# Microplastic and Phthalate Esters Occurrences in *Oreochromis niloticus* and *Clarias gariepinus* Obtained from Lagos and Epe Lagoons

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# Chapter One

# Introduction

## 1.1 Background of the Study

Plastics are ubiquitous environmental pollutants that accumulate in marine, freshwater, and terrestrial ecosystems[1,2]. In developing countries, the production, usage, and improper disposal of plastics have led to an increase in environmental pollution[3]. Over the years, plastic products have become widely and extensively used across various sectors, including agriculture, industry, and personal care[4]. In 2020, global plastic production surpassed 360 million tons and is projected to reach 1.1 billion tons by 2050. A significant portion of this plastic, over 79%, is classified as single-use and is often discarded into the environment[5]. This waste and its improper disposal have raised concerns about plastic waste pollution[6].

As plastics degrade, they fragment into smaller particles, with those less than 5 mm in size referred to as microplastics (MP)[7]. Microplastics can be produced by degrading fragments of major plastics or as raw material to manufacture plastics[8]. Several recent investigations have emphasized the potential environmental and health hazards microplastics pose, prompting further research into their distribution, sources, and persistence in different environments[8]. Due to their large surface area and hydrophobic nature, microplastics can act as vectors for various contaminants, including toxic heavy metals, pathogens, and chemical pesticides[9].

This increase in microplastics has led researchers to identify and classify plastics and microplastics as important threats to marine and aquatic environments[10]. In recent years, global plastic production has surged to 359 metric tons (Mt) from 348 (Mt) in just two years. More so, the global plastic manufacturing industry has expanded significantly from a mere 0.5 Mt per year in the 1960s to 348 Mt in 2017, paving the way for the present global abundance of plastic

items[1,2]. Projections suggest that by the year 2030, plastic emissions released into the aquatic environment will hit 53Mt across 173 countries of the world[3]. A significant portion of plastic waste (between 4.8 and 12.7 million Mt) will end up in the world's oceans, intensifying marine pollution[10]. Due to its wide-ranging impact on ecosystems, plastic pollution has become a dominant global issue. These plastic fragments are categorized by size, including nanoplastics (<0.001 mm), microplastics (MPs, ≥0.001 mm and <5 mm), mesoplastics (≥5 mm and <25 mm), and macroplastics (≥25 mm)[10]. The adverse effects of macroplastics through entanglement, choking, and strangulation of animals are some of the well-documented impacts of macroplastics[11]. Macroplastics are often considered the face of plastic pollution because of their large size, which makes them more recognizable and impactful in illustrating the scale of plastic waste[11].

Microplastics have been recognized as the most prevalent form of solid waste on Earth even as they are almost invisible to the naked eye[11]. Microplastics are complex, with many different constituent chemical mixtures and contaminants as components[10]. Due to their minute size, they are easily ingested by a wide range of marine organisms, including fish and plankton, leading to varying levels of ecological disruption[12]. The ingestion of microplastics can cause physical harm, reduce feeding, impair growth, and negatively affect reproduction in aquatic species[13].

Microplastics have been found to be vectors of poisonous substances, such as persistent organic pollutants (POPs), heavy metals and pathogenic microorganisms, which can enter the food web when marine organisms ingest these particles[14.] When marine species ingest these microplastics, they are also exposed to these toxins, which can bioaccumulate in their tissues, potentially leading to physiological stress, oxidative damage, and impaired immune function[15]. The consumption of contaminated seafood by humans raises concerns about health risks, including

inflammation, oxidative stress, and potential cellular toxicity[16]. Although the full extent of these health implications is still under scientific investigation, the existing findings suggest a need for further research into the origin, distribution, sources, and fate of microplastics in various environments[16].

One significant challenge in assessing the potential health hazards of microplastics to humans stems from the limited availability of extensive data regarding the extent to which people are exposed to microplastics[1]. This gap is largely due to the ongoing and continuous development of techniques for accurately measuring MPs in the air, water, food, and cosmetics[1]. Moreover, the current detection method, primarily based on spectroscopy, typically describes microplastics (such as items, fibres, or particles) in terms of their count, size, and shape, whereas in the field of exposure science, doses are usually expressed in terms of their mass[1]. This inconsistency presents a significant barrier to understanding fully the health risks posed by microplastics to human well-being[3].

The Lagos Lagoon in Nigeria, part of an extensive lagoon network, is highly susceptible to microplastic pollution[17]. This vulnerability is primarily due to the area's high population density and insufficient waste management systems. The lagoon is essential for local fisheries and aquaculture, serving as a significant source of livelihood and food security for the surrounding communities[17]. There is a lack of comprehensive data on the extent and effects of microplastic contamination in the Lagos Lagoon despite the lagoon's critical importance[18].

This challenge predominantly arises due to the ongoing refinement of methods used to accurately measure the presence of microplastics in the atmosphere, water, food, and cosmetics[3]. Food and water were thought to be the primary sources of microplastic exposure. However, several studies have reported a per capita intake of MPs through ingestion of food, water, dust and inhalation of

air of 74,000–121,000 items annually[1]. In another study, the annual per capita MP intake of 39,000–52,000 items, including 37–1000 from sea salt, 4000 from tap water, and 11,000 from shellfish[3].

Moreover, exposure to microplastics can have significant reproductive and developmental consequences for fish. Studies have demonstrated that microplastics can interfere with hormonal processes, leading to disrupted reproductive behaviors, reduced fertility, and adverse developmental outcomes in offspring[19]. For example, fish exposed to microplastic contaminants have shown altered reproductive behaviors and reduced egg production and hatch rates, suggesting that microplastic pollution could have long-term effects on fish populations[20]. The effects of microplastic pollution not only affect fish but also degrade water quality, posing broader ecological risks. Their vector ability encourages accumulation of chemical contaminants, including pesticides, heavy metals, and antibiotics, which they can absorb and transport throughout the water column[15]. Accumulation of these contaminants in water bodies can further degrade water quality and negatively impact aquatic habitats, affecting a wide range of plant and animal life that depend on clean water[15].

Additionally, microplastics can alter the physical properties of water. High concentrations of these particles can increase water turbidity, reducing light penetration and potentially impacting photosynthetic activity in aquatic plants[21]. This reduction in photosynthesis can lower oxygen levels in the water, which is crucial for the survival of many aquatic organisms, thereby disrupting entire food webs and ecosystem dynamics[21].

Microplastics also provide surfaces for microbial colonization, leading to the formation of biofilms that can change the composition of microbial communities in water bodies[22]. These

changes can affect key ecological processes such as nutrient cycling and organic matter decomposition, further impacting the overall health of aquatic ecosystems.

Moreover, microplastics can harbor pathogenic bacteria, which could pose health risks to aquatic organisms and, indirectly, to humans who consume contaminated water or seafood[23].

Probabilistic lifetime exposure model predicted an MP intake rate of 184 ng/capita/d for children and 583 ng/capita/d for adults, through nine different exposure sources. Mass (or weight) based estimates of annual MP ingestion were reported to be 15–287 g/person[1]. Through analysis of the faeces of cats and dogs, as surrogates for humans. Furthermore, a report by a study gave an estimate of 0.03–677 mg/week for pet animals[8].

Recent technological advancements have significantly improved the detection and analysis of microplastics in environmental samples. Developing techniques such as Fourier-transform infrared spectroscopy (FTIR), Raman spectroscopy, and pyrolysis-gas chromatography-mass spectrometry (Pyr-GC/MS) now enable researchers to identify polymer types and trace the sources of microplastics more accurately[24]. These methods provide critical insights into the distribution and composition of microplastics, supporting more robust environmental risk assessments[25].

Addressing the issue of microplastic pollution requires not only scientific research but also effective policy and management strategies. Several countries and regions have already implemented measures to reduce plastic waste, such as banning single-use plastics and promoting recycling[26.] Understanding the regional context and legislative frameworks is crucial for developing strategies to mitigate microplastic pollution in the Lagos Lagoon and similar environments[18].

Microplastic pollution has significant economic consequences, especially for communities surrounding the Lagos Lagoon that depend largely on fishing and aquaculture. The contamination of fish stocks threatens food security and diminishes the economic value of fisheries, thereby impacting livelihoods and local economies[27].

### 1.2 Statement of the Problem

Microplastics have been found and reported in different environmental matrices, ranging from freshwater, the ocean, sediments, soil, and the atmosphere, even in areas without human inhabitants[1]. Microplastic pollution is a pressing environmental concern, especially in aquatic ecosystems, where it risks both marine life and human health. The Lagos Lagoon in Nigeria, part of an extensive lagoon complex, is particularly susceptible to microplastic pollution due to its dense urban population, high levels of industrial activity, and inadequate waste management practices[17]. Despite its ecological and economic importance, there is a lack of comprehensive research on microplastics' presence, distribution, and impact in this lagoon, explicitly focusing on fish species, benthic environments, and surface waters[18].

This knowledge gap in microplastic research makes it challenging to fully understand the extent of microplastic pollution and its consequences for the health of the lagoon and the well-being of the communities that depend on it. In aquatic systems, microplastics can be suspended in the water column, settle in sediments, or be ingested by marine organisms, leading to physical and chemical harm. For instance, microplastics can cause internal blockages in fish and other marine animals, reduce nutrient absorption, and introduce toxic chemicals that adhere to their surfaces[27]. In the Lagos Lagoon, microplastic pollution is primarily driven by urban runoff, improper waste disposal, and industrial discharges[18]. However, limited data exist on the types, concentrations,

and distribution of microplastics in the water column and sediments of the lagoon[18]. Understanding the prevalence of microplastics in these areas is crucial, as these particles can affect not only aquatic organisms but also the water quality and overall health of the ecosystem.

Communities around the Lagos Lagoon heavily rely on fishing and aquaculture for their livelihoods and food security[17]. Contaminated fish stocks not only pose a threat to food safety but also reduce the economic value of fisheries, thereby affecting local economies and livelihoods[5].

Microplastics have been detected in edible fish species, indicating a possible route of human exposure through seafood consumption. Microplastics have also been identified to induce several types of impairments in species. This microplastic-induced impairment ranges from minimal biological system disturbance to very large undesirable effects that lead to death[29].

Microplastics have been reported in various fish and other species that are high up in the food chain[18.] In planktivorous fish, microplastics have also been found and biomagnified in bigger fish that prey on the planktivorous fish[1]. Human exposure to microplastics has been reported through various pathways, including seafood consumption, suggesting potential for bioaccumulation. Microplastics in the water column can adversely affect water quality and aquatic life. As microplastics accumulate in the water, they can act as vectors for other pollutants, such as heavy metals, pesticides, and persistent organic pollutants[28]. These pollutants can attach to the surfaces of microplastics, making them more readily available for uptake by aquatic organisms. The ingestion of microplastics by fish and other aquatic species can lead to a range of negative health effects, including reduced feeding, energy depletion, and impaired reproductive functions[29].

Moreover, the presence of microplastics in the water can affect photosynthesis in aquatic plants and disrupt the natural cycling of nutrients, further degrading the water quality and overall health of the lagoon[30]. Fish and benthic (bottom-dwelling) organisms are particularly vulnerable to microplastic pollution[28]. Fish can ingest microplastics directly from the water or indirectly through their diet, while benthic organisms are exposed to microplastics that settle in the sediment[28]. Ingestion of microplastics by fish has been linked to various harmful effects, such as inflammation, oxidative stress, and reproductive issues[13]. These effects not only threaten fish populations but can also disrupt the food web and ecosystem stability. While global studies have highlighted these risks, there is a lack of region-specific data on the socio-economic and health impacts of microplastic pollution in the Lagos and Epe Lagoons.

### 1.3 Justification of the Study

The Lagos Lagoon plays a pivotal role in Nigeria's coastal ecosystem and crucial in maintaining biodiversity and ecological balance. It is a habitat for numerous fish species, benthic organisms, and other aquatic life forms. However, the growing presence of microplastic pollution increasingly threatens the lagoon, with the potential to alter food chains and damage aquatic habitats[18]. There are various sources, including the breakdown of larger plastic debris, synthetic fibers from clothing, and microbeads from personal care products[4]. Marine organisms can ingest these particles, causing physical harm and introducing toxic substances into their bodies, which can subsequently affect higher trophic levels, including humans[5].

Lagos Lagoon is an important habitat for a wide array of estuarine and marine species, characterized by high biodiversity and feeding grounds for marine and coastal biota. It is also a source of fish supply supporting a large human population[17]. This lagoon also receives a lot of waste, some of which may include microplastics from numerous industrial and other

anthropogenic activities[18]. Moreover, there is growing evidence that microplastics in the marine environment can absorb and concentrate harmful chemicals from surrounding waters, such as persistent organic pollutants (POPs) and heavy metals[28].

When aquatic organisms ingest microplastics, these toxic substances can bioaccumulate and biomagnify through the food web, ultimately reaching humans who consume seafood. This has serious implications for public health, as prolonged exposure to such toxins can lead to various health issues, including endocrine disruption, carcinogenic effects, and other chronic health conditions[7].

This research can provide the evidence needed to engage communities and foster a collective approach to tackling plastic pollution, which is essential for achieving sustainable development goals related to clean water, marine life, and sustainable communities[8].

Despite the increasing global focus on microplastic pollution, there is a notable gap in localized data concerning the Lagos Lagoon and similar tropical and subtropical ecosystems in West Africa. While studies have documented microplastic pollution in several regions, the specific dynamics, sources, and ecological impacts in tropical lagoons like Lagos remain under-researched[17,18].

The ecological significance of the Lagos Lagoon, coupled with its vulnerability to pollution, underscores the necessity of this study. Understanding the distribution, concentration, and characteristics of microplastics in the lagoon's waters and sediments is essential for assessing their impact on aquatic life and ecosystem health. Additionally, this study will explore how environmental factors such as seasonality and sediment characteristics influence microplastic pollution, providing essential data to inform future mitigation and management efforts.

## 1.4 Aim and Objectives of the Study

This study is aimed at analyzing microplastic pollution in *Oreochromis niloticus and Clarias gariepinus* fishes, and also benthic and surface water from three high population density zones of Lagos (Makoko and Ikorodu axis) and Epe Lagoons.

The specific objectives of the study are to;

1. determine the morphometric characteristics of *Oreochromis niloticus* and *Clarias gariepinus* from the Lagos and Epe Lagoons
2. determine the physicochemical parameters in benthic and surface waters of the Lagos and Epe Lagoons.
3. determine the microplastic concentration in *Oreochromis niloticus* and *Clarias gariepinus* fishes from the Lagos and Epe Lagoons.
4. determine the phthalate ester concentration in *Oreochromis niloticus* and *Clarias gariepinus* fishes from the Lagos and Epe Lagoons.
5. determine seasonal variations in microplastic concentration in fishes from the Lagos and Epe Lagoons.
6. make policy recommendations for plastic waste management and environmental protection based on the findings.

## 1.5 Research Questions

The research is geared to resolve the following questions:

I. What are the morphometric characteristics of *Oreochromis niloticus* and *Clarias gariepinus* from the Lagos and Epe Lagoons?

II. What are the levels and types of microplastics found in *Oreochromis niloticus* and *Clarias gariepinus* fishes from the Lagos and Epe Lagoons?

III. What are the physicochemical properties and microplastic contamination levels of the benthic and surface waters from the Lagos and Epe Lagoons?

IV. What are the levels of phthalate esters present in the fish tissue?

V. Are there seasonal variations in microplastic contamination levels in fish?

VI. How can the findings of this study inform policy decisions related to plastic waste management and environmental protection?

## 1.6 Significance of the Study

This study seeks to address some of the critical gaps in current research by focusing on the impact of microplastics on fish, a key component of aquatic ecosystems and an important food source for humans. Through a combination of controlled laboratory experiments and rigorous data analysis, this research aims to provide new insights into the ways microplastics interact with aquatic organisms, particularly fish, and to assess the broader ecological implications of these interactions. The findings from this research will not only contribute to the scientific understanding of microplastics but also provide valuable information for policymakers, environmental managers, and public health officials working to address the growing challenge of plastic pollution.

## 1.7 Scope of the Study

The scope of this study is on the impact of microplastics on fish and encompasses several key areas. This study focuses on three aquatic environments, which include the Lagos lagoon (Makoko and Ikorodu axis) and Epe lagoon, where the fish species are collected for analysis. This study targets two fish species (*Oreochromis niloticus* and *Clarias gariepinus*) that are representative of the local ecosystem and are of particular interest due to their ecological significance and commercial value in Lagos State. The study spans a defined period during

which samples are collected and experiments are conducted. This period ranges from January to November, allowing for the observation of seasonal variations and long-term effects. This research involves both fieldwork and laboratory analysis, focusing on detecting and quantifying microplastics in fish and water. It also includes studying the physical and chemical impacts of microplastics on fish health, behavior, and physiology. The study examines the broader ecological implications of microplastic contamination in aquatic ecosystems, including potential impacts on food webs and ecosystem services. The research also considers the potential human health implications of microplastic contamination in fish, contributing to policy discussions on plastic pollution and food safety.

## 1.8 Limitation of the Study

This research focused on a limited number of fish species, which may not fully represent the diversity of responses to microplastics across all fish species or other aquatic organisms. The findings may not be generalizable to species not included in the study. The study is constrained by time, limiting the assessment of long-term effects of microplastic exposure on fish populations. However, it does capture seasonal variations in microplastic concentrations in the fish species, providing insight into the cumulative impact of microplastics over extended periods. Limited sample sizes due to resource constraints might affect the statistical power of the study, potentially leading to less robust conclusions. Laboratory experiments often involve controlled conditions that do not fully replicate the complexity of natural environments. As a result, the effects of microplastics observed in the lab might vary from those in the wild. Gaining access to the water bodies was also a challenge, as community members in the study locations were reluctant and demanded unreasonable rewards for granting access.

