southeast Asia. All macroalgal cultivation sites, however, present their own challenges for monitoring habitat provisioning, which has until now limited their study.[17]

Quantifying the habitat provided by macroalgal cultivation has received little attention, and therefore, no economic or ecological valuations of this potential benefit have yet been made.[16,17,25] In a recent review evaluating the available literature on the habitat value of bivalve and macroalgae cultivation of the 65 studies identified, only eight of these included macroalgal cultivation sites.[17] This review also only included the habitat value of aquaculture sites for wild macroinvertebrate and fish populations because of a lack of information on how macroalgae cultivation affects other species[17] (e.g. microorganisms, marine mammals or seabirds etc.). Macroalgal cultivation can, however, provide habitat for a diverse array of fauna and flora, similar to that of wild macroalgal populations,[26] through the provision of novel suspended and benthic three-dimensional substrates, food and enhanced reproduction and recruitment opportunities.[5,7,13,17] This in turn could potentially support secondary food production with spill-over benefits for fisheries.[16,17] It is unclear, however, how effective temporary habitats of seasonal macroalgal cultivation sites would be at maintaining biodiversity after harvests and the removal of macroalgal biomass. It is also unclear whether sites will simply aggregate wild populations instead of enhancing overall population size, production and viability through reproduction and juvenile recruitment.[5,7,16,17] Further issues relate to which species macroalgal farms will support, and whether these will differ from surrounding areas, thus potentially altering ecosystem dynamics or introducing invasive species.[27] Better monitoring of species at macroalgal cultivation sites is therefore needed to address these concerns and determine whether this form of aquaculture supports habitat provisioning while quantifying what its ecological (and economic) value may be.

Increased recognition and valuation of the habitat provisioned by macroalgal cultivation would enable better farm design and management to optimise potential environmental benefits, mitigate potential negative impacts and contribute towards sustainable development and an ecosystem approach to aquaculture (EAA).[8,16,25,28] EAA aims to design and integrate aquaculture within ecosystems to promote sustainable development, equity and ecosystem resilience while minimising any potential negative impacts.[29,30] In macroalgal cultivation, EAA may guide policy, financing and certification schemes towards promoting increased sustainable practices in mariculture development.[11] Accordingly, there is the potential for macroalgal aquaculture to lead the way as an example of sustainable EAA; however, more quantitative evidence on a wider range of the potential environmental benefits is needed, including habitat provisioning.[25]

This review aims to: (1) summarise evidence relating to how macroalgal cultivation could provision habitat for species spanning from microorganisms to megafauna, and (2) generate recommendations for standardised monitoring of habitat provisioning in and around macroalgal cultivation sites for these species. In turn, this could support the development and optimisation of practices for EAA, which will enable the ecological and economic value of macroalgal aquaculture to be assessed more holistically and help to inform its legislation and regulation in future. Where information on macroalgal cultivation is lacking, we draw upon some relevant studies from wild macroalgae populations and shellfish/finfish aquaculture to help guide in developing universal standardised monitoring techniques. Adopting this approach, we also hope to seek ways of standardising the monitoring of habitat provisioning between aquaculture species, which will be particularly useful given the increasing implementation of integrated multitrophic aquaculture (IMTA) systems.

2 | POTENTIAL HABITAT PROVISIONING OF MACROALGAL CULTIVATION FOR DIFFERENT TAXONOMIC AND FUNCTIONAL GROUPS AND METHODS ENABLING ITS QUANTIFICATION

As Figure 1 illustrates, complex interactions exist in macroalgal cultivation sites between species and between species and the environment, which will differ depending on farm scale, location and type (discussed further in Section 3). These complexities are more explicitly detailed in other recent reviews (e.g. Refs. 10,17), and in this section, we outline some of the ways macroalgal cultivation may affect different taxonomic and functional groups based on previous studies, why they are important to quantify and challenges relating to monitoring. We have grouped organisms based on their taxonomic and functional groups, how they inhabit or interact with macroalgal cultivation sites, and their monitoring requirements.

2.1 | Microorganisms (Bacteria, viruses, archaea, fungi, oomycetes and protists)

Bacteria, viruses, archaea, fungi, oomycetes (fungus-like eukaryotic water moulds) and protists are often overlooked in habitat evaluations and biodiversity assessments in aquaculture, despite their importance in maintaining ecosystem health and functioning through nutrient and carbon cycling, decomposing organic matter and helping prevent diseases or harmful algal blooms (HABs).[31–35]

Macroalgae host diverse associations of microorganisms that facilitate the health and function of the host plants, such as morphological development, disease protection and antifouling properties from epibionts[36–38] (see Section 4). As such, macroalgae and their microbiomes should be considered as synergistic ecological units or holobionts.[36,38] Additionally, the microorganisms associated with macroalgae are often sources of novel compounds that have industrial applications, for example, bacteria hosted on *Eucheuma* species produce enzymes that could be used in biofuel production.[39] However, changes in the microbiome structure and bacterial infections may also contribute to several diseases (summarised in Ref. 40), including ice–ice rotting syndrome, which has been known to decimate entire *Kappaphycus* and *Eucheuma* seaweed farms, causing significant economic losses.[34,41–47] Oomycete pathogens can also be responsible for extensive damage, as seen in Korean *Pyropia* seaweed farms,[48] and are more diverse and geographically widespread than currently acknowledged, posing threats to macroalgal cultivation in Asia and Europe.[49,50] The presence of harmful microorganisms and diseases at macroalgal cultivation sites also pose the risks of spreading to neighbouring wild macroalgal populations, with the potential to cause substantial ecological damage.[51] A greater understanding of the microbiome of macroalgal cultivation is therefore needed to determine both its ecological value in macroalgal cultivation,[8] and understand how the macroalgal microbiome is regulated by other associated biodiversity present at cultivation sites, which may mitigate disease outbreaks.[35,52] Macroalgal cultivation sites may also potentially affect microorganisms in the water column and benthic sediments, through production of soluble dissolved and particulate organic matter,[53] deposition of detritus and the potential attraction of waste-producing species such as fish (Figure 1). Microorganisms in the surrounding environment, however, are unlikely to be affected to the extent seen in other aquaculture species, particularly fishfarms,[54] as waste production is comparatively low (although less is known about the amounts produced at larger offshore sites). Determining interactions between macroalgal cultivation sites and microbial communities in the water column and benthos will be an important step in future research, given that microorganisms can act as powerful indicators monitoring ecosystem health.[54] Relatively little research has been conducted so far to investigate this.

Microorganisms can be quantified either from the surface of the macroalgae or from water or sediment samples in macroalgal cultivation sites, which can host distinct microbiomes from one another.[34] The diversity and community composition of microorganisms, such as fungi, can vary between tissue types (e.g. stipe, holdfasts and blades) of the same macroalgae,[55] so multiple samples from individuals should be collected to accurately capture their microbial diversity. Various methods can be used for microorganism quantification (Table 1), including microscopy, cell counts and RNA or DNA sequencing methods.[35,38,54,56] The main constraints for quantifying macroalgal farm microbiomes, however, are generally a lack of knowledge of the microbial ecology of these systems and because currently many are not culturable by common microbial methods[34,35,41,45,46]; however, the advancement of molecular methods may help to mitigate these issues[38,55] (Table 1). Additionally, eukaryotic microorganisms, including protists, are especially understudied within the seaweed holobiont, so particular focus needs to be more directed on determining their importance. If sampling effort of microorganisms in wild and cultivated macroalgal populations was increased, reference libraries should be compiled to make detecting and quantifying their abundance easier in future. It should also be recognised that better understanding of the dynamics and plasticity of these microbial communities is needed, as there will be similarities in their ecological and functional roles within ecosystems.

2.2 | Plankton (pico to macro)

Plankton provide important primary food sources in marine food webs, and regulate nutrient, carbon and oxygen cycles in the oceans. Therefore, plankton abundance and diversity are important measures of ecosystem productivity and health. Macroalgal cultivation sites may provide zooplankton with shelter and food[57]; however, plankton may also negatively affect cultivated macroalgae. For example, ‘diatom felt’ caused by settling diatoms has been shown to result in algal bleaching and significant economic losses for farmers in *Pyropia* farms in Korea.[48] Macroalgal cultivation sites may also benefit overall ecosystem health by mitigating eutrophication and harmful algal blooms (HABs) through improving water quality, stabilising the water column and assimilating excess nutrients[48,58–60] (Figure 1). The installation of macroalgal farms in China has been observed to alleviate eutrophication and ocean acidification, reduce turbidity and subsequently enhance phytoplankton diversity and biomass.[61,62] As primary producers, however, macroalgae and phytoplankton may compete for light and nutrients,[63–65] particularly at large-scale cultivation sites (>50 lines × 200 m)[7] or those with high macroalgal biomass density, like that in Sanggou Bay, China.[66–68] This competition in turn may reduce food availability for wild fish or shellfish populations and affect fisheries or shellfish production.[69–71] Small- to medium-scale (0–50 lines × 200 m)[7]macroalgal farms, however, are unlikely to have major impacts on phytoplankton assemblages as phytoplankton in most cases will likely pass through the site with current flow.[7,16] Farms should be sited in suitable locations with sufficient nutrient concentrations and tidal mixing, and therefore with adequate environmental carrying capacity (the maximum biomass of a farmed species that can be supported without exceeding the maximum acceptable impacts to the farmed stock and its environment)[72,73] (see Section 3.5). Indeed, no significant changes in plankton were predicted or detected in models of hydrodynamic and biogeochemical processes in United Kingdom and Dutch small- or large-scale macroalgal farms.[71,74] Thus, the effects of macroalgal cultivation on plankton assemblages are site-specific

TABLE 1 List of suitable monitoring techniques to assess habitat provisioning in macroalgal cultivation sites for different taxonomic and functional groups. Potential pros and cons for each technique are discussed with recommendations for improved use and examples of previous usage in macroalgal cultivation sites and other similar habitats

Monitoring technique & species group(s) it can be used for	Pros (+) and cons (−) for use in macroalgal cultivation sites to assess habitat provisioning	Recommendations for future use	Example uses in macroalgae (M), finfish (F), shellfish (S) aquaculture, wild macroalgal populations (W) or other (O)
Water sampling, plankton nets or trawls *Microorganisms and plankton*	Inexpensive, simple and highly replicable (+) Species can then be identified by several methods e.g.: • Microscopy – time-consuming and requires taxonomic expertise, but captures total biodiversity at a given time point (−/+) • Fluorescence microscopy – a fast and direct detection protocol for microorganisms (+) • Fluorometry – rapidly measures chlorophyll *a* concentrations as proxies for productivity and phytoplankton biomass (+). Does not identify species or detect zooplankton species (−) • Automated or semi-automated systems for quantifying plankton e.g. flow cytometry or FlowCAM (see Refs. 68,161,162) – faster but require established reference libraries or taxonomic expertise (+/−) • Molecular methods (see below) – faster but require established reference libraries that are currently lacking (+/−)	Should standardise depth and volume of samples, mesh size of nets, time of day and year of surveying, number of replicates taken and post-analysis methods for identification and enumeration of species Should create reference libraries of microorganism and plankton DNA or RNA in cultivation sites and wild populations to aid future quantification[34]	Microorganisms (reviews): e.g. Refs. 34 (M); 35,56 (F); 38 (W & O) Plankton: e.g. Refs. 68,76,120,163 (M); 164 (S, F & M); 165 (S); 161,162 (O)
Sediment and light traps *Microorganisms, plankton, pelagic invertebrates, fish and decapod larvae*	Sediment traps are deployed on the seabed and can collect plankton over longer time periods (e.g. monthly) for a more representative diversity (+) Sediment traps are not suitable for offshore farms due to depth limitations of setting up traps (−) Light traps can be deployed in the water column at night to collect species with nocturnal migration patterns, including amphipods and other major epibiont species that would otherwise not be present during the day[166] (+) Both are low-cost and have similar post-analysis methods used to water sampling methods (above)	Should set traps at fixed depths from the seabed or in the water column within the farm and at reference areas ahead of typical seasonal or predicted plankton blooms Timing and duration of light trap deployment should be recorded for standardisation, along with environmental conditions. Concurrent plankton trawls should be conducted to determine sampling bias[166]	Microorganisms: e.g. Ref. 167 (F) Plankton: e.g. Ref. 168 (M) Pelagic invertebrates, fish and decapod larvae: e.g. Ref. 166 (F)
Autonomous detection systems *Microorganisms and plankton*	Deployable in situ for continuous plankton monitoring and instantaneous harmful algal bloom (HAB) alerts,[75] but does not quantify total community assemblages (+/−) Currently further development is needed before real-time microorganism detection systems are available,[169] but portable sensors and combined modelling methods may aid in development (−/+) Commercial monitoring systems are expensive for small-scale farmers, but low-cost options are being developed (e.g. Refs. 170,171) (−/+) Requires good connectivity for instantaneous data transfer i.e. 4G networks, which may not be available in all farm locations (−)	Should standardise sensors used and their placement on farms Should invest further research and development into these systems to streamline data connectivity and lower production and running costs for use in small-scale farms Should ensure systems are simple to use for operation and maintenance by farmers	Microorganisms: e.g. Refs. 172 (O); 169 (review) Plankton: e.g. Refs. 170,171 (S)

(Continues)

TABLE 1 (Continued)

Monitoring technique & species group(s) it can be used for	Pros (+) and cons (–) for use in macroalgal cultivation sites to assess habitat provisioning	Recommendations for future use	Example uses in macroalgae (M), finfish (F), shellfish (S) aquaculture, wild macroalgal populations (W) or other (O)
Collecting species on macroalgal biomass *Microorganisms and epibionts*	Easy to collect samples on farm maintenance or harvest trips (+) Species can be then identified by several methods e.g.: • Microscopy or by eye – time-consuming and requires taxonomic expertise, but captures total biodiversity at a given time point above a visible size class (−/+) • Molecular methods (see below) – faster but requires established reference libraries (+/−) • Photographic analysis (e.g. using ImageJ or equivalent software) – standardises measurements for sessile epibionts, but excludes less abundant mobile species, and those obscured in holdfasts (+/−)	Should standardise methods for collection, preservation, species identification and enumeration Should assess all parts of the plant and account for habitat volume of holdfasts or surface area of blades and stipes, as species richness scales with plant size.[173] These methods are described in Refs. 88,105 Should combine image analysis and taxonomic identification (e.g. Ref. 83) for increased accuracy	Microorganisms: e.g. Refs 55,174–177 (W) Epibionts: e.g. Refs 88,101-105 (M);83,173,178,179 (W)
Benthic sediment grabs or diver collected cores *Benthic microorganisms and infauna*	Routinely practised in marine ecology so techniques are relatively standardised[180] (+) Constrained by depth and substrate type, e.g. sampling offshore farms or those suspended over bedrock is challenging (−) Species can be then identified via: • Taxonomy – expensive, time-consuming and requires expertise (−) • Molecular methods (see below) – faster but requires established reference libraries (+/−)	Should conduct pre-surveys via side-scan sonar or drop-down cameras to determine substrate types and any vulnerable or protected habitats to be avoided[5] Should use standardised monitoring methods, e.g. Refs. 181,182, and diverse biotic indices and statistical approaches (e.g. Refs. 117,183)	Benthic microorganisms: e.g. Refs. 184–186 (F) Benthic infauna: e.g. Refs. 100,101,118,120,121 (M); 184 (F)
Small beam trawls *Benthic epifauna*	Routinely conducted and replicable around different benthic sites and effective at assessing total community assemblages (+) Relatively destructive and may be hard to conduct around farm infrastructure or directly below farms (−) High levels of settled farm detritus may clog nets and hinder accurate quantification of epifauna (−) Can subsequently determine biometrics, age/size and gut contents of epifauna (+)	Should standardise methods (e.g. tow size, net size, tow length and duration, sample preservation, species identification and enumeration). Where trawl sizes differ, data should be standardised by area/volume Static sampling methods (e.g. trap or camera surveys) are more appropriate around aquaculture infrastructure	Benthic epifauna: e.g. Refs. 187 (S); 188 (F); 180 (O)
Traps or nets *Benthic epifauna and finfish*	Relatively simple to deploy below or around cultivation sites for crustaceans and fish species (e.g. Ref. 101) (+) Traps may be particularly effective for surveying species of commercial importance, but will be biased for certain species (+/−) Can subsequently determine biometrics, age/size and gut contents of caught individuals or tag them (see mark recapture or biologging below) (+)	Should be used with other survey methods to derive whole-site biodiversity Should standardise methods (e.g. net or trap size, length of deployment, baited or unbaited). Where length of deployment differs among studies, data should be standardised to catch per hour	Benthic epifauna: e.g. Refs. 101 (M); 187 (S) Finfish: e.g. Refs. 129 (M); 187,189 (S); 130 (S, F & O)

