

REVIEW OPEN ACCESS

A State-Of-The-Art Review of Aquatic eDNA Sampling Technologies and Instrumentation: Advancements, Challenges, and Future Prospects

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ABSTRACT

The field of environmental DNA (eDNA) analysis has revolutionized our ability to detect and monitor biodiversity in aquatic and terrestrial ecosystems. However, traditional eDNA sampling methods often present limitations in terms of temporal and spatial coverage, resulting in a loss of resolution associated with infrequent events or those prohibitive to onsite fieldwork. In recent years, the emergence of autonomous eDNA sampling technology has provided researchers with a powerful tool for collecting high-resolution genetic data, overcoming many of the challenges associated with manual sample acquisition. This review focuses exclusively on eDNA technologies designed for the collection and preservation of water samples, to provide a comprehensive overview of the current landscape of aquatic autonomous eDNA sampling technology and instrumentation. A new era of instrument development and capabilities is emerging; the result of knowledge gained through experience with long-tested marine biological observation instrumentation. Lastly, we highlight current research to develop an in situ eDNA analytical capability, as well as explore the challenges and future prospects associated with this rapidly evolving field.

1 | Introduction

Routine monitoring of biological communities is integral to characterizing ecosystem health, biodiversity, and providing information necessary for public health and resource management (Canonico et al. 2019; Forio and Goethals 2020). Traditionally,

researchers have relied on labor-intensive and invasive techniques, such as netting, trapping, and visual identifications, to identify and quantify species presence. While effective, these conventional methods often present many practical limitations, including constrained spatial coverage, inadequate temporal resolution, disturbance to sensitive habitats and species,

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and an inherent inability to capture the diversity of organisms present in an environment. In addition, the expertise and methodology required to conduct surveys are highly dependent on the organisms in question. In recent years, environmental DNA (eDNA) analysis has emerged as a complementary approach to traditional observational techniques (Rourke et al. 2022; Stat et al. 2017; Wang et al. 2024; Westgaard et al. 2024). This review specifically addresses eDNA sample collection from aquatic environments using technologies intended for the recovery of eDNA obtained from discrete water samples.

eDNA analysis involves the collection and analysis of genetic material shed by organisms into their surrounding environment (Taberlet et al. 2018). Multicellular organisms release DNA and RNA in a myriad of ways, such as by shedding skin cells, scales, mucus, feces, and gametes, all of which can be extracted and sequenced to identify species present within a given ecosystem. This approach offers several advantages over traditional survey techniques, including non-invasiveness, high sensitivity, and capacity to detect rare or elusive species—particularly those that are difficult to observe visually (Gold et al. 2021; Holman et al. 2019; Noble-James et al. 2023). Moreover, eDNA analysis enables comprehensive assessments of biodiversity over large spatial and temporal scales, providing valuable insights into community composition, species richness, and ecological dynamics (Preston et al. 2024; Searcy et al. 2022; Thomsen et al. 2012).

One of the main benefits of aquatic-based 'omics research (broadly referring to the study of DNA, RNA, proteins, and metabolites to characterize biological communities and ecosystem function) is that, despite enormous habitat diversity, the act of collecting samples is relatively straightforward. However, this is also a pivotal challenge to collect an appropriate sample set over relevant temporal and spatial scales. While analysis of eDNA data can be complex, eDNA filtration and preservation is universally accessible. Coverage using conventional eDNA sampling techniques can be relatively comprehensive in readily accessible marine settings (e.g., coastal), but it is also laborious as it typically involves onsite water collection followed by time-consuming sample processing in the lab. In contrast, sampling in remote environments, for example, when the use of crewed ships is required, significantly limits spatial and temporal sampling resolution, hindering the characterization of dynamic aquatic ecosystems. To address these challenges and leverage the ease of eDNA sample collection, autonomous samplers and in situ biomolecular sensors have emerged, offering a paradigm shift in our capacity to understand ecosystem dynamics broadly (Govindarajan et al. 2023; McQuillan and Robidart 2017). These new technologies not only enhance the precision and frequency of data collection but also enable researchers to delve deeper into the molecular mechanisms governing aquatic life in remote locations.

A critical but often overlooked consideration in the development of these technologies is the alignment between the diversity of end-user needs and the diversity of sampling systems. No single technology can meet the demands associated with all applications and use cases; thus, the details of instrument design generally reflect trade-offs between performance requirements, ease of usability, and cost. For instance, real-time detection

capabilities are particularly valuable for event-based sampling, such as during harmful algal blooms or pathogen outbreaks, whereas they are less critical for long-term biodiversity surveys. High-capacity, autonomous sampling systems are better suited for extended deployments or high-frequency data collection but may be excessive for short-term missions such as ROV-based exploration. Likewise, affordability is a key driver for community science and resource-limited monitoring programs, while fit-for-purpose tools may be prioritized by structured research initiative requirements, including long-term ecosystem observation or targeted oceanographic expeditions. Recognizing this diversity of applications, sampling environments, and resulting technological solutions helps to illuminate why no single approach or device is universally optimal and highlights the importance of encouraging the development of a flexible and interoperable instrumentation portfolio.

To that end, this review summarizes information shared during the Marine 'Omics Technology and Instrumentation Workshop, which was held October 10–12, 2023, supplemented with a subsequent literature review to synthesize the state of autonomous eDNA sampling technology and instrumentation. We explore the latest advancements in autonomous sampling instrumentation, including device design and capabilities, but limit this review to automated samplers, without consideration of the parallel expansion of passive eDNA sampling technologies (Bessey et al. 2021). Additionally, we discuss the integration of these sampling devices with various platforms, advanced in situ analytical capabilities, environmental contextual sensors, and imaging technologies, all of which collectively enhance the effectiveness and utility of eDNA sampling systems. Finally, we examine current challenges and opportunities associated with autonomous eDNA sampling, including applications, validation, and standardization, all of which are required for a coordinated and larger scale adoption of eDNA-based observations (Agersnap et al. 2022; Kelly et al. 2024).

2 | Methods

The overarching goal of this review is to comprehensively document insights and current advancements in the field of autonomous 'omics-based sampling instrumentation by integrating expert perspectives with published literature. Information was collected through the Marine 'Omics Technology and Instrumentation (MOTI) workshop (October 10–12, 2023), coordinated personal communication with domain experts, and a structured literature review.

The MOTI workshop, held at the Monterey Bay Aquarium Research Institute, brought together 55 experts, including instrument and technology developers from around the world representing academic as well as non-profit and for-profit entities, to foster collaboration and define roadmaps for next-generation biodiversity observation technologies. The motivation for this paper grew from those interactions. The workshop was structured around keynote presentations, panel discussions, and thematic breakout sessions. The viewpoints in the Challenges and Barriers, Use Case and Application, and Future Perspectives sections of this review stem from consensus reached during workshop-focused discussions. Additional details on the

workshop session structure, questions, and facilitation are provided in the [Supporting Information](#).

Between October 2023 and March 2024, coordinated personal communications with instrument developers and scientific eDNA experts continued via email and virtual meetings. A total of 15 experts were consulted. The selection of these experts was based on their significant contributions to the field, a majority

of whom have co-authored this review. The data for Figures 2 and 3 were provided by the authors of this paper. Upon receipt, the data were reviewed for completeness and consistency, with any discrepancies or missing values addressed through consultation with the authors. While some instruments were developed by this paper's authors and discussed at the workshop, not all instrument developers who participated in the workshop provided information included in this summary; contributions

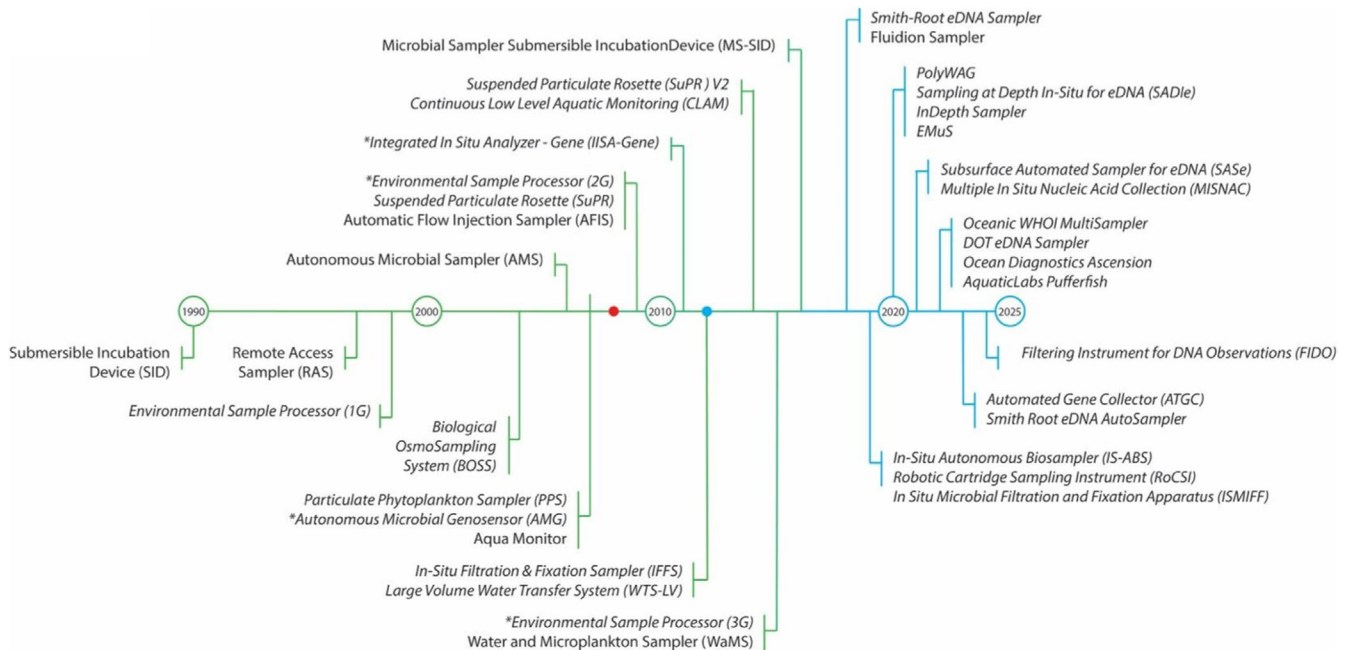


FIGURE 1 | Timeline of Aquatic Sampling Systems and Ecogenomic Sensors Development includes a green section representing the period of marine microbe and phytoplankton development. The blue section marks the period following the incorporation of eDNA in publications, highlighted by a red dot for Ficetola et al. (2008) and a blue dot for Foote et al. (2012). Note that in many cases, the sampling and preservation instrumentation for microbes can be used for eDNA. Italicized instrumentation are those instruments that incorporate filtration, non-italicized instruments are whole water bulk samples and * indicates in situ analytical capabilities.

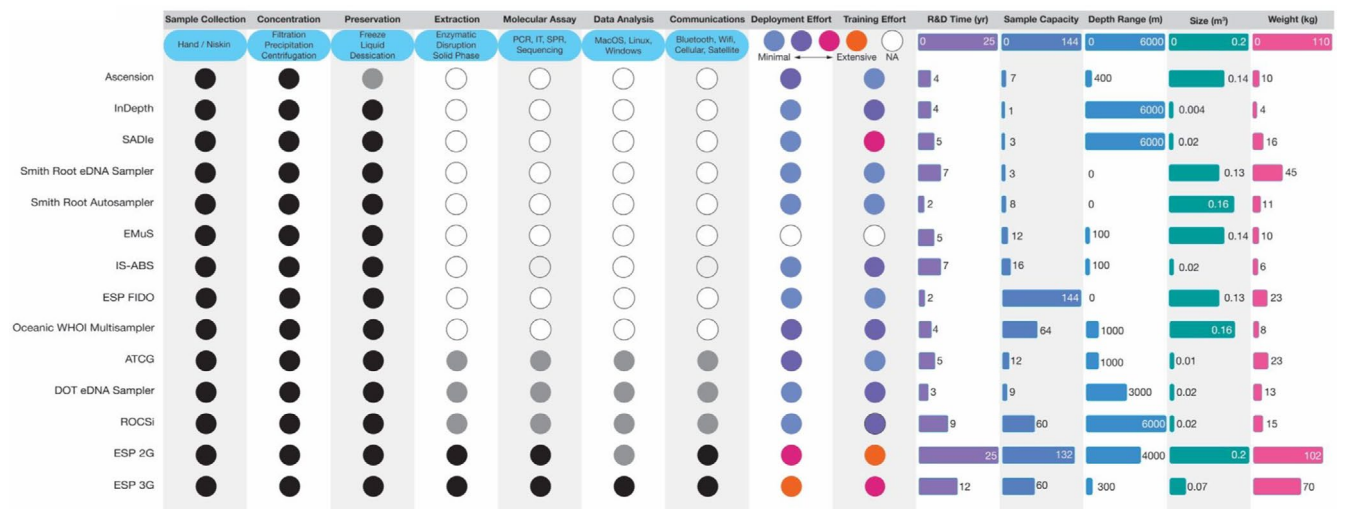


FIGURE 2 | Specifications and ratings of commercially available or in-development aquatic eDNA sampling instruments. The first seven columns indicate traditional laboratory methods with data analysis and communication capabilities: Black circles show existing functionality, gray circles indicate in-development, and open circles denote non-existent functionality. Effort categories are ranked from 1 (minimal effort) to 4 (extensive effort) or NA (open circles); NA icons represent values not available. R&D time represents the number of years in development. Sample capacity is the number of unique samples that can be collected on a single deployment. Depth range is in units of meters, size is in units of m³, weight is in units of kg. A complete list of instrument specifications is available in Table S1.