## 1.9 Operational Definition of Terms

**Microplastics:** Microplastics are small plastic particles, typically between 0.001 – 5 millimeters in size, that originate from the breakdown of larger plastic debris or are manufactured as microbeads, fibers, or pellets.

**Pollution:** This refers to the presence or introduction into the environment of a substance which have harmful or poisonous effects.

**Fish:** Fish are cold-blooded vertebrate animals with gills and fins living wholly in water.

**Benthic Water:** This refers to water from the bottom of a body of water.

**Surface Water:** This refers to the top layer of a body of water.

**Ingestion:** Ingestion is the consumption of a substance by an organism through the mouth.

**Contamination:** This is the addition of an undesirable and potentially harmful substance to the environment.

**Bioaccumulation:** Bioaccumulation refers to the gradual accumulation of substances, such as pesticides or other chemicals in an organism.

**Ecosystem:** Ecosystem is a community of living organisms in a particular area, interacting with each other and with their physical environment.

**Toxicity:** This is the degree to which a chemical substance or a mixture of substances can damage an organism.

**Sample:** A sample is any type of biological material derived from a living organism that can be studied and analyzed in a laboratory.

**Aquatic Environment:** This refers to any ecosystem found in and around a body of water.

**Marine Organisms:** These are organisms that live in or depend on the ocean and other saltwater environments for survival.

## Endnotes


1. A. O. Adeogun, O. R. Ibor, E. A. Khan, A. V. Chukwuka, E. D. Omogbemi & A. Arukwe, *Detection and Occurrence of Microplastics in the Stomach of Commercial Fish Species from a Municipal Water Supply Lake in Southwestern Nigeria*, **Environmental Science and Pollution Research**, 27, 2020, 31035–31045. doi: 10.1007/s11356-020-09031-5.

2. A. M. Elgarahy, A. Akhdhar & K. Z. Elwakeel, *Microplastics Prevalence, Interactions, and Remediation in the Aquatic Environment: A Critical Review*, **Journal of Environmental Chemical Engineering**, 9(5), 2021, 106224.

3. G. Thushari & J. D. M. Senevirathna, *Plastic Pollution in the Marine Environment*, **Heliyon**, 6, 2020, 8.

4. L. Yang, Y. Zhang, S. Kang, Z. Wang & C. Wu, *Microplastics in the Marine Environment: Sources, Distribution, and Pathways*, **Environmental Science: Processes & Impacts**, 23(2), 2021, 167–184.

5. S. Gupta, R. K. Singh, A. Kumar, S. Kumar & R. Sinha, *Plastic Pollution in the Marine Environment: A Review of its Impacts, Sources, and Mitigation Strategies*, **Journal of Environmental Management**, 302, 2022, 114908. https://doi.org/10.1016/j.jenvman.2021.114908.

6. S. Ali, S. R. Naqvi, M. Naqvi & M. Islam, *Global Plastic Waste Management: Challenges and Opportunities*, **Marine Pollution Bulletin**, 173, 2021, 112998.

7. L. V. Raamsdonk, M. van der Zande, A. A. Koelmans, R. L. A. P. Hoogenboom, R. J. B. Peters, M. J. Groot & A. A. C. Peijnenburg, *Current Insights into Monitoring, Bioaccumulation, and Potential Health Effects of Microplastics Present in the Food Chain*, **Foods**, 9(1), 2020, 72.

8. X. Zhang, X. Kang, X. Liu & J. Li, *Sources, Distribution, and Fate of Microplastics in River Sediments: A Global Review*, **Environmental Research**, 220, 2023, 115179.

9. X. Yang, Z. Wang, Y. Zhang, J. Li, Y. Wu & X. Zhao, *Microplastics in Freshwater Sediments: Current Status and Knowledge Gaps*, **Water Research**, 198, 2021, 117104.

10. W. Lau, Y. Shiran, R. M. Bailey, E. Cook, M. R. Stuchtey, J. Koskella, C. A. Velis, L. Godfrey, J. Boucher, M. B. Murphy, R. C. Thompson, E. Jankowska, A. C. Castillo, T. D. Pilditch, B. Dixon, L. Koerselman, E. Kosior, E. J. Favoino, J. Gutberlet & J. E. Palardy, *Evaluating Scenarios Toward Zero Plastic Pollution*, **Science (New York)**, 369(6510), 2020, 1455–1461.

11. S. Bhuyan, *Effects of Microplastics on Fish and in Human Health*, **Frontiers in Environmental Science**, 10, 2022, 827289.

12. J. Chen, J. Liu, X. Wei, P. Zhang & G. Zhuang, *How to Build a Microplastics-Free Environment: Strategies for Microplastics Degradation and Plastics Recycling*, **Advanced Science**, 9(6), 2022, 2103764.

13. Y. Lee, J. Cho, J. Sohn & C. Kim, *Health Effects of Microplastic Exposures: Current Issues and Perspectives in South Korea*, **Yonsei Medical Journal**, 64(5), 2023, 301.

14. D. He, Y. Luo, S. Lu, M. Liu, Y. Song & L. Lei, *Microplastics in Terrestrial Ecosystems: A Scientometric Analysis*, **Sustainability**, 12(20), 2020, 8739.

15. W. Wang, J. Ge & X. Yu, *Bioavailability and Toxicity of Microplastics to Fish Species: A Review*, **Ecotoxicology and Environmental Safety**, 173, 2019, 110–117.

16. J. C. Prata, J. P. da Costa, I. Lopes, A. C. Duarte & T. Rocha-Santos, *Environmental Exposure to Microplastics: An Overview on Possible Human Health Effects*, **Science of the Total Environment**, 702, 2020, 134455.

17. I. B. Ogundele, A. Adebayo & S. K. Ojo, *Microplastic Pollution in Nigerian Aquatic Environments: A Review of the Current State and Implications for Future Research*, **Journal of Environmental Science and Pollution Research**, 28(9), 2021, 11544–11557.

18. T. Ojo, I. O. Akinyemi & A. A. Ojo, *Microplastic Contamination in Lagos Lagoon, Nigeria: Sources, Distribution, and Ecological Implications*, **Environmental Pollution**, 307, 2023, 119580.

19. S. M. Brander, V. C. Renick, M. M. Foley, C. Steele, M. Woo, A. Lusher & S. Carr, *Microplastics Impact on Animal Behavior and Physiology: A Call for Integrated Research on Social-Ecological Systems*, **Environmental Toxicology and Chemistry**, 39(5), 2020, 1117–1130.

20. B. E. Trotter Bailey & J. C. Wingfield, *Reproductive Success and Embryonic Development are Impacted by Exposure to Microplastics in Aquatic Species*, **Environmental Science and Pollution Research**, 26(7), 2019, 6534–6543.

21. E. Besseling, P. E. Redondo-Hasselerharm, E. M. Foekema & A. A. Koelmans, *Quantifying Ecological Risks of Aquatic Micro- and Nanoplastic*, **Critical Reviews in Environmental Science and Technology**, 49(1), 2019, 32–80.

22. A. Amaral-Zettler, E. R. Zettler & T. J. Mincer, *Ecology of the Plastisphere*, **Nature Reviews Microbiology**, 18(3), 2020, 139–151.

23. A. Keswani, D. M. Oliver, T. Gutierrez & R. S. Quilliam, *Microbial Hitchhikers on Marine Plastic Debris: Human Exposure Risks at Bathing Waters and Beach Environments*, **Marine Environmental Research**, 146, 2019, 1–10.

24. M. Kiendrebeogo, L. Ceccato, B. Letten, A. De Girolamo, B. Teychene & L. De Lary, *Treatment of Microplastics in Water by Anodic Oxidation: A Case Study for Polystyrene*, **Environmental Pollution**, 269, 2021, 116168.

25. Y. Zhang, S. Kang, S. Allen, D. Allen, T. Gao & M. Sillanpää, *Atmospheric Microplastics: A Review on Current Status and Perspectives*, **Earth-Science Reviews**, 203, 2020, 103118.

26. European Commission, *Reducing Marine Litter: Action on Single-Use Plastics and Fishing Gear*, **European Union**, 2018. https://ec.europa.eu/environment/pdf/waste/plastics/reducing_marine_litter_brochure.pdf.

27. A. M. Barboza, A. D. Vethaak, B. R. B. Lavorante, A. K. Lundebye & L. Guilhermino, *Marine Microplastic Debris: An Emerging Issue for Food Security, Food Safety, and Human Health*, **Marine Pollution Bulletin**, 133, 2020, 336–348.

28. N. D. van der Hal, Angel & Z. Ronen, *Dynamics of Heavy Metals and Persistent Organic Pollutants in Plastic Debris*, **Marine Pollution Bulletin**, 146, 2020, 423–432.

29. C. M. Rochman, T. Kurobe, I. Flores & S. J. Teh, *Early Warning Signs of Endocrine Disruption in Adult Fish from the Ingestion of Polyethylene with and without Sorbed Chemical Pollutants From the Marine Environment*, **Science of the Total Environment**, 493, 2020, 656–661.

30. J. Alava, *Modeling the Bioaccumulation and Biomagnification Potential of Microplastics in a Cetacean Foodweb of the Northeastern Pacific: A Prospective Tool to Assess the Risk Exposure to Plastic Particles*, **Frontiers in Marine Science**, 7, 2020, 566101.

31. K. Kannan & V. Krishnamoorthi, *A Review of Human Exposure to Microplastics and Insights into Microplastics as Obesogens*, **Frontiers in Endocrinology**, 12, 2021, 724989.

# Chapter Two

# Literature Review

## 2.1 Plastic Production

Plastic production has exponentially increased since its inception in the early 20th century, making it one of modern society's most versatile and widely used materials. In 2021, global plastic production exceeded 390 million metric tons, marking a continuous upward trend in production volumes driven by the material's extensive application in various sectors such as packaging, automotive, construction, electronics, and consumer goods[1]. Plastic production is continually rising, making plastic pollution a critical modern environmental concern. Plastic is a highly functional material known for its durability, decomposition resistance, stability, shape versatility, and cost-effectiveness. As a result, it has gained significant importance in our daily lives as a standard material of choice[2].

Approximately 300 million tons of plastic materials are produced anually globally, marking a more than 20-fold increase over the past six decades[1]. In Europe, only around 30% of plastic materials are recycled. Roughly 10% of the yearly plastic production finds its way into the ocean, with about 8 million plastic pieces escaping from terrestrial sources into the oceans daily, primarily through rivers[1]. Plastic materials upon deposition in the ocean, can become entangled in ocean currents and carried far into the open sea[4].

In the natural environment, plastic is highly resilient and can take centuries to break down. Only a few naturally occurring microorganisms can decompose these synthetic materials, hence, the biodegradation of plastics occurs very slowly[1]. Sunlight, wind, and waves continuously fragment plastic into smaller particles. Although this photodegradation process can proceed to the molecular level, the resulting degradation products still consist of polymers and may contribute

to the dissolved organic carbon content in the ocean[5]. Plastics frequently incorporate additives to improve their mechanical characteristics, flexibility, longevity, stability, and color. During the weathering and deterioration of plastic materials, these additives can leach into the seawater and are then consumed by marine organisms[2].

Plastic products are majorly derived from fossil fuels, including natural gas and crude oil, which are extracted through refining and drilling. These petrochemical feedstocks, such as ethylene and propylene, form the basis of most conventional plastics. In recent years, efforts have been made to produce bioplastics from renewable biomass sources such as corn, sugarcane, or other plant-based resources[6]. While bioplastics are considered more environmentally friendly, their production still presents challenges regarding energy use and disposal[6]. Polymerization, a chemical process that transforms small molecules called monomers into long chains known as polymers, is the heart of plastic production. The two primary types of polymerization are addition polymerization and condensation polymerization[6].

In **addition polymerization**, monomers such as ethylene and propylene are added together in a chain reaction, creating common plastics like polyethylene, polypropylene, and polystyrene. Due to their lightweight and flexible properties, these materials are widely used in packaging, textiles, and consumer goods[7].

**Condensation polymerization**, on the other hand, occurs when monomers join and lose small molecules, such as water, during the reaction. This process produces materials like nylon and polyester, which are important for textiles and industrial applications. These polymerization methods create the base materials for plastics that can be shaped into numerous products[7]. Once polymers are produced, they are processed into pellets or granules to facilitate transportation and handling. These plastic pellets are sold to manufacturers who melt them down and shape them

into final products through various techniques, including extrusion, injection molding, and blow molding[8]. Plastics are often combined with various additives to enhance their properties. These additives can improve durability, flexibility, color, heat and UV light resistance. Common additives include stabilizers, plasticizers, colorants, and flame retardants[6]. While these additives increase the functionality of plastic products, they can also complicate recycling efforts and pose additional environmental risks when plastics are improperly disposed of[6].

Plastics are generally categorized based on their polymer structure and use. The most common types of plastics include polyethylene (PE), polypropylene (PP), polyvinyl chloride (PVC), polystyrene (PS), polyethylene terephthalate (PET), and polycarbonate (PC)[9]. PE and PP account for the largest share of global plastic production, primarily utilized in packaging materials such as plastic bags, bottles, and containers. PET is widely used in the production of beverage bottles, while PVC is extensively used in the construction industry for pipes, window frames, and flooring. Each type of plastic is synthesized through various polymerization processes, with the choice of process and raw materials significantly influencing the properties and applications of the final product[9].

The growth of plastic production over the decades has been influenced by a number of factors. Factors such as Economic development, population growth, and urbanization have propelled the demand for plastic products, particularly in emerging markets in Asia, Africa, and Latin America[8]. While technological advancements have expanded the applications of plastics in high-performance sectors such as electronics, aerospace, and medical devices, the increase in disposable incomes has led to higher consumption of packaged goods[7].

The automotive and packaging industries have further fueled the demand for plastics due to the shift towards lightweight and durable materials. Plastic is now used in the automotive industry, to reduce vehicle weight, improve fuel efficiency, and lower greenhouse gas emissions. Similarly, in the packaging industry, plastics are relied on for their barrier properties, which help extend the shelf life of food products and reduce food waste[6].

Furthermore, plastics persistence in the environment has led to widespread pollution, particularly in marine and freshwater ecosystems. Plastics are highly resistant to degradation, and as a result, they can accumulate in the environment for centuries, breaking down into smaller particles known as microplastics. These microplastics have now been found in virtually all environmental matrices, including oceans, rivers, soils, and even the air, posing risks to wildlife and human health[7].



## 2.2 Degradation of Plastics Materials into Microplastics

Plastics exhibit remarkable resistance to degradation when exposed to natural conditions. In marine environments, the breakdown of plastics is an incredibly slow process, taking several centuries[2]. This decomposition is driven by a combination of factors, including sunlight, air (oxygen), heat, and moisture. Photodegradation is a key process where the material transforms due to exposure to irradiation and the absorption of photons. As a result of degradation, plastics change color, physical properties, surface characteristics, the development of visible flaws like cracks, and the fragmentation of plastic into smaller particles, ultimately forming microplastics[10]. Exposure to sunlight, particularly ultraviolet light, is the primary catalyst for plastic degradation. The absorption of light with sufficient energy (shorter wavelengths) breaks chemical bonds, leading to both physical and chemical alterations (oxidation) and the fragmentation of the polymeric material[11]. Plastic degradation can also produce very tiny plastic fragments or even

dissolved polymer compounds that become bioavailable to microorganisms[12]. While these microorganisms can metabolize the polymer compounds into carbon dioxide ($CO_2$), this process is exceedingly slow.

A study observed numerous alterations in the characteristics of microplastics during a simulated ageing process[13]. These changes encompassed an increase in carbonyl content, an expansion of the specific surface area, and a color modification. Surface fragmentation and the development of cracks allowed light and oxygen to penetrate internal surfaces, leading to further oxidation of the microplastics. Additionally, this ageing process resulted in a heightened release of pigments[14]. Various polymer types exhibited varying degradation rates when exposed to natural marine conditions. For instance, polyvinyl chloride (PVC) swiftly released estrogenic compounds into seawater, whereas polyethene terephthalate (PET) displayed minimal surface alterations with no detected estrogenic activity. Meanwhile, poly (butylene adipate-co-terephthalate) (PBAT), a copolymer, exhibited a heterogeneous surface with some cavities after weathering[13]. Acrylonitrile butadiene styrene (ABS), upon exposure to a marine environment, underwent a smoother texture, discolouration, and developed a deformed, fractured, and fragmented structure. Additionally, the plastic surface accumulated fouling, and there was leaching of additives and pigments from the material matrix[14.] Over time in the marine environment, the molar mass of plastic particles decreased, indicating degradation of the polymer chain.

Studies have also shown that artificial photo degradation has been shown to result in an increase in VOCs, including Carbonyls, lactones, esters, acids, alcohols ethers, and aromatics[15]. An investigation into the impact of artificial seawater on beads made from polyethylene (PE) revealed notable structural and morphological transformations[1].The presence of artificial seawater caused substantial microcracking on the surface of the pellets, the generation of

oxidized functional groups, modifications in the thermal stability of the PE pellets, and an augmentation in the organic matter content of the water in which the pellets were immersed.

The degradation of many plastic materials led to a noticeable increase in the release of particles into the surrounding solution, with up to several million particles per milliliter detected after 112 days of degradation[2]. In a study focusing on the artificial photodegradation and fragmentation of polyethylene (PE) films in both air and water, it was found that fragmentation only occurred in water, even though exposure to air did result in higher levels of oxidation[14]. In addition, after 25 weeks of weathering in water, around 90% of the fragments measured under 1mm in size and exhibited very similar shapes, with micrometric fragments not yet being prevalent. As a result, weathering in water demands extended durations and multiple stages of fragmentation before micro-sized particles are generated[16].

The exposure of polystyrene (PS) microparticles to simulated sunlight irradiation, also known as photoaging, has been linked to the generation of reactive oxygen species such as singlet oxygen and hydrogen peroxide. These species are highly reactive with PS microparticles, causing chemical changes[17]. Prolonged photoaging results in the formation of more oxidative functional groups and increased negative charges on the particle surfaces. This leads to stronger electrostatic repulsion and slower settling of the microparticles in water[18,19].