TABLE 1 (Continued)

Monitoring technique & species group(s) it can be used for	Pros (+) and cons (−) for use in macroalgal cultivation sites to assess habitat provisioning	Recommendations for future use	Example uses in macroalgae (M), finfish (F), shellfish (S) aquaculture, wild macroalgal populations (W) or other (O)
Diver/snorkelling surveys using visual counts or handheld cameras *Epibionts, benthic epifauna, finfish, mammals, reptiles and seabirds*	Non-extractive, routinely conducted and replicable around benthic and pelagic sites (+) Can bias towards counting conspicuous and abundant species over cryptic or rare individuals that may be hard to spot within macroalgal farms[126] and may only capture species a set time point or time of day e.g. does not account for diurnal movements of organisms (−) Not suitable for benthic surveys in offshore farms that may be beyond diving depth limits (−) Divers presence may cause additional disturbance to mobile species affecting counts (−)	Should standardise methods for surveys e.g. either point counts, transect lengths and widths or manta tows etc. and describe search effort and time of day Should use cameras where possible to increase replicability and reduce individual observer bias (see remotely operated camera surveys below)	Benthic epifauna: e.g. Refs. 125 (S); 124,190 (F); 191–193 (O) Finfish: e.g. Refs. 75,135 (M); 194 (S); 27,131,195-203(F); 204,205 (O) General reviews of method: e.g. Ref 206
Remotely operated camera surveys (below surface) *Epibionts, benthic epifauna, finfish, marine mammals, reptiles and seabirds*	Efficient, non-extractive, relatively inexpensive and highly replicable[134,207] (+) Easy to use around farm infrastructure, in high energy environments and varied depths, including offshore systems[126,208,209] (+), but dependent on weather, sea state and visibility[209] (−) Surveys multiple species and their size classes simultaneously (+), but biased towards larger, more conspicuous individuals over smaller, or cryptic ones that may be obscured by macroalgae or present in holdfasts[134,210,211] (−). Can be baited to attract fauna, but this introduces bias[134] (−) Different camera specifications and post-analysis techniques affect estimates of species richness, density, cover and assemblage composition[212] (−) Post-analysis is time-consuming, but can be streamlined using motion detection software and machine learning (e.g. Refs. 209,213) (+), although this may be ineffective in seaweed farms that are in constant motion[208] (+/−)	Should create standardised monitoring protocols (e.g. describing camera specifications, set-up and deployment, baited or unbaited systems, duration and time of day for deployment, etc.) and video analysis guidelines to limit observer bias between studies.[126] Should be used in combination with other monitoring methods to detect cryptic or hidden species	Epibionts: e.g. Ref. 213 (O) Benthic epifauna: e.g. Refs. 126,214,215 (F); 191,193,210,216(O) Finfish: e.g. Refs. 208 (M); 209 (S); 217,218 (F); 211 (W) General reviews of method: e.g. Refs. 134,206,207,219

(Continues)

TABLE 1 (Continued)

Monitoring technique & species group(s) it can be used for	Pros (+) and cons (−) for use in macroalgal cultivation sites to assess habitat provisioning	Recommendations for future use	Example uses in macroalgae (M), finfish (F), shellfish (S) aquaculture, wild macroalgal populations (W) or other (O)
Aerial or drone surveys *Finfish, marine mammals, reptiles and seabirds*	Similar to remotely operated camera surveys (above), but can cover a wider area to facilitate monitoring low-density populations of large species (+) Restricted to near-surface species e.g. mammals, birds, reptiles and planktonic elasmobranchs, and may cause disturbance or be unsuitable near seabird colonies (−) May be obscured by dense macroalgal biomass, glare and seabed bottom type (−) Aerial surveys with planes are expensive (−), but lower cost drone surveys are increasingly common (+), however still requires a licensed drone pilot (−) Can simultaneously monitor crop production and success for farmers[220] (+)	Similar recommendations to remotely operated camera surveys (above) in terms of developing standardised protocols,[159] camera specifics and video analysis techniques. Should be used in combination with other monitoring methods to detect non-surfacing or smaller species	Finfish: e.g. Ref. 221 (F) Marine mammals, reptiles and seabirds: e.g. Refs 222–225 (F); 226,227 (O) Using drones around macroalgal cultivation sites in general: e.g. Refs. 220,228
Tagging or biologging/ telemetry *Benthic epifauna, finfish, marine mammals, reptiles and seabirds*	Monitors site fidelity, behaviour of tagged individuals (i.e. use of macroalgal farms as foraging, breeding or nursery grounds) and spill-over effects, to determine whether farms facilitate species recruitment or affect migration patterns (e.g. Refs. 149,189,229) (+) Relies on capturing individuals using traps, nets, trawls etc. and can be invasive, expensive and technical if using electronic tags, requiring specialist knowledge and permits, which may not be suitable for small-scale farmers (−)	Should be used in combination with other methods to determine abundance and diversity of species. Should adhere to tagging and ethical guidelines to limit harm to tagged individuals (e.g. Refs. 230,231)	Benthic epifauna: e.g. Refs. 232 (S); 233 (O) Finfish: e.g. Refs. 189 (S); 229,234 (F) Mammals, reptiles and seabirds & reviews of method: e.g. Refs. 236–238
Acoustic surveys: echosounders, hydrophones or passive acoustic monitoring *Benthic epifauna, finfish and marine mammals*	Efficient, non-extractive, inexpensive and replicable (+) Echosounders can detect morphological, geographical and behavioural data of fish around cultivation lines[239] (+) Can be conducted at any time of the day[240] and over a large area to assess how fish distribution is affected by cultivation on a larger scale[241] (+) Do not face constraints of visual surveys, but echosounders cannot identify species without other verification methods (e.g. capture) (+/−) Passive acoustic monitoring devices only detect species that vocalise, e.g. cetaceans (e.g. Ref. 242), but could readily be installed on farms (−/+)	Echosounders should be combined with hand-lining/netting to verify species present (e.g. Ref. 241).	Benthic epifauna: e.g. Ref. 120 (M) Finfish: e.g. Refs. 120 (M); 239 (S); 240,241 (F) Marine mammals: e.g. Ref. 242 (O)

TABLE 1 (Continued)

Monitoring technique & species group(s) it can be used for	Pros (+) and cons (−) for use in macroalgal cultivation sites to assess habitat provisioning	Recommendations for future use	Example uses in macroalgae (M), finfish (F), shellfish (S) aquaculture, wild macroalgal populations (W) or other (O)
Molecular methods: e.g. environmental DNA (eDNA), antibody techniques *All groups*	Non-extractive, low-cost, highly replicable and efficient method for multispecies and community analysis, particularly for detecting cryptic, rare or low abundance species that are hard to detect or identify via visual methods. Genetic material can be collected from water, sediment or macroalgal samples easily to detect current or recent presence of species (+) DNA may degrade rapidly under unfavourable environmental conditions (e.g. high temperatures, turbulence, salinity and UV radiation), or depending on the biology of the species.[243] Samples may also be contaminated (e.g. by faeces of non-target individuals or via currents from neighbouring areas) (−) Can distinguish community assemblages separated by only 60 m,[244] and estimate biomass and abundance from quantitative models, but may overestimate during spawning periods due to abundant genetic material[243] (+/−) Allows rapid detection of harmful microorganisms and HABs,[245,246] but dependent on reference libraries, which are currently lacking (+/−)	Need standardised survey and DNA extraction methods, e.g. the time of year for sampling, ensuring suitable replicates are collected to account for dilution in aquaculture, standardisation of filtration pore size for water and sediment samples, and ensuring standardised sequencing methods are used[247] Need improved reference libraries for effective screening of samples, particularly for metabarcoding (the screening of many taxa in one sample simultaneously) Should be deployed alongside other census methods to increase accuracy and confirm species present	Microorganisms and plankton: e.g. Refs. 55 (W); 54,248,249 (F); 247,250,251 (O); 246 (methods review) Finfish: e.g. Refs. 252 (O); 243 (methods review) Marine mammals: e.g. Ref. 242 (O) Multiple species: e.g. Refs. 253 (M); 244,247,254,255 (W); 256 (O)
Visual surface searches by experienced observers *Finfish, marine mammals, reptiles and seabirds*	Weather, sea state and daylight dependent, which may bias against nocturnal species or shorter day lengths[142,145,146] (−) Time-consuming and restricted to inshore farms unless boats are used to access offshore farms but have the additional cost of boat time and the risk of causing disturbance (−)	Should be conducted before potential disturbance caused by farming to gather baseline data, during any potentially disruptive events e.g. harvesting, and at set time points after disturbance to assess longevity of impact Should be used in combination with more continuous or in depth methods to gain a more holistic assessment of habitat value	Marine mammals, reptiles and seabirds: e.g. Refs. 140,142,143,146 (S); 145 (F); 257 (S & F)
Farmer surveys or interviews and citizen science observations or photos *Epibionts, benthic epifauna, finfish, marine mammals, reptiles and seabirds*	Source of low-cost data, where farmers or public contribute sightings/photos of species to central databases or in questionnaires (+) Can yield data on the size range, behaviour, seasonality and frequency occurrence of notable species comparable to other survey methods (+) Enables stakeholder engagement and educates and engages public in the potential benefits of macroalgal cultivation, facilitating social licencing (+) Farmers may visit cultivation sites at different frequencies depending on species cultivated and farm needs. High frequency visits may capture more data but cause additional disturbance to species, whereas more infrequent visits could result in sporadic reporting and could miss key visiting times for certain mobile or short-lived species (+/−) Citizen science surveys only applicable for publicly accessible coastal farms and reference sites (−)	Should be used as a first approach method to establish general species distribution and abundance in the area and then in combination with other more continuous and in depth methods to gain a more holistic assessment of habitat value Should provide training and simplified methodology guidelines to produce more rigorous and accurate data sets Should increase education and awareness of the importance of healthy marine ecosystems and the ESs they provide to motivate and incentivise surveys	Benthic: e.g. Refs. 258 (F); 259 (O) Finfish: e.g. Ref. 235 (F) Marine mammals, reptiles and seabirds: e.g. Refs. 260,261 (O)

and depend on available nutrient concentrations and the scale of cultivation. Differences between model predictions also indicate that further research and ground truthing via in situ data collection are required.

Quantifying plankton assemblages associated with macroalgal cultivation is still in its infancy.[7] Standardised in situ monitoring methods commonly deployed in shellfish and finfish farms could, however, be applied and adapted to better monitor plankton within macroalgal farms[75] (Table 1). This will help to generate more accurate models to quantify habitat value of macroalgal cultivation for plankton assemblages and how it may contribute to other ESs related to HAB mitigation and bioremediation. Better determination of plankton assemblages in macroalgal cultivation sites will also be useful to detect early larval stages of species that may settle on the macroalgae, such as some epibionts[76] (see Section 2.3).

2.3 | Epibionts

Epibionts, or biofouling species that colonise available surfaces in marine ecosystems, are some of the most studied organisms in aquaculture due to their detrimental impacts on crop production, through reducing crop quality and yield.[77–79] Macroalgal epibionts such as endophytic or epiphytic algae, bryozoans, amphipods and hydroids may consume or degrade biomass, inhibit photosynthesis and algal growth, encourage grazers, increase disease susceptibility, contaminate commercial products by introducing allergen or toxin risks and damage farm infrastructure, thereby costing the aquaculture industry US\$1.5–3 billion yr^{-1}.[12,40,76,79–88] Farming infrastructure may also facilitate the settlement and spread of invasive non-native epibionts, by offering space for colonisation and reduced biotic resistance.[11,89]

Nevertheless, for farms that intend to provide non-consumable products, for example, farms that offer the ES of bioremediation (e.g. Ref. 58) or carbon sequestration (e.g. Ref. 90), epibiont colonisation may instead enhance their ES value. Colonising species such as bryozoans, bivalves, sponges, tunicates and other algae may improve water quality and host-plant health through biofiltration and nutrient addition,[91] in turn benefitting shellfish growth,[92] enhancing primary production,[93,94] providing protection from predation,[95,96] encouraging settlement of commercially farmed shellfish[97,98] and mitigating for disease risk.[99] Epibionts also provide food sources for higher trophic level species, increasing the habitat value of a cultivation site and secondary production[78]; however, grazing interactions on epibionts are not currently well understood.

Various studies have found similar or higher levels of epibiont biodiversity associated with cultivated kelps compared to wild populations, which suggests that suspended macroalgal farms could provide novel habitat for epibionts.[100,101] Epibiont assemblages vary widely however between cultivation sites, dependent on latitude, temperature and exposure.[102–104] Therefore, established, standardised methods are needed to quantify epibionts effectively at different sites. More targeted assessments are also needed to quantify environmentally beneficial epibionts as well as detrimental species to crop production, which have been the focus of studies to date. A better understanding of how epibionts may affect or interact with the environment will also enable a more ecosystem directed view for future development of EAA.

Census techniques for quantifying epibionts are relatively straightforward compared to other farm-associated biodiversity (Table 1), as most species are slow moving or sessile so they can be identified and enumerated directly from macroalgal biomass samples.[76,77,86,105,106] Previous studies assessing epibiont diversity on macroalgal farms have generally focussed on either the holdfast (e.g. Ref. 105) or the blade (e.g. Refs 100–104) separately however, rather than as one sampling unit. Quantifying the total epibiont assemblage is required if the potential habitat value of macroalgal cultivation is to be fully evaluated.

2.4 | Benthos

Benthic or seabed communities are comprised of many bioindicator species that signal environmental health and ecosystem functioning, and thus, benthic habitat monitoring is a crucial part of environmental impact assessments supporting aquaculture licence applications.[107] Nevertheless, limited research has been conducted to assess the ecological status below macroalgal cultivation sites compared to the aquaculture of finfish and shellfish.[16,17] Macroalgal cultivation could affect benthic community structure through shading; changes to hydrodynamic flow; increased sedimentation, organic enrichment, microbial activity and smothering from farm detritus breaking off; and competition with other important neighbouring benthic habitats such as wild kelp forests, seagrass beds or coral reefs (Figure 1, as reviewed in Refs. 5–8,10,12,13). Faeces and pseudofaeces produced by epibionts or mobile organisms associated with macroalgae may also cause benthic impacts that are similar to, although less pronounced than, other aquaculture farm types.[88,100,105] Farmers and policy makers can help to mitigate negative impacts on benthos by ensuring macroalgal cultivation sites are deployed at appropriate depths (e.g. Ref. 108). Recent guidelines advising on minimum water depths recommend that farms should be at locations where the water depth is at least twice the depth of the cultivation infrastructure and placed in areas where the minimum water flow rates are >0.05 m s^{-1}.[109,110] Compared to longline systems that can be deployed offshore,[111] there is often little if any flexibility for cultivation depth for tropical off-bottom farming sites.[9] Clearly, a better understanding of benthic interactions will help inform appropriate siting of farms in suitable environmental conditions to help mitigate for any potential negative effects on the benthos.

Ecosystem models of intensive kelp cultivation scenarios have indicated minimal effects on benthic food webs, or the potential to alter them through the provision of habitat, food and detritus.[112] Effects on benthic species vary between macroalgal cultivation sites globally. In tropical waters, where shallow, bottom-growing macroalgal cultivation techniques are favoured, significant changes to neighbouring benthic habitats such as corals and seagrass beds and their associated assemblages are often reported (as reviewed in Ref. 10). In temperate waters, where suspended macroalgal cultivation is favoured, benthic impacts tend to be less severe or even negligible (e.g. Refs. 100,101); however, this will depend on water flow rates through the cultivation area and light penetration in the water column. In Sandu Bay, China, however, sedimentary acid volatile sulphide content (linked to lower benthic biodiversity) was greater below kelp cultivation lines than at control sites.[113] This emphasises that effects on the benthos need to be evaluated on a site-specific basis, as they will be highly dependent on local environmental conditions and farm type.