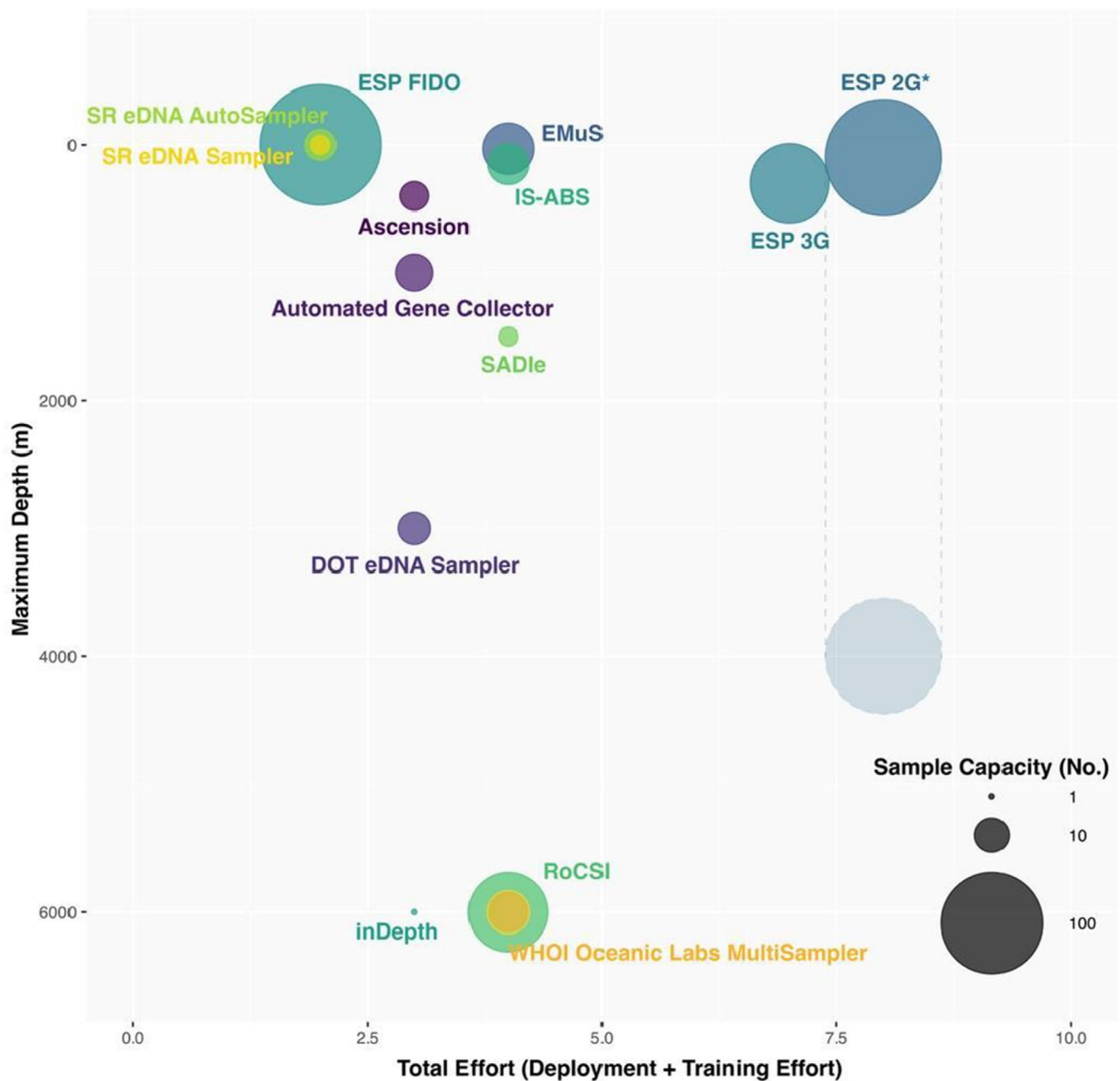


FIGURE 3 | The range of aquatic 'omics-based sampling instrumentation showcasing sampling capacity, required end-user effort (ranking 1 (easy) to 8 (difficult)), and depth capabilities. Maximum depth (m) is allowable deployment depth plotted against the required end user total effort required (based on the sum of training and deployment efforts (see Figure 2 and Table S2)). Bubble size indicates sample capacity (i.e., number of filters). *2nd generation ESP original design depth of 50 m and deployable to 4000 m with deep water sampling module.

to this work were based on the availability and completeness of technical specifications, not on workshop attendance alone. Contributors provided scores related to training and deployment effort based on a specific rubric. Training effort was assessed by the level and duration required to become proficient in operating the instrumentation: a value of 1 indicated less than 1 day of training, while a value of 4 indicated a week or more. Similarly, deployment effort was assessed by the level of effort, time, and equipment needed to deploy an instrument: a value of 1 indicated less than 3 h and minimal equipment, while a value of 4 indicated a week or more with specialized infrastructure (e.g., ship, mooring, etc.). The total effort, ranging from 2 to 8, was

the sum of training and deployment efforts. The complete rubric can be found in Table S2.

A literature review was conducted using the Web of Science database to identify and analyze existing research on autonomous marine molecular biology-based sampling instrumentation. The review focused on articles published up to January 1, 2024, to ensure the inclusion of the most recent and relevant studies. Search terms included "eDNA", "environmental DNA", "molecular biology", "sensors", "biosensors", "autonomous", "in situ", "ecogenomic sensor" and "instrumentation", and combinations of those terms.

3 | Background and History

The advent of autonomous eDNA sampling has been attributed to the convergence of scientific and engineering breakthroughs in environmental genomics and the expansion of autonomous water samplers and in situ incubation devices during the mid- to late 1980s (McQuillan and Robidart 2017; Ottesen 2016). Today's current eDNA sampling instrumentation stems from these pioneering investigations (Figure 1), driven by limitations associated with ship-based sampling and the need for unattended in situ long-term operations. Starting in 1986, Friederich et al. reported the development of a moored water sampler for the collection of 20 chemical or biological water samples at predetermined depths in the ocean (Friederich et al. 1986). This device allowed chemical and biological variability assessments over long periods without concurrent ship-based operations. It was later deployed in the California Current on a Lagrangian drifter to preserve phytoplankton (Abbott et al. 1990). In 1990, the Submersible Incubation Device (SID) established the idea of in situ laboratory experiments in the deep sea, with the ability to return samples to measure biogeochemical fluxes from in situ isotope incubations with the Microbial Sampler—Submersible Incubation Device (MS-SID) (Taylor and Doherty 1990). The SID inspired the development of several other instruments during the late 1990s through the mid-2010s that focused primarily on enabling remote collection and preservation of water samples that were returned to the laboratory for prokaryotic and phytoplankton analyses (Figure 1). These instruments can be categorized into three classes: whole bulk water samplers, filtration samplers, and ecogenomic sensors. The latter is distinguishable from the others as it includes the capacity for sample manipulation beyond initial collection, filtration, and (where applicable) primary preservation.

3.1 | Bulk Water Samplers

A number of devices were designed to collect unfiltered, native water samples for shore-side laboratory analyses, providing bulk (or “whole”) water samples for a variety of laboratory-based analyses. Some instruments only collect native (i.e., “live”) water, whereas others also can add a preservative. Many of these sampling systems were not designed for molecular biological analyses, and a particular drawback of non-preserved whole water sampling systems is that the material collected may not be suitable for molecular analyses (e.g., metatranscriptomics, eRNA) where the target analyte may be compromised through degradation, growth, or alterations in metabolic activities (e.g., mRNA expression). Adding a preservative to bulk water samples can help maintain sample integrity over longer deployments. However, this capability introduces additional fluidic handling complexity and may not be required in cases where target molecules are stable and/or samples can be promptly processed upon recovery.

The earliest bulk water sampler was the Remote Access Sampler (RAS), which was the first demonstration of unattended moored sampling operations in the ocean, designed to study planktonic diatoms (McKinney et al. 1997). In 2012, Feike et al. reported the development of the Automatic Flow Injection Sampler (AFIS), allowing the collection of a single whole water sample

in a tethered deployment configuration with in situ preservation via mixing with a concentrated preservative. The AFIS was validated for microbial metatranscriptomics, revealing a 30-fold increase in ammonia monooxygenase transcription compared to standard oceanographic sampling methods (Feike et al. 2012). The AFIS was subsequently upgraded to AFISsys to increase sample capacity to six samples. More recently, the Water and Microplankton Sampler (WaMS) was designed to enhance the capabilities of the Continuous Plankton Recorder (CPR) by autonomously acquiring whole water samples downstream of the CPR at predetermined intervals for later DNA analyses, first demonstrated with 18S rDNA metabarcoding (Stern et al. 2015).

In the mid-2000's, a few development efforts focused on incorporating water sampling systems on fully autonomous platforms with the goal of improving spatial and temporal coverage while reducing overall collection expense and effort. While mainly developed for biogeochemical measurements, these instruments also provided viable material for shoreside filtration and 'omics-based analyses. For example, the Gulper, a unique AUV payload system, was originally designed to collect up to 10 discrete water samples (1.8 L each) per deployment (Bird et al. 2007). Collections can be triggered adaptively based on the AUV's underway environmental contextual sensors (e.g., CTD, chlorophyll) or at preset locations and depths. The system has been used in tandem with multiple AUVs for coordinated and adaptive environmental sampling of zooplankton (Harvey et al. 2012). Green Eyes Environmental Observation Systems developed the AquaMonitor for deployment on moorings and autonomous vehicles with a capacity to collect 47 1-L samples with an option of dosing the water acquired with preservatives such as Lugol's or mercuric chloride (GreenEyes LLC 2025). FluidIon's sampler systems (surface and deep samplers) were designed to autonomously capture up to 26 whole water samples (100 mL) without a preservative. Those devices can be deployed in a number of configurations, including moorings to autonomous vehicles (FluidIon 2025). While collecting bulk water samples expands downstream analytical opportunities (e.g., measuring nutrients, trace metals, etc. from the same sample as 'omics analyses), it also requires moving and containing discrete large volumes of water which limits the total number of samples collected based on spatial and transport constraints.

3.2 | Filtration Samplers

By filtering water in situ and returning the concentrated material, it becomes much easier to acquire larger numbers of samples during a single instrument deployment. Similar to bulk water samples, instruments may or may not have a preservation mode and can vary in deployment platforms (i.e., fixed versus on a mobile vehicle). The deployment platform may determine whether the collected sample is time-integrated (at a fixed location) or time-and-space integrated (if on a moving vehicle). The combination of the collection system and platform also determines whether water is essentially a “grab sample” collected very quickly or time-integrated with filtration occurring over longer time periods. The majority of deployable sample filtration instruments were initially designed to return particulates to the laboratory for downstream biological analyses that were not feasible or practical to conduct in situ.

A variety of in situ water filtration instruments have been developed, showcasing the diversity of this technology and its applications in aquatic and biological observations. Taylor et al. (2006) devised the Autonomous Microbial Sampler (AMS) which collected 6 independent filtered or unfiltered samples from remotely operated vehicles, autonomous underwater vehicles, or crewed ships. This device proved the possibility of remotely capturing uncontaminated microbial samples from marine environments, specifically in the deep sea and in extreme environments (e.g., hydrothermal vents; Taylor et al. 2006). Similarly, the Particulate and Phytoplankton Sampler (PPS) is an autonomous filtration and preservation sampler coupled to a physical and chemical sensor system. The PPS can collect 24 individual water samples on 47 mm membrane filters and can be deployed for extended periods (14 months). Sequencing of 16S rRNA and rDNA revealed differences between ship-based collections and those conducted in situ with the PPS (Torres-Beltrán et al. 2019), highlighting the need to establish standardized and reproducible techniques for validating the performance of newly developed automated samplers in light of well-established manual (human-in-the-loop) “gold standards”. Likewise, the In situ Filtration and Fixation sampler (IFFS) was designed to avoid potential biases in studies of microbial transcriptomics that are introduced when using traditional Niskin bottle sampling followed by manual shipboard processing (Wurzbacher et al. 2012). The IFFS revealed the importance of in situ sampling and preservation to obtain metatranscriptomic profiles reflective of prevailing environmental conditions.

Long interval, time integrated sampling devices have been devised for investigations of dynamic and changing aquatic environments. For example, the Biological OsmoSampler System (BOSS) was built to continuously collect and preserve fluid samples over long temporal periods from a fixed location (Jannasch et al., 2004). It was successfully deployed to characterize the microbial diversity, protein expression, and geochemistry at deep sea hydrothermal vents (Robidart et al. 2013). A unique feature of the BOSS is that it uses an osmotic pump that does not require any electrical power. However, its relatively slow sampling rate of < 5 mL/day limits its applications. Another time integrated sampling system known as the Continuous Low-Level Aquatic Monitoring (CLAM) device was originally developed for capturing chemical pollutants through solid phase adsorption media (Ensminger et al. 2017), and has since been adapted for eDNA sampling (Aqualytical 2025). The CLAM allows for single sample filtration of large volumes (20–100 L) over 48 h in submerged fixed or drifting configurations.

To comprehensively characterize low particulate density in open ocean systems, large sample volumes (> 5 L) are often required. For example, the Suspended Particulate Rosette (SuPR) collects 30–100 L water samples on moorings and CTD rosettes for in situ optical analysis (Breier et al. 2014). The upgraded SuPR version 2 (v2) includes sample preservation capabilities and can be deployed on various platforms, such as ROVs and AUVs (Govindarajan et al. 2023, 2015). Both SuPR and SuPR v2 have been used to characterize microbial (Sheik et al. 2015) and mesozooplankton communities (Govindarajan et al. 2015). The design principles of SuPR were incorporated into *Clio*, a

large-volume autonomous sampling vehicle, which has enabled genomic and proteomic measurement across large geographical and depth scales (6000 m; Breier et al. 2020). The Large Volume Water Transfer System (WTS-LV) collects 2500 to 36,000 L of seawater by filtering through 142 mm membrane filters at flow rates of 0.5 to 30 L/min (Morrison et al. 2000). It can be autonomously deployed from ships to depths of 5500 m, capturing depth-resolved samples of suspended and dissolved particulates. The In Situ Microbial Filtration and Fixation (ISMIFF) device collects, filters, and preserves large-volume microbial samples at depths over 6000 m. It uses a 142 mm membrane filter, followed by liquid preservation and has been deployed on a hadal lander (Wang et al. 2019). When the ISMIFF was compared to Niskin samples collected in parallel, 16S rRNA amplicon sequencing revealed differences in community profiles (Wang et al. 2019). Similar to the Wurzbacher et al. (2012) findings, these results reveal the importance of applying in situ filtration and preservation as opposed to removing live organisms from their native environment and subjecting them to time-delay and disturbance before processing (Wang et al. 2019; Wurzbacher et al. 2012). While these instruments are well-suited for capturing low-abundance targets in remote or deep-sea environments, their size, power requirements, and deployment logistics may present challenges for routine or resource-limited applications.

Building on lessons learned with ISMIFF, the Multiple In Situ Nucleic Acid Collection (MISNAC) instrument was developed for the collection, extraction, and preservation of nucleic acids in the deep sea (3000 m depth rating) (Wei et al. 2020). This instrument enables the in situ extraction and stabilization of RNA and DNA from marine microorganisms, ensuring minimal degradation and preserving the integrity of the genetic material for subsequent analyses. The MISNAC is capable of filtering and extracting nucleic acids from 9 discrete samples, preserving the extracts on polymer columns. The performance of the MISNAC was compared to ISMIFF using 16S rRNA metabarcoding, revealing similar microbial communities (Wei et al. 2020). Moreover, for metatranscriptomics, the MISNAC showed higher recovery of 16S rRNA fragments than traditional Niskin-based samples. The MISNAC is an example of a device that highlights the potential for in situ laboratories in the deep ocean to study genetic and functional diversity in situ.