Different polymeric materials exhibit distinct degradation patterns. For instance, a study investigating the accelerated weathering of high-density polyethylene (HDPE), high-impact polystyrene (HIPS), nylon 6, and polypropylene (PP) under UV light in simulated seawater found that HDPE and nylon 6 broke down into microfibers, whereas HIPS and PP did not physically degrade[14,17].

Pellets made from PP, PE, and PS exposed to UV light in air, ultrapure water, and simulated seawater all displayed an increase in the number of oxidized functional groups[20]. The type of reaction medium was shown to influence the rates of photochemical weathering, although the formation of cracks and flakes was a common characteristic of weathering in all the media examined[21].

Another study noted that biodegradation is a natural phenomenon in which microorganisms break down or alter the composition of organic substances. This process is influenced by various factors, including environmental conditions (such as temperature, moisture, pH, nutrient availability), the characteristics of the substrate being degraded (like molecular weight, structure, functional groups, crystallinity, and cross-linking), and the specific microorganisms participating (including their extracellular enzymes, hydrophobicity, and population size)[20].

Plastics, due to their resistant nature, do not readily undergo biodegradation, and the natural breakdown of plastics in aquatic environments can take several years. The process by which plastics biodegrade in marine settings is highly intricate[22,14]. It begins with the formation of a microbial biofilm on the surface of the plastic, followed by the degradation and fragmentation of the polymers into smaller components like oligomers, dimers, and monomers through enzymatic activity[14]. The ultimate step involves the conversion of these smaller components into carbon dioxide ($CO_2$) and water ($H_2O$) through mineralization[23].

In a microcosm experiment by where a blend of naturally weathered plastic fragments was exposed to an indigenous pelagic community, several noteworthy outcomes were observed. There was an increase in the proportion of double bonds on the surface of microbially treated PE films, and changes in the profile of the PS films were also noted. As the incubation period

progressed, the molecular weight of the PE pieces went up, while the weight of PS pieces decreased[20]. The buoyancy of PS pieces underwent alterations due to the formation of biofilms. In addition, the microbial diversity associated with two different types of microplastics (PP and PVC) increased over time when they were exposed to seawater[13]. Furthermore, the microplastic biofilms displayed distinct microbial community structures in different marine environments. The surfaces of microplastics became enriched with bacteria capable of breaking down hydrocarbons. Similar findings were replicated in a mesocosm experiment exploring the role of hydrocarbon-degrading bacteria in the decomposition of PET[24].

Notably, substantial disparities in biofilm diversity were observed, with PET surfaces attracting unique communities of hydrocarbon-degrading bacteria[14]. There were also significant modifications in the surface chemistry and morphology of PET films, primarily attributed to the bacterial consortia enriched in tetradecane and diesel[25]. In another microcosm experiment that examined the degradation of weathered PS films under simulated marine conditions, diverse changes were observed. Specifically, two customized consortia effectively reduced the weight of PS films[26]. Fourier transform infrared (FTIR) spectrophotometry was employed to detect variations in the intensity of functional groups, in addition to indications of bio-erosion on the PS surface. These results indicate that adapted marine populations are capable of breaking down weathered PS fragments[14].



### 2.3 Types of Microplastics

Microplastics are generally classified into two main categories: primary and secondary microplastics. Each type has distinct origins and characteristics, contributing differently to environmental pollution. Both types are prevalent in various ecosystems, including marine, freshwater, and terrestrial environments.

Primary microplastics are small particles intentionally manufactured to be less than 5 mm in size. These particles are typically used in various commercial applications, including cosmetics, detergents, industrial scrubbers, and air-blasting technologies. Personal care products that contain microbeads like facial cleansers and toothpaste are typical examples of primary microplastics. These particles are introduced directly into water systems through wastewater and often bypass filtration systems in treatment plants[27]. Additionally, during washing, synthetic textile fibers are a significant source of primary microplastics. These fibers are shed from materials like polyester, nylon, and acrylic, contributing to pollution in water bodies[7].

Secondary microplastics are formed from the disintegration of larger plastic debris. Over time, environmental processes such as physical abrasion, UV radiation, and microbial activity cause plastics to break down into smaller fragments. These fragments include particles from everyday items like plastic bottles, packaging materials, and fishing gear[28]. Secondary microplastics are often formed when larger plastic products undergo wear and tear in the environment, making them more widespread than primary microplastics[28].

The degradation of plastics occurs in different stages, depending on factors like environmental exposure and the chemical makeup of the plastic. For example, polyethylene (PE) and polypropylene (PP), two of the most widely produced plastics, tend to break down into microplastics over time due to their widespread use in packaging and single-use products[6]. This process is accelerated in marine environments where plastics are exposed to saltwater, wave action, and sunlight. This contributes to the large quantities of secondary microplastics found in ocean gyres and coastal areas[29].

Fibrous microplastics are a specific form of microplastic, often originating from textiles. Synthetic fibers from clothing and fishing nets are key sources of fibrous microplastics, especially in urban and coastal environments. Depending on their source, these fibers can vary in length and diameter. Textile washing is considered one of the major contributors to microplastic pollution, as fibers are released into wastewater during laundry cycles and are not fully captured by wastewater treatment plants[27].

Fragmented microplastics are small pieces of plastic that are formed from the breakdown of larger plastic items. These fragments are often irregular in shape and can vary in size, with most measuring less than 5 mm. Fragmented microplastics typically originate from discarded plastic products like bottles, bags, and packaging, which degrade into smaller pieces due to mechanical forces or chemical reactions in the environment[28].

Pellet microplastics, often called nurdles, are small plastic granules used in the manufacturing of plastic products. They are shipped worldwide for industrial processing but can spill into the environment during transportation and handling. Pellets are typically spherical or cylindrical and are found in both marine and freshwater environments. They can absorb toxic pollutants from the environment due to their persistence and buoyancy which makes them particularly harmful [6].

## 2.4 Chemical Composition of Microplastics

Carcinogenic chemicals are now contained in many plastics as part of their production process. For example, polyvinyl chloride (PVC), a common plastic polymer, is manufactured using vinyl chloride monomer, a known human carcinogen[30]. Additionally, plastics often contain chemical additives like plasticizers (e.g., phthalates), flame retardants, and stabilizers, many of which are recognized as harmful to human health. Phthalates, for instance, are endocrine-disrupting

chemicals (EDCs) that have been linked to increased cancer risk, particularly breast and reproductive cancers[31]. When plastics degrade into microplastics, these toxic chemicals can leach into the environment, potentially increasing human exposure to carcinogenic substances through ingestion or inhalation[31].

### 2.5 Microplastics as Carriers of Carcinogenic Pollutants

Microplastics pose risks through their chemical composition and serve as carriers for other toxic compounds. Due to their hydrophobic nature, microplastics can adsorb dangerous contaminants, such as polycyclic aromatic hydrocarbons (PAHs), heavy metals, and persistent organic pollutants (POPs) from the surrounding environment. PAHs, in particular, are a class of chemicals formed during the incomplete combustion of organic materials and are known to be carcinogenic[32]. Studies have shown that microplastics act as vectors for these pollutants, transporting them into the food chain when ingested by marine organisms[6]. These pollutants can accumulate in tissues, posing potential cancer development risks for aquatic organisms and humans who consume contaminated seafood[32].

#### 2.5.1 Inhalation of Microplastics and Cancer Risk

Another significant route of human exposure to microplastics is through inhalation. Microplastics are present in both indoor and outdoor air, where they can be inhaled and accumulate in the lungs[26]. Fibrous microplastics, such as those derived from synthetic textiles, are particularly concerning because they can penetrate deep into the respiratory system. Studies have drawn parallels between inhaled microplastics and the development of respiratory diseases, including lung cancer, similar to the effects observed with asbestos fibers[26]. Inhaled microplastics may cause chronic inflammation, a known precursor to cancer, and their potential to transport toxic chemicals into lung tissues further exacerbates the risk[26].

### 2.5.2 Gastrointestinal Exposure and Carcinogenicity

With particles being detected in seafood, drinking water, and even table salt, ingestion is now noted as the primary route of human exposure to microplastics. Microplastics when ingested and passed through the gastrointestinal tract, can potentially cause physical damage to tissues, leading to inflammation. This chronic inflammation has become associated with an increased cancer risk, particularly in the digestive system[18]. Moreover leaching toxic chemicals from microplastics during digestion may disrupt cellular processes, promote oxidative stress, and damage DNA, contributing to carcinogenesis[33].

## 2.6 Review of Previous Studies on Microplastics

Research on the carcinogenic effects of microplastics is still in its infancy, but several animal studies have provided insights into their potential health impacts. Rodent studies have shown that exposure to microplastics can lead to inflammation, oxidative stress, and immune system disruption, all of which are risk factors for cancer development[18]. Moreover, studies on aquatic organisms have demonstrated the bioaccumulation of microplastics and associated toxic chemicals, raising concerns about transferring these risks through the food chain to humans[34].

The mechanisms through which microplastics may induce carcinogenesis in animal models are multifaceted. One proposed mechanism is generating reactive oxygen species (ROS) due to the oxidative stress induced by microplastic particles. ROS can damage cellular DNA, lipids, and proteins, leading to mutations that drive cancer development[35]. Additionally, microplastics may disrupt cell signaling pathways that regulate cell growth and apoptosis, potentially leading to uncontrolled cell proliferation, a hallmark of cancer[35].

Another important mechanism is chronic inflammation. Studies have shown that microplastic particles can induce swelling in a number of tissues, including the gastrointestinal tract and lungs[26,36]. Chronic inflammation creates a pro-carcinogenic environment by promoting cellular damage, altering tissue repair processes, and enhancing the survival of mutated cells[36].

### 2.7 Sources of Microplastics in the Aquatic Environment

Microplastics originate from two main sources: primary and secondary microplastics. Primary microplastics are manufactured in small sizes for cosmetics, detergents, and industrial plastic debris due to physical, chemical, and biological degradation processes[7]. Studies indicate that urban areas with inadequate waste management systems, such as those surrounding the Lagos Lagoon, significantly contribute to microplastic pollution through untreated wastewater discharge and stormwater runoff[8].

Research has highlighted the role of synthetic textiles in contributing to microplastic pollution. During washing, synthetic fibers from clothing release tiny plastic particles into the water, which ultimately find their way into rivers and oceans. Urban rivers and estuaries, like those leading into the Lagos Lagoon, are particularly vulnerable to this kind of pollution[27].

The primary sources by which microplastics enter the aquatic environment have been categorized as primary and secondary sources[37]. The primary source is the microbeads, which are small-sized (1mm) plastic particles that are included in cosmetics and toiletries that enhance their abrasive and cleaning ability[22]. Microbeads are contained in facial cleansers, facial scrubs, toilet soaps and toothpaste. Facial cleaners contain polyethylene microparticles with sizes ranging between 4.1 μm and 1.24 mm[38]. These microbeads are non-biodegradable and tiny plastic

particles end up in the open aquatic environment because they escape the treatment and filtration process for waste water[39].

The secondary source of microplastics is through the degradation of mesoplastics (5 to 25mm) and macroplastics (>25 mm). Plastics are highly resistant to microbial degradation in the aquatic environment[39]. However, they break down to smaller fragments through the fragmentation/degradation of large plastic items through weathering by abiotic factors such as ultraviolet light, water temperature, and wave and wind action[40,41]. Boring isopods damage expanded polystyrene floats of fishing docks and aquaculture facilities and expel large quantities of microplastics into the aquatic environment[39,42]. Polyester fibres from textiles also form sources of microplastics that enter the aquatic environment through domestic and industrial waste water[43]. More so, Secondary sources of microplastics encompass various scenarios, including deliberate discharge (such as illegal dumping), improper disposal of waste (like litter), or accidental losses (abandoned fishing gear or cargo lost from shipping vessels). The extent and significance of these various sources and pathways for microplastic release can differ between terrestrial, freshwater, and marine environments[22].

### 2.8 Marine Plastic Pollution

Plastic pollution is now recognized as a significant threat to marine biodiversity on a global scale. It is prioritized alongside other human-induced stressors like climate change, rising sea levels, overfishing, and the spread of invasive species, among scientists and the general public[44]. It is estimated that each year, approximately 10% of the total global plastic production is either intentionally or accidentally released into marine ecosystems[45.] Recent assessments have placed the annual input of plastic waste into the world's oceans in the range of 4.8 to 12.7 million tons[1]. Some estimates suggesting there are as many as 5 trillion individual plastic pieces currently

floating at the ocean's surface[2]. This unprecedented influx of plastic into marine environments has led to floating masses of debris accumulating in ocean gyres[2]. For instance, the Great Pacific Garbage Patch, located in the North Pacific gyre between California and Hawaii, covers an expansive area of 1.6 million square kilometers and is believed to contain over 79,000 metric tons of plastic waste[2]. It is estimated that about 80% of the plastic entering marine systems originates from sources on land, particularly single-use consumer items, while the remaining portion comes from other sources. An additional 20% of plastic pollution is attributed to shipping and fishing vessels[2].

The existing global challenge of inadequate recycling practices and disposable culture, emphasizes land-based plastic waste. Unexpectedly, only 9% of the total plastic production is effectively recycled, while the vast majority is either disposed of in landfills, incinerated, or discarded into the environment, where it can harm marine life and ecosystems[2].

## 2.9 Microplastics in Marine Organisms

### 2.9.1 Microplastics in Fish

In fisheries, there is hardly no aspect of fish harvest, fish farming, fish packaging and fish transport that does not involve the extensive use of plastics[39]. In the fish harvest sector, plastics are used in fish nets, trawls, lines, lures, traps, pingers, in boat construction, fish holds, buoys, floats and fish aggregating devices. In fish farming, plastics are used in cage fabrication, mesh screens, drain pipes, plastic paddle wheel aerators, pond plastic lining, crates as well as for feed and feed supplement packaging[46]. In fish processing, plastics are mainly used in insulated fish boxes, crates, conveyor belts and product packaging. The choice of material depends on the intended use as some plastic materials (polyethylene, polypropylene, expanded polystyrene) float

while some other forms of plastics (polyvinyl chloride, polyamide, polyethylene terephthalate) sink in seawater[47].

There has been a notable increase in research interest concerning the consumption of microplastics by fish. This attention is attributed to the significance of fish in human diets and the rising significance of aquaculture. Fish also hold a pivotal position in marine food chains and contribute significantly to the cycling of organic matter in the oceans. These investigations have explored the ingestion of microplastics by both wild and farmed fish, employing field studies and laboratory experiments conducted globally[48]. Microplastic contamination has been well documented across a range of habitats and for a large number of organisms in the marine environment[49]. Consequently, bioaccumulation, and in particular biomagnification of MPs and associated chemical additives, are often inferred to occur in marine food webs[39].

Reports of small plastic particle ingestion have been made in over 690 marine species of different trophic levels. These aquatic animals ingest microplastics passively due to their inability to identify the microplastics and differentiate it from their foods[50]. More so, these microplastics are highly attractive to the aquatic animals because of their minute size of microplastic particles, buoyancy of the plastic particles and attractive colour of the plastics[51].

According to findings from studies corroborated by previous studies that MP bioaccumulation occurs within each trophic level, while current evidence around bioaccumulation of associated chemical additives is much more ambiguous[48].

In contrast, MP biomagnification across a general marine food web is not supported by current field observations, while results from the few laboratory studies supporting trophic transfer are hampered by using unrealistic exposure conditions[52]. Furthermore, a lack of both field and

laboratory data precludes an examination of potential trophic transfer and biomagnification of chemical additives associated with MPs[52].

Previous findings had indicated that, although bioaccumulation of MPs occurs within trophic levels, no clear sign of MP biomagnification in situ was observed at the higher trophic levels. The Mediterranean Sea has received significant attention in research related to microplastics and their presence in fish[53]. A study investigated the occurrence of microplastics in two edible fish species, *Mullus barbatus* and *Merluccius merluccius*, across three distinct geographical sub-regions of the Mediterranean Sea[53]. Plastic fragments were identified in 23.3% of all the fish examined, with fibers being the primary type of microplastic detected. Notably, the two fish species exhibited variations in the frequency of plastic ingestion, with M. merluccius showing double the prevalence of microplastics[53].

Furthermore, the consumption of anthropogenic particles was examined in two regions within the western Mediterranean Sea[54]. This study included four fish species: *Trachurus mediterraneus*, *Sardina pilchardus*, *Engraulis encrasicolus*, and *Boops boops*. Anthropogenic particles were detected in the digestive tracts of 28% of the sampled fish[54]. Among these fish, *T. mediterraneus* had the highest incidence of these particles, while *E. encrasicolus* exhibited the lowest percentage of affected individuals. Additionally, some variations were observed between different study areas. In the central-western Adriatic Sea, marine litter has been found in the stomach contents of sardines (*S. pilchardus*) and anchovies (*E. encrasicolus*)[54].

Another research also identified microplastics litter in benthic flatfish *Solae solae* from the northern and Adriatic Sea. Microplastics were detected in 95% of the fish samples, and approximately 80% of the examined specimens contained more than one microplastic item. The most frequently identified polymers were PVC, PP, PE, polyester, and polyamide. Among the

microplastic particles, fragments accounted for 72% of the total, while fibers comprised the remaining 28%[55].

### 2.9.2 Microplastics in Mussels

Mussels are organisms that engage in filter feeding, processing substantial amounts of water (typically around 7 to 8 liters of seawater per hour). Consequently, they have the capacity to accumulate and concentrate various pollutants present in seawater. Mussels are widely distributed geographically, making them ideal subjects for extensive research on coastal regions[39]. Furthermore, they are stationary organisms that don't migrate, simplifying the process of sampling. This characteristic makes them valuable indicator species for assessing pollutants in seawater. As a result, they have garnered attention in efforts to monitor microplastics in marine environments[52]. Mussels, along with other bivalve mollusks, play significant roles in marine food chains and can contribute to the transfer of microplastics to organisms at higher trophic levels. Numerous studies conducted at various locations worldwide have focused on monitoring microplastic pollution using mussels.