Key indicators of benthic habitat health include sediment biogeochemistry (e.g. particle size, nutrient, heavy metal, oxygen-reduction-potential, carbon and organic matter content) and the biodiversity, composition and abundance of benthic infauna and epifauna. Infauna relates to organisms living in the sediment, whereas epifauna relates to organisms living on the seabed. Here, we consider benthic infauna and epifauna as two separate faunal categories, due to the different census techniques required to study them. We focus on both meiofauna (45 μm to 1 mm) and macrofauna (>1 mm) due to their important ecological roles and similar quantification methods.[114,115]

2.4.1 | Benthic infauna

Benthic infauna are comprised primarily of detritivores, grazers and filter feeders, such as polychaetes, flatworms, gastropods and bivalves, that all play key roles in recycling nutrients, filtering water and providing prey to epibenthic species.[114,116] The diversity and abundance of infaunal assemblages are used as bioindicators of contamination, eutrophication and hypoxia, due to the varying tolerances of species in the community.[116,117] It is therefore important to assess how macroalgal farms influence infaunal assemblage structure and function, to monitor the health of the cultivation site and of the wider habitat. Previous studies in off-bottom seaweed farms in Tanzania have found reduced infaunal biomass[118] or that the infaunal assemblages more closely resemble unvegetated areas rather than seagrass beds.[119] In contrast, at a Swedish longline farm, increases in infaunal species diversity and abundance have been found, indicating a positive effect of the farm on benthic health.[101] The marked differences in farming systems and environmental conditions between these locations (discussed further in Section 3.1) highlights how the effects of macroalgal cultivation on benthic infauna need to be evaluated in more detail. Sampling benthic infauna generally involves taking a sediment grab or core of the seabed and determining its associated fauna and biogeochemical properties[100,101,120,121] (Table 1). Benthic survey designs and approaches to the subsequent analysis of infaunal communities can vary greatly, however (Table 1). Standardised monitoring protocols, methods and analyses to quantify impacts from aquaculture on benthic infauna may help to regulate benthic monitoring between aquaculture types and locations in future.

2.4.2 | Benthic epifauna

Benthic epifauna includes macroinvertebrates (>1 mm), such as echinoderms (e.g. sea stars, urchins and sea cucumbers), crustaceans (e.g. lobsters and crabs) and benthic fish species (e.g. flatfish), many of which are of commercial or ecological importance. Wild kelp populations contribute on average US$48,600 to $141,000 ha^{-1} yr^{-1} to capture fisheries across the four major kelp genera globally, with nine of the top 10 valuable species being benthic invertebrates such as lobsters, abalone, urchins and gastropods.[122] Macroalgal cultivation may provide similar habitat value to epibenthic species via increased food availability from farm-derived nutrients and detritus (including epibiont cast-off) and the creation of a more heterogeneous habitat (via detritus accumulation or farm infrastructure such as mooring systems).[8,17,123–125] At a small temperate kelp farm in Sweden however, no effect on benthic macrofauna was reported,[101] whereas in Tanzania, lower abundances of macrofaunal or increases in sea urchin species, that could threaten to graze on the cultivated seaweed, were found.[118] Differing effects on epifauna reflect the diverse nature of macroalgal cultivation sites worldwide and highlights the need to increase monitoring across different systems.

Epifaunal assemblages may be more challenging to accurately quantify than infauna, as epifauna tend to be more mobile, patchily distributed and can also be cryptic.[126] Monitoring mobile epibenthic macrofauna (>1 mm) may however use similar methods to those used to quantify pelagic fish species (Section 2.5) or marine mammals, seabirds and reptiles (Section 2.6) and these have been summarised and reviewed in Table 1.

2.5 | Finfish

Finfish species include commercially important species for fisheries or for supporting wider food webs. Macroalgal cultivation sites could provide novel habitat for fish species by offering spawning substrate, shelter and food in the form of farm biomass or epibionts, similar to wild macroalgal populations (reviewed in Refs 10,17,127,128) (Figure 1). Potential benefits to fish species will however depend on farms being sited appropriately, to not replace natural nursery habitats such as seagrass beds, and the habitat complexity created by cultivation sites compared to what was in the area previously.[10,127,129] Mariculture infrastructure may also restrict fishing activities in an area, indirectly benefitting fish populations,[130] with potential spill-over benefits for fisheries.[16] Conversely, however, if macroalgal cultivation sites are poorly managed and regulated, they may act as 'ecological traps' whereby fish are attracted to farm infrastructure and become more vulnerable to capture from unregulated fisheries or natural predators, such as seals.[10,11,17,131] It is also unclear whether macroalgal farms enhance juvenile recruitment or simply aggregate existing adult fish populations.[16] The increased availability of macroalgae and novel epibiont prey on cultivated macroalgae may also alter fish assemblage composition. Illustrating this, increased herbivorous fish biomass was reported in farms in Tanzania compared to neighbouring seagrass beds.[129] Increases in herbivorous fish may be detrimental to farm yields, or conversely beneficial to fisheries in the area[129,132]; however, it is unclear as to how changes in fish assemblage composition will affect wider ecosystem function. Indeed, fish assemblages in *Eucheuma* farms in the Philippines were found to be significantly different to those in neighbouring marine protected areas and coral reefs, with more invertivores and fewer large herbivorous fish found in farms and greater biomass and diversity found in neighbouring coral reefs.[133] Conversely, multispecies macroalgal farms in Costa Rica attracted a larger number of fish species compared to control sites.[78] This highlights that the effects of macroalgal cultivation on fish populations can be highly variable between sites[132] and demonstrates the importance of establishing comparative reference sites in survey design (discussed further in Section 3.2).

Finfish species are generally highly mobile and can vary significantly in relation to timing of fish reproductive cycles, which makes accurately surveying their populations challenging. For example, at an Irish seaweed farm, juvenile mackerel and pollack that were found to be abundant in summer months were absent by September, whereas wrasse, which are more associated with the benthos, remained abundant below the site across the whole study period.[134] Inshore fish assemblages are also highly influenced by tides, with greater abundances observed during high tides.[135,136] Timing of seaweed harvesting is also important for monitoring finfish populations. For example, in wild kelp beds in Norway, juvenile gadoids were 92% less abundant in harvested areas compared to unharvested areas and remained 85% lower in areas one year after harvest.[137] Therefore, standardised medium- to long-term monitoring at multiple spatial scales is required to reliably assess the habitat value of macroalgal cultivation on fish populations and to inform better operational management practices and farm design. Various methods (Table 1) have been used to census fish in macroalgal cultivation sites, many of which are similar to those discussed in epibenthic surveys (Section 5.2). Models of macroalgal cultivation impacts on fish species may aid understanding of ecosystem-wide implications, but should be combined with long-term in situ studies to ground-truth model outputs.[128]

2.6 | Marine mammals, reptiles and seabirds

Marine mammals, reptiles and seabirds include many species of high conservation importance, so understanding how to manage marine industries to maximise environmental protection and minimise disturbance for these groups is hugely important.[138] These species also play major ecological roles[138] and are of particular value to marine industries such as tourism due to their charismatic value.[139] Here we have grouped these species as they will likely interact with macroalgal cultivation sites in similar ways as they share many life-history traits, and will also require similar surveying techniques to monitor (Table 1).

Potential increases in prey species of fish and macroinvertebrates in macroalgal farms could provide foraging grounds for mammals such as seals and dolphins, reptiles such as turtles, and seabirds, which are frequently observed around finfish and shellfish aquaculture sites (e.g. Refs. 140–146). Farm infrastructure, however, may interfere with the ability for mammals such as dolphins to aggregate fish prey and therefore affect their behaviour and habitat use (as discussed in Ref. 128). Macroalgal cultivation sites may also displace other mammals, reptiles and seabirds due to farm construction and operation activities (e.g. as seen in shellfish aquaculture[147–149]), which could lead to malnutrition if animals are displaced from their foraging grounds.[10] Minimal risks to cultivated macroalgae are perceived from vertebrate predators (unlike other aquaculture species), and they may instead help to maintain trophic balance and control grazing species as seen in wild populations.[150] Herbivorous species such as green turtles (*Chelonia mydas*, Linnaeus, 1758), however, have been found to consume some cultivated macroalgae,[151] which may attract them to cultivation sites, causing conflicts between turtles and farmers or increasing entanglement risk.[10,152,153] Other species are also susceptible to entanglement risk with farming infrastructure, as seen with stationary fishing gear[154,155]; or some mussel lines.[156,157] Lethal entanglement of critically endangered dugongs (*Dugong dugon*, Lacépède, 1799) has been reported in cultivation sites in the Philippines.[158] Entanglement risk is generally well understood however, and can be mitigated by increasing line thickness and tautness and avoiding placement in areas of known importance, such as migratory corridors.[7,156] Nevertheless, there are still considerable uncertainties about how large-scale farms that occupy large surface areas of coastal seas or entire bays influence marine mammals, reptiles and seabirds.

Census of marine mammals, reptiles and seabirds is challenging due to their generally low population densities and highly mobile existence. Furthermore, surveys need to be conducted over long timescales to fully assess impacts of cultivation sites on their populations, because many have long lifespans and slow population growth.[159] Monitoring of these species therefore requires immense survey effort or specialist behavioural knowledge, and is usually conducted from land, boats or air, often relying on species breaching or being at the surface of the water to be visible (Table 1). Currently, there are very limited studies on the interactions of marine mammals, reptiles or seabirds with macroalgal farms[5,10] and further research to address this knowledge gap is essential to enable better management and enhance potential habitat benefits of cultivation sites. Recent reviews have been published regarding census around marine renewable energy infrastructure (e.g. Ref. 160) and other aquaculture types,[128] which can inform monitoring in macroalgal cultivation sites (Table 1).

3 | GENERAL RECOMMENDATIONS ON STUDY DESIGN FOR MONITORING HABITAT PROVISIONING IN MACROALGAL CULTIVATION SITES

This review illustrates that it is not only the monitoring approach used that is important for quantifying habitat provisioning in macroalgal cultivation sites but also how and when these surveys are carried out. Standardised approaches in study design and implementation will ensure monitoring is more directly comparable across farm sites and scales, including IMTA and offshore systems. The effectiveness of different survey methods will differ between farm types, however, which we discuss further below (Section 3.1).

Here, we provide the first steps in standardising monitoring protocols for assessing habitat provisioning by macroalgal cultivation, with regard to accounting for differences in farm types, establishing reference sites, timing surveys appropriately and defining the necessary species metrics to be taken for relevant habitat value analysis. We also discuss how the collection of appropriate field data can be used in models to answer some of the broader, ecosystem-wide effects of the habitat value of macroalgal cultivation that extend beyond the footprint of the farms, and importantly how these data can be shared in open-access repositories to advance the sustainable design of macroalgal cultivation sites in EAA.

3.1 | Accounting for differences in macroalgal cultivation farm types

Globally, there is a wide diversity of macroalgal farm types and species cultivated, from offshore temperate kelp farms to off-bottom tropical carrageenophyte cultivation (Figure 1). This diversity makes creating standardised monitoring techniques to quantify habitat provisioning challenging, as their effectiveness will differ widely between cultivated species, locations and farm types. From the monitoring techniques outlined in Table 1, many of these are applicable to all macroalgal cultivation sites globally. For example, diver-conducted visual surveys of fish and pelagic species have been used successfully in both *Eucheuma* and *Kappaphycus* farms in the Philippines[133] and *Codium*, *Gracilaria*, *Sargassum* and *Ulva* farms in Costa Rica.[78] This survey method is straightforward and flexible due to divers or snorkelers being able to adapt their positions around farming infrastructure. Between farm types, where infrastructure differs greatly however, some monitoring techniques are not feasible to deploy universally. For instance, small beam trawls may be suitable for monitoring benthic species around kelp longline systems (as conducted in a Canadian mussel farm[187]); however, they are not suitable in most tropical macroalgal farms due to shallow depth limitations, high density of cultivation lines or use of mesh nets to seed (e.g. in *Pyropia* or *Ulva* farms).[9,10] To survey benthic species in shallow tropical farms, more targeted methods could be deployed, such as benthic drop cameras or traps (Table 1), which can also be used successfully in temperate systems.[101] Remote camera surveys would also cause less disturbance to both biodiversity and the cultivated species. To enable more effective comparison of habitat value between macroalgal cultivation sites globally, monitoring methods that are flexible in terms of their deployment should be favoured over those that will only work in certain cultivation scenarios.

Working collaboratively with farmers or other stakeholders, such as local fisheries, will enable suitable survey methods for the region to be developed. For example, in Tanzanian off-bottom *Eucheuma* and *Kappaphycus* farms, where trawling or netting is not possible, researchers used traditional 'madema' basket traps to survey fish populations, and were instructed on best deployment techniques by local fishermen.[129] Using local methods and knowledge-enabled successful catches of fish populations in relation to macroalgal cultivation sites and also to assess how the presence of macroalgal cultivation sites may affect the economic value of other marine industries in the area.[129] Engaging with farmers and other local industries may also help to improve awareness of the potential ecological and economic benefits of habitat provisioning by macroalgal cultivation[9] and therefore increase interest in contribution to farmer or citizen science observations or the uptake of habitat monitoring into farm management protocols[13] (Section 3.7).

3.2 | Surveying appropriate reference sites and environmental variables

Ideally, to fully assess the environmental effects of an aquaculture system, surveys should be conducted before farms are established in any given area, and then compared to results seen during and after implementation, as well as at control sites, thereby adopting a before-after-control-impact (BACI) design.[262] Beyond BACI studies may often include multiple, additional control sites away from the farm, which experience similar background environmental conditions to the farm site (e.g. depth, sediment, hydrology) but are at an appropriate distance away so as to not be affected by the presence of aquaculture species (e.g. Refs. 100,101). Where macroalgal cultivation sites have already been implemented before baseline conditions were established, habitat value has often been compared to other reference habitats, such as seagrass beds (e.g. Ref. 129) or wild macroalgae populations (e.g. Ref. 105). To understand any added habitat value created by macroalgal cultivation sites to an area, farms should also be compared to areas with similar environmental conditions where there is no structural habitat, as this is where farms are often implemented. Factoring in the monitoring of other variables such as macroalgal species and biomass, depth, light, temperature, sediment and water biogeochemistry (e.g. dissolved inorganic carbon, sediment size and oxygen-reduction-potential), nutrient availability and hydrodynamic activity, will also be required to accurately compare the habitat value of different farms and better understand the health of these systems.[7,13,87,88,101–103,105]

To assess the effect of habitat provisioning beyond the direct footprint of a cultivation site, reference sites can be set at incremental distances away from the site to determine a sphere of influence or radius of attraction for species (e.g. Ref. 124). In many aquaculture sites, wider effects can be relatively small, e.g. limited to <50 m for epibenthic macrofauna in Canadian blue mussel farms.[124] For different taxonomic groups however, it is likely that the influence of cultivation sites will extend further outside of the farm's footprint, such as for larger megafauna that may be deterred from foraging grounds, as observed with dolphins and shellfish aquaculture in New Zealand.[147,148]

3.3 | Timing of surveys

Previous studies have often only sampled at harvesting time points (e.g. Ref. 263), when successional communities may not be fully established. This is representative for the farming industry, which normally completely removes algal biomass at harvest; however, time-series data that extend beyond typical harvesting seasons could inform better harvesting practices to maximise the habitat value of macroalgae farms and inform EAA. For example, trials are being conducted to assess how partial harvesting techniques (whereby some of the holdfast and blade are left in situ) may influence epibiont communities and extend the biodiversity value and crop yield of the farms.[80,87] Surveying beyond the cultivation season, when macroalgal biomass is removed, will also help to determine to what extent the habitat value of the site has been removed in the absence of the algae and indeed whether it might be maintained, at least in part, by the presence of farm infrastructure in the area (if this remains in place).[79,264] Sampling should therefore be conducted at multiple or continuous set time points before, during and beyond multiple harvesting cycles, to help avoid temporal bias, allow comparability and more completely estimate the habitat value of cultivation sites, as well as determine whether or not they provide habitat only during cultivation seasons.