3.3 | Ecogenomic Sensors

The most complex instruments in this category can not only collect and filter water but also conduct in situ analysis in near real-time. However, these systems are typically limited to research-focused deployments due to their operational complexity, cost, and the need for specialized operations personnel. This class of device was dubbed “ecogenomic sensor” as first captured in an illustration by Hunter Hadaway at the University of Washington (Hadaway 2010), providing a conceptual representation of a device that integrates genetic-level sensing with larger-scale environmental characterization, providing a holistic view of the marine environment (Scholin et al. 2018). Hadaway's illustration captured what a number of investigators had been working on at that time—a device that not only collected material but also performed molecular analytical analyses in situ. The Autonomous Microbial Geosensor (AMG; Fries

and Paul 2003), the Environmental Sample Processor (ESP; e.g., Goffredi et al. 2006), and the Integrated In situ Analyzer-Gene (IISA-Gene; Fukuba and Fujii 2021) were among the first autonomous systems to attempt such analyses remotely in a variety of ocean settings.

The AMG was built to automate sample collection, filtration, RNA extraction, and in situ RNA analysis through Nucleic Acid Sequence Based Amplification (NASBA; Fries and Paul 2003). The AMG had the capability of performing 12 in situ analyses, where purified RNA is extracted in situ and detected using real-time NASBA chemistry. This was the first system to demonstrate microfluidic-based techniques for in situ detection and quantification of mRNA associated with *Karenia brevis*, a harmful algal species.

The ESP is similar to the AMG. It is an autonomous robotic device for carrying out sample filtration, homogenization, and conducting a variety of molecular analytical analyses in situ (Scholin et al. 2018). The ESP was the first device to enable real-time species detection using rRNA-targeted DNA probe arrays, cell metabolite (phycotoxin) detection using protein arrays (Doucette et al. 2009), as well as sample preservation for post-deployment analyses (Greenfield et al. 2008; Ottesen et al. 2011). Iterations of the ESP introduced the concept of instrument modularization, where specialized sampling and analytical functions can be added to or removed from the “core” ESP. One versatile example is the utilization of a separate microfluidic block to enable qPCR capabilities remotely, in situ for detection of a variety of microorganisms (Hansen et al. 2020; Preston et al. 2011; Ussler et al. 2013; Yamahara et al. 2015). Similarly, the deep-water sampling module made it possible to deploy the core ESP in the deep sea onboard an ROV as well as a standalone benthic elevator (Pargett et al. 2013). The development and application of the ESP are further explored in proceeding sections.

The IISA-Gene is an in situ molecular biological analyzer utilizing PCR-based analyses, in particular for ocean waters (Fukuba et al. 2011). This device utilizes microfluidic lab-on-chip architecture for fluidic manipulation and handling, performs sample lysis, DNA purification, PCR amplification, and optical detection. The system has been deployed at deep-sea hydrothermal vents on-board the ROV HYPER-DOLPHIN and successfully detected 16S rRNA genes, demonstrating the feasibility of a functional lab-on-chip device in the deep sea (Fukuba et al. 2011).

These early instruments span a range of sample collection, filtration, and in situ analytical use cases, resulting in multiple research papers; yet their main application to date rests primarily in a research setting. Only a small number of these early devices are commercially available (mostly from McLane Research Labs), including the RAS, PPS, SuPR, WTS-LV, and ESP (2nd generation); the rest have never been brought to market. This exemplifies a common challenge with in situ sampling and sample processing instruments. Most are developed for highly specialized use cases that offer limited market potential (e.g., deep sea, hydrothermal vent research). Further barriers for adoption in a commercial setting include significant developmental and operational costs, limited sample

capacity, reliability, or fidelity; and/or they are too technically demanding for an average technician to routinely deploy and operate.

4 | A New Era of eDNA Sampling Instrumentation

Advances in eDNA methods, techniques, and applications, along with an increasing market pull, have driven a renewed interest in the development of semi- to fully autonomous sampling systems (Figure 2). Similar to their earlier counterparts, these instruments span a range of sample capacity, depth rating, and respond to a variety of end user operational requirements (Figure 3). Over the last 10 years, a suite of instruments has been developed to parallel eDNA “front-end” sample handling techniques, utilizing filtration and in some instances nucleic acid preservation for subsequent laboratory-based molecular analyses (e.g., PCR, sequencing). These field-deployable instruments range from human-in-the-loop operated to fully autonomous devices that are deployable on uncrewed systems (UxS). Though the previous instruments could be *adapted* specifically for eDNA sampling use, the instruments reviewed in this section were designed specifically for eDNA sampling and ‘omics-based laboratory analyses and can be categorized into the following groups: terrestrial/shipboard (non-submersible), semi-autonomous Niskin-style (e.g., manually deployed), and fully autonomous. From the previous section, we include the ESP given the extensive use of these instruments specifically for eDNA sampling on a variety of fixed as well as crewed and uncrewed platforms (Den Uyl et al. 2022; Hansen et al. 2020; Preston et al. 2024; Sepulveda et al. 2020; Truelove et al. 2019; Yamahara et al. 2019; Zhang, Kieft, et al. 2021).

4.1 | Terrestrial and Shipboard Instrumentation (Non-Submersible)

To increase the efficiency of eDNA sample collections in the field, several sampling instruments have been developed to automate aquatic eDNA collections from terrestrial and shipboard environments. This group includes instruments that are not designed for submerged operations; the goal being to aid field technicians in sample collection and filtration of lentic and lotic systems, as well as to facilitate shipboard collections.

4.1.1 | Smith Root eDNA Sampler and Autosampler—Smith-Root (USA)

The SR-eDNA sampler, developed by Smith-Root, was the pioneering instrument specifically designed to assist field personnel in the precise collection and filtration of eDNA using specialized self-preserving filtration cartridges (Thomas et al. 2018, 2019). This system employs vacuum-based filtration using custom, single-use 47 mm self-preserving filter housings. It is controlled by a simple graphical user interface for programming and operation. Of note is its “trident” configuration, allowing for the simultaneous acquisition of three replicate samples. Recently, the eDNA Autosampler, an automated iteration of the SR-eDNA sampler, was introduced for

deployable, non-submersible instruments dedicated to regular eDNA monitoring. Utilizing the same self-preserving filter housings as its non-automated counterpart, this system can be pre-programmed to collect up to 8 eDNA samples via a graphical user interface (George et al. 2024). Deployment of this device enables high-frequency eDNA collection, revealing temporal variability and environmental associations with comparable or greater yields relative to what manual sampling affords (George et al. 2024).

Originally designed for terrestrial settings, these instruments are increasingly being adopted for use in marine environments due to their simple and effective sample filtration and preservation capabilities. For instance, the SR-eDNA sampler and autosampler have been employed aboard ships for direct processing of samples from Niskin bottles (NOAA Fisheries 2025); on ships of opportunity facilitating sample collection from underway water sources; and for dockside use. Other advantages include the reduced risk of contamination and reduced cleaning between deployments given the design of in-line sampling (for the non-auto sampler) where the filter is upstream of any reused tubing or parts, there is no separate reservoir of reagent(s) to manage, and the self-preservation method does not require handling the filter while operating in a field setting.

4.2 | Semi-Autonomous Niskin Style Instruments

The need for semi-autonomous Niskin-style instruments for environmental DNA (eDNA) collections arises from the growing demand for efficient and reliable shipboard sampling methods with collections at discrete depths. Here we define this class of instruments as semi-autonomous, as the instruments are generally deployed tethered and under end-user command and control. These instruments combine the precision of traditional Niskin bottle casts with automated features, reducing the labor and time required for sampling while ensuring consistent sample quality. Their ease of use and portability greatly expand the capacity for citizen science and the variety of crewed vessel operations. They are particularly valuable for eDNA studies due to their ability to collect water samples at multiple depths with minimal human intervention. This feature is crucial for capturing a more comprehensive snapshot of biodiversity in aquatic environments, where species distribution can vary with depth.

4.2.1 | Ascension—Ocean Diagnostics (CAN)

The Ocean Diagnostics Ascension eDNA sampler is engineered for straightforward deployment and user-friendliness, featuring a compact, lightweight device akin to a Niskin rosette (Ocean Diagnostics 2024). This design enables end-users to lower the Ascension sampler to multiple depths down to 400m, allowing for the collection and in situ filtration of up to 7 samples, most commonly using 47mm diameter disc filters. Notably, the Ascension sampler's advantage lies in its size and portability (i.e., hand-deployable by a single person), eliminating the need for costly hardware typically associated with traditional Niskin rosette sampling from ships (Missen and Ayad 2025).

The deployed system captures real-time environmental data (depth, salinity, temperature), providing contextual information alongside eDNA samples through a computer interface. Additionally, Ascension can be programmed ahead of time and deployed in an autonomous configuration on a mooring or in other stationary deployment configurations.

4.2.2 | Sampling at Depth in Situ for eDNA (SADIE)—NOAA (USA)

The SADIE instrument arose from a collaborative effort between the National Oceanic and Atmospheric Administration (NOAA) Northwest Fisheries Science Center (NWFSC) and University of Washington Applied Physics Laboratory. It is designed to enhance the efficiency of conventional Niskin-based sampling during NOAA Fisheries Survey Cruises while reducing the resources associated with post-collection filtration of thousands of samples per cruise. A programmable instrument capable of filtering and preserving 3 samples on 47mm diameter filters, SADIE is primarily deployed on CTD rosettes from ships; however, it can also be independently deployed to depths of 1500m (Parsons et al. 2022).

4.3 | Fully Autonomous Deployable Instruments

Many groups have focused on developing fully automated submersible 'omics-based sampling instruments. These instruments can collect, filter, and preserve samples at precise locations and depths without any human intervention, enhancing the efficiency and scope of 'omics-based research. These devices offer the means to reach hard-to-access areas and over expanded space and time domains compared to the systems summarized above, providing a scalable capability for achieving a persistent monitoring presence in aquatic environments. Looking to the future, this class of instrument offers a means for advancing our understanding of aquatic biodiversity and informing effective public health and resource management strategies for applications that require having an extended, "hands-off" presence in environments where labor and logistical constraints hamper routine sample collections.

4.3.1 | inDEPTH—Applied Genomics (UK)

The Applied Genomics inDEPTH sampler is a compact, programmable device capable of filtering a single large volume sample (up to 50L) on custom filtration cartridges to depths up to 6000m (Applied Genomics Ltd 2024). It features programmable sampling duration to collect time integrated samples from hours to days. Samples collected with the inDEPTH sampler have been utilized for eDNA monitoring of inshore fish populations utilizing metabarcoding of mitochondrial cytochrome B (cytB) (Marsh 2020; Mynott and Marsh 2020). These results suggest the potential for detecting changes in fish communities over large spatial scales with consistent temporal sampling. The inDEPTH instrumentation is currently available through Applied Genomics as a service, which provides instrumentation rental and deployment along with sample processing, analytical, and data solutions.

4.3.2 | EMuS and Quokka—CSIRO (AUS)

The EMuS and Quokka are automated eDNA sampling instruments under development by the Australian National Science Agency, the Commonwealth Scientific and Industrial Research Organization (CSIRO). The Quokka is designed for ship-board sampling, and the EMuS for in-water sampling (P. Craw, personal communication, November 2023). Both instruments automate sample filtration and preservation of 24 samples. The “EMuS vessel” version utilizes flexible filter connections capable of accepting a range of filter types, including Sterivex or 47 mm filter housings, while “EMuS” is restricted to Sterivex filters only due to instrument size constraints. The Quokka is designed to automate eDNA sampling and preservation from a vessel’s underway water system to ease the collection and staffing needs for eDNA collections aboard ships. EMuS was designed for in-water sampling to a depth of 20 m and has been deployed on moorings and above-water infrastructure (P. Craw, personal communication, November 2023).

4.3.3 | RoCSI—NOC/McLane Labs (UK/US)

The Robotic Cartridge Sampling Instrument (RoCSI) developed at the National Oceanography Centre (NOC) is commercially available through McLane Research Labs. It filters and preserves up to 48 samples on a continuous bandolier of cartridges. Technically, the number of samples is only restricted by the storage space (J. Robidart, personal communication, November 2023). The instrument utilizes Sterivex filters to which a liquid preservative is applied upstream of a return for shore-side interrogation. It also includes an optional programmable dilute-bleach decontamination step to reduce sample-to-sample contamination. The RoCSI is rated to 6000 m and has been deployed on the ROV Isis (The National Oceanography Centre 2023), the Autosub 6000 and Autosub Long Range AUVs, and utilized to sample underway flowing seawater from research vessels (Huvenne 2024). The instrument has been utilized to characterize N₂-fixing communities in the North Atlantic Ocean, where 1.5 to 4 L water samples were collected and preserved for laboratory qPCR and high-throughput sequencing of *nifH*, 16S, and 18S rRNA genes (Tang et al. 2020). qPCR-derived *nifH* quantifications in this study were consistent between RoCSI- and manually-collected samples filtered with a peristaltic pump.

4.3.4 | ATGC—JAMSTEC (JPN)

The Japan Agency for Marine-Earth Science and Technology (JAMSTEC) has developed the Automated Gene Collector (ATGC) to enhance the efficiency of eDNA sample collection from aquatic environments. This instrument, characterized by its compact design, utilizes 3D printing technology to integrate 12 Sterivex filters (Fukuba et al. 2022). It employs a valveless piston pump coupled with a stepper motor, facilitating precise filtration volumes and improved operational reliability compared to earlier prototypes. Filter preservation is achieved using liquid-based preservatives such as RNAlater, while decontamination between samples is achieved by using a cleaning protocol based on sterilized seawater. The modular design allows for scalability

in sample capacity and adaptability to various filter cartridge types. Fukuba et al. (2022) evaluated the ATGC deployed on a buoy and achieved a 90% DNA recovery as compared to traditional manual sampling methods. The ATGC development team is currently extending their advancements to include an in situ eDNA extraction and qPCR analysis system based on the ATGC architecture (T. Fukuba, personal communication, November 2023). The improved version of this device is scheduled to be commercially available from OceanFluidics Co. Ltd. in 2025.

4.3.5 | eDNA Sampler—Dartmouth Ocean Technologies (CAN)

Dartmouth Ocean Technologies Inc. (DOT) and Dalhousie University have collaborated and developed a single-person-portable eDNA Sampler that features a modular design. This device utilizes 3 independent and detachable sections that house the filtration cartridges, electronics, and fluidic storage. The DOT eDNA Sampler is compact in design and can collect up to 9 samples using 25 mm filter housings (Hendricks et al. 2023). This device provides preservation and decontamination capabilities in the form of liquid reagents (e.g., RNAlater and 5% Hydrochloric Acid). The modular design of the filter cartridge housing allows for quick servicing and resupply. Further, the modularity of the DOT design enables different filters to be adapted to the sampler fluid automation section with a custom cartridge or filter holder. For instance, the DOT and NatureMetrics (NM) version of the sampler houses 9 of the NM 50 mm diameter filters for increased volume filtering capabilities (5 L) (Luy et al. 2024). All versions of the DOT eDNA sampler are available in 3 depth configurations for 20 m, 200 m, and 3000 m. Field tests of the instrumentation alongside Niskin sampling and manual filtration methods revealed the DOT eDNA sampler recovered similar communities and counts of 16S rRNA amplicon sequence variants (ASVs) (Hendricks et al. 2023). Deployment of the DOT sampler for over 3 months showed effective DNA preservation, demonstrating the DOT sampler is suitable for long-term deployments (Van Wyngaarden et al. 2024).