For example, a research discovered eight different types of plastic materials in blue mussels (*Mytilus edulis*) from the French Atlantic coast, with PE and PP being the most prevalent. The majority of these microplastics were in the form of fragments, and on average, one particle per mussel was detected. In a study of wild *M. edulis* in coastal waters of the United Kingdom, an average of six items per individual were found[56].

However, micro-FTIR spectroscopy revealed that only about half of these were actually microplastics, with rayon and cotton particles comprising the second most abundant group[2]. In Norwegian waters, a comparison was made between mussels from the relatively remote Barents

Sea and those from the highly urbanized Oslofjord[57].The mussels were found to contain 13 different types of polymers, with cellulose-based polymers and small black rubbery particles, potentially from road dust, being the most common. On average, 1.5 particles per individual were detected, and fibers accounted for 83% of the identified particles.

The study found no significant differences between the two sites in terms of particle amounts. However, differences in microplastic levels were observed in Mediterranean mussels (*M. galloprovincialis*) collected from coastal and offshore areas in the northern and central Adriatic Sea[57]. Coastal waters had microplastic loads approximately double those found further offshore. Smaller particles (20–40 μm) were the most prevalent in both areas, with the most common polymer types being PE, PP, and PET, followed by smaller but equal amounts of PS, PA, and PVC[58].

In the Bizerte lagoon in northern Tunisia, where significant anthropogenic pressures are present, much higher numbers of microplastic particles were detected in mussels[59]. The average number of particles per mussel ranged from 2.6 to 12. The predominant polymer found in both mussels and seawater was PE, followed by PP. Areas highly polluted with fibers and PE were associated with a higher incidence of particle ingestion or deposition.

Similarly, filaments (fibers) were the primary form of microplastics in *M. galloprovincialis* studied in various regions of Italy[60]. The average number of particles per mussel ranged from 3.0 to 12.4, and no significant differences were observed between cultivated and wild mussels[59]. Interestingly, cooking was found to reduce the presence of microplastics by up to 50%. In contrast, lower numbers of particles were detected in the same species along the Turkish coasts with an average of 0.69 items per mussel[61]. The majority of these particles were fragments,

followed by fibers and a smaller portion of films. Twelve different polymer types were identified, with PET, PP, and PE making up 80% of the total microplastics. Lastly, nearly half of the *M. galloprovincialis* sampled in the northern Ionian Sea contained microplastics[61]. Most of these microplastics were in the form of fragments, and the primary polymeric material was PE.

### 2.10 Microplastic Pollution in Marine Waters

The presence of microplastics in surface and subsurface waters has been documented across various ecosystems. Microplastics in water bodies can be transported over long distances by currents, and their accumulation in the water column poses a threat to aquatic organisms. Research has shown that microplastics tend to concentrate in coastal areas and estuaries due to human activity, making lagoons such as Lagos Lagoon hotspots for pollution[6].

In a study conducted in the Mediterranean, it was reported that the concentration of microplastics in surface waters ranged from 0.3 to 0.9 items per cubic meter. This highlights the widespread nature of microplastic contamination in coastal environments, with similar concentrations expected in highly urbanized and industrialized regions like Lagos[62]. The physical and chemical properties of microplastics, such as density and size, influence their distribution in water bodies, with lighter plastics tending to float and accumulate at the surface, while denser particles settle in the sediment[8].

### 2.11 Microplastic Accumulation in Sediments

Microplastics are not only found in water and within organisms but also accumulate in sediments. Research has shown that sediments act as a sink for microplastics, where heavier particles settle. In a study, it was reported that sediment samples from estuaries and coastal areas revealed high concentrations of microplastics, particularly in regions with high human activity[63]. The

persistence of microplastics in sediments means they can remain in the environment for long periods, continuously affecting benthic organisms.

A comparative study conducted in various global estuaries, including the Thames (UK) and Yangtze (China), revealed that sediment samples contained up to 1,500 microplastic particles per kilogram of dry sediment[63]. Similar concentrations are likely in urbanized water bodies like the Lagos Lagoon, where waste management systems are often inadequate, leading to high levels of plastic debris entering the water.

## 2.12 Environmental and Health Impacts of Microplastics

The environmental impacts of microplastic pollution are extensive, affecting aquatic organisms at all trophic levels. Beyond ingestion by fish, microplastics can be ingested by plankton, filter feeders, and other marine organisms, leading to bioaccumulation and biomagnification throughout the food web[6]. The attachment of toxic substances, such as persistent organic pollutants (POPs), to microplastic particles further amplifies their harmful effects, as these toxins can be transferred to organisms that ingest the particles[6].

From a human health perspective, the ingestion of microplastics by fish and other seafood poses potential risks. Although research on the direct health effects of microplastic consumption by humans is still ongoing, concerns have been raised about the potential for toxic chemicals associated with microplastics to accumulate in human tissues. Recent studies have shown that microplastics can enter the human body through the consumption of contaminated seafood, drinking water, and even air[7].

## 2.13 Phthalate Esters

Phthalate esters, commonly known as phthalates, are a group of chemical compounds used primarily as plasticizers, substances added to plastics to increase their flexibility, durability, and longevity. They are most commonly incorporated into polyvinyl chloride (PVC) plastics, making them essential in the production of numerous plastic products, including packaging materials, medical devices, toys, and personal care products[64]. Despite their widespread use, phthalates are not chemically bound to the plastics they soften, meaning they can leach out into the environment, leading to widespread exposure in humans and animals[32].

Phthalates are divided into two primary categories based on their molecular weight: high-molecular-weight phthalates and low-molecular-weight phthalates. High-molecular-weight phthalates, such as di(2-ethylhexyl) phthalate (DEHP), are typically used in the production of more durable plastic goods, including vinyl flooring, cables, and automotive components. Low-molecular-weight phthalates, such as diethyl phthalate (DEP) and dibutyl phthalate (DBP), are frequently found in personal care products like perfumes, lotions, and shampoos[65].

Phthalates are ubiquitous in the environment due to their extensive use in industrial and consumer products. They are often detected in air, water, soil, and sediments. Phthalates can enter the environment through various pathways, including industrial discharge, improper waste disposal, and the degradation of plastic materials. Once released into the environment, phthalates are persistent and can accumulate in living organisms, leading to potential health risks[66].

Human exposure to phthalates occurs primarily through ingestion, inhalation, and dermal absorption. For instance, phthalates can migrate from food packaging into food, leading to dietary exposure[67]. Similarly, indoor air and dust in homes and workplaces may contain phthalates, particularly in spaces with vinyl flooring or plastic furnishings. Phthalates in personal care products can be absorbed through the skin, contributing to cumulative exposure[68].

One of the primary concerns surrounding phthalate exposure is their potential to act as endocrine disruptors. Phthalates can interfere with the normal function of hormones in the body, particularly those involved in reproductive development. Several studies have linked phthalate exposure to adverse reproductive health effects, such as reduced sperm quality in men and altered hormonal levels in women[69]. Phthalates have also been implicated in the development of birth defects, including cryptorchidism and hypospadias, in newborn boys exposed in utero[64].

### 2.13.1 Phthalate Esters in Aquatic Environment

Phthalates enter aquatic environments from a range of sources, including industrial wastewater, urban runoff, sewage treatment plants, and plastic waste degradation. Improper disposal of plastic materials, such as plastic bags, packaging, and industrial products, is a primary contributor to PAE pollution. For instance, a significant amount of phthalates in aquatic environments can be traced back to municipal waste and landfill leachate[70]. In addition, PAEs can be released into water bodies through atmospheric deposition, where phthalates volatilize into the air from plastic surfaces and later settle into aquatic environments.

Once in aquatic systems, phthalates tend to adsorb onto suspended particles due to their hydrophobic nature. Over time, they accumulate in sediments, creating long-term reservoirs of these pollutants. Their persistence in sediments is concerning because they can be re-released into the water under specific conditions, such as changes in pH, temperature, or salinity[71]. PAEs exhibit varying degrees of water solubility depending on their molecular weight, with low-molecular-weight phthalates being more water-soluble, allowing them to move more easily through water systems[71].

In aquatic environments, the concentration of PAEs can vary depending on factors like the proximity to pollution sources, water flow, and the physical-chemical characteristics of the water. Studies have reported high concentrations of phthalates in coastal areas, rivers near industrial zones, and densely populated urban regions[72]. Their persistence in these ecosystems raises concerns about their potential to bioaccumulate in aquatic organisms and disrupt ecosystems.

### 2.13.2 Health Impacts of Phthalate Esters Contamination

Phthalates have been shown to have toxic effects on aquatic organisms, including fish, invertebrates, and amphibians. These compounds can induce endocrine disruption, reproductive toxicity, and developmental abnormalities in aquatic species. For example, exposure to di(2-ethylhexyl) phthalate (DEHP), one of the most widely used phthalates, has been linked to abnormal development and reduced fertility in fish and amphibians[73]. These effects are particularly concerning in species that are part of the human food chain, as phthalates can bioaccumulate and be transferred through trophic levels[73].

In addition to their effects on aquatic life, phthalates have been found to impact water quality. Due to their persistence, PAEs can degrade into more toxic by-products over time, such as phthalate monoesters, which are known to exhibit higher toxicity levels[74]. The potential for bioaccumulation and biomagnification of phthalates in aquatic food webs further heightens the risk of human exposure through the consumption of contaminated seafood, drinking water, or recreational activities[74].

### 2.13.3 Review Studies of Phthalate Esters in the Aquatic Environment

Several studies have investigated the presence of phthalates in various aquatic systems, including rivers, lakes, estuaries, and oceans. A study conducted on urban rivers in China reported high concentrations of PAEs, particularly di(2-ethylhexyl) phthalate (DEHP) and dibutyl phthalate (DBP), in sediments and water samples. The concentrations of these phthalates were linked to industrial discharges and urban runoff, indicating that human activities contribute significantly to phthalate contamination in water bodies[72].

Similarly, another study analyzed the occurrence of phthalates in riverine and estuarine sediments along the Pearl River Delta in China. The study found that DEHP and DBP were the

most prevalent phthalates, with higher concentrations detected in sediments near industrial zones and wastewater treatment plants. The findings suggested that wastewater effluent is a major pathway for phthalates entering aquatic environments, where they accumulate in sediments and pose a long-term threat to the ecosystem[75].

In a study of coastal waters in the Mediterranean, elevated levels of phthalates in both surface water and sediment samples were found. The research highlighted that phthalates' hydrophobic nature allows them to adsorb onto suspended particles and sediments, where they can persist for long periods. The study also emphasized the importance of understanding sediment dynamics in assessing the risks posed by phthalates, as these compounds can be remobilized into the water column under specific conditions, such as changes in temperature, pH, and salinity[76].

### 2.14 Current Research on Microplastics in Lagos and Epe Lagoons

The Lagos Lagoon, located in one of the most densely populated urban areas in Africa, is highly vulnerable to microplastic contamination. A significant contributor to this pollution is the improper disposal of plastic waste, with urban drainage systems, rivers, and stormwater channels acting as conduits for plastic debris entering the lagoon[77]. Lagos has one of the highest plastic consumption rates in Nigeria, and much of this plastic waste is either improperly disposed of or ends up in landfills, from which it can leak into nearby water bodies. According to a research, single-use plastics, such as polyethylene bags, plastic bottles, and food packaging, dominate the waste stream in Lagos, with a high proportion of this waste eventually degrading into microplastics in the lagoon[78].

The Epe Lagoon, although situated further away from the central city, is also impacted by microplastic pollution. Agricultural runoff, artisanal fishing activities, and domestic waste disposal have been identified as significant sources of microplastics in this lagoon[79]. The rise of

aquaculture and other local economic activities in the surrounding communities has also increased the amount of plastic entering the lagoon, leading to elevated levels of microplastic contamination, particularly in areas close to human settlements.

Several recent studies have focused on detecting and quantifying microplastics in both the Lagos and Epe lagoons. One study conducted an extensive survey of the Lagos Lagoon, which found that microplastics were prevalent in surface waters, sediment, and even within the tissues of aquatic organisms. Their research identified polyethylene, polypropylene, and polystyrene as the most common types of microplastics in the lagoon, consistent with findings in other urban water bodies worldwide[80]. Sampling across different locations within the lagoon revealed that microplastic concentrations were highest near urbanized and industrialized regions.

Similarly, another research in the Epe Lagoon highlighted the significant presence of microplastics in both surface and benthic waters. The study found that microplastics were not only present in the water column but had also settled in the sediment layers, where they posed risks to benthic organisms[81]. Their findings suggested that the distribution of microplastics in the lagoon is influenced by factors such as water currents, proximity to pollution sources, and the nature of the surrounding land use. The study further reported that microplastics in the Epe Lagoon primarily consisted of fragments and fibers, which are often derived from the breakdown of larger plastic items and textiles[81].

The ecological impacts of microplastics in the Lagos and Epe lagoons are becoming more evident as research into their effects on aquatic organisms grows. A recent study investigated the ingestion of microplastics by fish species in the Lagos Lagoon and found that many fish had ingested small plastic particles[82]. The ingestion of microplastics can cause physical damage to the digestive systems of fish, reduce their ability to absorb nutrients, and lead to blockages,

which may ultimately result in starvation. Moreover, the ingestion of microplastics raises concerns about the transfer of these particles up the food chain, with potential consequences for human health due to the consumption of contaminated seafood[82].

In the Epe Lagoon, it was also reported that microplastics were found in the stomach contents of several commercially important fish species. The presence of these plastics in fish not only poses risks to the health of aquatic organisms but also threatens the economic viability of local fisheries[79]. The accumulation of microplastics in aquatic food chains can lead to bioaccumulation, where higher trophic-level organisms, including humans, are exposed to increasing concentrations of microplastics and associated toxic substances over time[79].

### 2.15 Heavy Metals

A metal is classified as a "heavy metal" when its density exceeds 5–6 g/cm³[83]. Certain heavy metals, which are naturally present in organisms in trace amounts, are often termed "trace metals." This term can imply that the metal is essential for the organism, even if it occurs in very small quantities[84].Heavy metals such as lead (Pb), cadmium (Cd), mercury (Hg), zinc (Zn), chromium (Cr), nickel (Ni), and copper (Cu) are commonly found in aquatic environments due to anthropogenic activities, including industrial discharges, agricultural runoff, mining, urbanization, and improper waste disposal[8,63]. Once introduced into aquatic systems, these metals persist in water columns and sediments, affecting the health of aquatic organisms and ecosystems.

**Lead** is one of the most prevalent heavy metals found in the Lagos Lagoon due to its widespread use in industrial processes, vehicle emissions, and urban runoff. Studies have shown that lead concentrations often exceed safe levels, posing risks to aquatic organisms and humans through the consumption of contaminated fish and shellfish. Lead exposure is known to cause neurological, reproductive, and developmental issues in both animals and humans[85].

**Mercury**, although less commonly detected at high levels compared to other heavy metals, can still be found in the lagoons, especially near industrial zones. Mercury contamination poses a particular threat due to its ability to bioaccumulate in the food chain, leading to higher concentrations in fish and other aquatic organisms. Methylmercury, the organic form of mercury, is especially toxic and can cause severe neurological damage in humans[86].

**Cadmium** is another heavy metal of concern in the Lagos and Epe Lagoons. It is often released from industrial activities, particularly in metal plating and plastic production. Cadmium exposure is harmful to aquatic organisms, affecting growth and reproduction, and in humans, it can lead to kidney damage and bone demineralization[87]

**Chromium**, especially in its hexavalent form (Cr(VI)), is a highly toxic heavy metal commonly associated with industrial discharge from tanneries, dyeing operations, and metal finishing industries. Chromium can cause damage to aquatic organisms, including gill damage and reproductive issues, while exposure to Cr(VI) in humans is linked to carcinogenic effects[88]

**Zinc** is an essential trace metal for living organisms but becomes toxic at elevated levels. It enters the Lagos and Epe Lagoons mainly through industrial discharge and urban runoff. Zinc pollution can affect the metabolic processes of aquatic species and reduce biodiversity. While zinc is less toxic to humans compared to other heavy metals, long-term exposure can still lead to health issues, including gastrointestinal distress and immune system dysfunction[89].

**Copper** is used in numerous industrial processes and is also present in antifouling paints used on ships. In the Lagos Lagoon, copper levels have been found to fluctuate depending on proximity to industrial areas. Copper toxicity in aquatic ecosystems can lead to reduced growth and survival rates in fish and invertebrates, and in humans, excessive exposure can cause liver and kidney damage[90].

**Nickel** contamination in the Lagos and Epe Lagoons is primarily due to industrial effluents from metalworking industries. Nickel is moderately toxic to aquatic life, particularly affecting fish and invertebrates, and chronic exposure to nickel in humans can lead to respiratory issues and skin dermatitis[91].

### 2.15.1 Sources of Heavy Metals

**Industrial Effluents**: Industries such as manufacturing, mining, and petrochemicals are major contributors of heavy metals in aquatic systems. Effluents containing metals like mercury and lead are often discharged directly into water bodies[86].

**Agricultural Runoff**: Fertilizers and pesticides used in agricultural practices can contain heavy metals such as cadmium, zinc, and copper, which are washed into aquatic systems through runoff[92].

**Atmospheric Deposition**: Heavy metals from vehicle emissions and industrial activities can settle into water bodies via atmospheric deposition, increasing metal concentrations in surface waters[93].

Once in the aquatic environment, heavy metals are known to exhibit toxic effects, including bioaccumulation in aquatic organisms and biomagnification in the food chain. Heavy metals can affect aquatic life by interfering with biological processes, causing reproductive issues, organ damage, and even death in fish, invertebrates, and other marine organisms. In humans, the consumption of seafood contaminated with heavy metals has been linked to neurological damage, cancer, and other health complications[6].