3.4 | Species metrics to be taken and statistical analyses of habitat value

Quantifying the habitat value of macroalgal cultivation sites does not only rely on determining the species present in the site and their abundances but also on understanding their usage of and behavioural interactions with the site at various life stages in the long term, as well as monitoring their physiological condition and fitness (e.g. juvenile recruitment). Juvenile recruitment success underpins population fitness and biodiversity, so it should be monitored within cultivation sites and surrounding areas to address whether macroalgal farms enhance wild populations through juvenile recruitment or simply aggregate individuals from surrounding areas. Quantifying the size and nominal age of individual organisms is also needed to understand juvenile recruitment.[8,124] Biomass measurements are also important to determine nutrient and energy flows in ecosystem-wide and food web models[120] (see Section 3.5), but can be estimated for wild fish using published length–weight conversions (e.g. ecoCEN[265]). Many of the methods outlined in Table 1 are capable of determining size and age classes of individuals, through the direct measurements of captured, from photographed organisms using a scale, or through visual estimates from experienced divers or observers (e.g. Ref. 200). Visual methods are also often necessary to determine the behavioural activity of species at macroalgal cultivation sites, for example, feeding, breeding and sheltering or avoidance of the site. Tagging or biologging species also allows the behaviour of individuals to be monitored and site fidelity to be established.[238] Feeding behaviour can also be determined through gut content or isotopic analysis (e.g. Ref. 266), which can establish food web effects of the farm. Better knowledge of species' activity and feeding behaviours will help determine what attracts them to farming infrastructures, and the significance of the farm habitat. This will furthermore help to improve the understanding of how farming practices can be better designed to maximise the habitat value of macroalgal cultivation sites and mitigate disturbance on key life stages of inhabiting species, through EAA.

The use of diverse biotic indices and statistical approaches can also provide various insights into overall community and ecosystem health, determining the habitability of an area for different species. For example, to quantify infaunal biodiversity, a large variety of statistical approaches[183,267,268] and biotic indices are used (summarised in Refs. 117,269). Indices lend themselves to standardisation and they can be fine-tuned to detect certain types of pressure, for example, the Infaunal Trophic Index (ITI), which takes into account species sensitivity to organic enrichment[270] could be used to quantify the impact of organic exudates and cast-off from macroalgal farms. The Infaunal Quality Index (IQI) is also used to assess sediment quality and would be useful for detecting disturbance below macroalgal cultivation sites.[269] When multiple indices are employed, they generate a wider picture of the impacts of aquaculture.[117,271–273] The choice of data analysis tools is therefore important and should be considered carefully when designing habitat value surveys to ensure they fulfil assessment objectives and are comparable to other sites.

For comparability of habitat value between cultivation sites and aquaculture species, a standardised set of variables, biotic indices and statistical approaches should be produced, which would enable better quantification of the habitat value of macroalgal cultivation.[117] Such assessment criteria exist for reviewing the impacts of human activities on the marine environment, such as the Marine Evidence Based Sensitivity Assessment (MarESA).[274–276] MarESA can be applied to monitoring different forms of aquaculture and their impacts in terms of magnitude, extent, duration and frequency of the effect, so that pressures from different activities can be compared on an equal footing.

3.5 | Modelling the wider ecosystem effects of habitat provisioning for an ecosystem approach to aquaculture (EAA)

Despite the increasing number of survey methods available for quantifying habitat provisioning in macroalgal cultivation sites, the resources available to conduct such field surveys are limited, particularly at small-scale farms and they tend to focus within a farm's footprint.

Models may be used instead to predict ecosystem-wide effects based on established relationships with primary productivity, nutrient and energy flows and readily available environmental and species data.

At macroalgal cultivation sites, models have already been run for determining effects on plankton assemblages at different farm scales (e.g. Refs. 65,67,71,73,74) and ecosystem-wide effects on food web dynamics (e.g. Ref. 120). Models of carrying capacity in aquaculture systems assess the maximum production potential of a cultivated species that can be supported in an area based on environmental conditions, optimal stocking density, cultivation approaches and environmental impact.[72,277] Carrying capacity models can be used in the development of EAA to assess ecosystem impacts beyond the direct footprint of the cultivation site and ensure sustainability.[277,278] In Sanggou Bay, China, carrying capacity models have been used to assess production limits of cultivated kelp and oysters based on species growth dynamics and environmental data[279]; however, these did not focus on the ecological carrying capacity of the site, rather on maximising production. Ecological carrying capacity in macroalgal cultivation sites should be investigated further to balance production with ecosystem management goals, as has been outlined for shellfish and finfish cultivation previously.[73,277,278]

Although models rely on initial species input data from field surveys to be calibrated, ground truthed and verified, data from other prepublished studies and reports can also be used.[120] As ecosystems are complex, models can tend to oversimplify aquaculture–environmental relationships[71]; however, as our understanding of these systems improves, the usefulness of habitat provisioning models will also increase. Models can also be used to assess cumulative ecosystem impacts of IMTA systems or multipurpose platforms such as integrated aquaculture and marine renewable energy sites to inform marine spatial planning and policy related to EAA.[280]

3.6 | Data distribution and access

In order to inform decisions on optimising the habitat value of macroalgal cultivation sites in EAA, high quality comparable data from multiple sites will need to be used. To facilitate this, global standardised data sets should be generated with available data on habitat provisioning in macroalgal farms or other aquaculture types. Conceptual frameworks such as the Essential Biodiversity Variables (EBVs)[281] could aid in creating interoperable data sets based on data collected using common methodologies. These frameworks could then be made available in open-access repositories to facilitate habitat value or biodiversity assessments.[282]

3.7 | Integration of monitoring techniques into farm management protocols and policy

Currently, policy relating to macroalgal cultivation at either national or international level is not well established, and farm management protocols are often focused on reducing waste, pollution, disease outbreaks and damaging epibionts, rather than maximising the ecological value of the site. Standardised quantification of habitat provisioning of macroalgal cultivation sites would be greatly facilitated through the creation of clear monitoring criteria and guidance from regulatory and accreditation bodies through farm management protocols or policy.[5,13,107] Monitoring techniques should adhere to licencing and certification standards, whilst also benefitting farmers to incentivise their usage, for example, via optimising harvest schedules and increasing product yield and grade or facilitating regulation and licencing of farms. Several sustainability and organic certification standards for macroalgal cultivation production (outlined in Ref. 9) discuss the need for farmers to assess the positive and negative environmental impacts of their farms and establish sustainable management plans to enable their products to be accredited; however, very little direct guidance on evidence-based monitoring is given. For example, the Aquaculture Stewardship Council (ASC)-Marine Stewardship Council (MSC) sustainable seaweed standard for both wild and farmed seaweeds sets a number of requirements for farmers to demonstrate that they are actively reducing any potential negative environmental impacts of their farms, including on native species and habitats.[283] Elements of the ASC-MSC standard include habitat, ecosystem structure and function, species status, species management, waste management and pollution control, energy efficiency, disease and pest management practices and introduced species management. The ASC-MSC standard does not, however, detail any specific monitoring techniques required to provide evidence for this. If certification standards could encourage this form of standardised data collection as a requirement for certification, it would greatly incentivise farmers to integrate habitat provisioning monitoring techniques into their current farm management protocols. Standardising methods for quantifying habitat provisioning and the associated ecological (and economic) value is undoubtedly more challenging than for data collection related to assessing and managing impacts around waste management and pollution control, energy efficiency, disease and pest management practices. However, encouraging monitoring of habitat provisioning is essential for realising an EAA and assuring the sustainability of the industry.[72]

Marine licencing bodies could also set environmental and habitat monitoring as a legal requirement to grant farming permissions, particularly in newly emerging regions of macroalgal cultivation, such as Europe. The EU strategic guidelines for a more sustainable and competitive EU aquaculture in 2021–2030 highlights the limited data reported on environmental indicators related to aquaculture and the need to obligate farmers to monitor and report environmental data to licencing systems and for regulating bodies to enforce sanctions for non-compliance.[72] The environmental data currently reported relates mostly to water quality and pollution levels[72]; however, reporting could be expanded to include biodiversity or habitat monitoring.[13] To engage other stakeholders and facilitate social licencing, monitoring techniques could also be targeted at verifying potential economic benefits to other marine industries, including fisheries, by identification of commercially important species such as edible crabs, fish and lobsters.[72]

4 | CONCLUSIONS

Macroalgal cultivation may provide several key ESs, which could increase cultivation value beyond that of biomass production alone. Macroalgae cultivation sites could provide important habitat for a diverse number of species of ecological, conservation and/or commercial importance. However, the habitat provisioning of macroalgal farms largely remains unquantified and consequently, placing economic values on this environmental benefit remains uncertain. If the habitat value of macroalgal cultivation sites can be effectively quantified, this would incentivise farmers to design cultivation sites or harvesting schedules to maximise habitat value and contribute towards sustainable EAA. With growing exploitation of the marine environment and increasing fragmentation and degradation of marine habitats, the strategic implementation of restorative EAA may provide important wildlife corridors to reconnect habitats and support complementary communities of fish and fishing grounds, which may otherwise take decades to recover unaided.[284] Better understanding of the species that occupy macroalgae cultivation sites is therefore important and requires comprehensive and robust census techniques and methods specifically designed for use in macroalgal farms, due to the unique challenges these habitats pose for monitoring. With the increasing proportion of large-scale offshore or IMTA sites, census techniques should be designed to survey in a variety of environmental conditions and at differing cultivation scales globally. Standardising methods for quantifying habitat provisioning and its ecological (and economic) value is challenging due to wide ranging taxonomic and functional groups that can inhabit seaweed farms; however, this work is essential for realising EAA and assuring the sustainability of the industry. A future challenge for quantifying the value of habitat provisioning will be to harness the data generated from employing standardised methods and move beyond summative assessments or indices of biodiversity to more ecosystem-based assessments, which take into account species–species and species–environment interactions and synergies.

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AUTHOR CONTRIBUTIONS

Sophie Corrigan: Conceptualization; Investigation; Writing – original draft. **A. Ross Brown:** Conceptualization; Funding acquisition; Supervision; Writing – original draft; Writing – review & editing. **Ian G. C. Ashton:** Funding acquisition; Supervision; Writing – review & editing. **Dan A. Smale:** Funding acquisition; Supervision; Writing – review & editing. **Charles R. Tyler:** Conceptualization; Funding acquisition; Supervision; Writing – original draft; Writing – review & editing.

DATA AVAILABILITY STATEMENT

No data were generated for this review.

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REFERENCES

1. FAO. *The State of World Fisheries and Aquaculture 2020. Sustainability in Action*. FAO; 2020. doi:10.4060/ca9229en
2. Schlarb-Ridley B, Parker B. A road map for algal technologies in the UK. 2013. https://connect.innovateuk.org/documents/3312976/3726818/AB_SIG+Roadmap.pdf
3. Capuzzo E, McKie T. Seaweed in the UK and abroad – status, products, limitations, gaps and Cefas role. *Cefas*. 2016. https://assets.publishing.service.gov.uk/government/uploads/system/uploads/attachment_data/file/546679/FC002I__Cefas_Seaweed_industry_report_2016_Capuzzo_and_McKie.pdf
4. Buschmann AH, Camus C, Infante J, et al. Seaweed production: overview of the global state of exploitation, farming and emerging research activity. *Eur J Phycol*. 2017;52(4):391-406. doi:10.1080/09670262.2017.1365175
5. Wood D, Capuzzo E, Kirby D, Mooney-McAuley K, Kerrison P. UK macroalgae aquaculture: what are the key environmental and licensing considerations? *Mar Policy*. 2017;83:29-39. doi:10.1016/j.marpol.2017.05.021
6. Hasselström L, Visch W, Gröndahl F, Nylund GM, Pavia H. The impact of seaweed cultivation on ecosystem services – a case study from the west coast of Sweden. *Mar Pollut Bull*. 2018;133:53-64. doi:10.1016/j.marpolbul.2018.05.005
7. Campbell I, Macleod A, Sahlmann C, et al. The environmental risks associated with the development of seaweed farming in Europe – prioritizing key knowledge gaps. *Front Mar Sci*. 2019;6:107. doi:10.3389/fmars.2019.00107
8. Langton R, Augyte S, Price N, et al. An ecosystem approach to the culture of seaweed. *NOAA Tech Memo NMFS-F/SPO-195*. July 2019:24. https://www.researchgate.net/publication/338711001 Accessed September 2, 2021
9. Waters T, Jones R, Theuerkauf S, et al. Coastal conservation and sustainable livelihoods through seaweed aquaculture in Indonesia: a guide for buyers, conservation practitioners, and farmers. *Nat Conserv*. 2019;1:1-47.
10. Kelly ELA, Cannon AL, Smith JE. Environmental impacts and implications of tropical carrageenophyte seaweed farming. *Conserv Biol*. 2020;34(2):326-337. doi:10.1111/COBI.13462/FORMAT/PDF
11. Alleway HK, Gillies CL, Bishop MJ, Gentry RR, Theuerkauf SJ, Jones R. The ecosystem services of marine aquaculture: valuing benefits to people and nature. *Bioscience*. 2019;69(1):59-68. doi:10.1093/biosci/biy137
12. Buschmann AH, Correa JA, Westermeier R, del Hernández-González MC, Norambuena R. Red algal farming in Chile: a