4.3.6 | Oceanics-WHOI MultiSampler—Woods Hole Oceanographic Institution (WHOI) and Oceanic Labs (USA)

The Oceanics-WHOI eDNA sampling instrument is designed for the collection of eDNA samples from marine environments, particularly in the deep sea. This instrument can collect up to 16 samples per unit utilizing either Pall Mini Kleenpak filters or Waterra filter cartridges with a maximum flow rate of ~2 L/min (A. Govindarajan, personal communication, November 2023). Filtration time is adjustable with no maximum limit but is typically set at 15–20 min. Depending on deployment configuration, up to 3 sampler units can be potentially integrated for a maximum of 48 samples (Govindarajan et al. 2023). The sampling system is depth-rated to 1000 m and has been deployed on a number of oceanographic platforms, such as the Deep-See towed vehicle, the AUV *Mesobot*, and ROVs *SuBastion* and *Hercules*, and a CTD rosette (Govindarajan et al. 2023; Wagner 2023). The original version of this instrument was deployed alongside traditional Niskin sampling to assess the recovery of 18S rRNA

metabarcoding ASVs, revealing the importance of acquiring large volume water samples in the mesopelagic and deep sea (Govindarajan et al. 2022). During recent *Mesobot* expeditions, sampling was initiated via communications from the autonomous surface vehicle *DriX* (Hayden et al. 2023; Mayer and Schmidt 2023).

4.3.7 | Biosampler IS-ABS—INSEC TEC (PRT)

The Biosampler IS-ABS was developed through the Institute for Systems and Computer Engineering, Technology and Science (INSEC TEC) and the Interdisciplinary Centre of Marine and Environmental Research (CIIMAR), and represents an updated version of the In situ Automatic Bio-sampler System (IS-ABS) (Carneiro et al. 2023; Ribeiro et al. 2019). The instrument was specifically designed for incorporation into autonomous vehicle platforms with a depth rating of 150 m. It utilizes Sterivex filters to collect up to 16 samples and incorporates a liquid preservation protocol for the stabilization of nucleic acids. The original IS-ABS unit was validated using parallel laboratory vacuum filtrations, followed by 16S and 18S rRNA metabarcoding. The validation results revealed the IS-ABS filtered the same (3 L) volume of water twice as fast as conventional filtration methods and provided equivalent recoveries of DNA. Additionally, 16S and 18S rRNA metabarcoding revealed that the IS-ABS provides similar community and taxonomic composition for both prokaryote and eukaryote populations (Ribeiro et al. 2019). The Bio-sampler IS-ABS has recently been deployed aboard the AUV IRIS for collection of eDNA samples in remote Arctic waters (INSEC Brussels 2024; Martins et al. 2020). Currently, the instrument has a depth rating of 150 m, with a 1000 m depth-rating design in progress (A. Martins, personal communication, January 2024).

4.3.8 | Environmental Sample Processor 2nd, 3rd Generations and Filtering Instrument for DNA Observation (FIDO)—MBARI (USA)

The Environmental Sample Processor (ESP) is one of the pioneering instruments that helped to establish the feasibility of autonomous in situ sampling and real-time molecular analytical sensing. Developed by MBARI, the ESP research and development program began in the early 1990s to provide real-time detection of microorganisms and biological toxins, addressing the need for a persistent presence in the ocean capable of near real-time analyte analyses and data dissemination (Scholin et al. 2018). Currently, three versions of the ESP are in operation: the second and third generations, and the newly devised Filtering Instrument for DNA Observations (FIDO) eDNA-specific sampler (J. Birch, personal communication, November 2023). All three instruments are completely autonomous and can be controlled and transmit data via cellular, WiFi, and satellite communications.

The second-generation ESP's (2G ESP) electromechanical fluidic system allows users to collect and filter water samples, either preserving them for later analysis (e.g., Ottesen et al. 2014, 2013, 2011) or immediately applying molecular detection technologies such as quantitative PCR (qPCR) (Preston et al. 2011; Robidart

et al. 2014; Ussler et al. 2013; Yamahara et al. 2015), rRNA sandwich hybridization (Greenfield et al. 2008; Jones et al. 2008; Preston et al. 2009), or competitive ELISA (Doucette et al. 2009; Ritzenthaler et al. 2016) to identify particular microorganisms or genes and associated toxins (metabolites). The 2G ESP utilizes custom filtration housings (pucks) to collect either 132 (standard puck) or 198 (short puck) samples on 25 mm filters (NOAA NCCOS 2019). Sample numbers are reduced for in situ analyses, as the filtration housings are used as reaction chambers for lysis and processing analytical DNA and protein probe arrays. The 2G ESP has been successfully deployed in diverse marine environments, from coastal waters to open ocean to deep-sea seeps and hydrothermal vents (Scholin et al. 2018). More recently, the 2G ESP has been utilized in freshwater environments for monitoring invasive and threatened species (Jones, Clements, and Sepulveda 2024; Searcy et al. 2022; Sepulveda et al. 2020), as well as cyanobacterial blooms in the Great Lakes (Ritzenthaler et al. 2016). The 2G ESP's continuous, high-resolution monitoring capabilities have contributed valuable data for basic research as well as applied public health and resource management initiatives.

Building on these successes, the 3G ESP incorporates a reduction in size with its development oriented towards use onboard autonomous platforms for mobile operations (Pargett et al. 2015). The 3G ESP operates based on a cartridge and toroidal ring design that allows for the collection and processing of 60 samples with a depth rating of 300 m. Two different cartridge configurations allow either sample preservation or in situ lysis and downstream analytical processing. The instrumentation has been validated for the collection and preservation of samples from marine systems using qPCR for prokaryotes and eukaryotes (Yamahara et al. 2019) and metabarcoding of rRNA 12S and 16S, mitochondrial CO1, and nuclear 18S rRNA (Truelove et al. 2022), as well as from freshwater systems (Den Uyl et al. 2022). The 3G ESP has been deployed on a number of uncrewed platforms (UxS) including MBARI's Long Range Autonomous Underwater Vehicle (LRAUV; Den Uyl et al. 2022; Truelove et al. 2022; Yamahara et al. 2019; Zhang, Ryan, et al. 2021), Saildrone's Surveyor Autonomous Surface Vehicle (ASV; Preston et al. 2024), and the SeaTrac ASV (NOAA NCCOS 2024). A mobile capability that allows for event response affords opportunities for targeting sample collections in dynamic environments over large spatial areas and extended periods of time. Additionally, the 3G ESP functions as an ecogenomic sensor with near real-time in situ analysis of microcystin toxins in Lake Erie (Ussler III et al. 2024) as well as domoic acid in marine settings (B. Ussler, personal communication, November 2023, (NOAA NCCOS 2023)).

While not the original intent of the device, the 2G ESP has been used to successfully demonstrate the utility of high frequency, long duration automated eDNA sampling in freshwater systems (Searcy et al. 2022; Sepulveda et al. 2020). Although effective for demonstration purposes, that device enables more analytical capabilities and meets strict requirements for ocean deployments that are not required for these specific freshwater use cases (e.g., shore-side eDNA preservation). The experience gained pointed to a need for simplified and cost effective eDNA sampling instrumentation that led to the development of the Filtration Instrument for DNA Observations (FIDO) instrument. FIDO is currently in development for use with the US Geological

Survey's Early Detection and Rapid Response framework (Jones, Augustine, et al. 2024) and is tailored for shore-based lentic and lotic deployments. FIDO's core design objectives include high-capacity sampling, full autonomy, and user-friendly functionality. FIDO is engineered to procure 144 samples through 47mm user-defined filters with liquid preservation (J. Birch, personal communication, July 2024). To facilitate easy access and communication with the instrument, a web-based graphical user interface (GUI) has been crafted, utilizing WiFi, cellular, and satellite connectivity for remote command and control. FIDO has also recently been deployed onboard a crewed vessel for sample collection using the ships flow-through seawater system (J. Birch, personal communication, May, 2025).

5 | Other Samplers and Ecogenomic Sensors

There has been a surge in the development of new deployable 'omics instrumentation in recent years, with some presented only online at the time of this publication, with limited details and technical performance specifications. Efforts to develop these instruments are duly acknowledged, and their successful demonstration in the future is eagerly anticipated. The most advanced of these instruments is the Nucleic Sensing Systems Tracker (NS2), which offers real-time eDNA analytical capabilities by targeting "free-eDNA" (Nucleic Sensing Systems 2024). Aquatic Labs has developed the Pufferfish instrument family for eDNA sample collections. The Pufferfish is advertised as an easy-to-use, compact, and adaptive system for the collection and preservation of up to 192 eDNA samples (Aquatic Labs 2024), and has been deployed on ROVs and profilers (Govindarajan et al. 2025). Through Citizens of the Sea, theDNAutic is a citizen science sampler that has been developed for eDNA collection on racing yachts in conjunction with Sequench Ltd. (Citizens of the Sea 2025). As eDNA methods become more integral to routine monitoring and resource management, and the demand for eDNA collection and analysis increases on a commercial scale, new samplers and ecogenomic sensor systems are sure to arise.

6 | Emerging Analytical Capabilities

The majority of the recent engineering efforts have focused on the "front-end" of eDNA workflows (i.e., sampling, filtration and preservation), with the return of samples for subsequent laboratory-based processing. There are only a few instruments that have achieved in situ, real-time analytical capability as chronicled in peer-reviewed literature (ESP, AMG, and IISA-Gene). These devices vary in complexity and cost depending on the specific use case, technical requirements, and the contextual data needed for deployment. While some systems may have higher operational costs, they should be evaluated in comparison to manual alternatives, including the requirements for accessing sample locations, on a per-sample basis to assess their overall cost-effectiveness given specific use cases. Purpose-built devices designed for particular applications can optimize this balance—some use cases may never allow for extremely low-cost solutions, while others may benefit from significant cost reductions. Advancements in technologies such as microfluidic systems and material science and manufacturing are enabling the development of new in situ analytical systems at smaller

scales with the potential for mass production. For example, nanopore sequencing, CRISPR-based detection systems, digital PCR, and isothermal techniques offer opportunities for enhancing the sensitivity, specificity, and efficiency of eDNA detection both in a laboratory and remotely in situ. Although these analytical technologies show great promise, many remain in prototype or early deployment stages. Significant miniaturization, numerous engineering refinements, and extensive validation experiments are needed before they become broadly accessible for routine field use, especially in the context of fully autonomous systems capable of extended field deployments.

Although technologically challenging, the use of qPCR to detect a variety of targets in situ, in near real-time, was proven feasible roughly 15 years ago using the 2nd generation ESP (Preston et al. 2011). This device utilized a chaotrope-based sample lysis protocol that provided input material for both sandwich hybridization DNA probe arrays for detecting specific rRNA targets as well as for qPCR. For the latter, a portion of the lysate was passed to a specialized fluidic system that purified DNA and then manipulated microliter quantities of extracted DNA and reagents (primer/probe, polymerase) in order to perform qPCR assays serially using a custom-built thermal cycler and optical detection system (Preston et al. 2011). This low-power system was capable of conducting up to six assays per sample but was technically difficult to set up and operate.

More recently, biotechnology innovations are renewing interest in PCR-based devices by coupling lab-on-a-chip devices with point-of-care medical diagnostics. Lab-on-a-chip (LOC) technologies are enabling the development of miniaturized PCR instrumentation, such as the IISA-gene instrument, which has been successfully deployed on an ROV to amplify 16S rRNA genes at deep sea hydrothermal vents (Fukuba and Fujii 2021). Prototypes of microfluidic qPCR instruments currently in development include JAMSTEC's eDNA analyzer (T. Fukuba, personal communication, November 2023) and Dartmouth Ocean Technology in situ qPCR module (Sonnichsen et al. 2024). The eDNA analyzer capitalizes on LOC microfluidics and advanced Peltier cooling to analyze up to 12 samples. DOT's eDNA qPCR module is being developed for incorporation into its eDNA sampler and features amplification via 9 qPCR chambers on a lab-on-chip, housed in an A-sized sonobuoy form factor submersible instrument (i.e., a cylindrical canister approximately 4.875 in. in diameter and 36 in. in length; V. Sieben, personal communication, November 2023). Digital PCR is an emerging technology that uses microliter-scale reaction volumes, provides quantitative results without standard curves, and is compatible with lab-on-a-chip devices (Xu et al. 2023) consistent with ecogenomic sensor applications (Hatch et al. 2014).

Portable nanopore sequencing has been successfully used to sequence eDNA in the field (Edwards et al. 2022; Krehenwinkel et al. 2019; Pomerantz et al. 2018). Field portable sequencing methodologies have been demonstrated on-board open-ocean research vessels for the detection of white sharks and could be used to improve sampling strategies while at sea (Truelove et al. 2019). The miniaturization of the technology, for example with devices like Oxford Nanopore's MinION, is stimulating the development of lab-on-chip systems that can accept a sample homogenate from an upstream sampler, carry out PCR

for metabarcoding, and complete library preparation for subsequent MinION sequencing in situ (P. Thielen, personal communication, November 2023). Given the enormous data generated through sequencing, in situ sequencing will require low power on-board processing capabilities that enable real-time data analysis in order to transmit manageable data packets over low bandwidth communications links typical of those currently used for remotely deployed platforms (Morganti et al. 2018).

Isothermal amplification methods and point of care (POC) diagnostics are increasingly being applied in eDNA use cases. Biosecurity surveillance is one area these methods have garnered considerable attention for rapid analysis to inform actionable decision making (Bowers et al. 2021; Williams et al. 2017). Loop-mediated isothermal amplification (LAMP) and recombinase polymerase amplification (RPA) are well suited for in situ diagnostics because they do not require thermal cycling and operate at much lower temperatures than other methods, reducing power consumption and technical complexity. Additionally, these isothermal methods are amenable to CRISPR-based detection systems, which have been shown to be rapid, highly sensitive, and specific (Baerwald et al. 2023).