One of the most concerning aspects of microplastic pollution is its role as a vector for transporting heavy metals in aquatic environments. Microplastics have been found to adsorb and

concentrate heavy metals from the surrounding water due to their surface properties, particularly their large surface area and chemical composition[94]. Once microplastics adsorb heavy metals, they can transport these contaminants over long distances and make them bioavailable to aquatic organisms, which can lead to greater bioaccumulation and biomagnification within the food web[94].

### 2.15.2 **Adsorption of Heavy Metals**

The hydrophobic surface of microplastics allows them to adsorb heavy metals from the surrounding water. Studies have shown that microplastics can accumulate heavy metals such as lead, cadmium, and mercury on their surfaces[95]. Microplastics, being lightweight, can transport adsorbed heavy metals over long distances, potentially redistributing contaminants from polluted to less-polluted areas[92].

Microplastics can be ingested by a variety of aquatic organisms, including fish, mollusks, and zooplankton. When these organisms ingest microplastics containing adsorbed heavy metals, the metals are released into their digestive systems, making them bioavailable and increasing their uptake into the body[7].

The combination of microplastic and heavy metal pollution in aquatic ecosystems poses significant risks to both ecological health and human populations. Aquatic organisms that ingest microplastics with adsorbed heavy metals face the dual threat of physical harm from plastic particles and chemical toxicity from the metals. Over time, these contaminants can bioaccumulate and biomagnify through the food web, leading to higher concentrations of heavy metals in top predators, including humans who consume contaminated seafood[8].

In humans, the ingestion of seafood contaminated with both microplastics and heavy metals has been linked to various health risks, including carcinogenic, neurotoxic, and endocrine-disrupting effects. The presence of heavy metals on microplastic surfaces can exacerbate the toxicological impacts, increasing the potential for long-term chronic health issues[94.]

### 2.15.3 Recent Studies on Heavy Metals Interaction with Microplastics

Recent studies have documented the interaction between microplastics and heavy metals in various aquatic environments. A recent research investigated the adsorption behavior of heavy metals on microplastics in marine environments, demonstrating that microplastics can adsorb significant amounts of lead, cadmium, and copper, depending on the type of plastic and environmental conditions[95]. Similarly, another research by showed that microplastics in freshwater systems can concentrate heavy metals and serve as vectors for their transport into aquatic organisms[7].

A study focused on the role of microplastics in the bioaccumulation of heavy metals in fish species in contaminated waters, finding that fish exposed to both microplastics and heavy metals had higher concentrations of metals in their tissues compared to fish exposed to metals alone. This highlights the compounding effect of microplastics on heavy metal toxicity in aquatic organisms[93].

### 2.15.4 Environmental Impacts of Heavy Metals

**Bioaccumulation and Biomagnification**: One of the major concerns with heavy metal contamination is bioaccumulation, where metals accumulate in the tissues of organisms over time, and biomagnification, where the concentration of these metals increases as they move up the food chain[94]. Fish and shellfish in the Lagos and Epe Lagoons can accumulate significant

levels of heavy metals, posing risks to human health when consumed. Long-term exposure to metals like mercury and cadmium can cause chronic illnesses, including cancer, neurological damage, and kidney failure[86].

**Impact on Aquatic Life**: The devastating effects of heavy metals on aquatic ecosystems are numerous. Metals such as lead, mercury, and cadmium can cause acute toxicity, leading to reduced reproduction, growth, and survival rates in fish, mollusks, and crustaceans. Sediment contamination by heavy metals also affects benthic organisms that play essential roles in nutrient cycling and ecosystem functioning[87].

## 2.16 Physicochemical Properties of Surface and Benthic Waters and their Influence on Microplastic Distribution

The physicochemical properties of aquatic ecosystems are essential in determining the distribution and behavior of microplastics in both surface and benthic waters. Benthic waters refer to the lower layers of water near or on the bottom sediment, while surface waters are those at or near the water's surface. Understanding these properties can provide critical insights into the pathways of microplastics in aquatic systems, their persistence, and their interactions with other contaminants[95].

Key parameters such as temperature, pH, dissolved oxygen (DO), salinity, and turbidity not only define the health of aquatic systems but also have direct effects on the movement, fragmentation, and settlement of microplastics. This essay discusses the role of these physicochemical properties in determining the behavior of microplastics in both benthic and surface waters, with a focus on their influence on the ecological dynamics of microplastic pollution.

### 2.16.1 Temperature

Temperature plays a crucial role in determining the buoyancy, distribution, and degradation rates of microplastics. Microplastics, which are primarily composed of polymers like polyethylene (PE), polypropylene (PP), and polystyrene (PS), respond to temperature changes due to their thermoplastic nature. As the temperature increases, the viscosity of water decreases, and microplastic particles may become more buoyant, leading to their accumulation in surface waters[96]. On the other hand, lower temperatures may cause microplastics to sink more easily, resulting in their deposition in benthic zones. Studies have also shown that temperature can influence the rate of plastic degradation, where higher temperatures enhance the breakdown of larger plastics into microplastics due to increased photodegradation and thermal processes[97].

Temperature gradients between surface and benthic waters create stratification, which affects the vertical movement of microplastics. In tropical regions, such as Lagos Lagoon, where surface waters are often warmer, microplastics are more likely to be concentrated at the surface, while cooler benthic waters may trap denser particles[81]. Understanding temperature dynamics is therefore key to predicting the spatial distribution of microplastics in both the water column and sediments.

### 2.16.2 pH

The pH level of aquatic systems can significantly affect the chemical properties of microplastics and their interactions with other contaminants. In more acidic or basic environments, microplastics may undergo changes in surface charge, which influences their tendency to adsorb or desorb pollutants such as heavy metals, organic pollutants, and microbial contaminants[72]. The adsorption of toxic substances on microplastics can exacerbate their ecological impact, as these particles become vehicles for transporting hazardous chemicals through the water column.

Variations in pH between surface and benthic waters can also affect the behavior of microplastics. Benthic waters tend to have more stable pH levels, while surface waters are more susceptible to fluctuations due to atmospheric interactions and photosynthetic activity. The interaction between microplastics and their environment can become more complex in areas where pH levels fluctuate, with the potential for greater adsorption of harmful chemicals at certain pH levels[98].

### 2.16.3 Dissolved Oxygen (DO)

Dissolved oxygen (DO) is another critical parameter in aquatic ecosystems that influences the behavior of microplastics, particularly in benthic zones. High levels of dissolved oxygen in surface waters promote aerobic conditions that facilitate the degradation of certain types of plastics, particularly biodegradable ones[19]. In contrast, low DO levels, often found in benthic waters due to poor circulation and organic matter decomposition, create anaerobic conditions that can slow down the degradation of plastics, allowing microplastics to persist for longer periods[99].

Additionally, low DO levels in benthic waters can lead to the accumulation of microplastics in sediments, where they are less likely to degrade. This creates potential long-term reservoirs of plastic pollution that can be resuspended into the water column during turbulent conditions or through bioturbation by benthic organisms[100]. The interaction between DO levels and microplastic persistence highlights the importance of monitoring oxygen dynamics in aquatic ecosystems to understand the full extent of microplastic pollution.

### 2.16.4 Salinity

Salinity, the concentration of dissolved salts in water, also plays a significant role in microplastic distribution, particularly in estuarine and coastal environments like the Lagos and Epe lagoons. The density of plastics is affected by salinity levels, with higher salinity generally increasing the buoyancy of microplastics. As a result, microplastics may remain suspended in surface waters in saline environments, while in freshwater systems, they are more likely to settle[101]. In estuarine systems where salinity gradients exist, microplastics can migrate between surface and benthic waters depending on local salinity variations.

Furthermore, salinity can influence the interaction of microplastics with other pollutants. For example, studies have shown that in higher salinity conditions, microplastics have an increased affinity for hydrophobic organic pollutants, such as polycyclic aromatic hydrocarbons (PAHs) and polychlorinated biphenyls (PCBs)[102]. This increased sorption in saline environments raises concerns about the role of microplastics as vectors for transporting toxic chemicals in marine and estuarine ecosystems.

### 2.16.5 Turbidity and Sedimentation

Turbidity, a measure of the cloudiness or haziness of water, is directly related to the amount of suspended solids in the water column. In highly turbid waters, such as those found in the Lagos Lagoon, microplastics can become entrapped in suspended particulate matter and sink to the bottom more readily[80]. Increased sedimentation rates in turbid environments contribute to the burial of microplastics in benthic sediments, where they can persist for long periods.

Sedimentation processes can also lead to the resuspension of microplastics during periods of high turbulence, such as during storms or tidal movements. This cyclical process of deposition and resuspension complicates efforts to assess the overall burden of microplastics in aquatic

systems and underscores the importance of turbidity as a factor influencing microplastic distribution[100].

### 2.17 Global and Local Efforts to Reduce Plastic and Microplastic Pollution

Several global initiatives have been launched to mitigate plastic pollution. One of the most significant actions has been the adoption of the United Nations Environment Programme (UNEP) resolutions, which focus on combating marine litter and microplastic pollution. The 2019 report emphasized the need for coordinated global action to reduce plastic use, improve waste management, and encourage innovation in biodegradable plastics[103].

The European Union (EU) has taken a leading role in reducing plastic waste through policies such as the European Strategy for Plastics in a Circular Economy. This strategy, launched in 2018, aims to make all plastic packaging recyclable by 2030, reduce single-use plastics, and encourage the development of sustainable alternatives[104]. The EU also passed the Single-Use Plastics Directive, which bans certain single-use plastic items and imposes extended producer responsibility (EPR) on manufacturers for plastic products' life cycle[105].

Another global initiative is the G7 Ocean Plastics Charter, endorsed by several G7 member countries in 2018. The charter focuses on promoting sustainable plastic use, developing waste management solutions, and fostering international collaboration to address plastic pollution[106]. The International Maritime Organization (IMO) has also implemented regulations through the International Convention for the Prevention of Pollution from Ships (MARPOL), which aims to reduce plastic waste from ships entering the ocean[107].

Countries across the globe have implemented local mitigation strategies to tackle microplastic pollution. In Nigeria, local initiatives include bans on single-use plastics in some states, public

awareness campaigns, and partnerships between the government and non-governmental organizations (NGOs) to address plastic pollution in water bodies such as the Lagos and Epe lagoons[108]. Local governments in Nigeria have been working with community-based organizations to promote plastic recycling and encourage the use of reusable materials.

In Kenya, a ban on plastic bags, introduced in 2017, has led to a significant reduction in plastic pollution. According to research, this measure has decreased plastic waste by up to 80% in some regions[109]. South Africa has also adopted extended producer responsibility (EPR) measures, which hold manufacturers accountable for the environmental impacts of plastic products throughout their life cycle[110].

In Asia**,** China has implemented a ban on the import of foreign plastic waste since 2018, forcing countries to take responsibility for their plastic waste domestically[111]. The Plastic Waste Reduction Program in India has been working towards reducing plastic consumption by focusing on public awareness and promoting the use of biodegradable alternatives[112].

## 2.18 Policy Responses

Effective policy responses are crucial for reducing microplastic pollution. Many countries have implemented regulations targeting the reduction of plastic waste at the source, such as banning or restricting the use of single-use plastics and microbeads in personal care products. In 2015, the United States passed the Microbead-Free Waters Act, which prohibits the use of microbeads in cosmetic products[113]. Similarly, Canada introduced legislation to ban microbeads in toiletries and over-the-counter drugs in 2018[114].

Several countries have also adopted extended producer responsibility (EPR) policies, which hold manufacturers accountable for the collection and recycling of plastic products. EPR shifts the

financial burden of waste management from consumers to producers, encouraging companies to design products that are easier to recycle and have lower environmental impacts. The European Union and Japan have successfully implemented EPR programs, resulting in higher recycling rates and reduced plastic pollution[105].

Additionally, international organizations are working towards developing a global treaty on plastic pollution. In 2022, the United Nations Environment Assembly (UNEA) passed a historic resolution to begin negotiations on a legally binding international agreement to address plastic pollution from its production to disposal[115]. This treaty aims to establish global standards for reducing plastic production, improving waste management, and curbing plastic leakage into the environment.

### 2.19 Waste Management and Recycling

Improving waste management systems is essential for reducing plastic pollution, particularly in low- and middle-income countries where infrastructure for waste collection and recycling is often inadequate. Effective waste management involves the implementation of the 3Rs (Reduce, Reuse, Recycle) principle to minimize plastic waste and promote sustainable consumption patterns[9].

Recycling has been identified as one of the most effective strategies to combat plastic pollution. Countries with well-established recycling systems, such as Germany and South Korea, have achieved recycling rates of over 50% for plastic packaging[116]. However, global plastic recycling rates remain low, with less than 10% of plastics being recycled globally[21]. This highlights the need for improved recycling infrastructure, particularly in developing nations.

Innovative waste management technologies, such as waste-to-energy plants and pyrolysis, are being explored as potential solutions for reducing plastic waste. Pyrolysis is a process that converts plastic waste into fuels and other valuable products, offering a potential avenue for dealing with non-recyclable plastics[105].

## 2.20 Remediation Techniques

Several remediation techniques are being developed to address existing microplastic pollution in aquatic environments. Physical removal methods, such as the use of filtration systems and skimmer vessels, have been employed to capture floating microplastics from water bodies. Bioremediation, which involves the use of microorganisms to degrade plastic pollutants, is another promising approach for mitigating microplastic contamination[117].

Additionally, nanotechnology is being explored as a potential tool for detecting and removing microplastics from the environment. Nanomaterials, such as graphene oxide and metal oxides, can adsorb microplastics and facilitate their removal from water bodies[6]. However, these technologies are still in the early stages of development and require further research before they can be implemented on a large scale.

## Endnotes


1. Plastics Europe, *Plastics – the Facts 2022: An Analysis of European Plastics Production, Demand and Waste Data*, **Plastics Europe**, 2022.

2. G. Thushari & J.D.M. Senevirathna, *Plastic Pollution in the Marine Environment*, **Heliyon**, 6, 2020, 8e04709, https://doi.org/10.1016/j.heliyon.2020.e04709.

3. C. M. Rochman, E. Hoh, T. Kurobe, & S. J. Teh, *Ingested Plastic Transfers Hazardous Chemicals to Fish and Induces Hepatic Stress*, **Scientific Reports**, 3, 2015, 3263. https://doi.org/10.1038/srep03263.

4. C. Le Guern, *When The Mermaids Cry: The Great Plastic Tide*, accessed January 13, 2024, https://plasticpollution.org/.

5. Y. Lee, J. Cho, J. Sohn, & C. Kim, *Health Effects of Microplastic Exposures: Current Issues and Perspectives in South Korea*, **Yonsei Medical Journal**, 64(5), 2023, 301.

6. S. Gupta, R. K. Singh, A. Kumar, S. Kumar, & R. Sinha, *Plastic Pollution in the Marine Environment: A Review of its Impacts, Sources, and Mitigation Strategies*, **Journal of Environmental Management**, 302, 2022, 114908.

7. Y. Zhang, X. Kang, X. Liu, & J. Li, *Sources, Distribution, and Fate of Microplastics in River Sediments: A Global Review*, **Environmental Research**, 220, 2023, 115179.

8. S. S Ali, T. Elsamahy, E. Koutra, M. Kornaros, & M. E El-Naggar, *Environmental Impact of Bioplastics: A Review*, **Science of The Total Environment**, 806, 2021, 150998.

9. C. Schimdt, T. Krauth, & S. Wagner, *Export of Plastic Debris by Rivers into the Sea*, **Environmental Science & Technology**, 51(21), 2021, 12246-12253.

10. K. Omoyajowo, M.O. Raimi, T.O. Waleola, O.E. Odipe, & A.L. Ogunyebi, *Public Awareness, Knowledge, Attitude and Perception on Microplastics Pollution around Lagos Lagoon*, **Research Square**, 2021. https://doi.org/10.21203/rs.3.rs-506361/v1.

11. M. Landon-Lane, *Corporate Social Responsibility in Marine Plastic Debris Governance*, **Marine Pollution Bulletin**, 127, 2018, 310-319.

12. H. Luo, Y. Zhang, D. He, Y. Li, & Y. Zhao, *Aging of Microplastics Affects their Surface Properties, Thermal Decomposition, Additives Leaching and Interactions in Simulated Fluids*, **Science of The Total Environment**, 714, 2020, 136862.

13. A. Bacha, I. Nabi, & L. Zhang, *Mechanisms and the Engineering Approaches for the Degradation of Microplastics*, **ACS ES&T Engineering**, 1(11), 2021, 1481-1501.

14. J. Duan, Y. Li, J. Gao, R. Cao, E. Shang & W. Zhang, *ROS-mediated Photoaging Pathways of Nano-and Micro-plastic Particles Under UV Irradiation*, **Water Research**, 216, 2022, 118320.

15. T. Lomonaco, J. La Nasa, A. Corti, E. Manco, & S. Ghimenti, *Release of Harmful Volatile Organic Compounds (VOCs) from Photo-degraded Plastic Debris: a neglected source of environmental pollution*, **Journal of Hazardous Materials**, 394, 2020, 122596.

16. L. Lebreton, B. Slat, F. Ferrari, B. Sainte-Rose, J. Aitken, R. Marthouse, S. Hajbane, S. Cunsolo, A. Schwarz, A. Levivier, K. Noble, P. Debeljak, H. Maral, R. Schoeneich-Argent, R. Brambini, & J. Reisser, *Evidence that the Great Pacific Garbage Patch is Rapidly Accumulating Plastic*, **Scientific Reports**, 8(1), 2018, 4666.