review. *Aquaculture*. 2001;194(3–4):203-220. doi:10.1016/S0044-8486(00)00518-4
13. Wilding C, Tillin H, Corrigan S, et al. *Seaweed aquaculture and mechanical harvesting: An evidence review to support sustainable management*; 2021. Natural England Commissioned Reports. Natural England Report NECR378. Available: http://nepubprod.appspot.com/publication/5191133599891456
14. Kim JK, Yarish C, Hwang EK, Park M, Kim Y. Seaweed aquaculture: Cultivation technologies, challenges and its ecosystem services. *Algae*. 2017;32(1):1-13. doi:10.4490/algae.2017.32.3.3
15. Hasselström L, Thomas J-B, Nordström J, et al. Socioeconomic prospects of a seaweed bioeconomy in Sweden. *Sci Rep*. 2020;10(1):1-7. doi:10.1038/s41598-020-58389-6
16. Gentry RR, Alleway HK, Bishop MJ, Gillies CL, Waters T, Jones R. Exploring the potential for marine aquaculture to contribute to ecosystem services. *Rev Aquac*. 2020;12(2):499-512. doi:10.1111/raq.12328
17. Theuerkauf SJ, Barrett LT, Alleway HK, Costa-Pierce BA, St GA, Jones RC. Habitat value of bivalve shellfish and seaweed aquaculture for fish and invertebrates: Pathways, synthesis and next steps. *Rev Aquac*. 2022;14(1):54–72. doi:10.1111/RAQ.12584
18. Doumeizel V, Aass K, McNevin A, et al. Seaweed revolution: A manifesto for a sustainable future. 2020:1–16. https://ungc-communications-assets.s3.amazonaws.com/docs/publications/The-Seaweed-Manifesto.pdf
19. Costanza R, de Groot R, Braat L, et al. Twenty years of ecosystem services: How far have we come and how far do we still need to go? *Ecosyst Serv*. 2017;28:1-16. doi:10.1016/j.ecoser.2017.09.008
20. Haines-Young R, Potschin M. Common International Classification of Ecosystem Services (CICES) V5.1 and guidance on the application of the revised structure. *CICES*. 2018:53. https://cices.eu/resources/
21. Costanza R, d'Arge R, de Groot R, et al. The value of the world's ecosystem services and natural capital. *Nature*. 1997;387(6630):253-260. doi:10.1038/387253a0
22. Millennium Ecosystem Assessment M. *Ecosystems and Human Well-Being: Synthesis*. United States Am Isl Press. 2005;5.
23. TEEB. *Mainstreaming the economics of nature: a synthesis of the approach, conclusions and recommendations of TEEB*. Earthscan, London Washingt; 2010.
24. McVittie A, Hussain S. The economics of ecosystems and biodiversity – valuation database manual. *Econ Ecosyst Biodivers Geneva*; 2013.
25. Weitzman J. Applying the ecosystem services concept to aquaculture: a review of approaches, definitions, and uses. *Ecosyst Serv*. 2019;35:194-206. doi:10.1016/J.ECOSER.2018.12.009
26. Smale DA, Burrows MT, Moore P, O'Connor N, Hawkins SJ. Threats and knowledge gaps for ecosystem services provided by kelp forests: a northeast Atlantic perspective. *Ecol Evol*. 2013;3(11):4016-4038. doi:10.1002/ece3.774
27. Dempster T, Sanchez-Jerez P, Bayle-Sempere JT, Giménez-Casalduero F, Valle C. Attraction of wild fish to sea-cage fish farms in the south-western Mediterranean Sea: spatial and short-term temporal variability. *Mar Ecol Prog Ser*. 2002;242:237-252. doi:10.3354/MEPS242237
28. Custódio M, Villasante S, Calado R, Lillebø AI. Valuation of Ecosystem Services to promote sustainable aquaculture practices. *Rev Aquac*. 2020;12(1):392-405. doi:10.1111/RAQ.12324
29. Soto D, Aguilar-Manjarrez J, Hishamunda N (Eds). Building an ecosystem approach to aquaculture. FAO/Universitat de les Illes Balears Expert Workshop. In: *FAO/Universitat de Les Illes Balears Expert Workshop*. Palma de Mallorca, Spain.: No. 14. FAO, Rome; 2007. http://www.fao.org/3/a-i0339e.pdf
30. FAO. Aquaculture development. 4. ecosystem approach to aquaculture. In: *FAO Technical Guidelines for Responsible Fisheries*. No. 5, Sup. Rome: FAO; 2010. http://www.fao.org/docrep/013/i1750e/i1750e00.htm
31. Hyde KD, Jones EBG, Leaño E, Pointing SB, Poonyth AD, Vrijmoed LLP. Role of fungi in marine ecosystems. *Biodivers Conserv*. 1998;7(9):1147-1161. doi:10.1023/A:1008823515157
32. Arrigo KR. Marine microorganisms and global nutrient cycles. *Nature*. 2005;437(7057):349-355. doi:10.1038/nature04159
33. Imai I, Yamaguchi M, Hori Y. Eutrophication and occurrences of harmful algal blooms in the Seto Inland Sea, Japan. *Plankt Benthos Res*. 2006;1(2):71-84. doi:10.3800/PBR.1.71
34. Gachon CMM, Sime-Ngando T, Strittmatter M, Chambouvet A, Kim GH. Algal diseases: spotlight on a black box. *Trends Plant Sci*. 2010;15(11):633-640. doi:10.1016/j.tplants.2010.08.005
35. Bentzon-Tilia M, Sonnenschein EC, Gram L. Monitoring and managing microbes in aquaculture – towards a sustainable industry. *Microb Biotechnol*. 2016;9(5):576-584. doi:10.1111/1751-7915.12392
36. Egan S, Harder T, Burke C, Steinberg P, Kjelleberg S, Thomas T. The seaweed holobiont: understanding seaweed-bacteria interactions. *FEMS Microbiol Rev*. 2013;37(3):462-476. doi:10.1111/1574-6976.12011
37. Agostini VO, Steigleder KM, Islabão C, José A. Evaluation of epibiont community on seaweed *Ulva lactuca*: biofilm and its relationship with the larval settlement of invertebrates. In: *Congresso Brasileiro de Oceanografia*. Itajai, Santa Catarina; 2014. doi:10.13140/RG.2.2.11600.71686
38. Trevathan-Tackett SM, Sherman CDH, Huggett MJ, et al. A horizon scan of priorities for coastal marine microbiome research. *Nat Ecol Evol*. 2019;3(11):1509-1520. doi:10.1038/s41559-019-0999-7
39. Satheeja Santhi V, Bhagat AK, Saranya S, Govindarajan G, Jebakumar SRD. Seaweed (*Eucheuma cottonii*) associated microorganisms, a versatile enzyme source for the lignocellulosic biomass processing. *Int Biodeterior Biodegradation*. 2014;96:144-151. doi:10.1016/J.IBIOD.2014.08.007
40. Bernard M. Seaweed diseases and pests. *Wageningen Marine Research, Proseaweed Dossier (project AF-16202)*. https://edepot.wur.nl/475726 Published 2018 Accessed November 21, 2021.
41. Loureiro R, Gachon CMM, Rebours C. Seaweed cultivation: Potential and challenges of crop domestication at an unprecedented pace. *New Phytol*. 2015;206(2):489-492. doi:10.1111/nph.13278
42. Valero M, Guillemin M-L, Destombe C, et al. Perspectives on domestication research for sustainable seaweed aquaculture. *Perspect Phycol*. 2017;4(1):33-46. doi:10.1127/pip/2017/0066
43. Campbell I, Kambey CSB, Mateo JP, et al. Biosecurity policy and legislation for the global seaweed aquaculture industry. *J Appl Phycol*. 2019;32(4):2133-2146. doi:10.1007/S10811-019-02010-5
44. Cottier-Cook EJ, Nagabhatla N, Badis Y, et al. Safeguarding the future of the global seaweed aquaculture industry. *United Nations Univ Scottish Assoc Mar Sci Policy Brief*. 2016:1–12.
45. Barbier M, Araújo R, Rebours C, Jacquemin B, Holdt SL, Charrier B. Development and objectives of the PHYCOMORPH European Guidelines for the Sustainable Aquaculture of Seaweeds (PEGASUS). *Bot Mar*. 2020;61(1):5-16.
46. Capuzzo E, Mengo E, Kennerley A. Seaweed farming feasibility within the Dorset and East Devon FLAG area. Report 1 – Potential species, farming methods, end uses and benefits. *Cefas*. 2019;204.
47. Ward GM, Faisan JP, Cottier-Cook EJ, et al. A review of reported seaweed diseases and pests in aquaculture in Asia. *J World Aquac Soc*. 2020;51(4):815-828.
48. Kim GH, Moon KH, Kim JY, Shim J, Klochkova TA. A revaluation of algal diseases in Korean *Pyropia (Porphyra)* sea farms and their economic impact. *ALGAE*. 2014;29(4):249-265. doi:10.4490/ALGAE.2014.29.4.249

49. Badis Y, Klochkova TA, Brakel J, et al. Hidden diversity in the oomycete genus *Olpidiopsis* is a potential hazard to red algal cultivation and conservation worldwide. *Eur J Phycol*. 2020;55(2):162-171. doi:10.1080/09670262.2019.1664769
50. Badis Y, Klochkova TA, Strittmatter M, et al. Novel species of the oomycete *Olpidiopsis* potentially threaten European red algal cultivation. *J Appl Phycol*. 2019;31(2):1239-1250. doi:10.1007/s10811-018-1641-9
51. Bouwmeester MM, Goedknegt MA, Poulin R, Thieltges DW. Collateral diseases: aquaculture impacts on wildlife infections. *J Appl Ecol*. 2021;58(3):453-464. doi:10.1111/1365-2664.13775
52. Keesing F, Holt RD, Ostfeld RS. Effects of species diversity on disease risk. *Ecol Lett*. 2006;9(4):485-498. doi:10.1111/J.1461-0248.2006.00885.X
53. Paine ER, Schmid M, Boyd PW, Diaz-Pulido G, Hurd CL. Rate and fate of dissolved organic carbon release by seaweeds: a missing link in the coastal ocean carbon cycle. *J Phycol*. 2021;57(5):1375-1391. doi:10.1111/jpy.13198
54. Stoeck T, Frühe L, Forster D, Cordier T, Martins CIM, Pawlowski J. Environmental DNA metabarcoding of benthic bacterial communities indicates the benthic footprint of salmon aquaculture. *Mar Pollut Bull*. 2018;127:139-149. doi:10.1016/J.MARPOLBUL.2017.11.065
55. Bonthond G, Barilo A, Allen RJ, Cunliffe M, Krueger-Hadfield SA. Fungal endophytes vary by species, tissue type, and life cycle stage in intertidal macroalgae. *J Phycol*. 2022;0–2, Epub ahead of print. doi:10.1111/jpy.13237
56. Dittmann KK, Rasmussen BB, Castex M, Gram L, Bentzon-Tilia M. The aquaculture microbiome at the centre of business creation. *Microb Biotechnol*. 2017;10(6):1279-1282. doi:10.1111/1751-7915.12877
57. Pakhomov E, Kaehler S, McQuaid C. Zooplankton community structure in the kelp beds of the sub-Antarctic Prince Edward Archipelago: are they a refuge for larval stages? *Polar Biol*. 2002;25(10):778-788. doi:10.1007/S00300-002-0411-X
58. Kim JK, Kraemer GP, Yarish C. Use of sugar kelp aquaculture in Long Island Sound and the Bronx River Estuary for nutrient extraction. *Mar Ecol Prog Ser*. 2015;531:155-166. doi:10.3354/MEPS11331
59. Seghetta M, Tørring D, Bruhn A, Thomsen M. Bioextraction potential of seaweed in Denmark — An instrument for circular nutrient management. *Sci Total Environ*. 2016;563–564:513-529. doi:10.1016/J.SCITOTENV.2016.04.010
60. Rosman JH, Monismith SG, Denny MW, Koseff JR. Currents and turbulence within a kelp forest (*Macrocystis pyrifera*): Insights from a dynamically scaled laboratory model. *Limnol Oceanogr*. 2010;55(3):1145-1158. doi:10.4319/LO.2010.55.3.1145
61. Jiang Z, Liu J, Li S, et al. Kelp cultivation effectively improves water quality and regulates phytoplankton community in a turbid, highly eutrophic bay. *Sci Total Environ*. 2020;707:135561. doi:10.1016/j.scitotenv.2019.135561
62. Xiao XI, Agustí S, Yu Y, et al. Seaweed farms provide refugia from ocean acidification. *Sci Total Environ*. 2021;776:145192. doi:10.1016/J.SCITOTENV.2021.145192
63. Solidoro C, Dejak C, Franco D, Pastres R, Pecenik G. A model for macroalgae and phytoplankton growth in the Venice Lagoon. *Environ Int*. 1995;21(5):619-626. doi:10.1016/0160-4120(95)00080-5
64. Xiao XI, Agusti S, Lin F, et al. Nutrient removal from Chinese coastal waters by large-scale seaweed aquaculture. *Sci Rep*. 2017;7(1):1-6. doi:10.1038/srep46613
65. Aldridge J, van der Molen J, Forster R. Wider ecological implications of macroalgae cultivation. Crown Estate, 2012;95.
66. Lüning K, Pang S. Mass cultivation of seaweeds: current aspects and approaches. *J Appl Phycol*. 2003;15(2):115-119. doi:10.1023/A:1023807503255
67. Shi J, Wei H, Zhao L, Yuan Y, Fang J, Zhang J. A physical–biological coupled aquaculture model for a suspended aquaculture area of China. *Aquaculture*. 2011;318:412-424. doi:10.1016/J.AQUACULTURE.2011.05.048
68. Zhao L, Zhao Y, Xu J, et al. Distribution and seasonal variation of picoplankton in Sanggou Bay, China. *Aquac Environ Interact*. 2016;8:261-271. doi:10.3354/AEI00168
69. Gibbs MT. Interactions between bivalve shellfish farms and fishery resources. *Aquaculture*. 2004;240(1–4):267-296. doi:10.1016/J.AQUACULTURE.2004.06.038
70. Préat N, De Troch M, van Leeuwen S, et al. Development of potential yield loss indicators to assess the effect of seaweed farming on fish landings. *Algal Res*. 2018;35:194-205. doi:10.1016/j.algal.2018.08.030
71. Aldridge JN, Mooney K, Dabrowski T, Capuzzo E. Modelling effects of seaweed aquaculture on phytoplankton and mussel production. Application to Strangford Lough (Northern Ireland). *Aquaculture*. 2021;536:736400. doi:10.1016/J.AQUACULTURE.2021.736400
72. European Commission. *Strategic guidelines for a more sustainable and competitive EU aquaculture for the period 2021–2030*. European Commission; 2021. https://eur-lex.europa.eu/resource.html?uri=cellar:bab1f9a7-b30b-11eb-8aca-01aa75ed71a1.0022.02/DOC_1&format=PDF
73. Zollmann M, Rubinsky B, Liberzon A, Golberg A. Multi-scale modeling of intensive macroalgae cultivation and marine nitrogen sequestration. *Commun Biol*. 2021;4(1):1–11. 10.1038/s42003-021-02371-z
74. van der Molen J, Ruardij P, Mooney K, et al. Modelling potential production of macroalgae farms in UK and Dutch coastal waters. *Biogeosciences*. 2018;15(4):1123-1147. doi:10.5194/BG-15-1123-2018
75. Brown AR, Lilley M, Shutler J, et al. Assessing risks and mitigating impacts of harmful algal blooms on mariculture and marine fisheries. *Rev Aquac*. 2019;12(3):1-26. doi:10.1111/raq.12403
76. Førde H, Forbord S, Handå A, et al. Development of bryozoan fouling on cultivated kelp (*Saccharina latissima*) in Norway. *J Appl Phycol*. 2016;28(2):1225-1234. doi:10.1007/s10811-015-0606-5
77. Park CS, Hwang EK. Seasonality of epiphytic development of the hydroid *Obelia geniculata* on cultivated *Saccharina japonica (Laminariaceae, Phaeophyta)* in Korea. *J Appl Phycol*. 2011;24(3):433-439. doi:10.1007/S10811-011-9755-3
78. Radulovich R, Umanzor S, Cabrera R, Mata R. Tropical seaweeds for human food, their cultivation and its effect on biodiversity enrichment. *Aquaculture*. 2015;436(October):40-46. doi:10.1016/j.aquaculture.2014.10.032
79. Bannister J, Sievers M, Bush F, Bloecher N. Biofouling in marine aquaculture: a review of recent research and developments. *Biofouling*. 2019;35(6):631-648. doi:10.1080/08927014.2019.1640214
80. Bak UG, Mols-Mortensen A, Gregersen O. Production method and cost of commercial-scale offshore cultivation of kelp in the Faroe Islands using multiple partial harvesting. *Algal Res*. 2017;2018(33):36-47. doi:10.1016/j.algal.2018.05.001
81. Gutow L, Poore AGB, Díaz Poblete MA, Villalobos V, Thiel M. Small burrowing amphipods cause major damage in a large kelp. *Proc R Soc B Biol Sci*. 2020;287(1926):20200330. doi:10.1098/rspb.2020.0330
82. Anderson DM, Fensin E, Gobler CJ, et al. Marine harmful algal blooms (HABs) in the United States: History, current status and future trends. *Harmful Algae*. 2021;102:101975. doi:10.1016/J.HAL.2021.101975
83. da Gama BAP, de Santos RPA, Pereira RC. The effect of epibionts on the susceptibility of the red seaweed *Cryptonemia seminervis* to herbivory and fouling. *J Bioadhesion Biofilm Res*. 2008;24(3):209-218. doi:10.1080/08927010802041253
84. Krumhansl KA, Lee JM, Scheibling RE. Grazing damage and encrustation by an invasive bryozoan reduce the ability of kelps to