Portable Surface Plasmon Resonance (SPR) devices are increasingly being developed for pathogen detection in water quality and toxin analyte detection in seafood (Quintanilla-Villanueva et al. 2023; Zhang et al. 2016). Recently, Ussler et al. utilized a portable SPR device for in situ detection of cyanobacterial microcystins on-board the 3G-ESP coupled to a long range AUV (Ussler et al. 2024). However, the current deployable SPR device is limited in its dynamic range and number of analytes that can be sensed from a given sample. Next generation optical and nanophotonic biosensors potentially offer a much more innovative means for detecting biological targets in situ. This class of sensors can support the detection of multiple analytes simultaneously or serially, which the current deployable SPR system cannot. These sensors leverage a range of photonic technologies, such as fluorescence, surface plasmon resonance, and Raman spectroscopy, to detect changes in light properties—intensity, wavelength, or polarization—upon interaction with target molecules (Javaid et al. 2024). The integration of nanomaterials and innovative surface chemistries enhances the selectivity and sensitivity of these sensors, enabling rapid, real-time analysis with minimal sample preparation and allowing detection of low-abundance targets in complex matrices. Miniaturized optical sensors utilizing nanophotonics are being developed to increase the selective, rapid, and sensitive detection of biomolecules in marine ecosystems (Balch et al. 2024). A major advantage of optical biosensors is the capacity for device regeneration, which provides the ability for continuous measurements without employing additional consumables (Park et al. 2022; Vashist 2012), which is ideal for incorporation with ecogenomic sensors.

7 | Use Cases, Applications, and Deployment Platforms

Autonomous eDNA sampling instrumentation and ecogenomic sensors have the potential to transform the field of ecological monitoring and biodiversity assessment. These technologies address a diverse suite of use cases, including tracking biodiversity,

detecting invasive species, monitoring water quality, managing endangered species, and assessing ecosystem health across various habitats. They are particularly valuable for use in remote or hazardous locations where traditional sampling methods are logistically difficult or impractical. Platforms for deploying these instruments are equally diverse—ranging from benthic landers and stationary moorings to ROVs and UxS (AUVs, ASVs)—all of which allow for flexible and targeted environmental monitoring across a wide variety of operational settings. Such technical and operational advancements are opening a new era of persistent and high-resolution temporal and spatial sampling that will be essential for scaling the application of eDNA observations in the future.

Instrument selection must be grounded in the intended use case, environmental context, and deployment logistics. Given the growing variety of instruments available and the wide range of emergent use cases, it is unlikely that any single device will ever be universally superior. Matching the right tool to the task demands collaboration between end users and engineers to align sampling requirements with specific capabilities. End users face numerous choices in defining these requirements in relation to situation-specific tradeoffs, such as sample collection depth(s), volume, frequency, replicates, and spatial coverage, as well as practical limitations tied to cost, logistics, and operational duration. Similarly, instrument developers must navigate technological limitations and competing design priorities, which are rarely aligned with all user needs—particularly for remotely operable or autonomous deployments.

One approach to facilitate alignment is to group instrumentation by generalized categories of use cases—for example, surface, midwater, deep water; mobile or stationary; short- or long-duration—alongside performance and logistical characteristics. For instance, some applications benefit from event-triggered sampling or adaptive scheduling, while others rely on predetermined sampling intervals or fixed-depth deployments. Table 1 provides a framework that organizes available instruments by shared operational attributes and deployment contexts, offering a high-level reference to help guide fit-for-purpose decision-making.

A key consideration across instruments is the trade-off between sampling frequency and deployment duration, governed by sample capacity and autonomous capabilities. High-frequency sampling (e.g., daily or hourly) often limits deployment length, while long-term deployments reduce sampling rates or rely on advanced autonomy to extend performance. These trade-offs are central to aligning instrument capabilities with mission duration, platform constraints, and monitoring goals.

In sum, end-users must clearly define their sampling objectives and constraints with respect to actionable outcomes, while instrument developers must be transparent about device capabilities, limitations, operational complexity, and support needs to ensure that new instruments are fit-for-purpose or that existing tools are appropriately matched to use cases. Instrument performance alone does not determine suitability—deployment platform compatibility, environmental conditions, and mission duration all play decisive roles. For example, ship-based biodiversity observations that leverage flow-through seawater access

TABLE 1 | Generalized use case framework for omics' sampling technology.

Deployment environment	Platform type	Autonomy level	Representative instruments	Example applications
Nearshore/coastal	Shipboard, Dockside	Low/assisted	SR-eDNA Sampler & AutoSampler, DOT eDNA Sampler, ESP FIDO	Routine biodiversity monitoring, invasive species detection
Surface/shelf	ASVs, Moored Surface Buoys, Shipboard	Moderate/autonomous	DOT eDNA Sampler, RoCSI, ESP-3G, ESP-2G, ESP-FIDO, MISNAC	Coastal time-series, HAB and toxin surveillance
Midwater/pelagic	AUVs, Gliders, Shipboard	High/semi-autonomous	SADIE, MISNAC, ESP-3G, DOT eDNA Sampler, RoCSI, Ascension Sampler	Broad-scale ocean surveys, water column profiling
Deep ocean/Seafloor	ROVs, Deep Moored Systems, Shipboard	Moderate/assisted	Ascension Sampler, RoCSI, inDEPTH, ISMIF, ESP-3G, MBARI Deep ESP, MISNAC	Targeted benthic sampling, deep-sea biodiversity assessments
Multi-purpose sample return	Shipboard	Low/assisted	AFIS, WaMS, CTD-rosette-based filtration, bag/bottle samplers	eDNA/eRNA + nutrient/toxin analyses, laboratory-based workflows
High-frequency observatory	Cabled or Long-term Moorings	High/autonomous	ESP-2G, ESP-3G, ESP-FIDO, RoCSI	Continuous monitoring, real-time or near-real-time data transmission

Note: Generalized use case framework for autonomous eDNA samplers. Instrument classes are organized by deployment environment, platform type, and autonomy level, with representative instruments drawn from both commercial and research-grade systems. While individual instruments are listed under primary use cases, many span multiple categories depending on integration context, mission constraints, and support infrastructure. The overlap reflects the inherent variability in eDNA sampling needs and reinforces that no single instrument is universally optimal—effective deployment depends on fit-for-purpose matching between user requirements and platform compatibility.

offer opportunities to reduce instrument complexity and cost, with the added benefit of human oversight during deployment and recovery. In contrast, use cases that demand strict hands-off operation under more extreme conditions or over extended durations introduce added complexity and increase the probability of failure—necessitating autonomous fault tolerance and redundancy. One approach to coordinate feature prioritization is MoSCoW (Must have, Should have, Could have and Wont have) analysis (Clegg and Barker 1994). Such an approach could provide a standardized means for gathering input from intended end-user groups to more systematically determine essential sampling or analysis requirements.

In addition to meeting end users' needs for first-order sample acquisition (e.g., sample volume, filter size/type, depth, etc.), the combined use of eDNA sampling instrumentation with a variety of oceanographic platforms has significantly enhanced capabilities for observing and monitoring marine ecosystems in ways that were not possible until recently. A number of instruments have been integrated with autonomous and remotely operated vehicles (Figure 4) to provide comprehensive and multi-dimensional data collection. This integration allows for simultaneous gathering of genetic material and water column parameters such as temperature, salinity, and current patterns, providing a holistic view of the marine environment. For example, ROVs equipped with eDNA samplers can target specific deep-sea locations (Govindarajan et al. 2025; Satoh et al. 2025; The National Oceanography Centre 2023), while AUVs can perform extensive, long-duration missions across the ocean's depths (Aquatic Sensors 2024; Den Uyl et al. 2022; Truelove et al. 2022; Yamahara et al. 2019; Zhang, Ryan, et al. 2021). Autonomous surface vehicles enable broad, surface-level monitoring, particularly in remote areas (NOAA AOML Communications 2023; ON&T 2024; Preston et al. 2024). Moorings, as a backbone of

sustained ocean observing systems, present an ideal platform for eDNA integration—supporting continuous, co-located biological and environmental measurements. Moored systems may relax constraints on size, weight, and power, allowing for deployment of more complex or higher throughput sampling systems, enabling extended operation and high-resolution temporal data. The versatility in platform availability ensures that eDNA sampling can be conducted in diverse and challenging settings, from coastal zones to the deep ocean, leading to more accurate and detailed assessments of biodiversity, ecosystem health, and the detection of both native and invasive species. The autonomous nature of these platforms also allows for continuous, high-resolution data collection, reducing the need for human intervention and enabling more efficient monitoring operations. However, the integration of samplers and deployment platforms often requires platform-specific engineering adaptations, and the suitability of a given instrument is highly dependent on compatibility with platform constraints such as power, size, data handling capacity, stability, etc. Again, no one-size-fits-all.

While this review summarizes technical capabilities and deployment characteristics, cost was not explicitly assessed, as it is highly use-case dependent. Sampling depth, mobility, platform constraints, and required contextual data all influence total operational costs. The goal remains to reduce cost and complexity while maximizing reliability and usability—but instruments must still meet specific deployment requirements. Ultimately, instrument selection must be grounded in the intended use case, environmental context, and deployment logistics—but no matter the scenario, platform compatibility remains a defining constraint. Nearly all deployments currently require some level of customization, and despite promising advances in modularity and interoperability, there remains no one-size-fits-all approach. Purpose-built devices tailored to specific applications continue

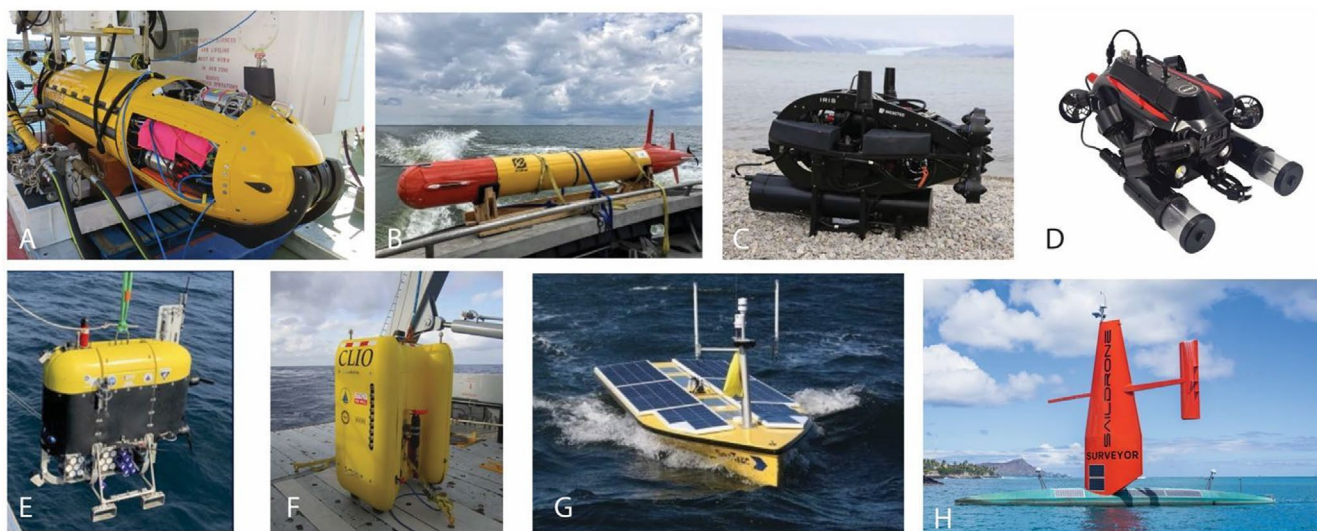


FIGURE 4 | Images of autonomous platforms with autonomously deployed 'omics-based sampling systems and ecogenomics sensors. (A) Autosub 6000 containing a RoCSI (Matt Kingsland, 2022), (B) LRAUV with a 3G-ESP (MBARI, 2016), (C) IRIS with IS-ABS (Alfredo Martin), (D) Okinawa MiniROV with two Samplers, (E) Mesobot with a WHOI-Oceanic Labs multisampler (F) CLIO and SUPR, (G) A SeaTrac ASV (SeaTrac Systems) and (H) Saildrone Surveyor, both carrying a 3G ESP. Image Attributions/Credits are listed in parentheses above. Images (E, F and H) were sourced from published journal articles¹⁻³, no modifications of the images were made. Image (C, D and G) used in this figure are sourced from publicly available phys.org and glos.org websites: Image C ©INESC Brussels Hub, 2024, Image D ©Okinawa Institute of Science and Technology, 2024 and Image G ©SeaTrac Systems, 2021.

to offer the most practical path forward. Acknowledging these limitations is essential for setting realistic expectations and for planning the true cost—financial, technical, and logistical—of deploying molecular observation technologies at scale.

8 | Challenges and Barriers: Developmental Gaps and Needs

While the potential of new eDNA instrumentation is very promising, there still are many challenges and barriers to both instrument development and uptake. Trust and validation remain some of the largest challenges, particularly because there is not necessarily a “gold standard” to compare to and that it can be difficult to parse differences between samples due to sampling instrumentation versus true biological variation and observation variance. Technology Readiness is typically defined for specific use cases, and requirements for invasive species detection, for example, are distinct from biodiversity assessments, each requiring independent investment for evaluation. Finally, some of the different types of instruments discussed above have fundamentally different sampling mechanisms where some collect a nearly instantaneous water sample, while others collect time or space (or both) integrated samples. Here, we discuss some of these challenges and barriers and suggestions for moving forward.

8.1 | Validation, Deployment Controls and Standard Reporting Metrics

Transparency is key to building trust and uptake of eDNA instrumentation as instrument development progresses. This requires developers to adequately test and validate instrumentation and report these results in an accessible manner (e.g., open-access peer-reviewed publication or technical specifications data sheet). Reporting instrumentation accuracy and precision, as well as points of failure such as propensity for contamination and limited sample stability, will give end-users the ability to assess instrument capability and applicability relative to meeting their specific needs.

One important aspect of deploying autonomous samplers and ecogenomic sensors is incorporating both positive and negative controls in order to fully evaluate the validity of material and/or data returned. In some cases, it is desirable to run controls while the instrument is deployed, to ensure it is operating properly. Positive controls provide information on both instrument state and confidence that negative results are truly negative (i.e., not false-negatives or degradation of the signal) over the course of a deployment. Negative controls assess the cleanliness of an instrument during a deployment and provide critical information about sample cross- and/or carry-over contamination. Guidelines have not been established on the number and timing of controls, but these are essential in providing transparency for the adoption of eDNA instrumentation on larger scales.

There are currently no standard protocols for validating autonomous environmental DNA (eDNA) instruments, including the processes of filtration, nucleic acid extraction, and in situ analytical techniques. It is also important to recognize that even

traditional eDNA sampling methods often lack full standardization, with variability in protocols depending on target taxa, sampling environment, and regional practices. The absence of standardized validation protocols presents challenges, particularly the risk of variability in data quality and reporting, which can compromise result comparability across different instruments and applications, and thus widespread adoption. Validation is still reliant on accepted standard protocols from the end-user community, which can vary depending on the target species or use case in question. Moreover, there is a need for standardized reference material (e.g., cell lines or mock communities) that would enable validation from sampling to analysis irrespective of the system and analytical approach employed. Until those community standard protocols and reference samples are established, it is recommended that instrument developers utilize protocols that are in widespread use to benchmark and validate their instrumentation processes.