17. K. Mengen, Y. Li, H. Wang, L. Yu, & W. Zhang, *Physiological Toxicity and Antioxidant Mechanism of Photoaging Microplastics on Pisum sativum L. Seedlings*, **Toxics**, 11(3), 2023, 242.

18. S. L. Wright, & F.J. Kelly, *Plastic and Human Health: A Micro Issue?*, **Environmental Science & Technology**, 53(4), 2020, 1748-1765.

19. A. L. Andrady, *The Plastic in Microplastics: A Review*, **Marine Pollution Bulletin**, 119(1), 2017, 12-22. https://doi.org/10.1016/j.marpolbul.2017.01.082.

20. J. Lu, Z. Dong, J. Zhou & R. Jin, *Adsorption/Desorption Behavior of Ciprofloxacin on Aged Biodegradable Plastic PLA Under Different Exposure Conditions*, **Journal of Environmental Chemical Engineering**, 11(1), 2023, 109256.

21. R. Geyer, J. R. Jambeck, & K. L. Law, *Production, Use, and Fate of all Plastics Ever Made*, **Science Advances**, 3(7), 2017, e1700782. https://doi.org/10.1126/sciadv.1700782.

22. A. A. Horton, C. Svendsen, R. J. Williams, D. J. Spurgeon, & E. Lahive, *The Influence of Exposure and Physiology on Microplastic Ingestion by the Freshwater Fish Rutilus rutilus (roach) in the River Thames, UK*, **Environmental Pollution**, 236, 2018, 188-194.

23. M. Anik, A. Arefin, M.M. Rahman, K. Hossain, & M.N. Islam, *Role of Reactive Oxygen Species in Aging and Age-Related Diseases: A Review*, **ACS Applied Biomaterials**, 2022, doi:10.1021/acsabm.2c00411.

24. K. Birnie-Gauvin, D. Costantini, S. J. Cooke, & W.G. Willmore, *A Comparative and Evolutionary Approach to Oxidative Stress in Fish: A Review*, **Fish and Fisheries**, 18(5), 2017, 928-942.

25. Z. Li, J. Zhu, T. Wang, X. Huang, S. Wang, *Is Microplastic an Oxidative Stressor? Evidence from a Meta-analysis on Bivalves*, **Journal of Hazardous Materials**, 423, 2022, 127211.

26. J. C. Prata, J. P. da Costa, I. Lopes, A. C. Duarte, & T. Rocha-Santos, *Environmental Exposure to Microplastics: An Overview on Possible Human Health Effects*, **Science of The Total Environment**, 702, 2021, 134455.

27. I. E. Napper, A. Bakir, S. J. Rowland, & R. C. Thompson, *Characterization, Quantity, and Sorptive Properties of Microplastics Extracted from Cosmetics*, **Marine Pollution Bulletin**, 99(1-2), 2021, 178-185.

28. I. Li, H. Liu, & J. P. Chen, *Microplastics in Freshwater Systems: A Review on Occurrence, Environmental Effects, and Methods for Microplastics Detection*, **Water Research**, 137, 2021, 362-374.

29. A. L. Lusher, N. A. Welden, P. Sobral, & M. Cole, *Sampling, Isolating and Identifying Microplastics Ingested by Fish and Invertebrates.* **Analytical Methods**, 9, 1346–1360. DOI: 10.1039/C6AY02415G

30. T. S. Galloway, M. Cole, & C. Lewis, *Interactions of Microplastic Debris Throughout the Marine Ecosystem*, **Nature Reviews Earth & Environment**, 2(3), 2017, 105-118.

31. C. M. Rochman, M. A. Browne, A .J. Underwood, J. A. van Franeker, R. C. Thompson, & L. A. Amaral-Zettler, *The Ecological Impacts of Marine Debris: Unraveling the Demonstrated Evidence From What is Perceived*, **Ecology**, 94(3), 2021, 248-264.

32. A. Wang, X. Liu, Y. Li, T. Powell, X. Wang, G. Wang, & P. Zhang, *Microplastics as Contaminants in the Soil Environment: A Mini Review*, **Science of the Total Environment**, 785, 2022, 147403.

33. C. Campanale, C. Massarelli, I. Savino, V. Locaputo, & V.F. Uricchio, *A Detailed Review Study on Potential Effects of Microplastics and Additives of Concern on Human Health*, **International Journal of Environmental Research and Public Health**, 17(4), 2020, 1212.

34. L .G. A. Barboza, A. D. Vethaak, B. R. Lavorante, A. K. Lundebye, & L. Guilhermino, *Marine Microplastic Debris: An Emerging Issue for Food Security, Food Safety and Human Health*, **Marine Pollution Bulletin**, 133, 2020, 336-348.

35. J. Li, K. Zhang, & H. Zhang, *Adsorption of Antibiotics on Microplastics*, **Environmental Pollution**, 237, 2020, 460-467.

36. Y. Deng, Y. Zhang, B. Lemos, & H. Ren, *Tissue Accumulation of Microplastics in Mice and Biomarker Responses Suggest Widespread Health Risks of Exposure*, **Scientific Reports**, 7(1), 2017, 46687.

37. H. C. Vo & M. H. Pham, *Ecotoxicological Effects of Microplastics on Aquatic Organisms: A Review*, **Environmental Science and Pollution Research**, 28, 2021, 44716-44725.

38. L. S. Fendall, & M. A. Sewell, *Contributing to Marine Pollution by Washing your Face: Microplastics in Facial Cleansers*, **Marine Pollution Bulletin**, 58(8), 2009, 1225-1228.

39. I. Rao, *Microplastics in the Aquatic Environment: Implications for Post-harvest Fish Quality*, **Indian Journal of Fisheries**, 66(10), 2019, 142-152.

40. A. Andrady, *Persistence of Plastic Litter in the Oceans*, **Marine Anthropogenic Litter**, 2015, 57-72.

41. H. S. Auta, E. Chijioke, & H. Shahu, *Distribution and Importance of Microplastics in the Marine Environment: A Review of the Sources, Fate, Effects, and Potential Solutions*, **Environment International**, 102, 2017, 165-176.

42. T. Davidson, *Boring Crustaceans Damage Polystyrene Floats Under Docks Polluting Marine Waters with Microplastic*, **Marine Pollution Bulletin**, 64(9), 2012, 1821-1828.

43. A. Šaravanja, T. Pusic, T. Dekanic, *Microplastics in Wastewater by Washing Polyester Fabrics*, **Materials (Basel, Switzerland)**, 15(7), 2022, 2683.

44. R. Stafford, P. Jones, & S. Jones, *Viewpoint – Ocean Plastic Pollution: A Convenient but Distracting Truth*, **Marine Policy**, 103, 2019, 187-191.

45. K. Mattsson, E.V. Johnson, A. Malmendal, S. Linse, L.A. Hansson, & T. Cedervall, *Brain Damage and Behavioural Disorders in Fish Induced by Plastic Nanoparticles Delivered Through the Food Chain*, **Scientific Reports**, 7(1), 2017, 11452.

46. W. d'Ambrières, *Plastics Recycling Worldwide: Current Overview and Desirable Changes*, **Field Actions Science Reports**, 19, 2019, 12-21.

47. R. C. Hale, M. E. Seeley, M. J. La Guardia, L. Mai, & E. Y. Zeng, *A Global Perspective on Microplastics*, **Journal of Geophysical Research: Oceans**, 125(1), 2020, e2018JC014719. https://doi.org/10.1029/2018JC014719.

48. I. Miller, M. Hamann, & F. J. Kroon, *Bioaccumulation and Biomagnification of Microplastics in Marine Organisms: A Review and Meta-analysis of Current Data*, **PloS One**, 15(10), 2020, e0240792.

49. K. Davidson, & S. E. Dudas, *Microplastic Ingestion by Wild and Cultured Manila Clams (Venerupis philippinarum) from Baynes Sound, British Columbia*, **Archives of Environmental Contamination and Toxicology**, 71, 2016, 147-156.

50. Plastics Europe, *Plastics - the Facts 2020: An Analysis of European Plastics Production, Demand and Waste Data*, PlasticsEurope. https://www.plasticseurope.org/en/resources/publications/4312-plastics-facts-2020.

51. J. Lavers, & A. L. Bond, *Exceptional and Rapid Accumulation of Anthropogenic Debris on One of the World's Most Remote and Pristine Islands*, **Proceedings of the National Academy of Sciences**, 114(23), 2017, 6052-6055.

52. A. Rodríguez-Seijo, J. Santos, T. da Silva, S. Cachada, S. Pereira, P. Carvalho, A. Duarte, F.F.R. Moreira, & A.C. Duarte, *Oxidative Stress, Energy Metabolism and Molecular*

*Responses of Earthworms (Eisenia fetida) Exposed to Low-density Polyethylene Microplastics*, **Environmental Science and Pollution Research**, 25, 2018, 33599–33610. https://doi.org/10.1007/s11356-018-3255-3.

53. D. Giani, L. Baini, E. Galli, L. Casini, & M. Fossi, *Microplastics Occurrence in Edible Fish Species (Mullus barbatus and Merluccius merluccius) Collected in Three Different Geographical Sub-areas of the Mediterranean Sea*, **Marine Pollution Bulletin**, 140, 2019, 129–137. https://doi.org/10.1016/j.marpolbul.2019.01.005.

54. I. Renzi, C. Guerranti, & A. Blašković, *Marine Litter in Stomach Content of Small Pelagic Fishes from the Adriatic Sea: Sardines (Sardina pilchardus) and Anchovies (Engraulis encrasicolus)*, **Environmental Science and Pollution Research**, 26, 2019, 2771–2781. https://doi.org/10.1007/s11356-018-3886-8.

55. G. Pellini, A. Gomiero, T. Fortibuoni, C. Ferra, F. Grati, A. N. Tassetti, P. Polidori, & G. Fabi, *Characterization of Microplastic Litter in the Gastrointestinal Tract of Solea solea from the Adriatic Sea*, **Environmental Pollution**, 234, 2018, 943–952. https://doi.org/10.1016/j.envpol.2017.12.038.

56. N. N. Phuong, L. Poirier, Q. T. Pham, F. Lagarde, & A. Zalouk-Vergnoux, *Quantification and Characterization of Microplastics in Blue Mussels (Mytilus edulis): Protocol Setup and Preliminary Data on the Contamination of the French Atlantic Coast*, **Environmental Science and Pollution Research**, 25, 2018, 6135–6144. https://doi.org/10.1007/s11356-017-8862-3.

57. J. Li, C. Green, A. Reynolds, H. Shi, & J. M. Rotchell, *Microplastics in Mussels Sampled from Coastal Waters and Supermarkets in the United Kingdom*, **Environmental Pollution**, 241, 2018, 35–44. https://doi.org/10.1016/j.envpol.2018.05.038.

58. L. Yin, B. Chen, B. Xia, X. Shi, K. Qu, & T. Zhang, *Impacts of Microplastics on Fish: What we Know and What we Need to Know*, **Environmental Science & Technology**, 55(4), 2021, 2189-2206.

59. T. Wakkaf, A. Dhib, R. El Zrelli, I. Zouari, L. Rabaoui, & H. Beyrem, *Microplastics in Edible Mussels from a Southern Mediterranean Lagoon: Preliminary Results on Seawater-Mussel Transfer and Implications for Environmental Protection and Seafood Safety*, **Marine Pollution Bulletin**, 158, 2020, 111355. https://doi.org/10.1016/j.marpolbul.2020.111355.

60. F. Provenza, G. Russo, M. Falco, & G. Maffioli, *Chemical Pollution and Ecotoxicological Effects of High-density Polyethylene Microplastics in Mytilus galloprovincialis from Two Italian Lagoon Ecosystems*, **Environmental Toxicology and Pharmacology**, 98, 2023, 104075. https://doi.org/10.1016/j.etap.2023.104075.

61. K. Gedik and A. Raif Eryaşar, *Microplastic Pollution Profile of Mediterranean Mussels (Mytilus galloprovincialis) Collected A long the Turkish Coasts*, **Chemosphere** 260, 2020, 127570.

62. Y. Sun, Y. Liu, & Z. Huang, *Microplastic Contamination in the Surface Waters of the Mediterranean Sea*, **Science of The Total Environment**, 725, 2020, 138594.

63. S. Liu, Y. Liu, Y. He, & F. Zhao, *Inhaled Microplastics Induce Systemic Immune and Metabolic Disruption in Mice*, **Journal of Hazardous Materials**, 408, 2021, 124840.

64. H. Frederiksen, L. Aksglaede, K. Sørensen, N. E. Skakkebæk, A. Juul, A. M. Andersson, *Urinary Excretion of Phthalate Metabolites in 129 Healthy Danish Children and Adolescents: Estimation of Daily Phthalate Intake.* **Environmental Research**, 111, 2011, 656–663.doi: 10.1016/j.envres.2011.03.005

65. D. Gao, Z. Li, H. Wang, & H. Liang, *An Overview of Phthalate Acid Ester Pollution in China over the Last Decade: Environmental Occurrence and Human Exposure.***Science of The Total Environment**, 645, 2018, 1400–1409. doi: 10.1016/j.scitotenv.2018.07.093

66. M. Zarean, P. Poursafa, M. H. Tajadini, M. Ghaffari, & M. M. Amin, *Phthalates Exposure, Human Health Risk Assessment, and Their Reproductive Health Toxicity*, **Environmental Toxicology and Pharmacology**, 89, 2022, 103742.

67. S. E. Serrano, J. Braun, L. Trasande, R. Dills, & S. Sathyanarayana, *Phthalates and Diet: A Review of the Food Monitoring and Epidemiology Data*, **Environmental Health**, 13(1), 2014, 43.

68. C. G. Bornehag, F. Carlstedt, B. A. Jönsson, C. H. Lindh, T. K. Jensen, A. Bodin, & C. Jonsson, *Prenatal Phthalate Exposures and Anogenital Distance in Swedish Boys*, **Environmental Health Perspectives**, 123(1), 2015, 101-107. doi: 10.1289/ehp.1408163

69. L. Sun, M. Lu, X. Zhu, & L. Wu, *Exposure to Phthalates and Human Reproductive Health: Insights from Epidemiology and Experimental Studies*, **Environmental Pollution**, 297, 2022, 118753.

70. M. Chen, L. Zhang, J. Wang, & L. Zheng, *Phthalate Bioaccumulation in Aquatic Food Webs and Human Health Risks*, **Environmental Pollution**, 276, 2021, 116661.

71. F. Zhao, X. Wang, Z. Liang, & D. Yuan, *Phthalate Removal from Wastewater: Advances in Treatment Technologies*, **Water Research**, 204, 2022, 117573.

72. L. Wang, X. Zhang, Y. Zhang, & Q. Wang, *Occurrence of Phthalate Esters in River Sediments: Ecological Risks and Human Exposure*, **Journal of Hazardous Materials**, 408, 2021, 124982.

73. S. Kim, Y. Choi, Y. Kim, & Y. Kim, *Endocrine Disrupting Effects of DEHP on Reproduction in Aquatic Organisms*, **Environmental Toxicology and Pharmacology**, 77, 2020, 103356.

74. Q. Zeng, G. Chen, & Y. Xu, *Toxic Effects of Phthalate Esters on Freshwater Invertebrates: Oxidative Stress and Bioaccumulation*, **Environmental Toxicology**, 37(3), 2022, 456-468.

75. C. Gao, Y. Wang, X. Zhang, & Q. Zhang, *Phthalate Esters in Riverine and Estuarine Sediments: Distribution, Risk Assessment, and Management Strategies*, **Science of the Total Environment**, 769, 2021, 145186.

76. I. S. Fatoki, M. Bornman, & E. J. Ncube, *Phthalates in Coastal Water Environments: Sources, Risks, and Management*, **Marine Pollution Bulletin**, 159, 2020, 111489.

77. D. T. Ogundele, A. O. Ojo, & O. A. Adeyemi, *Microplastics in Lagos Lagoon: An Emerging Threat to the Aquatic Ecosystem*, **Journal of Environmental Management**, 255(2), 2021, 109834.

78. I. S. Ogunola, A. O. Adeogun, & E. O. Akindele, *Microplastics in Tropical Aquatic Environments: The Case of Epe Lagoon, Nigeria*, **Ecotoxicology and Environmental Safety**, 244(1), 2022, 113443.

79. A. O. Ojo, F. A. Akande, & D. T. Ogundele, *Distribution and Characterization of Microplastics in Surface Waters of Lagos Lagoon*, **Science of the Total Environment**, 859(4), 2023, 160341.

80. A. O. Adeogun, O. S. Ogunola, & E. O. Akindele, *Microplastic Contamination in Sediment and Water of a Tropical Lagoon*, **Marine Pollution Bulletin**, 184(2), 2022, 1009-1022.

81. O. A. Adada, & A. O. Adeogun, *Microplastics in Carnivorous Fish Species, Water and Sediments of a Nigerian Lagoon Ecosystem.* **Environmental Science and Pollution Research**, 30(3), 2023, 2045–2058. https://doi.org/10.1007/s11356-023-26410-w

82. O. B. Akintan, J. A. Olusola, & A. M. Azeez, *Heavy Metals Loads in Soil, Farmlands and Plant Crop at Open Dumpsite*, **Global J. Environ. Sci. Manage.**, 5(4), 2019, 449-460.

83. A. O. Afolayan, *Accumulation of Heavy Metals from Battery Waste Topsoil Surface Water, and Garden Grown Maize at Omilende Area, Olodo, Nigeria*, **Global Challenges**, 2, 2018, 1-12.

84. A. T. Ibrahim, D. O. Odulate, & K. O. Olayinka, *Distribution of Heavy Metals in Water and Sediments from Lagos Lagoon, Nigeria*, **Journal of Health and Pollution**, 9(24), 2019, 191211. https://doi.org/10.5696/2156-9614-9.24.191211

85. J. C. Akan, F. I. Abdulrahman, G. A. Dimari, & V. O. Ogugbuaja, *Heavy Metal Contamination in Urban and Peri-urban Aquatic Ecosystems: A Review of Sources, Pathways, and Effects*, **Journal of Environmental Sciences**, 98, 2020, 141-159.