withstand breakage by waves. *J Exp Mar Bio Ecol*. 2011;407(1):12-18. doi:10.1016/J.JEMBE.2011.06.033
85. Fitridge I, Dempster T, Guenther J, de Nys R. The impact and control of biofouling in marine aquaculture: a review. *Biofouling*. 2012;28(7):649-669. doi:10.1080/08927014.2012.700478
86. Peteiro C, Freire Ó. Biomass yield and morphological features of the seaweed *Saccharina latissima* cultivated at two different sites in a coastal bay in the Atlantic coast of Spain. *J Appl Phycol*. 2013;25(1):205-213. doi:10.1007/s10811-012-9854-9
87. Rolin C, Inkster R, Laing J, McEvoy L. Regrowth and biofouling in two species of cultivated kelp in the Shetland Islands, UK. *J Appl Phycol*. 2017;29(5):2351-2361. doi:10.1007/s10811-017-1092-8
88. Walls AM, Edwards MD, Firth LB, Johnson MP. Successional changes of epibiont fouling communities of the cultivated kelp *Alaria esculenta*: Predictability and influences. *Aquac Environ Interact*. 2017;9(1):57-71. doi:10.3354/aei00215
89. Bishop MJ, Mayer-Pinto M, Airoldi L, et al. Effects of ocean sprawl on ecological connectivity: impacts and solutions. *J Exp Mar Bio Ecol*. 2017;492:7-30. doi:10.1016/J.JEMBE.2017.01.021
90. Chung IK, Beardall J, Mehta S, Sahoo D, Stojkovic S. Using marine macroalgae for carbon sequestration: a critical appraisal. *J Appl Phycol*. 2011;23(5):877-886. doi:10.1007/s10811-010-9604-9
91. Hurd KM, Durante F-S, Harrison C-R J, et al. Effect of bryozoan colonization on inorganic nitrogen acquisition by the kelps *Agarum fimbriatum* and *Macrocystis integrifolia*. *Mar Biol*. 1994;121(1):167–173. doi:10.1007/BF00349486
92. Dalby JE, Young CM. Variable effects of ascidian competitors on oysters in a Florida epifaunal community. *J Exp Mar Bio Ecol*. 1993;167(1):47-57. doi:10.1016/0022-0981(93)90183-O
93. Leblanc A, Landry T, Miron G. Fouling organisms of the blue mussel *Mytilus edulis*: Their effect on nutrient uptake and release. *J Shellfish Res*. 2003;22(3):633-638.
94. Ross KA, Thorpe JP, Norton TA, Brand AR. Fouling in scallop cultivation: help or hindrance? *J Shellfish Res*. 2002;21(2):539-548.
95. Wahl M, Hay ME, Enderlein P. Effects of epibiosis on consumer-prey interactions. *Hydrobiologia*. 1997;355(1–3):49-59. doi:10.1007/978-94-017-1907-0_6
96. Manning LM, Lindquist N. Helpful habitant or pernicious passenger: interactions between an infaunal bivalve, an epifaunal hydroid and three potential predators. *Oecologia*. 2003;134(3):415-422. doi:10.1007/S00442-002-1134-Y
97. Fitridge I. The ecology of hydroids (Hydrozoa: Cnidaria) in Port Phillip Bay, Australia, and their impacts as fouling species in longline mussel culture. 2011. http://minerva-access.unimelb.edu.au/handle/11343/36276 Accessed September 2, 2021
98. Hickman N, Sause B. *Culture of the Blue Mussel (Mytilus Edulis Planulatus) in Port Phillip Bay, Victoria Australia. III: Larval Settlement. Report No. 75*. Victoria (Australia); 1984.
99. Paclibare JO, Evelyn TPT, Albright LJ, Prosperi-Porta L. Clearing of the kidney disease bacterium *Renibacterium salmoninarum* from seawater by the blue mussel *Mytilus edulis*, and the status of the mussel as a reservoir of the bacterium. *Dis Aquat Organ*. 1994;18(2):129-133. doi:10.3354/DAO018129
100. Walls AM, Kennedy R, Edwards MD, Johnson MP. Impact of kelp cultivation on the Ecological Status of benthic habitats and *Zostera marina* seagrass biomass. *Mar Pollut Bull*. 2017;123(1–2):19-27. doi:10.1016/j.marpolbul.2017.07.048
101. Visch W, Kononets M, Hall POJ, Nylund GM, Pavia H. Environmental impact of kelp (*Saccharina latissima*) aquaculture. *Mar Pollut Bull*. 2020;155:110962. doi:10.1016/j.marpolbul.2020.110962
102. Matsson S, Christie H, Fieler R. Variation in biomass and biofouling of kelp, *Saccharina latissima*, cultivated in the Arctic, Norway. *Aquaculture*. 2018;2019(506):445-452. doi:10.1016/j.aquaculture.2019.03.068
103. Visch W, Nylund GM, Pavia H. Growth and biofouling in kelp aquaculture (*Saccharina latissima*): the effect of location and wave exposure. *J Appl Phycol*. 2020;32(5):3199–3209. doi:10.1007/s10811-020-02201-5
104. Forbord S, Matsson S, Brodahl GE, et al. Latitudinal, seasonal and depth-dependent variation in growth, chemical composition and biofouling of cultivated *Saccharina latissima (Phaeophyceae)* along the Norwegian coast. *J Appl Phycol*. 2020;32(4):2215-2232. doi:10.1007/s10811-020-02038-y
105. Walls AM, Kennedy R, Fitzgerald RD, Blight AJ, Johnson MP, Edwards MD. Potential novel habitat created by holdfasts from cultivated *Laminaria digitata*: assessing the macroinvertebrate assemblages. *Aquac Environ Interact*. 2016;8:157-169. doi:10.3354/AEI00170
106. Airoldi L, Turon X, Perkol-Finkel S, Rius M. Corridors for aliens but not for natives: effects of marine urban sprawl at a regional scale. *Divers Distrib*. 2015;21(7):755-768. doi:10.1111/ddi.12301
107. Wilding TA, Gill AB, Boon A, et al. Turning off the DRIP ('Data-rich, information-poor') – rationalising monitoring with a focus on marine renewable energy developments and the benthos. *Renew Sustain Energy Rev*. 2017;74:848-859. doi:10.1016/J.RSER.2017.03.013
108. Camus C, Infante J, Buschmann AH. Overview of 3 year precommercial seafarming of *Macrocystis pyrifera* along the Chilean coast. *Rev Aquac*. 2018;10(3):543-559. doi:10.1111/RAQ.12185
109. Belle SMNC. Better management practices for net-pen aquaculture. In: Tucker CSHJ, ed. *Environmental Best Management Practices for Aquaculture*. Wiley-Blackwell; 2008:261–330. Accessed December 6, 2021.
110. Froehlich HE, Smith A, Gentry RR, Halpern BS. Offshore aquaculture: I know it when I see it. *Front Mar Sci*. 2017;4:154. doi:10.3389/FMARS.2017.00154/BIBTEX
111. Kerrison PD, Stanley MS, Edwards MD, Black KD, Hughes AD. The cultivation of European kelp for bioenergy: site and species selection. *Biomass Bioenerg*. 2020;2015(80):229-242. doi:10.1016/j.biombioe.2015.04.035
112. Wu Z, Zhang X, Lozano-Montes HM, Loneragan NR. Trophic flows, kelp culture and fisheries in the marine ecosystem of an artificial reef zone in the Yellow Sea. *Estuar Coast Shelf Sci*. 2016;182:86-97. doi:10.1016/J.ECSS.2016.08.021
113. Zhou J. Impacts of mariculture practices on the temporal distribution of macrobenthos in Sandu Bay, South China. *Chinese J Oceanol Limnol*. 2011;30(3):388-396. doi:10.1007/S00343-012-1150-7
114. Schratzberger M, Ingels J. Meiofauna matters: The roles of meiofauna in benthic ecosystems. *J Exp Mar Bio Ecol*. 2018;502:12-25. doi:10.1016/J.JEMBE.2017.01.007
115. Balsamo M, Semprucci F, Frontalini F, Coccioni R. Meiofauna as a tool for marine ecosystem biomonitoring. *Mar Ecosyst*. 2012;4:77-104.
116. Weisberg SB, Thompson B, Ranasinghe JA, et al. The level of agreement among experts applying best professional judgment to assess the condition of benthic infaunal communities. *Ecol Indic*. 2008;8(4):389-394. doi:10.1016/J.ECOLIND.2007.04.001
117. Borja Á, Rodríguez JG, Black K, et al. Assessing the suitability of a range of benthic indices in the evaluation of environmental impact of fin and shellfish aquaculture located in sites across Europe. *Aquaculture*. 2009;293(3–4):231-240. doi:10.1016/J.AQUACULTURE.2009.04.037
118. Lyimo TJ, Mvungi EF, Mgaya YD. Abundance and diversity of seagrass and macrofauna in the intertidal areas with and without seaweed farming. *J Sci*. 2009;34:1-14.
119. Eklöf JS, De La Torre CM, Adelsköld L, Jiddawi NS, Kautsky N. Differences in macrofaunal and seagrass assemblages in seagrass beds with and without seaweed farms. *Estuar Coast Shelf Sci*. 2005;3(63):385-396. doi:10.1016/J.ECSS.2004.11.014
120. Xu S, Chen Z, Li S, He P. Modeling trophic structure and energy flows in a coastal artificial ecosystem using mass-balance Ecopath

Model. *Estuaries Coasts*. 2011;34(2):351-363. doi:10.1007/s12237-010-9323-0
121. Kraufvelin P, Díaz ER. Sediment macrofauna communities at a small mussel farm in the northern Baltic proper. *Boreal Environ Res*. 2015;20(3):378-390.
122. Eger A, Marzinelli E, Baes R, et al. The economic value of fisheries, blue carbon, and nutrient cycling in global marine forests. *EcoEvoRxiv*. 2021, Epub ahead of print. doi:10.32942/OSF.IO/N7KJS
123. Morrisey DJ, Cole RG, Davey NK, et al. Abundance and diversity of fish on mussel farms in New Zealand. *Aquaculture*. 2006;252(2–4):277-288. doi:10.1016/j.aquaculture.2005.06.047
124. D'Amours O, Archambault P, McKindsey CW, Johnson LE. Local enhancement of epibenthic macrofauna by aquaculture activities. *Mar Ecol Prog Ser*. 2008;371:73-84. doi:10.3354/MEPS07672
125. Drouin A, Archambault P, Clynick B, Richer K, McKindsey CW. Influence of mussel aquaculture on the distribution of vagile benthic macrofauna in îles de la Madeleine, eastern Canada. *Aquac Environ Interact*. 2015;6(2):175-183. doi:10.3354/AEI00123
126. Mabrouk G, Bungay T, Drover D, Hamoutene D. Use of remote video survey methodology in monitoring benthic impacts from finfish aquaculture on the south coast of Newfoundland (Canada). *Can Sci Advis Secr*. 2014;39:15.
127. Bergman KC, Svensson S, Öhman MC. Influence of algal farming on fish assemblages. *Mar Pollut Bull*. 2001;42(12):1379-1389. doi:10.1016/S0025-326X(01)00168-0
128. Callier MD, Byron CJ, Bengtson DA, et al. Attraction and repulsion of mobile wild organisms to finfish and shellfish aquaculture: a review. *Rev Aquac*. 2018;10(4):924-949. doi:10.1111/raq.12208
129. Eklöf JS, de la Torre-Castro M, Nilsson C, Rönnbäck P. How do seaweed farms influence local fishery catches in a seagrass-dominated setting in Chwaka Bay, Zanzibar? *Aquat Living Resour*. 2006;19(2):137-147. doi:10.1051/ALR:2006013
130. Wang Z, Chen Y, Zhang S, et al. A comparative study of fish assemblages near aquaculture, artificial and natural habitats. *J Ocean Univ China*. 2015;14(1):149-160. doi:10.1007/s11802-015-2455-x
131. Bacher K, Gordoa A. Does marine fish farming affect local small-scale fishery catches? A case study in the NW Mediterranean Sea. *Aquac Res*. 2016;47(8):2444-2454. doi:10.1111/ARE.12692
132. Hehre EJ, Meeuwig JJ. A global analysis of the relationship between farmed seaweed production and herbivorous fish catch. *PLoS One*. 2016;11(2):1-17. doi:10.1371/journal.pone.0148250
133. Hehre EJ, Meeuwig JJ. Differential response of fish assemblages to coral reef-based seaweed farming. *PLoS One*. 2015;10(3):e0118838. doi:10.1371/JOURNAL.PONE.0118838
134. Bicknell AWJ, Sheehan EV, Godley BJ, Doherty PD, Witt MJ. Assessing the impact of introduced infrastructure at sea with cameras: A case study for spatial scale, time and statistical power. *Mar Environ Res*. 2018;2019(147):126-137. doi:10.1016/j.marenvres.2019.04.007
135. Childs A-R, Cowley PD, Næsje TF, et al. Do environmental factors influence the movement of estuarine fish? A case study using acoustic telemetry. *Estuar Coast Shelf Sci*. 2008;78(1):227-236. doi:10.1016/J.ECSS.2007.12.003
136. Meynecke JO, Poole GC, Werry J, Lee SY. Use of PIT tag and underwater video recording in assessing estuarine fish movement in a high intertidal mangrove and salt marsh creek. *Estuar Coast Shelf Sci*. 2008;79(1):168-178. doi:10.1016/J.ECSS.2008.03.019
137. Lorentsen SH, Sjøtun K, Grémillet D. Multi-trophic consequences of kelp harvest. *Biol Conserv*. 2010;143(9):2054-2062. doi:10.1016/J.BIOCON.2010.05.013
138. Pimiento C, Leprieur F, Silvestro D, et al. Functional diversity of marine megafauna in the Anthropocene. *Sci Adv*. 2020;6(16):eaay7650. doi:10.1126/SCIADV.AAY7650
139. Mazzoldi C, Bearzi G, Brito C, et al. From sea monsters to charismatic megafauna: changes in perception and use of large marine animals. *PLoS One*. 2019;14(12):e0226810. doi:10.1371/JOURNAL.PONE.0226810
140. Caldow R, Beadman HA, McGrorty S, et al. Effects of intertidal mussel cultivation on bird assemblages. *Mar Ecol Prog Ser*. 2003;259:173-183. doi:10.3354/meps259173
141. Nemtzov SC, Olsvig-Whittaker L. The use of netting over fishponds as a hazard to waterbirds. *Waterbirds*. 2003;26(4):416-423.
142. Roycroft D, Kelly TC, Lewis LJ. Birds, seals and the suspension culture of mussels in Bantry Bay, a non-seaduck area in Southwest Ireland. *Estuar Coast Shelf Sci*. 2004;61(4):703-712. doi:10.1016/J.ECSS.2004.07.012
143. Žydelis R, Esler D, Kirk M, Boyd WS. Effects of off-bottom shellfish aquaculture on winter habitat use by molluscivorous sea ducks. *Aquat Conserv Mar Freshw Ecosyst*. 2009;19(1):34-42. doi:10.1002/AQC.977
144. Northridge S, Coram A, Gordon J. Investigations on seal depredation at Scottish fish farms report to Marine Scotland. *Edinburgh Scottish Gov*. 2013;1-79.
145. Díaz López B, Marini L, Polo F. The impact of a fish farm on a bottlenose dolphin population in the Mediterranean Sea. *Thalassas*. 2005;21(2):65–70.
146. Díaz López B, Methion S. The impact of shellfish farming on common bottlenose dolphins' use of habitat. *Mar Biol*. 2017;164:83. doi:10.1007/s00227-017-3125-x
147. Markowitz TM, Harlin AD, Würsig B, McFadden CJ. Dusky dolphin foraging habitat: overlap with aquaculture in New Zealand. *Aquat Conserv Mar Freshw Ecosyst*. 2004;14(2):133-149. doi:10.1002/AQC.602
148. Pearson HCH, Vaughn-Hirshorn RLR, Srinivasan M, Würsig B. Avoidance of mussel farms by dusky dolphins (*Lagenorhynchus obscurus*) in New Zealand. *New Zeal J Mar Freshw Res*. 2012;46(4):567-574. doi:10.1080/00288330.2012.712977
149. Watson-Capps JJ, Mann J. The effects of aquaculture on bottlenose dolphin (*Tursiops* sp.) ranging in Shark Bay, Western Australia. *Biol Conserv*. 2005;124(4):519-526. doi:10.1016/J.BIOCON.2005.03.001
150. Estes JA, Danner EM, Doak DF, et al. Complex trophic interactions in kelp forest ecosystems. *Bull Mar Sci*. 2004;74(3):621-638.
151. Russell DJ, Balazs GH. Increased use of non-native algae species in the diet of the Green Turtle (*Chelonia mydas*) in a primary pasture ecosystem in Hawaii. *Aquat Ecosyst Health Manag*. 2015;18(3):342-346. doi:10.1080/14634988.2015.1027140
152. Vanar M. Four more sea turtles found killed. The Star. 2021. Published 2014. Accessed September 2 https://www.thestar.com.my/news/nation/2014/04/17/four-more-sea-turtles-found-killed-carcasses-seen-floating-in-waters-off-semporna
153. Teitelbaum A. *Farming Seaweed in Kiribati: A Practical Guide for Seaweed Farmers*. Secretariat of the Pacific Community; 2003.
154. Kirkwood JK, Bennett PM, Jepson PD, Kuiken T, Simpson VR, Baker JR. Entanglement in fishing gear and other causes of death in cetaceans stranded on the coasts of England and Wales. *Vet Rec*. 1997;141(4):94-98. doi:10.1136/VR.141.4.94
155. Read AJ, Drinker P, Northridge S. Bycatch of marine mammals in U.S. and global fisheries. *Conserv Biol*. 2006;20(1):163-169. doi:10.1111/J.1523-1739.2006.00338.X
156. Young OM. Marine animal entanglements in mussel aquaculture gear: documented cases from mussel farming regions of the world including first-hand accounts from Iceland. May 2015.
157. Price CS, Morris JA, Keane EP, Morin DM, Vaccaro C, Bean DW. Protected species and marine aquaculture interactions. *NOAA Tech Memo NOS NCCOS*. 2017;211:1-82. doi:10.7289/V5/TM-NOS-NCCOS-211
158. Poonian C, Lopez D. Small-scale mariculture: a potentially significant threat to dugongs (*Dugong dugon*) through incidental