Device specification reporting for autonomous eDNA instrumentation is currently underdeveloped and lacks standardization. Reported specifications should encompass critical metrics such as sensitivity, specificity, detection limits, sample throughput, operational range, and environmental tolerances. The absence of standardized reporting metrics poses significant challenges by preventing meaningful comparisons between different instruments and hindering the assessment of their suitability for various applications. In parallel, the eDNA community is advancing efforts to standardize data and metadata reporting (e.g., FAIR principles, MxS, Darwin Core) (Berry et al. 2021; Takahashi et al. 2024), and engineering technologies can play an active role in supporting and accelerating these developments. For instance, enabling instruments to export standardized output formats—such as EMMI-formatted files for qPCR data—could facilitate alignment with global repositories like NCBI SRA and GBIF, while simplifying metadata management for users. Minimum reporting metrics are essential to ensure transparency, reproducibility, and reliability in the deployment of autonomous eDNA systems. Engaging the instrumentation community in these emerging standards discussions will help ensure that technologies are not only analytically robust but also interoperable, archivable, and scalable for long-term biodiversity monitoring.

To promote transparency, interoperability, and reproducibility in marine 'omics instrumentation, the adoption of a community-driven specification reporting framework is recommended. A best practices table of instrument specifications is provided in Table S3 and S4, outlining a minimal set of standardized fields that should be consistently reported across sampling and analytical devices to enable meaningful comparison, validation, and integration across platforms and studies. These critical parameters encompass instrument configuration, sampling and preservation methods, analytical performance metrics, environmental operating limits, power and data handling capabilities, and quality assurance controls including positive and negative controls.

One way to address knowledge gaps is the development of consensus guidelines by expert panels, the establishment of standardized reporting frameworks and reference materials provided by relevant scientific and regulatory bodies, and the

promotion of open data and sample sharing practices among the research community. Implementing these solutions would facilitate device comparison, improve data quality, and ultimately enhance the credibility and effectiveness of autonomous eDNA technologies in environmental monitoring and research.

8.2 | Standardized Interfaces

As ecogenomic sensing capabilities develop and sampling instrumentation advances towards commercialization, it is likely that modularization will become the norm and thus, the need for standard fluidic, electrical, and communication interfaces will become essential. These interfaces are crucial for ensuring interoperability among various instrument modules, such as sample collection, filtration, lysis, analytical modules, as well as data processing. Standardized connections would enable seamless integration between a diverse array of sampling instruments, analytical devices, and oceanographic platforms, simplifying deployments and operations. This would enhance the reliability and reproducibility of scientific findings and accelerate technological innovation.

9 | Future Perspectives and Conclusion

The development and deployment of autonomous eDNA instrumentation is poised to transform aquatic biodiversity monitoring and resource management. Ongoing technological innovation is expected to improve the sensitivity, specificity, and operational capabilities of eDNA instruments. Realizing this potential will depend on continued validation, user training, cost optimization, and development of interoperable systems suited to diverse deployment environments. Integration with a variety of deployment platforms and other sensory systems, bolstered by artificial intelligence and machine learning algorithms, will enhance targeted sampling and data analysis, enabling real-time monitoring and rapid response to ecological changes absent human-in-the-loop intervention. Additionally, advancements in miniaturization and energy efficiency will allow for longer deployments and broader geographic coverage, facilitating comprehensive and continuous environmental assessments. As we move forward, embracing interdisciplinary approaches and fostering global collaborations will be crucial to unlocking the full potential of these technologies in real-world applications.

In conclusion, the development of autonomous eDNA sampling technology and ecogenomic sensors holds great promise for advancing our understanding of biodiversity dynamics in aquatic and terrestrial ecosystems. By overcoming the limitations of traditional sampling methods, these technological innovations offer unprecedented opportunities for comprehensive and efficient biological surveys. The aim of this review was to provide researchers, practitioners, and managers with a comprehensive overview of the current state of autonomous eDNA sampling technology, highlighting its potential for revolutionizing ecological monitoring and conservation practices in the years to come. There is no doubt that the future holds great promise, but the field of deployable 'omics technologies is still very much in its infancy, and much work remains if we are to capitalize on the full potential of what is possible.

Author Contributions

K.M.Y. and E.A.A. conceptualized the review and designed the structure of the manuscript. K.M.Y., E.A.A., J.R., and W.H.W. conducted the literature review, interviews and drafted the initial manuscript. P.C., E.E., I.B.E., T.F., A.F.G., A.M.M., K.M.P., V.J.S., A.T., I.W., and J.M.B. provided technical input on instrument design and functionality, and contributed to specific sections by synthesizing relevant literature and providing critical insights and edits. C.A.S. reviewed and edited the manuscript, offering substantial intellectual contributions to refine the arguments and ensure accuracy. All authors reviewed and approved the final version of the manuscript.

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Ethics Statement

This article is a review of previously published literature and insights obtained through personal communications with experts during the Marine 'Omics Technology and Instrumentation (MOTI) Workshop. All contributions were provided voluntarily, and no sensitive personal data were collected. As the communications involved professional opinions and posed minimal risk, formal ethics committee approval was not required.

Conflicts of Interest Statement

The authors declare no competing financial interests. Although some authors (A.T., E.E., I.B.E., V.J.S.) are employed by companies that manufacture and sell instrumentation, these affiliations had no influence on the design, execution, or reporting of the research presented in this paper.

Data Availability Statement

Data sharing is not applicable to this article as no datasets were generated or analyzed during the current study. Any information gathered through the literature review or personal communications that underlies the main findings is included in the [Supporting Information](#): Tables provided with the manuscript.

References

- Abbott, M. R., K. H. Brink, C. R. Booth, et al. 1990. "Observations of Phytoplankton and Nutrients From a Lagrangian Drifter Off Northern California." *Journal of Geophysical Research: Oceans* 95, no. C6: 9393–9409. <https://doi.org/10.1029/JC095iC06p09393>.
- Agersnap, S., E. E. Sigsgaard, M. R. Jensen, et al. 2022. "A National Scale "BioBlitz" Using Citizen Science and eDNA Metabarcoding for

- Monitoring Coastal Marine Fish." *Frontiers in Marine Science* 9: 824100. <https://doi.org/10.3389/fmars.2022.824100>.
- Applied Genomics Ltd. 2024. "Industry-leading eDNA Sampling Technologies." <https://appliedgenomics.co.uk/sample>.
- Aqualytical. 2025. "Environmental DNA Extraction | eDNA Filter [WWW Document]. Environ. DNA Extr." <https://aqualytical.com/environmental-dna-extraction/>.
- Aquatic Labs. 2024. "Aquatic Labs [WWW Document]. Aquat. Labs." <https://www.aquatic-labs.com>.
- Aquatic Sensors. 2024. "YUCO-eDNA [WWW Document]." <https://www.aquaticsensors.com/product/yuco-edna/>.
- Baerwald, M. R., E. C. Funk, A. M. Goodbla, et al. 2023. "Rapid CRISPR-Cas13a Genetic Identification Enables New Opportunities for Listed Chinook Salmon Management." *Molecular Ecology Resources* 25: e13777. <https://doi.org/10.1111/1755-0998.13777>.
- Balch, H., S. Dagli, J. Hu, et al. 2024. Silicon Nanophotonics for in situ Multi-omic Marine Detection.
- Berry, O., S. Jarman, A. Bissett, et al. 2021. "Making Environmental DNA (eDNA) Biodiversity Records Globally Accessible." *Environmental DNA* 3: 699–705. <https://doi.org/10.1002/edn3.173>.
- Bessey, C., S. Neil Jarman, T. Simpson, et al. 2021. "Passive eDNA Collection Enhances Aquatic Biodiversity Analysis." *Communications Biology* 4: 1–12. <https://doi.org/10.1038/s42003-021-01760-8>.
- Bird, L. E., A. Sherman, and J. Ryan. 2007. "Development of an Active, Large Volume, Discrete Seawater Sampler for Autonomous Underwater Vehicles, in: OCEANS 2007." *Presented at the OCEANS 2007*: 1–5. <https://doi.org/10.1109/OCEANS.2007.4449303>.
- Bowers, H. A., X. Pochon, U. von Ammon, et al. 2021. "Towards the Optimization of eDNA/eRNA Sampling Technologies for Marine Biosecurity Surveillance." *Water* 13, no. 8: 1113. <https://doi.org/10.3390/w13081113>.
- Breier, J. A., M. V. Jakuba, M. A. Saito, et al. 2020. "Revealing Ocean-Scale Biochemical Structure With a Deep-Diving Vertical Profiling Autonomous Vehicle." *Science Robotics* 5: eabc7104. <https://doi.org/10.1126/scirobotics.abc7104>.
- Breier, J. A., C. S. Sheik, D. Gomez-Ibanez, et al. 2014. "A Large Volume Particulate and Water Multi-Sampler With in Situ Preservation for Microbial and Biogeochemical Studies." *Deep-Sea Research, Part A. Oceanographic Research Papers* 94: 195–206. <https://doi.org/10.1016/j.dsr.2014.08.008>.
- Canonico, G., P. L. Buttigieg, E. Montes, et al. 2019. "Global Observational Needs and Resources for Marine Biodiversity." *Frontiers in Marine Science* 6: 367. <https://doi.org/10.3389/fmars.2019.00367>.
- Carneiro, A., G. Silva, P. Marques, et al. 2023. *Biosampler IS-ABS: eDNA Auto Filtration Unit for Vehicle Integration (v2.0)*, in: *OCEANS 2023—Limerick. Presented at the OCEANS 2023—Limerick*, 1–4. IEEE. <https://doi.org/10.1109/OCEANS2023Limerick52467.2023.10244269>.
- Citizens of the Sea. 2025. "Citizens of the Sea [WWW Document]. Citiz. Sea." <https://www.citizensofthesea.org/news/revolutionizing-ocean-research-with-innovative-tools-for-citizen-scientists>.
- Clegg, D., and R. Barker. 1994. *CASE Method Fast-Track: A RAD Approach*. Addison-Wesley Publishing Company. ISBN: 0-201-62432-X.
- Den Uyl, P. A., L. R. Thompson, R. M. Errera, et al. 2022. "Lake Erie Field Trials to Advance Autonomous Monitoring of Cyanobacterial Harmful Algal Blooms." *Frontiers in Marine Science* 9: 1021952. <https://doi.org/10.3389/fmars.2022.1021952>.
- Doucette, G. J., C. M. Mikulski, K. L. Jones, et al. 2009. "Remote, Subsurface Detection of the Algal Toxin Domoic Acid Onboard the Environmental Sample Processor: Assay Development and Field Trials." *Harmful Algae* 8: 880–888. <https://doi.org/10.1016/j.hal.2009.04.006>.
- Edwards, A., A. Soares, A. Debbonaire, and S. M. Edwards Rassner. 2022. "Before You Go: A Packing List for Portable DNA Sequencing of Microbiomes and Metagenomes." *Microbiology* 168: 001220. <https://doi.org/10.1099/mic.0.001220>.
- Ensminger, M. P., M. Vasquez, H.-J. Tsai, et al. 2017. "Continuous Low-Level Aquatic Monitoring (CLAM) Samplers for Pesticide Contaminant Screening in Urban Runoff: Analytical Approach and a Field Test Case." *Chemosphere* 184: 1028–1035.
- Feike, J., K. Jürgens, J. T. Hollibaugh, S. Krüger, G. Jost, and M. Labrenz. 2012. "Measuring Unbiased Metatranscriptomics in Suboxic Waters of the Central Baltic Sea Using a New *in Situ* Fixation System." *ISME Journal* 6: 461–470. <https://doi.org/10.1038/ismej.2011.94>.
- Ficetola, G. F., C. Miaud, F. Pompanon, and P. Taberlet. 2008. "Species Detection Using Environmental DNA From Water Samples." *Biology Letters* 4, no. 4: 423–425. <https://doi.org/10.1098/rsbl.2008.0118>.
- FluidIon. 2025. "Fluidion Deep Water Sampler [WWW Document]." <https://fluidion.com/products/samplers/deep-water-sampler>.
- Foote, A. D., P. F. Thomsen, S. Sveegaard, et al. 2012. "Investigating the Potential Use of Environmental DNA (eDNA) for Genetic Monitoring of Marine Mammals." *PLoS One* 7, no. 8: e41781. <https://doi.org/10.1371/journal.pone.0041781>.
- Forio, M. A. E., and P. L. M. Goethals. 2020. "An Integrated Approach of Multi-Community Monitoring and Assessment of Aquatic Ecosystems to Support Sustainable Development." *Sustainability* 12, no. 14: 5603. <https://doi.org/10.3390/su12145603>.
- Friederich, G. E., P. J. Kelly, and L. A. Codispoti. 1986. *An Inexpensive Moored Water Sampler for Investigating Chemical Variability, Tidal Mixing and Plankton Dynamics, Tidal Mixing and Plankton Dynamics*. Springer-Verlag. https://doi.org/10.1007/978-1-4612-4966-5_19.
- Fries, D., and J. Paul. 2003. *Autonomous Microbial Genosensor*. Defense Technical Information Center. <https://doi.org/10.21236/ADA629476>.
- Fukuba, T., and T. Fujii. 2021. "Lab-On-a-Chip Technology for in Situ Combined Observations in Oceanography." *Lab On a Chip* 21: 55–74. <https://doi.org/10.1039/D0LC00871K>.
- Fukuba, T., S. Goto, M. K.-S. Wong, et al. 2022. "Development and Evaluation of Automated Gene Collector—ATGC-12S for Environmental DNA Sample Archive at Aquatic Environments." In *OCEANS 2022, Hampton Roads. Presented at the OCEANS 2022, Hampton Roads*, 1–5. IEEE. <https://doi.org/10.1109/OCEANS47191.2022.9977101>.
- Fukuba, T., A. Miyaji, T. Okamoto, T. Yamamoto, S. Kaneda, and T. Fujii. 2011. "Integrated in Situ Genetic Analyzer for Microbiology in Extreme Environments." *RSC Advances* 1: 1567–1573. <https://doi.org/10.1039/C1RA00490E>.
- George, S. D., A. J. Sepulveda, P. R. Hutchins, et al. 2024. "Field Trials of an Autonomous eDNA Sampler in Lotic Waters." *Environmental Science & Technology* 58: 20942–20953. <https://doi.org/10.1021/acs.est.4c04970>.
- Goffredi, S. K., W. J. Jones, C. A. Scholin, R. Marin, and R. C. Vrijenhoek. 2006. "Molecular Detection of Marine Invertebrate Larvae." *Marine Biotechnology* 8: 149–160. <https://doi.org/10.1007/s10126-005-5016-2>.
- Gold, Z., J. Sprague, D. J. Kushner, E. Z. Marin, and P. H. Barber. 2021. "eDNA Metabarcoding as a Biomonitoring Tool for Marine Protected Areas." *PLoS One* 16: e0238557. <https://doi.org/10.1371/journal.pone.0238557>.
- Govindarajan, A., A. Adams, E. Allan, et al. 2023. "Advances in Environmental DNA Sampling for Observing Ocean Twilight Zone Animal Diversity." *Oceanography* 36: 80–86. <https://doi.org/10.5670/oceanog.2023.s1.27>.
- Govindarajan, A., N. Yang, V. Brown, et al. 2025. *Deep Sea Biodiversity Exploration in American Samoa With Environmental DNA*, edited by D. Wagner, 40–41. Ocean Exploration Trust 2024 Field Season. <https://doi.org/10.62878/unc751>.