86. A. A. Otitoloju, A. O. Olagoke & O. Oladipo, *Heavy Metal Contamination and Its Ecological Risk Assessment in Sediments of the Lagos Lagoon, Nigeria*, **Environmental Monitoring and Assessment**, 191(5), 2019, 305. https://doi.org/10.1007/s10661-019-7445-8

87. O. A. Adeniyi, & K. A. Yusuf, *Microplastic Contamination in Lagos Lagoon, Nigeria: The Role of Urbanization and Waste Management*, **Science of the Total Environment**, 685, 2019, 577-583. https://doi.org/10.1016/j.scitotenv.2019.05.303

88. J. A. O. Oyekunle, O. S. Olatunji, O. S. Fatoki, & A. O. Ogunfowokan, *Occurrence and Distribution of Microplastics in Surface Waters and Sediments of Lagos Lagoon, Nigeria*, **Environmental Science and Pollution Research**, 28, 2021, 41067-41080. https://doi.org/10.1007/s11356-021-13691-3

89. O. S. Ogunola, O. A. Onada, & A. E. Falaye, *Microplastics in Marine Environment: Current Trends in Environmental Pollution and Mechanisms of Toxicological Profile*, **Environmental Science and Pollution Research**, 25(9), 2018, 9293-9304. https://doi.org/10.1007/s11356-018-1232-5

90. O. A. Nubi, O. A. Osibote, & O. S. Amuda, *Microplastic Pollution in Lagos Lagoon: Abundance, Characteristics, and Sources*, **Environmental Monitoring and Assessment**, 192(5), 2020, 315. https://doi.org/10.1007/s10661-020-8225-8

91. A. Jiang, L. Yin, L. Li, X. Wen, X. Luo, & J. Wang, *Microplastics as Vectors of Toxic Chemicals: Insights into Their Mechanisms and Environmental Impacts*, **Journal of Hazardous Materials**, 426, 2022, 128112. https://doi.org/10.1016/j.jhazmat.2021.128112

92. O. S. Ogunola, O. A. Onada, & A. E. Falaye, *Impacts of Microplastics on Fish and Aquatic Organisms: The Nigerian Perspective*, **Environmental Science and Pollution Research**, 27(22), 2020, 27661-27673. https://doi.org/10.1007/s11356-020-09218-3

93. A. M. Rochman, A. Tahir, S. L. Williams, D. V. Baxa, R. Lam, J. T. Miller, F. C. Teh, S. Werorilangi, & S. J. Teh, *Anthropogenic Debris in Seafood: Plastic Debris and Fibers from Textiles in Fish and Bivalves Sold for Human Consumption*, **Scientific Reports**, 5, 2019, 14340. https://doi.org/10.1038/srep14340

94. X. Xu, W. Liu, W. Wei & Z. Lu, *Microplastics in the Freshwater Environment: Occurrence, Distribution, and Risks*. **Science of the Total Environment**, 536, 2016, 739-744. https://doi.org/10.1016/j.scitotenv.2015.12.060

95. A. Koelmans, E. Besseling, & E. M. Foekema, *Leaching of Plastic Additives to Marine Organisms*. **Environmental Pollution**, 212, 2016, 238-249. https://doi.org/10.1016/j.envpol.2016.01.012

96. M. Jang, W. J. Shim, G. M. Han, M. Rani, Y. K. Song & S. H. Hong, *Styrofoam Debris as a Source of Hazardous Additives for Marine Organisms*. **Environmental Science & Technology**, 50(10), 2020, 4951-4960. https://doi.org/10.1021/acs.est.6b02602

97. M. Kumar, M., X. Xiong, M. He, D. C. W. Tsang, J. Gupta, E. Khan, & S. Harrad, *Microplastics as Pollutants in Aquatic Ecosystems: Determination, Toxicological Implications, and Remediation*. **Journal of Hazardous Materials**, 403, 2021, 123910. https://doi.org/10.1016/j.jhazmat.2020.123910

98. F. Liu, K. B. Olesen, A. R. Borregaard, & J. Vollertsen, *Microplastics in Urban and Highway Stormwater Retention Ponds*. **Science of the Total Environment**, 707, 2020, 136010. https://doi.org/10.1016/j.scitotenv.2019.136010

99. A. Naji, Z. Esmaili, & F. R. Khan, *Plastic Debris and Microplastics Along the Beaches of the Strait of Hormuz, Persian Gulf*. **Marine Pollution Bulletin**, 114(4), 2021, 1057-1062. https://doi.org/10.1016/j.marpolbul.2016.10.001

100. K. Zhang, H. Shi, J. Peng, Y. Wang, X. Xiong, C. Wu, & P. K. S. Lam, *Microplastic Pollution in China's Inland Water Systems: A Review of Findings, Methods, Characteristics, Effects, and Management*. **Science of the Total Environment**, 579, 2020, 1484-1495. https://doi.org/10.1016/j.scitotenv.2016.11.137

101. S. L. Wright, & F. J. Kelly, *Plastic and Human Health: A Micro Issue?* **Environmental Science & Technology**, 51(12), 2020, 6634-6647. https://doi.org/10.1021/acs.est.7b00423

102. United Nations Environment Programme (UNEP), *Plastic Pollution in Marine Environments: Status and Trends*. UNEP, 2019. https://www.unep.org

103. European Commission, *A European Strategy for Plastics in a Circular Economy*. European Union, 2018. https://ec.europa.eu/environment/circular-economy/pdf/plastics-strategy-brochure.pdf

104. K. R. Vanapalli, K. R., H. B. Sharma, V. P. Ranjan, B. Samal, J. Bhattacharya, B. K. Dubey, & S. Goel, *Challenges and Strategies for Effective Plastic Waste Management During and Post COVID-19 Pandemic*. **Science of the Total Environment**, 750, 2021, 141514. https://doi.org/10.1016/j.scitotenv.2020.141514

105. G7, *Ocean Plastics Charter*. G7 Summit 2018. https://www.canada.ca/en/environment-climate-change/services/managing-reducing-waste/international-commitments/ocean-plastics-charter.html

106. International Maritime Organization (IMO), *Action Plan to Address Marine Plastic Litter From Ships*. IMO 2019. https://www.imo.org/en/MediaCentre/PressBriefings/Pages/24-marine-litter-action-plan-.aspx

[107.] O. J. Ogunbiyi, *Impacts of Microplastics on Lagos Lagoon Ecosystems*. **Journal of Environmental Studies**, 15(4), 2021, 234-245. https://doi.org/10.1016/j.envst.2021.234

[108.] National Emergency Management Agency (NEMA), *Annual Report on Environmental Hazards and Pollution Control in Nigeria*. **NEMA**, 2020. https://www.nema.gov.ng

[109.] O. S. Alimi, J. F. Budarz, L. M. Hernandez, & N. Tufenkji, *Microplastics and Nanoplastics in Aquatic Environments: Aggregation, Deposition, and Enhanced Contaminant Transport*. **Environmental Science & Technology**, 52(4), 2018, 1704-1724.

[110.] C. Zhang, J. Ding, Y. Liu, L. Wang, X. Zhang, & P. Christie, *Plastic Particles Reduce Microbial Activity and Carbon Utilization Efficiency in Agricultural Soils*. **Environmental Pollution**, 284, 2022, 117152. https://doi.org/10.1016/j.envpol.2021.117152

[111.] S. Mukherjee, S. Chatterjee, M. Kumar, & P. Basu, *Microplastic Pollution in the Environment and its Implications for Human Health*. **Environmental Science & Pollution Research**, 28(16), 2021, 20544-20556. https://doi.org/10.1007/s11356-021-12778-1

[112.] United States Environmental Protection Agency (US EPA), *Aquatic Life Ambient Water Quality Criteria for Cadmium 2016*. US EPA, 2016. https://www.epa.gov

[113.] Government of Canada, *Taking Action to Reduce Plastic Pollution: Canada's Plastic Science Agenda*. Government of Canada, 2019. https://www.canada.ca/en/environment-climate-change/services/managing-reducing-waste/reduce-plastic-pollution/plastic-science-agenda.html

[114.] United Nations Environment Assembly (UNEA), *Resolution to End Plastic Pollution: A Global Response*. UNEP, 2022. https://www.unep.org/events/un-environment-assembly/resolution-end-plastic-pollution-global-response

[115.] Organisation for Economic Co-operation and Development (OECD), *Global Plastic Waste Management Outlook*. OECD, 2020. https://doi.org/10.1787/8891b2a1-en

[116.] M. Tiwari, A. Rathore, & S. K. Pandey, *Microplastics in the Environment: Occurrence, Detection, and Remediation Techniques*. **Environmental Science and Pollution Research**, 28(15), 2021, 18725-18740. https://doi.org/10.1007/s11356-021-13215-7

# Chapter Three

# Methodology

## 3.1 Research Design

This research was conducted in several stages and aimed to identify microplastics and phthalate ester concentrations in *Oreochromis niloticus* and *Clarias gariepinus*, assess the physicochemical properties of benthic and surface waters, and determine the levels of heavy metals in both water types, as well as the seasonal variations in these pollutants. The first stage involved site visitation, location profiling, and sample collection. Samples were collected on four separate occasions between January and November to allow for seasonal variation analysis. The second stage comprised sample preparation, laboratory procedures, and analytical work.

## 3.2 Study Area

The Lagos and Epe Lagoons are integral parts of a larger network of water bodies located in southwestern Nigeria. These lagoons are vital to the ecology and economy of Lagos State, one of the most densely populated and economically significant states in Nigeria[1,2]. Together with other coastal water systems, they form the Lagos Lagoon System, which extends from the Ogun River in the west to the Lekki Lagoon in the east, covering approximately 635 square kilometers[1,2]. The Lagos Lagoon lies between latitudes 6°26′ N and 3°24′ E. This lagoon acts as a major waterway connecting inland rivers to the Atlantic Ocean and is highly urbanized, particularly around Apapa, Ikoyi, and Victoria Island[3]. Rapid urbanization and industrialization around this lagoon have led to high levels of pollution from industrial discharges and urban runoff [4].

The Epe Lagoon, located to the east of Lagos Lagoon, is more rural and spans approximately 225 square kilometers. Fed mainly by the Osun River, it provides a less polluted environment due to

its distance from industrial activities[5]. The primary human activities here include agriculture and artisanal fishing[6].

The ecological conditions of Lagos and Epe Lagoons differ, largely influenced by variations in the intensity and nature of human activities within their surrounding areas. The Lagos Lagoon sustains a brackish water ecosystem due to its proximity to the Atlantic Ocean[1]. However, it faces serious ecological challenges from urban development, with the encroachment on mangrove swamps that serve as breeding grounds for many aquatic species[1]. The lagoon's biodiversity has been negatively affected by pollution, leading to periodic algal blooms and other ecological imbalances[7].

The Epe Lagoon supports a more balanced ecosystem compared to Lagos Lagoon, with extensive mangrove forests and swampy areas that provide habitats for various aquatic species[8]. Being a rural environment, Epe Lagoon experiences less pollution, which helps sustain its biodiversity, especially for fish species important to local fisheries[8].

In contrast, Epe Lagoon experiences more stable hydrological conditions, with freshwater dominating throughout the year due to the limited influence of tides[9]. Water quality monitoring in both lagoons indicates that Lagos Lagoon is significantly more polluted, with higher concentrations of dissolved oxygen, total suspended solids (TSS), and chemical oxygen demand (COD)[4]. In Epe Lagoon, nutrient levels are increasing, particularly phosphates and nitrates, which exacerbate algal blooms and oxygen depletion[9].

Runoff from nearby farms introduces excessive nutrients like phosphates and nitrates into the lagoon, resulting in eutrophication. Although Epe Lagoon is less industrialized, sand mining and

artisanal fishing have increased in recent years, raising concerns about habitat destruction and unsustainable resource use[9].

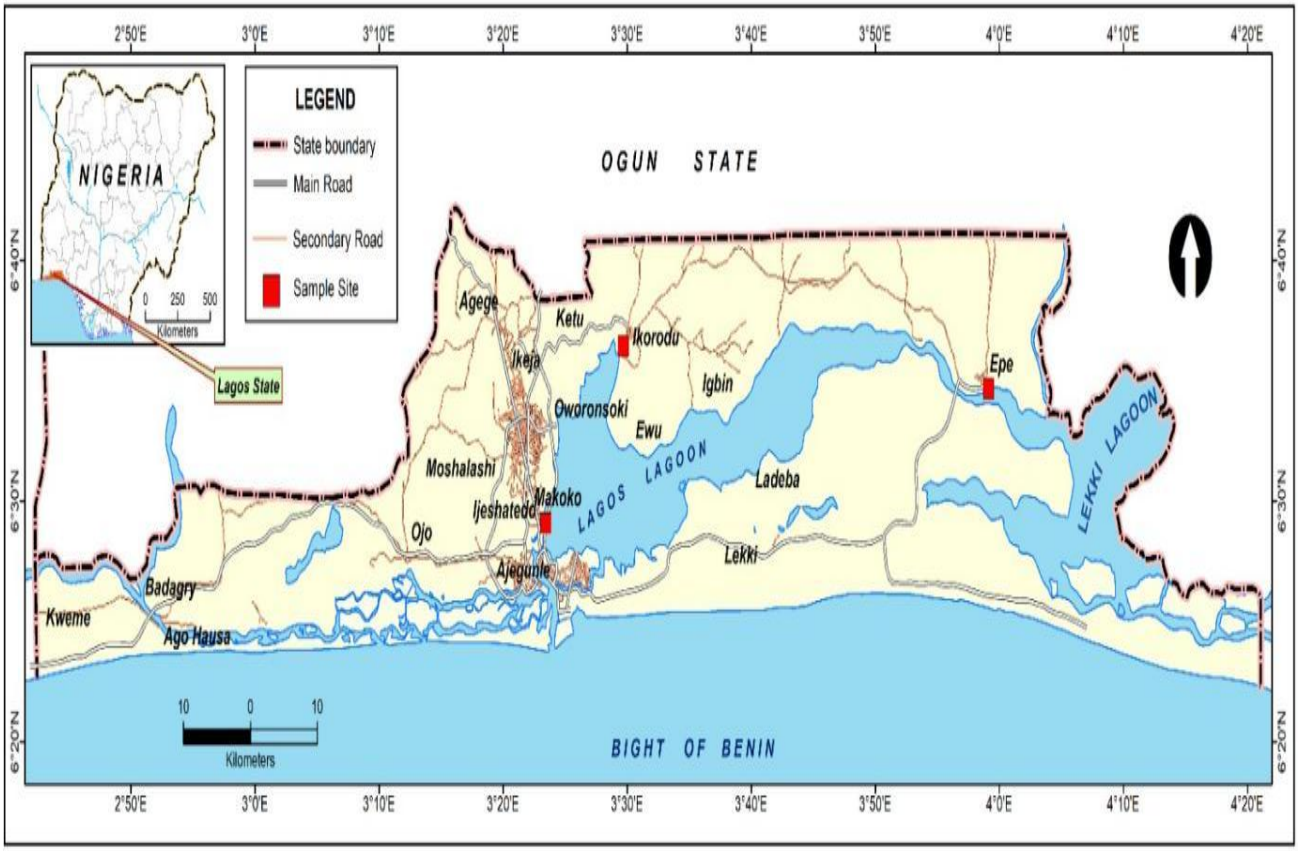


**Figure 3.1:** The Map of Lagos and Epe Lagoons

**Source:** Author's Field Work, 2025


**Figure 3.2:** Makoko Axis of the Lagos Lagoon

**Source**: Author's Field Work, 2025

**Figure 3.3:** Ikorodu Axis of the Lagos Lagoon

**Source**: Author's Field Work, 2025

**Figure 3.4:** The Epe Lagoon

**Source**: Author's Field Work, 2025

## 3.3 Sample Collection

### 3.3.1 Water Sample Collection

Water samples were collected from both the surface and subsurface regions of the lagoons using clean, sterilized glass bottles after ensuring they were rinsed three times with sample water before final collection[10].

At each sampling location, three 5-litre bottles of surface water and three 5-litre bottles of benthic water were collected during each of the four seasonal sampling cycles. This provided six water samples per site per cycle, ensuring adequate volume for subsequent physicochemical and heavy metal analysis.

To minimize the influence of surface films and floating debris, water samples were collected at a depth of approximately 30 cm below the surface using a clean sterilised, glass water sampler. In situ parameters like temperature, pH, and dissolved oxygen were measured immediately using portable field instruments[4].

Water samples were stored in ice-filled coolers and transported promptly to the laboratory for analysis to prevent changes in chemical composition. Proper labelling of the sample location, date, and time was carried out for tracking[9].

Seasonal variations in water samples will be determined from the results of all water sample analysis conducted after monitoring for one full cycle (Dry and rainy season).

### 3.3.2 Fish Sample Collection

Fish samples were collected from each of the three designated study sites(Makoko, Itamarun and Ipakodo), representing two commercially important species: *Oreochromis niloticus* (Tilapia) and *Clarias gariepinus* (African catfish). For each species, five individuals were obtained per site

during each of the four seasonal sampling cycles. This resulted in a total of 15 individuals per species per cycle, corresponding to 30 fish per cycle and an overall total of 120 specimens over the course of the study.

Fish samples of both species were obtained from artisanal fishermen early in the morning when fish activity is high and water conditions are stable[11]. Samples are collected early in the morning or late in the evening to align with peak fish activity. Captured fish are measured for morphometrics, and samples are preserved for further analysis, ensuring minimal contamination or sample degradation during transport[9]. The samples were stored in a cooler filled with ice and transported to the department of marine biology, University of Lagos for taxonomic identification by Mr Monday a fish expert, and for preparation and further analysis.

Seasonal variations in fish samples will be determined from results of all fish sample analysis conducted.