entanglement. *Aquat Mamm*. 2016;42(1):56-59. doi:10.1578/AM.42.1.2016.56
159. Castelblanco-Martínez DN, Blanco-Parra MP, Charruau P, et al. Detecting, counting and following the giants of the sea: a review of monitoring methods for aquatic megavertebrates in the Caribbean. *Wildl Res*. 2019;46(7):545-556. doi:10.1071/WR19008
160. Isaksson N, Masden EA, Williamson BJ, et al. Assessing the effects of tidal stream marine renewable energy on seabirds: A conceptual framework. *Mar Pollut Bull*. 2020;157:111314. doi:10.1016/J.MARPOLBUL.2020.111314
161. Álvarez E, Moyano M, López-Urrutia Á, Nogueira E, Scharek R. Routine determination of plankton community composition and size structure: a comparison between FlowCAM and light microscopy. *J Plankton Res*. 2014;36(1):170-184. doi:10.1093/PLANKT/FBT069
162. Wang Y, Castillo-Keller M, Eustance E, Sommerfeld M. Early detection and quantification of zooplankton grazers in algal cultures by FlowCAM. *Algal Res*. 2017;21:98-102. doi:10.1016/J.ALGAL.2016.11.012
163. Yang Y, Liu Q, Chai Z, Tang Y. Inhibition of marine coastal bloom-forming phytoplankton by commercially cultivated *Gracilaria lemaneiformis (Rhodophyta)*. *J Appl Phycol*. 2014;27(6):2341-2352. doi:10.1007/S10811-014-0486-0
164. Jiang Z-B, Chen Q-Z, Zeng J-N, Liao Y-B, Shou L, Liu J. Phytoplankton community distribution in relation to environmental parameters in three aquaculture systems in a Chinese subtropical eutrophic bay. *Mar Ecol Prog Ser*. 2012;446:73-89. doi:10.3354/MEPS09499
165. Jiang Z, Du P, Liao Y, et al. Oyster farming control on phytoplankton bloom promoted by thermal discharge from a power plant in a eutrophic, semi-enclosed bay. *Water Res*. 2019;159:1-9. doi:10.1016/J.WATRES.2019.04.023
166. Fernandez-Gonzalez V, Fernandez-Jover D, Toledo-Guedes K, Valero-Rodriguez JM, Sanchez-Jerez P. Nocturnal planktonic assemblages of amphipods vary due to the presence of coastal aquaculture cages. *Mar Environ Res*. 2014;101(1):22-28. doi:10.1016/j.marenvres.2014.08.001
167. Skilton DC, Saunders RJ, Hutson KS. Parasite attractants: Identifying trap baits for parasite management in aquaculture. *Aquaculture*. 2020;516:734557. doi:10.1016/J.AQUACULTURE.2019.734557
168. Lei Y, Feng P, Du X, Jiang S. Diatom assemblages from sediment traps in response to large seaweed *Gracilaria* cultivation off Nan'ao island, South China. *Mar Pollut Bull*. 2021;165:112157. doi:10.1016/J.MARPOLBUL.2021.112157
169. Offenbaume KL, Bertone E, Stewart RA. Monitoring approaches for faecal indicator bacteria in water: visioning a remote real-time sensor for *E. coli* and *Enterococci*. *Water*. 2020;12(9):2591. doi:10.3390/W12092591
170. Schmidt W, Raymond D, Parish D, et al. Design and operation of a low-cost and compact autonomous buoy system for use in coastal aquaculture and water quality monitoring. *Aquac Eng*. 2017;2018(80):28-36. doi:10.1016/j.aquaeng.2017.12.002
171. UnifAI Technology. Harbour and Coastal Waters, UK. https://www.unifaitechnology.com/harbour-and-coastal-waters Published 2021 Accessed September 2, 2021
172. Burnet JB, Dinh QT, Imbeault S, Servais P, Dorner S, Prévost M. Autonomous online measurement of β-D-glucuronidase activity in surface water: is it suitable for rapid *E. coli* monitoring? *Water Res*. 2019;152:241-250. doi:10.1016/J.WATRES.2018.12.060
173. Anderson MJ, Diebel CE, Blom WM, Landers TJ. Consistency and variation in kelp holdfast assemblages: spatial patterns of biodiversity for the major phyla at different taxonomic resolutions. *J Exp Mar Bio Ecol*. 2005;320(1):35-56. doi:10.1016/j.jembe.2004.12.023
174. Cundell AM, Sleeter TD, Mitchell R. Microbial populations associated with the surface of the brown alga *Ascophyllum nodosum*. *Microb Ecol*. 1977;4:81-91.
175. Bolinches J, Lemos ML, Barja JL. Population dynamics of heterotrophic bacterial communities associated with *Fucus vesiculosus* and *Ulva rigida* in an estuary. *Microb Ecol*. 1988;15:345-357.
176. Armstrong E, Rogerson A, Leftley JW. The abundance of heterotrophic protists associated with intertidal seaweeds. *Estuar Coast Shelf Sci*. 2000;50(3):415-424. doi:10.1006/ECSS.1999.0577
177. Bengtsson MM, Sjøtun K, Øvreås L. Seasonal dynamics of bacterial biofilms on the kelp *Laminaria hyperborea*. *Aquat Microb Ecol*. 2010;60(1):71-83. doi:10.3354/AME01409
178. Teagle H, Hawkins SJ, Moore PJ, Smale DA. The role of kelp species as biogenic habitat formers in coastal marine ecosystems. *J Exp Mar Bio Ecol*. 2017;492:81-98. doi:10.1016/J.JEMBE.2017.01.017
179. Teagle H, Moore PJ, Jenkins H, Smale DA. Spatial variability in the diversity and structure of faunal assemblages associated with kelp holdfasts (*Laminaria hyperborea*) in the northeast Atlantic. *PLoS One*. 2018;13(7):1-25. doi:10.1371/journal.pone.0200411
180. Eleftheriou A, Moore DC. Macrofauna Techniques. In: Eleftheriou A (Ed.), *Methods for the Study of Marine Benthos*, 4th edition. John Wiley & Sons; 2013. 10.1002/9781118542392.ch5
181. Hansen PK, Ervik A, Schaanning M, et al. Regulating the local environmental impact of intensive, marine fish farming: II. The monitoring programme of the MOM system (Modelling–Ongrowing fish farms–Monitoring). *Aquaculture*. 2001;194(1–2):75-92. doi:10.1016/S0044-8486(00)00520-2
182. Zhang J, Hansen PK, Fang J, Wang W, Jiang Z. Assessment of the local environmental impact of intensive marine shellfish and seaweed farming—Application of the MOM system in the Sungo Bay, China. *Aquaculture*. 2009;287(3–4):304-310. doi:10.1016/J.AQUACULTURE.2008.10.008
183. Clarke KR, Gorley RN, Somerfield PJ, Warwick RM. *Change in Marine Communities: An Approach to Statistical Analysis and Interpretation*, 3rd ed. Plymouth: PRIMER-e Ltd; 2014.
184. La Rosa T, Mirto S, Mazzola A, Danovaro R. Differential responses of benthic microbes and meiofauna to fish-farm disturbance in coastal sediments. *Environ Pollut*. 2001;112(3):427-434. doi:10.1016/S0269-7491(00)00141-X
185. Verhoeven JTP, Salvo F, Knight R, Hamoutene D, Dufour SC. Temporal bacterial surveillance of salmon aquaculture sites indicates a long lasting benthic impact with minimal recovery. *Front Microbiol*. 2018;3054: doi:10.3389/FMICB.2018.03054
186. Frühe L, Dully V, Forster D, et al. Global trends of benthic bacterial diversity and community composition along organic enrichment gradients of salmon farms. *Front Microbiol*. 2021;853. doi:10.3389/FMICB.2021.637811
187. Clynick BG, McKindsey CW, Archambault P. Distribution and productivity of fish and macroinvertebrates in mussel aquaculture sites in the Magdalen islands (Québec, Canada). *Aquaculture*. 2008;283(1–4):203-210. doi:10.1016/j.aquaculture.2008.06.009
188. Mente E, Martin JC, Tuck I, et al. Mesoscale effects of aquaculture installations on benthic and epibenthic communities in four Scottish sea lochs. *Aquat Living Resour*. 2010;23(3):267-276. doi:10.1051/ALR/2010030
189. Tallman JC, Forrester GE. Oyster grow-out cages function as artificial reefs for temperate fishes. *Trans Am Fish Soc*. 2007;136(3):790-799. doi:10.1577/t06-119.1
190. Grant J, Simone M, Daggett T. Long-term studies of lobster abundance at a salmon aquaculture site, eastern Canada. *Can J Fish Aquat Sci*. 2019;76(7):1096-1102. doi:10.1139/CJFAS-2017-0547
191. Assis J, Claro B, Ramos A, Boavida J, Serrão EA. Performing fish counts with a wide-angle camera, a promising approach reducing divers' limitations. *J Exp Mar Bio Ecol*. 2013;445:93-98. doi:10.1016/J.JEMBE.2013.04.007

192. Wehkamp S, Fischer P. Impact of coastal defence structures (tetrapods) on a demersal hard-bottom fish community in the southern North Sea. *Mar Environ Res*. 2013;83:82-92. doi:10.1016/j.marenvres.2012.10.013
193. Bethoney ND, Cleaver C, Asci SC, Bayer SR, Wahle RA, Stokesbury KDE. A comparison of drop camera and diver survey methods to monitor Atlantic sea scallops (*Placopecten magellanicus*) in a small fishery closure. *J Shellfish Res*. 2019;38(1):43-51. doi:10.2983/035.038.0104
194. Šegvić-Bubić T, Grubišić L, Karaman N, Tičina V, Jelavić KM, Katavić I. Damages on mussel farms potentially caused by fish predation-Self service on the ropes? *Aquaculture*. 2011;319(3–4):497-504. doi:10.1016/j.aquaculture.2011.07.031
195. Ballester-Moltó M, Sanchez-Jerez P, García-García B, Aguado-Giménez F. Husbandry and environmental conditions explain temporal variability of wild fish assemblages aggregated around a Mediterranean fish farm. *Aquac Environ Interact*. 2015;7(3):193-203. doi:10.3354/AEI00148
196. Dempster T, Sanchez-Jerez P, Sempere JB, Kingsford M. Extensive aggregations of wild fish at coastal sea-cage fish farms. *Hydrobiologia*. 2004;525(1):245-248. doi:10.1023/B:HYDR.0000038870.13985.0F
197. Valle C, Bayle-Sempere JT, Dempster T, Sanchez-Jerez P, Giménez-Casalduero F. Temporal variability of wild fish assemblages associated with a sea-cage fish farm in the south-western Mediterranean Sea. *Estuar Coast Shelf Sci*. 2007;72(1–2):299-307. doi:10.1016/j.ecss.2006.10.019
198. Fernandez-Jover D, Sanchez-Jerez P, Bayle-Sempere JT, Valle C, Dempster T. Seasonal patterns and diets of wild fish assemblages associated with Mediterranean coastal fish farms. *ICES J Mar Sci*. 2008;65(7):1153-1160. doi:10.1093/ICESJMS/FSN091
199. Sudirman, Halide H, Jompa J, Zulfikar, Iswahyudin, McKinnon AD. Wild fish associated with tropical sea cage aquaculture in South Sulawesi, Indonesia. *Aquaculture*. 2009;286(3–4):233-239. doi:10.1016/j.aquaculture.2008.09.020
200. Arechavala-Lopez P, Uglem I, Sanchez-Jerez P, Fernandez-Jover D, Bayle-Sempere JT, Nilsen R. Movements of grey mullet *Liza aurata* and *Chelon labrosus* associated with coastal fish farms in the western Mediterranean Sea. *Aquac Environ Interact*. 2010;1(2):127-136. doi:10.3354/AEI00012
201. Arechavala-Lopez P, Sanchez-Jerez P, Bayle-Sempere J, et al. Direct interaction between wild fish aggregations at fish farms and fisheries activity at fishing grounds: a case study with *Boops boops*. *Aquac Res*. 2011;42(7):996-1010. doi:10.1111/J.1365-2109.2010.02683.X
202. Bacher K, Gordoa A, Sagué O. Spatial and temporal extension of wild fish aggregations at Sparus aurata and Thunnus thynnus farms in the north-western Mediterranean. *Aquac Environ Interact*. 2012;2(3):239-252. doi:10.3354/AEI00042
203. Özgül A, Angel D. Wild fish aggregations around fish farms in the Gulf of Aqaba, Red Sea: implications for fisheries management and conservation. *Aquac Environ Interact*. 2013;4(2):135-145. doi:10.3354/AEI00076
204. Clynick BG. Assemblages of fish associated with coastal marinas in north-western Italy. *J Mar Biol Assoc UK*. 2006;86(4):847-852. doi:10.1017/S0025315406013786
205. Langlois TJ, Harvey ES, Fitzpatrick B, Meeuwig JJ, Shedrawi G, Watson DL. Cost-efficient sampling of fish assemblages: comparison of baited video stations and diver video transects. *Aquat Biol*. 2010;9(2):155-168. doi:10.3354/AB00235
206. Mallet D, Pelletier D. Underwater video techniques for observing coastal marine biodiversity: a review of sixty years of publications (1952–2012). *Fish Res*. 2014;154:44-62. doi:10.1016/j.fishres.2014.01.019
207. Langlois T, Williams J, Monk J, et al. Marine sampling field manual for benthic stereo BRUVS (Baited Remote Underwater Videos) [Version 1]. In: Przeslawski R, Foster S, eds. *Field Manuals for Marine Sampling to Monitor Australian Waters, Version 1*. NESP Marine Biodiversity Hub; 2018:82–104. doi:10.11636/9781925297669
208. Tonk L, Bernard MS, Jansen H. The use of video-techniques for monitoring and quantification of mobile fauna in marine cultivation systems. *Wageningen Univ Res Rep C017/19*. 2019. 10.18174/471068
209. Sheehan EV, Bridger D, Nancollas SJ, Pittman SJ. PelagiCam: a novel underwater imaging system with computer vision for semi-automated monitoring of mobile marine fauna at offshore structures. *Environ Monit Assess*. 2020;192(1):1–13. doi:10.1007/s10661-019-7980-4
210. Sheehan EV, Cartwright AY, Witt MJ, Attrill MJ, Vural M, Holmes LA. Development of epibenthic assemblages on artificial habitat associated with marine renewable infrastructure. *ICES J Mar Sci*. 2020;77(3):1178-1189. doi:10.1093/icesjms/fsy151
211. Davis GE, Anderson TW. Population estimates of four kelp forest fishes and an evaluation of three in situ assessment techniques. *Bull Mar Sci*. 1989;44(3):1138-1151.
212. Firth LB, Knights AM, Bridger D, et al. Ocean sprawl: challenges and opportunities for biodiversity management in a changing world. *Oceanogr Mar Biol an Annu Rev*. 2016;54:193-269. doi:10.1201/9781315368597
213. Gormley K, McLellan F, McCabe C, et al. Automated image analysis of offshore infrastructure marine biofouling. *J Mar Sci Eng*. 2018;6(1):2. doi:10.3390/JMSE6010002
214. Tanner JE, Williams K. The influence of finfish aquaculture on benthic fish and crustacean assemblages in Fitzgerald Bay, South Australia. *PeerJ*. 2015;3(9):e1238. doi:10.7717/PEERJ.1238
215. Hamoutene D, Salvo F, Bungay T, et al. Assessment of finfish aquaculture effect on Newfoundland epibenthic communities through video monitoring. *Chang Publ Wiley*. 2015;77(2):117-127. doi:10.1080/15222055.2014.976681
216. Sheehan EV, Vaz S, Pettifer E, et al. An experimental comparison of three towed underwater video systems using species metrics, benthic impact and performance. *Methods Ecol Evol*. 2016;7(7):843-852. doi:10.1111/2041-210X.12540
217. Dempster T, Uglem I, Sanchez-Jerez P, et al. Coastal salmon farms attract large and persistent aggregations of wild fish: An ecosystem effect. *Mar Ecol Prog Ser*. 2009;385:1-14. doi:10.3354/meps08050
218. Dempster T, Sanchez-Jerez P, Fernandez-Jover D, et al. Proxy measures of fitness suggest coastal fish farms can act as population sources and not ecological traps for wild gadoid fish. *PLoS One*. 2011;6(1):e15646. doi:10.1371/JOURNAL.PONE.0015646
219. Bicknell AWJ, Godley BJ, Sheehan EV, Votier SC, Witt MJ. Camera technology for monitoring marine biodiversity and human impact. *Front Ecol Environ*. 2016;14(8):424-432. doi:10.1002/fee.1322
220. Bell TW, Nidzieko NJ, Siegel DA, et al. The utility of satellites and autonomous remote sensing platforms for monitoring offshore aquaculture farms: a case study for canopy forming kelps. *Front Mar Sci*. 2020;7:1083. doi:10.3389/FMARS.2020.520223
221. Torao M. Counting of salmon escapements from aerial video images captured using a drone. *Aquac Sci*. 2017;65(2):157-159. doi:10.11233/AQUACULTURESCI.65.157
222. Jacobs S, Terhune J. Harbor seal (*Phoca vitulina*) numbers along the New Brunswick coast of the Bay of Fundy in autumn in relation to aquaculture. *Northeast Nat*. 2000;7(3):289-296.
223. Dorr BS, Burger LW, Barras SC. Evaluation of aerial cluster sampling of double-crested cormorants on aquaculture ponds in Mississippi. *J Wildl Manage*. 2008;72(7):1634-1640. doi:10.2193/2007-308
224. Burr PC, Avery JL, Street GM, Strickland BK, Dorr BS. Historic and contemporary use of catfish aquaculture by piscivorous birds in the Mississippi Delta. *Condor*. 2020;122(4):1-13. doi:10.1093/CONDOR/DUAA036