- Govindarajan, A. F., L. McCartin, A. Adams, et al. 2022. "Improved Biodiversity Detection Using a Large-Volume Environmental DNA Sampler With in Situ Filtration and Implications for Marine eDNA Sampling Strategies." *Deep Sea Research, Part I: Oceanographic Research Papers* 189: 103871. <https://doi.org/10.1016/j.dsr.2022.103871>.
- Govindarajan, A. F., J. Pineda, M. Purcell, and J. A. Breier. 2015. "Species- and Stage-Specific Barnacle Larval Distributions Obtained From AUV Sampling and Genetic Analysis in Buzzards Bay, Massachusetts, USA." *Journal of Experimental Marine Biology and Ecology* 472: 158–165. <https://doi.org/10.1016/j.jembe.2015.07.012>.
- GreenEyes LLC. 2025. "Aqua Monitor [WWW Document]." <https://gescience.com/water-sampler/>.
- Greenfield, D. I., R. Marin III, G. J. Doucette, et al. 2008. "Field Applications of the Second-Generation Environmental Sample Processor (ESP) for Remote Detection of Harmful Algae: 2006–2007." *Limnology and Oceanography: Methods* 6: 667–679. <https://doi.org/10.4319/lom.2008.6.667>.
- Hadaway, H. 2010. "Ecogenomic Sensor." http://www.cev.washington.edu/story/Ecogenomic_Sensor.html.
- Hansen, B. K., M. W. Jacobsen, A. L. Middelboe, et al. 2020. "Remote, Autonomous Real-Time Monitoring of Environmental DNA From Commercial Fish." *Scientific Reports* 10: 13272. <https://doi.org/10.1038/s41598-020-70206-8>.
- Harvey, J. B. J., J. P. Ryan, R. Marin, et al. 2012. "Robotic Sampling, in Situ Monitoring and Molecular Detection of Marine Zooplankton." *Journal of Experimental Marine Biology and Ecology* 413: 60–70. <https://doi.org/10.1016/j.jembe.2011.11.022>.
- Hatch, A. C., T. Ray, K. Lintecum, and C. Youngbull. 2014. "Continuous Flow Real-Time PCR Device Using Multi-Channel Fluorescence Excitation and Detection." *Lab On a Chip* 14: 562–568. <https://doi.org/10.1039/C3LC51236C>.
- Hayden, E., D. Yoerger, A. Govindarajan, et al. 2023. "Mesobot Operations From E/V Nautilus." *Oceanography* 36: 34–35.
- Hendricks, A., C. M. Mackie, E. Luy, et al. 2023. "Compact and Automated eDNA Sampler for in Situ Monitoring of Marine Environments." *Scientific Reports* 13: 5210. <https://doi.org/10.1038/s41598-023-32310-3>.
- Holman, L. E., M. de Bruyn, S. Creer, G. Carvalho, J. Robidart, and M. Rius. 2019. "Detection of Introduced and Resident Marine Species Using Environmental DNA Metabarcoding of Sediment and Water." *Scientific Reports* 9: 11559. <https://doi.org/10.1038/s41598-019-47899-7>.
- Huvenne, V. 2024. *RRS James Cook Expedition JC237, 6 AUGUST—4 SEPTEMBER 2022. CLASS—Climate-Linked Atlantic Sector Science Whittard Canyon and Porcupine Abyssal Plain Fixed Point Observatories (Publication—Report)*. National Oceanography Centre.
- INESC Brussels. 2024. "Scientists Collect eDNA Samples in the Extreme Environment of the Arctic Melting Glaciers Autonomously [WWW Document]." <https://phys.org/news/2024-01-scientists-edna-samples-extreme-environment.html>.
- Javaid, Z., M. A. Iqbal, S. Javeed, et al. 2024. "Reviewing Advances in Nanophotonic Biosensors." *Frontiers in Chemistry* 12: 1449161. <https://doi.org/10.3389/fchem.2024.1449161>.
- Jones, D., K. Clements, and A. Sepulveda. 2024. "A Workshop to Advance Invasive Species Early Detection Capacity of the Rapid Environmental DNA Assessment and Deployment Initiative & Network (READI-Net)." *Management of Biological Invasions* 15: 159–167. <https://doi.org/10.3391/mbi.2024.15.1.10>.
- Jones, D. N., B. C. Augustine, P. Hutchins, et al. 2024. "Autonomous Samplers and Environmental DNA Metabarcoding: Sampling Day and Primer Choice Have Greatest Impact on Fish Detection Probabilities." *Metabarcoding Metagenomics* 8: e122375. <https://doi.org/10.3897/mbmg.8.122375>.
- Jones, W. J., C. M. Preston, R. Marin III, C. A. Scholin, and R. C. Vrijenhoek. 2008. "A Robotic Molecular Method for in Situ Detection of Marine Invertebrate Larvae." *Molecular Ecology Resources* 8: 540–550. <https://doi.org/10.1111/j.1471-8286.2007.02021.x>.
- Kelly, R. P., D. M. Lodge, K. N. Lee, et al. 2024. "Toward a National eDNA Strategy for the United States." *Environmental DNA* 6: e432. <https://doi.org/10.1002/edn3.432>.
- Krehenwinkel, H., A. Pomerantz, J. B. Henderson, et al. 2019. "Nanopore Sequencing of Long Ribosomal DNA Amplicons Enables Portable and Simple Biodiversity Assessments With High Phylogenetic Resolution Across Broad Taxonomic Scale." *GigaScience* 8: giz006. <https://doi.org/10.1093/gigascience/giz006>.
- Luy, E., N. Gerdali, N. Horwood, et al. 2024. "Automated Environmental DNA (eDNA) Sampling Using an Optimized Filter Cassette for High Volume Filtration." In *OCEANS 2024—Halifax. Presented at the OCEANS 2024—Halifax*, 1–7. IEEE. <https://doi.org/10.1109/OCEANS55160.2024.10754477>.
- Marsh, M. 2020. *Pilot Study to Validate an Environmental DNA Sampler for Monitoring Inshore Fish Communities (No. NECR287)*. Natural England.
- Martins, A., C. Almeida, P. Lima, et al. 2020. "A Robotic Solution for NETTAG Lost Fishing Net Problem." In *Global Oceans 2020: Singapore – U.S., 1–6*. Gulf Coast. Presented at the Global Oceans 2020. <https://doi.org/10.1109/IEEECONF38699.2020.9389464>.
- Mayer, L., and V. Schmidt. 2023. "DriX Operations From E/V Nautilus in New Frontiers in Ocean Exploration: The Ocean Exploration Trust 2022 Field Season." *Oceanography* 36: 36–37.
- McKinney, E. S. A., C. E. Gibson, and B. M. Stewart. 1997. "Planktonic Diatoms in the North-West Irish Sea: A Study by Automatic Sampler." *Biology and Environment: Proceedings of the Royal Irish Academy* 97B: 197–202.
- McQuillan, J. S., and J. C. Robidart. 2017. "Molecular-Biological Sensing in Aquatic Environments: Recent Developments and Emerging Capabilities." *Current Opinion in Biotechnology* 45: 43–50. <https://doi.org/10.1016/j.copbio.2016.11.022>.
- Missen, L., and M. Ayad. 2025. "Scaling-Up Marine Biodiversity Monitoring: Automation of eDNA Workflows From Sampling to Sequencing." eDNA Conference, Wellington, New Zealand, February 20. <https://ednaconference.com.au/2025-program>.
- Morganti, L., D. Cesini, E. Corni, et al. 2018. "Low-Power Storage Bricks and Bioinformatics on Systems-On-Chip, in: 2018 26th Euromicro International Conference on Parallel, Distributed and Network-Based Processing (PDP)." In *Presented at the 2018 26th Euromicro International Conference on Parallel, Distributed and Network-based Processing (PDP)*, 635–638. IEEE. <https://doi.org/10.1109/PDP2018.2018.00106>.
- Morrison, A. T., J. D. Billings, and K. W. Doherty. 2000. "The McLane WTS-LV: A Large Volume, High Accuracy, Oceanographic Sampling Pump, in: OCEANS 2000 MTS/IEEE Conference and Exhibition. Conference Proceedings (Cat. No.00CH37158)." In *Presented at the OCEANS 2000 MTS/IEEE Conference and Exhibition. Conference Proceedings (Cat. No.00CH37158)*, vol. 2, 847–852. IEEE. <https://doi.org/10.1109/OCEANS.2000.881365>.
- Mynott, S., and M. Marsh. 2020. Development of a novel (DNA-based) method for monitoring inshore fish communities using a programmable large-volume marine eDNA sampler (No. NECR330).
- NOAA AOML Communications. 2023. "Failing Upwards: Developing an Autonomous Surface Vehicle to Advance 'Omics Research—NOAA/AOML' [WWW Document]. NOAAs Atl. Oceanogr. Meteorol. Lab." <https://www.aoml.noaa.gov/developing-autonomous-vehicle-to-advance-omics-research/>.
- NOAA Fisheries. 2025. "Environmental DNA Survey Continues in the Southern New England Wind Energy Area | NOAA Fisheries [WWW Document]. NOAA." <https://www.fisheries.noaa.gov/science-blog/>

environmental-dna-survey-continues-southern-new-england-wind-energy-area.

NOAA NCCOS. 2019. "Sharing Advances in Performance and Affordability of the Second-Generation Environmental Sample Processor [WWW Document]. NCCOS—Natl. Cent. Coast." *Ocean Science*. <https://coastalscience.noaa.gov/news/sharing-advances-in-performance-and-affordability-of-the-second-generation-environmental-sample-processor/>.

NOAA NCCOS. 2023. "'Team' of Uncrewed Vehicles Reveals Extent, Toxicity of Harmful Algal Bloom in Monterey Bay [WWW Document]. NCCOS—Natl. Cent. Coast. Ocean Sci." <https://coastalscience.noaa.gov/news/a-team-of-uncrewed-vehicles-reveals-the-extent-and-toxicity-of-a-harmful-algal-bloom-in-monterey-bay-ca/>.

NOAA NCCOS. 2024. "Enhancing Third-generation Environmental Sample Processor for HAB Toxin Detection Through Integration with Autonomous Surface Vehicle [WWW Document]. NCCOS—Natl. Cent. Coast. Ocean Sci." <https://coastalscience.noaa.gov/project/enhancing-the-capabilities-of-the-3rd-generation-environmental-sample-processor-for-hab-toxin-detection-through-integration-with-an-autonomous-surface-vehicle/>.

Noble-James, T., R. Bullimore, F. McBreen, et al. 2023. "Monitoring Benthic Habitats in English Marine Protected Areas: Lessons Learned, Challenges and Future Directions." *Marine Policy* 157: 105852. <https://doi.org/10.1016/j.marpol.2023.105852>.

Nucleic Sensing Systems. 2024. "Nucleic Sensing Systems." <https://NS2CO.com/index.html>.

Ocean Diagnostics. 2024. "Ascension Microplastics and eDNA Sampler." <https://oceandiagnostics.com/ascension>.

ON&T. 2024. "A New Wave of Collaboration: Advancing eDNA Sampling Through Uncrewed Platforms." *Ocean News and Technology* 4: 40.

Ottesen, E. A. 2016. "Probing the Living Ocean With Ecogenomic Sensors." *Current Opinion in Microbiology, Environmental Microbiology* * Special Section: Megaviruses 31: 132–139. <https://doi.org/10.1016/j.mib.2016.03.012>.

Ottesen, E. A., R. Marin III, C. M. Preston, et al. 2011. "Metatranscriptomic Analysis of Autonomously Collected and Preserved Marine Bacterioplankton." *ISME Journal* 5: 1881–1895. <https://doi.org/10.1038/ismej.2011.70>.

Ottesen, E. A., C. R. Young, J. M. Eppley, et al. 2013. "Pattern and Synchrony of Gene Expression Among Sympatric Marine Microbial Populations." *Proceedings of the National Academy of Sciences* 110: E488–E497. <https://doi.org/10.1073/pnas.1222099110>.

Ottesen, E. A., C. R. Young, S. M. Gifford, et al. 2014. "Multispecies Diel Transcriptional Oscillations in Open Ocean Heterotrophic Bacterial Assemblages." *Science* 345: 207–212. <https://doi.org/10.1126/science.1252476>.

Pargett, D. M., J. M. Birch, C. M. Preston, J. P. Ryan, Y. Zhang, and C. A. Scholin. 2015. "Development of a Mobile Ecogenomic Sensor." In *OCEANS 2015—MTS/IEEE Washington*, 1–6. Presented at the OCEANS 2015—MTS/IEEE. <https://doi.org/10.23919/OCEANS.2015.7404361>.

Pargett, D. M., S. D. Jensen, B. A. Roman, et al. 2013. "Deep Water Instrument for Microbial Identification, Quantification, and Archiving." In *2013 OCEANS—San Diego*, 1–6. Presented at the 2013 OCEANS. <https://doi.org/10.23919/OCEANS.2013.6741066>.

Park, J.-H., Y.-W. Cho, and T.-H. Kim. 2022. "Recent Advances in Surface Plasmon Resonance Sensors for Sensitive Optical Detection of Pathogens." *Biosensors* 12: 180. <https://doi.org/10.3390/bios12030180>.

Parsons, K., P. LaMothe, A. Wells, et al. 2022. Introducing SADIE: A Semi-Autonomous Sampler for In Situ Filtering of Seawater at Depth.

Pomerantz, A., N. Peñafiel, A. Arteaga, et al. 2018. "Real-Time DNA Barcoding in a Rainforest Using Nanopore Sequencing: Opportunities for Rapid Biodiversity Assessments and Local Capacity Building." *GigaScience* 7: giy033. <https://doi.org/10.1093/gigascience/giy033>.

Preston, C., K. Yamahara, D. Pargett, et al. 2024. "Autonomous eDNA Collection Using an Uncrewed Surface Vessel Over a 4200-Km Transect of the Eastern Pacific Ocean." *Environmental DNA* 6: e468. <https://doi.org/10.1002/edn3.468>.

Preston, C. M., A. Harris, J. P. Ryan, et al. 2011. "Underwater Application of Quantitative PCR on an Ocean Mooring." *PLoS One* 6: e22522. <https://doi.org/10.1371/journal.pone.0022522>.

Preston, C. M., R. Marin III, S. D. Jensen, et al. 2009. "Near Real-Time, Autonomous Detection of Marine Bacterioplankton on a Coastal Mooring in Monterey Bay, California, Using rRNA-Targeted DNA Probes." *Environmental Microbiology* 11: 1168–1180. <https://doi.org/10.1111/j.1462-2920.2009.01848.x>.

Quintanilla-Villanueva, G. E., J. Maldonado, D. Luna-Moreno, J. M. Rodríguez-Delgado, J. F. Villarreal-Chiu, and M. M. Rodríguez-Delgado. 2023. "Progress in Plasmonic Sensors as Monitoring Tools for Aquaculture Quality Control." *Biosensors* 13: 90. <https://doi.org/10.3390/bios13010090>.

Ribeiro, H., A. Martins, M. Gonçalves, et al. 2019. "Development of an Autonomous Biosampler to Capture in Situ Aquatic Microbiomes." *PLoS One* 14: e0216882. <https://doi.org/10.1371/journal.pone.0216882>.

Ritzenthaler, A., C. Mikulski, R. Marin, et al. 2016. "Development of the First Autonomous, In-Situ Microcystin Immunoassay for the Inaugural Freshwater Deployment on the Environmental Sampler Processor." In *OCEANS 2016 MTS/IEEE Monterey*, 1–4. Presented at the OCEANS 2016 MTS/IEEE Monterey. <https://doi.org/10.1109/OCEANS.2016.7761003>.

Robidart, J., S. J. Callister, P. Song, C. D. Nicora, C. G. Wheat, and P. R. Girguis. 2013. "Characterizing Microbial Community and Geochemical Dynamics at Hydrothermal Vents Using Osmotically Driven Continuous Fluid Samplers." *Environmental Science & Technology* 47: 4399–4407. <https://doi.org/10.1021/es3037302>.

Robidart, J. C., M. J. Church, J. P. Ryan, et al. 2014. "Ecogenomic Sensor Reveals Controls on N₂-Fixing Microorganisms in the North Pacific Ocean." *ISME Journal* 8: 1175–1185. <https://doi.org/10.1038/ismej.2013.244>.

Rourke, M. L., A. M. Fowler, J. M. Hughes, et al. 2022. "Environmental DNA (eDNA) as a Tool for Assessing Fish Biomass: A Review of Approaches and Future Considerations for Resource Surveys." *Environmental DNA* 4: 9–33. <https://doi.org/10.1002/edn3.185>.

Satoh, N., F. Sinniger, H. Narisoko, et al. 2025. "Using Underwater Mini-ROV for Coral eDNA Survey: A Case Study in Okinawan Mesophotic Ecosystems." *Coral Reefs* 44: 209–219. <https://doi.org/10.1007/s00338-024-02597-3>.

Scholin, C. A., J. Birch, S. Jensen, R. M. Iii, and E. Massion. 2018. "The Quest to Develop Ecogenomic Sensors: A 25-Year History of the Environmental Sample Processor (ESP) as a Case Study." *Oceanography* 30: 100–113. <https://doi.org/10.5670/oceanog.2017.427>.

Searcy, R. T., A. B. Boehm, C. Weinstock, et al. 2022. "High-Frequency and Long-Term Observations of eDNA From Imperiled Salmonids in a Coastal Stream: Temporal Dynamics, Relationships With Environmental Factors, and Comparisons With Conventional Observations." *Environmental DNA* 4: 776–789. <https://doi.org/10.1002/edn3.293>.

Sepulveda, A. J., J. M. Birch, E. P. Barnhart, et al. 2020. "Robotic Environmental DNA Bio-Surveillance of Freshwater Health." *Scientific Reports* 10: 14389. <https://doi.org/10.1038/s41598-020-71304-3>.

Sheik, C. S., K. Anantharaman, J. A. Breier, J. B. Sylvan, K. J. Edwards, and G. J. Dick. 2015. "Spatially Resolved Sampling Reveals Dynamic Microbial Communities in Rising Hydrothermal Plumes Across a

- Back-Arc Basin." *ISME Journal* 9: 1434–1445. <https://doi.org/10.1038/ismej.2014.228>.
- Sonnichsen, C., E. Luy, A. Hendricks, et al. 2024. "Towards an Automated, in-Situ Environmental DNA Sensor for Detection of Marine Species." In *OCEANS 2024—Halifax. Presented at the OCEANS 2024—Halifax*, 1–7. IEEE. <https://doi.org/10.1109/OCEANS55160.2024.10753699>.
- Stat, M., M. J. Huggett, R. Bernasconi, et al. 2017. "Ecosystem Biomonitoring With eDNA: Metabarcoding Across the Tree of Life in a Tropical Marine Environment." *Scientific Reports* 7: 12240. <https://doi.org/10.1038/s41598-017-12501-5>.
- Stern, R. F., K. T. Picard, K. M. Hamilton, et al. 2015. "Novel Lineage Patterns From an Automated Water Sampler to Probe Marine Microbial Biodiversity With Ships of Opportunity Progress." In *Oceanography the UK Western Channel Observatory: Integrating Pelagic and Benthic Observations in a Shelf Sea Ecosystem*, vol. 137, 409–420. Elsevier. <https://doi.org/10.1016/j.pocean.2015.04.015>.
- Taberlet, P., A. Bonin, L. Zinger, and E. Coissac. 2018. *Environmental DNA: For Biodiversity Research and Monitoring*. Oxford University Press. <https://doi.org/10.1093/oso/9780198767220.001.0001>.
- Takahashi, M., T. Frøslev, G. Jenkins, et al. 2024. "Best Practice for Publishing Environmental DNA (eDNA) Data According to FAIR Principles." *Biodiversity Information Science and Standards* 8: e137742. <https://doi.org/10.3897/biss.8.137742>.
- Tang, W., E. Cerdán-García, H. Berthelot, et al. 2020. "New Insights Into the Distributions of Nitrogen Fixation and Diazotrophs Revealed by High-Resolution Sensing and Sampling Methods." *ISME Journal* 14: 2514–2526. <https://doi.org/10.1038/s41396-020-0703-6>.
- Taylor, C. D., and K. W. Doherty. 1990. "Submersible Incubation Device (SID), autonomous Instrumentation for the *in Situ* Measurement of Primary Production and Other Microbial Rate Processes." *Deep-Sea Research Part A, Oceanographic Research Papers* 37: 343–358. [https://doi.org/10.1016/0198-0149\(90\)90132-F](https://doi.org/10.1016/0198-0149(90)90132-F).
- Taylor, C. D., K. W. Doherty, S. J. Molyneaux, et al. 2006. "Autonomous Microbial Sampler (AMS), a Device for the Uncontaminated Collection of Multiple Microbial Samples From Submarine Hydrothermal Vents and Other Aquatic Environments." *Deep Sea Research, Part I: Oceanographic Research Papers* 53: 894–916. <https://doi.org/10.1016/j.dsr.2006.01.009>.
- The National Oceanography Centre. 2023. "ROV Isis on SMARTEX Blog." <https://blogs.noc.ac.uk/jc241-smartex/isis-rov>.
- Thomas, A. C., J. Howard, P. L. Nguyen, T. A. Seimon, and C. S. Goldberg. 2018. "eDNA Sampler: A Fully Integrated Environmental DNA Sampling System." *Methods in Ecology and Evolution* 9: 1379–1385. <https://doi.org/10.1111/2041-210X.12994>.
- Thomas, A. C., P. L. Nguyen, J. Howard, and C. S. Goldberg. 2019. "A Self-Preserving, Partially Biodegradable eDNA Filter." *Methods in Ecology and Evolution* 10: 1136–1141. <https://doi.org/10.1111/2041-210X.13212>.
- Thomsen, P. F., J. Kielgast, L. L. Iversen, P. R. Møller, M. Rasmussen, and E. Willerslev. 2012. "Detection of a Diverse Marine Fish Fauna Using Environmental DNA From Seawater Samples." *PLoS One* 7: e41732. <https://doi.org/10.1371/journal.pone.0041732>.
- Torres-Beltrán, M., A. Mueller, M. Scofield, et al. 2019. "Sampling and Processing Methods Impact Microbial Community Structure and Potential Activity in a Seasonally Anoxic Fjord: Saanich Inlet, British Columbia." *Frontiers in Marine Science* 6: 132. <https://doi.org/10.3389/fmars.2019.00132>.
- Truelove, N. K., E. A. Andruszkiewicz, and B. A. Block. 2019. "A Rapid Environmental DNA Method for Detecting White Sharks in the Open Ocean." *Methods in Ecology and Evolution* 10: 1128–1135. <https://doi.org/10.1111/2041-210X.13201>.
- Truelove, N. K., N. V. Patin, M. Min, et al. 2022. "Expanding the Temporal and Spatial Scales of Environmental DNA Research With Autonomous Sampling." *Environmental DNA* 4: 972–984. <https://doi.org/10.1002/edn3.299>.
- Ussler, W., III, G. J. Doucette, C. M. Preston, et al. 2024. "Underway Measurement of Cyanobacterial Microcystins Using a Surface Plasmon Resonance Sensor on an Autonomous Underwater Vehicle." *Limnology and Oceanography: Methods* 22: 681–699. <https://doi.org/10.1002/lom3.10627>.
- Ussler, W. I., C. Preston, P. Tavormina, et al. 2013. "Autonomous Application of Quantitative PCR in the Deep Sea: In Situ Surveys of Aerobic Methanotrophs Using the Deep-Sea Environmental Sample Processor." *Environmental Science & Technology* 47: 9339–9346. <https://doi.org/10.1021/es4023199>.
- Van Wyngaarden, M., N. W. Jeffery, E. Horne, et al. 2024. "Evaluating an Autonomous eDNA Sampler for Marine Environmental Monitoring: Short- and Long-Term Applications." In *OCEANS 2024—Halifax*, 1–8. Presented at the OCEANS 2024. <https://doi.org/10.1109/OCEANS55160.2024.10753841>.
- Vashist, S. K. 2012. "A Method for Regenerating Gold Surface for Prolonged Reuse of Gold-Coated Surface Plasmon Resonance Chip." *Analytical Biochemistry* 423: 23–25. <https://doi.org/10.1016/j.ab.2011.12.045>.
- Wagner, D. 2023. "New Frontiers in Ocean Exploration: The Ocean Exploration Trust 2022 Field Season." *Oceanography* 36: 1–54. <https://doi.org/10.5670/oceanog.2023.s2>.
- Wang, B., L. Jiao, L. Ni, M. Wang, and P. You. 2024. "Bridging the Gap: The Integration of eDNA Techniques and Traditional Sampling in Fish Diversity Analysis." *Frontiers in Marine Science* 11: 132–137. <https://doi.org/10.3389/fmars.2024.1289589>.
- Wang, Y., Z.-M. Gao, J. Li, et al. 2019. "Hadal Water Sampling by *in Situ* Microbial Filtration and Fixation (ISMIF) Apparatus." *Deep Sea Research. Part A, Oceanographic Research Papers* 144: 132–137. <https://doi.org/10.1016/j.dsr.2019.01.009>.
- Wei, Z.-F., W.-L. Li, J. Li, et al. 2020. "Multiple *in Situ* Nucleic Acid Collections (MISNAC) From Deep-Sea Waters." *Frontiers in Marine Science* 7: 81. <https://doi.org/10.3389/fmars.2020.00081>.
- Westgaard, J.-I., K. Præbel, P. Arneberg, et al. 2024. "Towards eDNA Informed Biodiversity Studies – Comparing Water Derived Molecular Taxa With Traditional Survey Methods." *Progress in Oceanography* 222: 103230. <https://doi.org/10.1016/j.pocean.2024.103230>.
- Williams, M. R., R. D. Stedtfeld, C. Engle, et al. 2017. "Isothermal Amplification of Environmental DNA (eDNA) for Direct Field-Based Monitoring and Laboratory Confirmation of *Dreissena* sp." *PLoS One* 12: e0186462. <https://doi.org/10.1371/journal.pone.0186462>.
- Wurzbacher, C., I. Salka, and H. Grossart. 2012. "Environmental Actinorhodopsin Expression Revealed by a New *in Situ* Filtration and Fixation Sampler." *Environmental Microbiology Reports* 4: 491–497. <https://doi.org/10.1111/j.1758-2229.2012.00350.x>.
- Xu, D., W. Zhang, H. Li, N. Li, and J.-M. Lin. 2023. "Advances in Droplet Digital Polymerase Chain Reaction on Microfluidic Chips." *Lab On a Chip* 23: 1258–1278. <https://doi.org/10.1039/D2LC00814A>.
- Yamahara, K. M., E. Demir-Hilton, C. M. Preston, et al. 2015. "Simultaneous Monitoring of Faecal Indicators and Harmful Algae Using an In-Situ Autonomous Sensor." *Letters in Applied Microbiology* 61: 130–138. <https://doi.org/10.1111/lam.12432>.
- Yamahara, K. M., C. M. Preston, J. Birch, et al. 2019. "In Situ Autonomous Acquisition and Preservation of Marine Environmental DNA Using an Autonomous Underwater Vehicle." *Frontiers in Marine Science* 6: 373. <https://doi.org/10.3389/fmars.2019.00373>.
- Zhang, P., Y.-P. Chen, W. Wang, Y. Shen, and J.-S. Guo. 2016. "Surface Plasmon Resonance for Water Pollutant Detection and Water Process

Analysis." *TrAC Trends in Analytical Chemistry* 85: 153–165. <https://doi.org/10.1016/j.trac.2016.09.003>.

Zhang, Y., B. Kieft, B. W. Hobson, et al. 2021. "Persistent Sampling of Vertically Migrating Biological Layers by an Autonomous Underwater Vehicle Within the Beam of a Seabed-Mounted Echosounder." *IEEE Journal of Oceanic Engineering* 46: 497–508. <https://doi.org/10.1109/JOE.2020.2982811>.

Zhang, Y., J. P. Ryan, B. W. Hobson, et al. 2021. "A System of Coordinated Autonomous Robots for Lagrangian Studies of Microbes in the Oceanic Deep Chlorophyll Maximum." *Science Robotics* 6: eabb9138. <https://doi.org/10.1126/scirobotics.abb9138>.

Supporting Information

Additional supporting information can be found online in the Supporting Information section. **Table S1:** Detailed Instrumentation Specification Table. **Table S2:** Evaluation Framework for Instrument Training and Deployment Effort Scales. **Table S3:** Suggested Sampling Instrumentation Specifications Reporting. **Table S4:** Suggested Analytical Device Specifications Reporting. **Data S1:** edn370170-sup-0005-Supinfo.zip.