225. Burr PC, Avery JL, Street GM, Strickland BK, Dorr BS. Piscivorous bird use of aquaculture and natural water bodies in Mississippi. *J Wildl Manage*. 2020;84(8):1560-1569. doi:10.1002/JWMG.21948
226. Alves MDDO, Schwamborn R, Borges JCG, et al. Aerial survey of manatees, dolphins and sea turtles off northeastern Brazil: correlations with coastal features and human activities. *Biol Conserv*. 2013;161:91-100. doi:10.1016/J.BIOCON.2013.02.015
227. Ampela K, Jefferson TA, Smultea MA. Estimation of in-water density and abundance of harbor seals. *J Wildl Manage*. 2021;85(4):706-712. doi:10.1002/JWMG.22019
228. Sabetian A, Breen D, Vavia A, et al. SINOPSIS – the Solomon Islands National Ocean Planning Spatial Information System: Spatial decision support tools for seaweed aquaculture planning in the Solomon Islands. *New Zeal Inst Pacific Res*., 2019. https://openrepository.aut.ac.nz/handle/10292/12578 Accessed September 2, 2021
229. Otterå H, Skilbrei OT. Possible influence of salmon farming on long-term resident behaviour of wild saithe (*Pollachius virens L*.). *ICES J Mar Sci*. 2014;71(9):2484-2493. doi:10.1093/ICESJMS/FSU096
230. Hawkins P. Bio-logging and animal welfare: practical refinements. *Mem Natl Inst Polar Res Spec Issue*. 2004;58:58-68.
231. Horning M, Haulena M, Tuomi PA, et al. Best practice recommendations for the use of fully implanted telemetry devices in pinnipeds. *Anim Biotelemetry*. 2017;5(1):1-15. doi:10.1186/S40317-017-0128-9
232. Comeau LA, Sonier R, Hanson JM. Seasonal movements of Atlantic rock crab (*Cancer irroratus Say*) transplanted into a mussel aquaculture site. *Aquac Res*. 2012;43(4):509-517. doi:10.1111/J.1365-2109.2011.02856.X
233. Hunter E, Eaton D, Stewart C, Lawler A, Smith MT. Edible crabs "go west": migrations and incubation cycle of *Cancer pagurus* revealed by electronic tags. *PLoS One*. 2013;8(5):e63991. doi:10.1371/JOURNAL.PONE.0063991
234. Papastamatiou YP, Itano DG, Dale JJ, et al. Site fidelity and movements of sharks associated with ocean-farming cages in Hawaii. *Mar Freshw Res*. 2010;61(12):1366-1375. doi:10.1071/MF10056
235. Arechavala-Lopez P, Borg JA, Šegvić-Bubić T, Tomassetti P, Özgül A, Sanchez-Jerez P. Aggregations of wild Atlantic Bluefin Tuna (*Thunnus thynnus L*.) at Mediterranean offshore fish farm sites: Environmental and management considerations. *Fish Res*. 2015;164:178-184. doi:10.1016/J.FISHRES.2014.11.011
236. McIntyre T. Trends in tagging of marine mammals: a review of marine mammal biologging studies. *African J Mar Sci*. 2014;36(4):409-422. doi:10.2989/1814232X.2014.976655
237. Williams CL, Ponganis PJ. Diving physiology of marine mammals and birds: the development of biologging techniques. *Philos Trans R Soc B*. 2021;376(1830):20200211. doi:10.1098/RSTB.2020.0211
238. Chung H, Lee J, Lee WY. A review: Marine bio-logging of animal behaviour and ocean environments. *Ocean Sci J*. 2021;56(2):117-131. doi:10.1007/S12601-021-00015-1
239. Brehmer P, Gerlotto F, Guillard J, Sanguinède F, Guénnegan Y, Buestel D. New applications of hydroacoustic methods for monitoring shallow water aquatic ecosystems: the case of mussel culture grounds. *Aquat Living Resour*. 2003;16(3):333-338. doi:10.1016/S0990-7440(03)00042-1
240. Giannoulaki M, Machias A, Somarakis S, Karakassis I. Wild fish spatial structure in response to presence of fish farms. *J Mar Biol Assoc United Kingdom*. 2005;85:1271-1277.
241. Goodbrand L, Abrahams MV, Rose GA. Sea cage aquaculture affects distribution of wild fish at large spatial scales. *Can J Fish Aquat Sci*. 2013;70(9):1289-1295. doi:10.1139/cjfas-2012-0317
242. Foote AD, Thomsen PF, Sveegaard S, et al. Investigating the potential use of environmental DNA (eDNA) for genetic monitoring of marine mammals. *PLoS One*. 2012;7(8):e41781. doi:10.1371/JOURNAL.PONE.0041781
243. Wang S, Yan Z, Hänfling B, et al. Methodology of fish eDNA and its applications in ecology and environment. *Sci Total Environ*. 2021;755:142622. doi:10.1016/J.SCITOTENV.2020.142622
244. Port JA, O'Donnell JL, Romero-Maraccini OC, et al. Assessing vertebrate biodiversity in a kelp forest ecosystem using environmental DNA. *Mol Ecol*. 2016;25(2):527-541. doi:10.1111/mec.13481
245. Jo H, Kim D-K, Park K, Kwak I-S. Discrimination of spatial distribution of aquatic organisms in a coastal ecosystem using eDNA. *Appl Sci*. 2019;9(17):3450. doi:10.3390/APP9173450
246. Liu QI, Zhang Y, Wu H, et al. A review and perspective of eDNA application to eutrophication and hab control in freshwater and marine ecosystems. *Microorganisms*. 2020;8(3):417. doi:10.3390/MICROORGANISMS8030417
247. Stat M, Huggett MJ, Bernasconi R, et al. Ecosystem biomonitoring with eDNA: metabarcoding across the tree of life in a tropical marine environment. *Sci Rep*. 2017;7(1):1–11. doi:10.1038/s41598-017-12501-5
248. Bastos Gomes G, Hutson KS, Domingos JA, et al. Use of environmental DNA (eDNA) and water quality data to predict protozoan parasites outbreaks in fish farms. *Aquaculture*. 2017;479:467-473. doi:10.1016/J.AQUACULTURE.2017.06.021
249. Doucette GJ, Medlin LK, McCarron P, Hess P. Detection and surveillance of harmful algal bloom species and toxins. *Harmful Algal Bloom*. 2018:39–114. doi:10.1002/9781118994672.CH2
250. Pearman JK, Keeley NB, Wood SA, et al. Comparing sediment DNA extraction methods for assessing organic enrichment associated with marine aquaculture. *PeerJ*. 2020;8:e10231. doi:10.7717/PEERJ.10231
251. Dully V, Balliet H, Frühe L, et al. Robustness, sensitivity and reproducibility of eDNA metabarcoding as an environmental biomonitoring tool in coastal salmon aquaculture – An inter-laboratory study. *Ecol Indic*. 2021;121: doi:10.1016/J.ECOLIND.2020.107049. 107049.
252. Stat M, John J, DiBattista JD, Newman SJ, Bunce M, Harvey ES. Combined use of eDNA metabarcoding and video surveillance for the assessment of fish biodiversity. *Conserv Biol*. 2019;33(1):196-205. doi:10.1111/COBI.13183
253. Bruhn A, Boderskov T, Pedersen MF, et al. Ecosystem impact of large-scale macroalgae cultivation — Welcome to DTU Research Database. In: *Ecosystem Impact of Large-Scale Macroalgae Cultivation*; 2019. https://orbit.dtu.dk/en/publications/ecosystem-impact-of-large-scale-macroalgae-cultivation Accessed September 2, 2021
254. Andruszkiewicz EA, Starks HA, Chavez FP, Sassoubre LM, Block BA, Boehm AB. Biomonitoring of marine vertebrates in Monterey Bay using eDNA metabarcoding. *PLoS One*. 2017;12(4):e0176343. doi:10.1371/JOURNAL.PONE.0176343
255. Ortega A, Geraldi NR, Díaz-Rúa R, et al. A DNA mini-barcode for marine macrophytes. *Mol Ecol Resour*. 2020;20(4):920-935. doi:10.1111/1755-0998.13164
256. Cordier T, Frontalini F, Cermakova K, et al. Multi-marker eDNA metabarcoding survey to assess the environmental impact of three offshore gas platforms in the North Adriatic Sea (Italy). *Mar Environ Res*. 2019;146:24-34. doi:10.1016/J.MARENVRES.2018.12.009
257. Methion S, López BD. Natural and anthropogenic drivers of foraging behaviour in bottlenose dolphins: Influence of shellfish aquaculture. *Aquat Conserv Mar Freshw Ecosyst*. 2019;29(6):927-937. doi:10.1002/AQC.3116
258. Grant J. Coastal communities, participatory research, and far-field effects of aquaculture. *Aquac Environ Interact*. 2010;1(2):85-93. doi:10.3354/AEI00009
259. Raoult V, David PA, Dupont SF, et al. GoPros^{TM} as an underwater photogrammetry tool for citizen science. *PeerJ*. 2016;4(4):e1960. doi:10.7717/PEERJ.1960
260. Harvey GKA, Nelson TA, Paquet PC, Ferster CJ, Fox CH. Comparing citizen science reports and systematic surveys of marine

mammal distributions and densities. *Biol Conserv*. 2018;226:92-100. doi:10.1016/J.BIOCON.2018.07.024
261. Hann CH, Stelle LL, Szabo A, Torres LG. Obstacles and opportunities of using a mobile app for marine mammal research. *ISPRS Int J Geo-Information*. 2018;7(5):169. doi:10.3390/IJGI7050169
262. Underwood AJ. On beyond BACI: sampling designs that might reliably detect environmental disturbances. *Ecol Appl*. 1994;4(1):3-15. doi:10.2307/1942110
263. Peteiro C, Freire Ó. Epiphytism on blades of the edible kelps *Undaria pinnatifida* and Saccharina latissima farmed under different abiotic conditions. *J World Aquac Soc*. 2013;44(5):706-715. doi:10.1111/JWAS.12065
264. Walls AM, Edwards MD, Firth LB, Johnson MP. Ecological priming of artificial aquaculture structures: kelp farms as an example. *J Mar Biol Assoc United Kingdom*. 2019;99(4):729-740. doi:10.1017/S0025315418000723
265. Bayle-Sempere JT, Valle C, Verdu A. *ecoCEN: A database for fish counts*. FAO-AECI. http://www.ua.es/ecocen Published 2002
266. Walls AM, Graham CT, Edwards MD, Johnson MP. Incorporation of cultivated kelp (*Alaria esculenta* and *Saccharina latissima*) into the diet of the blue mussel Mytilus edulis: a stable isotope study. In: *Ecosystem Services and Environmental Impacts Associated with Commercial Kelp Aquaculture*. Galway: National University of Ireland Galway; 2017:140–161.
267. Quintino V, Elliott M, Rodrigues AM. The derivation, performance and role of univariate and multivariate indicators of benthic change: Case studies at differing spatial scales. *J Exp Mar Bio Ecol*. 2006;330(1):368-382. doi:10.1016/J.JEMBE.2005.12.040
268. ter Braak CJF. Canonical correspondence analysis: a new Eigenvector technique for multivariate direct gradient analysis. *Ecology*. 1986;67(5):1167-1179. doi:10.2307/1938672
269. Phillips GR, Anwar A, Brooks L, Martina LJ, Miles AC, Prior A. *Infaunal Quality Index: Water Framework Directive Classification Scheme for Marine Benthic Invertebrates*. Environ Agency; 2014.
270. Maurer D, Nguyen H, Robertson G, Gerlinger T. The Infaunal Trophic Index (ITI): Its suitability for marine environmental monitoring. *Ecol Appl*. 1999;9(2):699-713. doi:10.1890/1051-0761(1999)009
271. Borja A, Dauer DM. Assessing the environmental quality status in estuarine and coastal systems: comparing methodologies and indices. *Ecol Indic*. 2008;8(4):331-337. doi:10.1016/J.ECOLIND.2007.05.004
272. Tomassetti P, Gennaro P, Lattanzi L, et al. Benthic community response to sediment organic enrichment by Mediterranean fish farms: Case studies. *Aquaculture*. 2016;450:262-272. doi:10.1016/j.aquaculture.2015.07.019
273. Qiu B, Zhong X, Liu X. Assessment of the benthic ecological status in the adjacent waters of Yangtze River Estuary using marine biotic indices. *Mar Pollut Bull*. 2018;137:104-112. doi:10.1016/J.MARPOLBUL.2018.10.006
274. Tillin HM, Hull SC, Tyler-Walters H. *Development of a sensitivity matrix (pressures-MCZ/MPA features)*; 2010.
275. Tillin H, Tyler-Walters H. *Assessing the sensitivity of subtidal sedimentary habitats to pressures associated with marine activities. Phase 2 Report – Literature review and sensitivity assessments for ecological groups for circalittoral and offshore level 5 biotopes*; 2014. www.marlin.ac.uk/publications
276. Tyler-Walters H, Tillin HM, d'Avack EAS, Perry F, Stamp T. *Marine Evidence-Based Sensitivity Assessment (MarESA) – A Guide*. Plymouth, Marine Life Information Network; 2018:1-94.
277. Ross LG, Telfer TC, Falconer L, Soto D, Aguilar-Manjarrez J. Site selection and carrying capacities for inland and coastal aquaculture. *FAO/Institute Aquac Univ Stirling, Expert Work 6–8 December 2010 Stirling, United Kingdom Gt Britain North Ireland FAO Fish Aquac Proc No 21 Rome, FAO*; 46: 2013.
278. Smaal AC, van Duren LA. Bivalve aquaculture carrying capacity: concepts and assessment tools. In: Smaal, AC, Ferreira, J, Grant, J, Petersen, J, Strand, Ø, eds. *Goods and Services of Marine Bivalves*. Cham: Springer; 2019:451–483. doi:10.1007/978-3-319-96776-9_23
279. Fan L, Meirong D, Hui L, Jianguang F, Lars A, Zengjie J. A physical-biological coupled ecosystem model for integrated aquaculture of bivalve and seaweed in Sanggou bay. *Ecol Modell*. 2020;431:109181. doi:10.1016/J.ECOLMODEL.2020.109181
280. Serpetti N, Benjamins S, Brain S, et al. Modeling small scale impacts of multi-purpose platforms: an ecosystem approach. *Front Mar Sci*. 2021;8:778. doi:10.3389/FMARS.2021.694013/BIBTEX
281. Muller-Karger FE, Miloslavich P, Bax NJ, et al. Advancing marine biological observations and data requirements of the complementary essential ocean variables (EOVs) and essential biodiversity variables (EBVs) frameworks. *Front Mar Sci*. 2018;5:211. doi:10.3389/FMARS.2018.00211
282. Duffy JE, Benedetti-Cecchi L, Trinanes J, et al. Toward a coordinated global observing system for seagrasses and marine macroalgae. *Front Mar Sci*. 2019;6:317: doi:10.3389/FMARS.2019.00317
283. Aquaculture Stewardship Council and Marine Stewardship Council, ASC-MSC Seaweed (Algae) Standard, 2018, V 1.0. Accessed February 2, 2022. https://www.asc-aqua.org/wp-content/uploads/2017/11/ASC-MSC-Seaweed-Standard.pdf
284. Rees SE, Sheehan EV, Stewart BD, et al. Emerging themes to support ambitious UK marine biodiversity conservation. *Mar Policy*. 2020;117:103864. doi:10.1016/J.MARPOL.2020.103864

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