**Figure 3.5:** Tilapia Fish *(Oreochromis niloticus)*

**Source*:*** Author's Field Work, 2025

**Figure 3.6:** African Cat Fish *(Clarias gariepinus)*

**Source**: Author's Field Work, 2025

### 3.4 Laboratory Analyses

### 3.5 Determination of Morphometric Characteristics

Collected fish specimens were identified to species level based on distinct morphometric characteristics, including Total Length (TL), Standard Length (SL), Head Length (HL), Body Depth (BD), Pre-dorsal Length (PDL) and Pre-anal Length (PAL).

Each fish specimen was weighed using a digital weighing balance to the nearest 0.01g to record the total body weight. Other measurements were taken using a digital calliper (precision of 0.01mm), and each trait was measured three times to ensure precision. The scales were recalibrated before every measurement, and samples were measured multiple (5) times to ensure accuracy and reproducibility of results[12].

### 3.6 Determination of Physicochemical Properties of Surface and Subsurface Water

The water samples were analyzed according to the USEPA 1669 standard method for water analysis. Parameters measured include temperature, pH, turbidity, dissolved oxygen, salinity, total dissolved solids, total suspended solids and conductivity. Parameters like temperature was measured directly using a calibrated thermometer.

The pH was measured with a pH meter. It was calibrated with standard buffer solutions at pH 4, 7, and 10. The electrode was rinsed with distilled water before each reading, after which it was submerged in the water sample, ensuring the electrode tip was fully immersed. The pH reading was allowed to stabilize, and the value was recorded. Repeated calibration was needed between samples to maintain accuracy[13].

Temperature measurement was taken by submerging a thermometer probe directly into the water sample. It was allowed to stabilize for a few seconds, and the temperature was recorded in degrees Celsius (°C).

Turbidity was measured by calibrating a Nephelometric Turbidity Unit (NTU) meter using prepared standards. It was gently inverted into the water sample to ensure homogeneity. The sample was poured into a clean vial, avoiding air bubbles, then inserted into the NTU meter, and the lid closed. Readings were recorded in NTUs[13].

The Total Dissolved Solids (TDS) was measured by calibrating the TDS meter according to the manufacturer's instructions. The probe was rinsed with distilled water and submerged into the water sample until fully immersed. After waiting for the reading to stabilize, the TDS value was recorded in (mg/L)[13].

The dissolved oxygen (DO) was measured by calibrating the DO meter with the zero oxygen and air-saturated solutions. The probe was then submerged into the water sample without introducing air bubbles. The reading was allowed to stabilize, and the DO concentration was recorded in mg/L[14].

The Electrical Conductivity of the sample was measured by calibrating the conductivity meter using a known standard solution. The probe was rinsed with distilled water between samples then immersed in the water sample, ensuring complete submersion. The reading was allowed to stabilize, and the conductivity recorded in µS/cm.

The Total Suspended Solids was measured by filtering 500 mL of the sample through a pre-weighed filter. The filter was dried in the oven at 103–105°C until a constant weight was

achieved, and the dried filter was weighed on an analytical balance. The TSS (mg/L) was calculated using the difference between the pre- and post-dried weights[13].

### 3.7 Determination of Heavy Metals

The heavy metals of both benthic and surface waters were analysed using the USEPA method 200.8. The water samples were analysed for heavy metals including Cadmium (Cd), Chromium (Cr), Lead (Pb), Nickel (Ni), and Copper (Cu) using an atomic absorption spectrometer (AAS)[15].

#### 3.7.1 Digestion of Samples

Water samples were digested following a standardized acid suitable for trace metals analysis according to United States Environmental Protection Agency (USEPA) method 3050b[1]. 100ml of each water sample was measured into a 250ml conical flask. To each flask, 10ml of concentrated nitric acid ($HNO_3$) was added and heated gently on a hot plate until the volume of the mixture reduced to 25ml. 2ml of hydrogen peroxide was added continuously in batches. Heating continued until the solution became clear/bright yellow then it was allowed to cool. After cooling, the digested solution was filtered with Whatman No. 42 filter paper to remove any particulate. The filtrates were then preserved in a 100ml volumetric flask and made up to volume with deionized water. Samples were stored at $4^0$C until analysis for the following heavy metals using Atomic Absorption Spectrophotometer.

#### 3.7.2 Heavy Metal Determination

The filtrates obtained from digestion were being analyzed for the following heavy metals, Pb, Cu, Ni, Cd and Cr, using atomic absorption spectrophotometer. The digested sample was fed into the AAS. The reading of the metals concentration for each sample was obtained and recorded.

### 3.7.3 AAS Condition

The model of the Atomic Absorption Spectrophotometer (AAS) employed in the analysis of heavy metals is: Alpha 4 AAS, made by Buck Scientific. The analytical condition of the AAS is summarized in Table 3.7.3 below

**Table 3.1:** Alpha 4 Atomic Absorption Spectroscopy (AAS) Working Conditions

| Element | Wavelength (nm) | Slit Width (nm) | Working Range (µg/mL) | Sensitivity (µg/mL) | Lamp Current | Flame Type |
|---|---|---|---|---|---|---|
| **Pb** | 283.3 | 0.7 | 0.5-5 | 45 | 10 | Air-$C_2H_2$ |
| **Cd** | 228.8 | 0.5 | 0.5-5 | 0.03 | 4 | Air-$C_2H_2$ |
| **Cu** | 324.8 | 0.2 | 2.0-20 | 0.2 | 7 | Air- $C_2H_2$ |
| **Ni** | 213.9 | 0.5 | 4.0-40 | 0.2 | 5 | Air- $C_2H_2$ |
| **Cr** | 357.9 | 0.5 | 0.5-5 | 0.03 | 3 | Air- $C_2H_2$ |

**Source**: Author's Field Work, 2025

### 3.8 Determination of Microplastic Concentration

#### 3.8.1 Digestion of Fish Samples

The fish samples were thawed and carefully dissected to extract the gastrointestinal tract and other target tissues. The tissues were rinsed thoroughly with distilled water to remove external debris and potential surface contaminants. To prevent contamination, all procedures were conducted using non-plastic tools and containers. The dissected tissues were then digested using a microwave-assisted tissue digestion protocol adapted from USEPA Method 3546, which allowed removal of organic matter prior to filtration and microplastic identification.

5g of the tissue sample was placed in a glass beaker, and 10% potassium hydroxide (KOH) was added to break down the organic material. The beaker was covered with foil and allowed to digest at room temperature for 48 hours until the tissue was fully digested. The digested solution was filtered through a 0.45 µm stainless steel mesh, and the mesh was rinsed with distilled water to ensure that all the microplastics were captured[16].

#### 3.8.2 Microplastic Occurence Analysis

Visual identification was carried out using optical microscopy. Size, shape and colour were observed and recorded. Agilent Fourier-Transform Infrared Spectroscopy (FTIR) was used to identify and characterize the microplastics and the chemical composition of the microplastics by comparing the spectra to known polymer libraries. Typical polymers include polyethylene (PE), polypropylene (PP), polystyrene (PS), and polyvinyl chloride (PVC). The identified microplastics were classified based on their size, shape and colour[16].

### 3.9 Determination of Phthalate Esters in Fish Tissues

Fish tissue samples were collected using solvent-rinsed stainless-steel instruments. The samples were homogenized and immediately stored in pre-cleaned, phthalate-free glass containers at -

20°C. 10–20 g of homogenized fish tissue was weighed into a pre-cleaned glass container. The internal standard solution was added to correct for extraction efficiency and instrument response[17].

### 3.9.1 Solvent Extraction of PAEs

20 mL of a hexane-acetone mixture (1:1, v/v) was added to the sample. The mixture was sonicated for 30 minutes or in a shaker for 1 hour, then centrifuged at 4,000 × g for 15 minutes, and then the organic phase was decanted. This extraction was repeated two additional times, combining the organic phases[17].

### 3.9.2 Sample Clean-up

The combined extracts were dried with anhydrous sodium sulfate, and the extract was passed through a pre-conditioned SPE cartridge packed with Florisil or silica gel to remove lipids and impurities. The elution was concentrated to approximately 1 mL under a gentle nitrogen stream. The final extract was sealed and kept for further analysis[17].

### 3.9.3 Instrumental Analysis

The sample was reconstituted in 1 mL of hexane. 1 µL was injected into the GC-MS system.

A temperature-programmed GC oven was used with the following conditions:

**Initial temperature:** 50°C, hold for 2 minutes.

**Ramp:** 10°C/min to 280°C, hold for 10 minutes.

The compounds were analysed using the SIM mode, monitoring characteristic ions of the target phthalates[17].

### 3.10 Quality Control/Quality Assurance

Throughout the experiment, rigorous quality assurance and quality control protocols were strictly adhered to, ensuring the reliability and accuracy of the results. Safety was a paramount consideration during all phases of the research, from field sampling to laboratory analysis. Appropriate sampling equipment, containers, and preservation methods were meticulously selected and utilized to avoid contamination of the samples. Special care was taken to handle all materials under clean and controlled conditions[19].

All glassware used for metal analysis underwent thorough pre-treatment by soaking in 14% nitric acid ($HNO_3$) for 24 hours, followed by multiple rinses with deionized and filtered water to eliminate any potential residues or contaminants. This pre-treatment step was critical to ensuring the integrity of subsequent analytical processes. Additionally, all reagents employed during the experiment were of analytical grade, further minimizing the risk of introducing external contaminants into the samples[19].

The use of plastic materials was avoided throughout the process. Only pre-cleaned glassware and solvent-rinsed tools were used.

Recovery studies were performed using spiked fish samples. Acceptable recovery ranges were between 70–120%. Duplicate samples were analysed to assess precision, aiming for a relative standard deviation (RSD) below 15%. EPA's guidelines for sample preservation (e.g., keeping samples cool and analyzing within specified holding times) was strictly followed[17].

To address potential sources of error and improve data accuracy, reagent blanks were included in the analyses to correct for any background interference. Multiplicity of samples for each determination was implemented to enhance reproducibility, enabling reliable statistical analysis

of the results. Measures were also taken to minimize and eliminate all possible sources of instrumental and procedural contamination, ensuring that data obtained were reflective of the actual conditions under study[20].

To verify the robustness of the analytical methods, spiked blanks, reagent blanks, and appropriate standard reference materials were analyzed alongside each set of samples. The inclusion of spiked blanks involved the addition of known quantities of analytes to blank samples to assess the recovery rates of the methods employed. Reagent blanks served to account for any contamination originating from the chemicals used in the analytical process, while standard reference materials provided a benchmark for accuracy and method validation. This multi-faceted approach ensured the integrity of the analytical techniques and the reliability of the results obtained[21].

### 3.11 Data Analysis

#### 3.11.1 Statistical Analysis of Results

Descriptive statistics, such as standard deviation, and variance, were calculated to summarize the data and identify trends. Inferential statistics were used to test hypotheses and draw conclusions about the relationships between variables. The specific tests employed included:

i. **Analysis of Variance (ANOVA)**: To determine whether there were significant differences in microplastic concentrations between the sampled locations and seasonal variations.

ii. **T-tests**: For comparing paired datasets, such as differences in microplastic concentrations between benthic and surface waters.

iii. **Correlation Analysis**: To evaluate potential associations between physicochemical variables and microplastic abundance, correlation analysis was carried out using standard statistical procedures.

iv. **Regression Analysis**: To model the interaction between dependent and independent variables, such as the impact of environmental factors on microplastic concentration.

All statistical tests were performed at a significance level of 0.05 ($p < 0.05$).

### 3.11.2 Contamination Factor

The Contamination Factor (CF) is a key metric for assessing the degree of heavy metal pollution in this study. It quantifies the level of contamination by comparing the concentration of heavy metals in different samples to a baseline or reference concentration. By calculating CF, this research will provide a clear and quantifiable measure of heavy metals contamination, enabling effective communication of pollution severity and aiding decision-making for mitigation efforts.

$CF = C_{sample} / C_{background}$ ..... Equation 3.1

Where: $C_{sample}$ = The concentration of heavy metal in the sample (e.g., Benthic water, or Surface water).

$C_{background}$ = A reference or baseline concentration of the different heavy metals, typically derived from less contaminated or natural environments.

Contamination factor has four categories which include:

<1= low contamination;

1-3= moderate contamination;

3 – 6= considerable contamination;

>6= very high contamination factor[21]

### 3.11.3 Degree of Contamination

The degree of contamination of the sampling sites was evaluated using two indices, Contamination factor and Pollution Load index.

This is the sum of all the contamination factors of all the elements in the sample[11]. It is indicated in Equation 3.2:

$C_{deg} = \Sigma CF$ ……Equation 3.2

Where

CF= Contamination factor of each element of interest;

Four categories have been defined for the degree of contamination which includes:

<8=low degree of contamination;

8-16=moderate degree of contamination;

16- 32=considerable degree of contamination;

>32=very high degree of contamination[21].

### 3.11.4 Pollution Index (PI)

A pollution index was employed to categorize the pollution status of the study sites based on the heavy metals' levels. The PI was calculated using a normalized scale, allowing for easy comparison of pollution levels between benthic and surface waters, as well as between seasons.

### 3.11.5 Software Tools

All statistical analyses were performed using the Statistical Package for the Social Sciences (SPSS, version 25) and Microsoft Excel 2019 for data visualization and preliminary calculations. For complex modelling and multivariate analyses, R (version 4.3.0) was utilized. Graphical

presentations, including bar charts, line graphs, and scatter plots, were created to illustrate findings clearly and effectively.

### 3.11.6 Quality Assurance in Data Analysis

To ensure data integrity, the following measures were implemented:

i. Data cleaning and verification processes were conducted to identify and correct errors or inconsistencies in raw data.

ii. Outlier analyses were performed to detect and address anomalous values that could skew results.

iii. Replicate measurements and validation tests were carried out to confirm the reproducibility and accuracy of results.

## Endnotes


1. D. I. Nwankwo, *Euglenoids of Some Polluted Storm-water Channels in Lagos, Nigeria*, **Tropical Freshwater Biology**, 4, 1995, 29–39.

2. E. A. Ajao, E. O. Oyewo, & J. P. Unyimadu, *A Review of the Pollution of Coastal Waters in Nigeria*, **NIOMR Technical Paper**, Nigerian Institute for Oceanography and Marine Research, Lagos, Nigeria, 107, 1996.

3. K. Kusemiju, *A Study of the Catfish (Clarias lazera) in the Lekki Lagoon, Lagos, Nigeria*, **Journal of Fish Biology**, 5(5), 1973, 473–478.

4. M. S. Bhuyan, M. A. Bakar, A. Akhtar, M. B. Hossain, M. M. Ali, & M. S. Islam, *Distribution of Heavy Metals in Surface Sediments of the Buriganga River and Ecological Risk Assessment.* **Chemosphere**, 287, 2022, 132147, https://doi.org/10.1016/j.chemosphere.2021.132147

5. A. A. Olatunde, *Some Aspects of the Biology of Synodontis Species in Jebba Lake, Nigeria*, **Journal of Aquatic Sciences**, 4, 1989, 49–54.

6. M. M. Ali, M. L. Ali, M. S. Islam, & M. Z. Rahman, *Assessment of Heavy Metal Contamination in Water, Sediment, and Fish Tissues from a Freshwater River in Bangladesh.* **Environmental Science and Pollution Research**, 28, 2021, 12329–12343. https://doi.org/10.1007/s11356-020-11155-1

7. L. U. Onyia, F. A. Okoh, & E. O. Ayotunde, *Ecological Characterization and Abundance of Clarias gariepinus in Cross River System, Nigeria.* **Egyptian Journal of Aquatic Biology & Fisheries**, 26(1), 2022, 217–234, https://doi.org/10.21608/ejabf.2022.215341

8. O. O. Olawusi-Peters, & J. R. Solomon, *Environmental Parameters and Fish Assemblages of the Badagry-Lagos Coastal Lagoon System, Nigeria,* **West African Journal of Applied Ecology**, 29(2), 2021, 1–14. https://doi.org/10.4314/wajae.v29i2.1.

9. C. O. Akagha, A. O. Edegbene, & A. E. Igbehinse, *Pollution Assessment in Surface Waters of Lagos Lagoon using Biotic Indices*, **Nigerian Journal of Fisheries and Aquatic Sciences**, 17(1), 2019, 37–44.

10. American Public Health Association. *Standard Methods for the Examination of Water and Wastewater* (21st ed.), APA, 2005

11. Food and Agriculture Organization (FAO), *Code of Conduct for Responsible Fisheries*, **Food and Agriculture Organization of the United Nations**, Rome, 2005.

12. United States Environmental Protection Agency (US EPA), *Field Sampling Procedures for Fish. In National Rivers and Streams Assessment*, **Field Operations Manual**, EPA-841-B-12-009, 2013.

13. United States Environmental Protection Agency (US EPA), *Clean Water Act Analytical Methods. Method 1669*, 2023. Available at: https://www.epa.gov

14. United States Environmental Protection Agency (US EPA), *Ambient Water Quality Criteria for Dissolved Oxygen. Method 360.1*, **EPA Report**, EPA 440/5-86-003, 1984.

15. United States Environmental Protection Agency (US EPA), *Determination of Trace Elements by Stabilized Temperature Graphite Furnace Atomic Absorption. Method 200.9*, **EPA Report**, Revision 2.2, EPA/600/R-94/111, 1994.

16. United States Environmental Protection Agency (US EPA), *Microwave Assisted Acid Digestion of Sediments, Soils, and Oils. Method 3051A*, **EPA Report**, 1997.

17. United States Environmental Protection Agency (US EPA), *Semi-Volatile Organic Compounds by Gas Chromatography/Mass Spectrometry (GC/MS). Method 8270D*, **EPA Office of Solid Waste**, Washington, DC, 2007.

18. H. Barrett, J. Cheeseman, & B. D. Hardesty, *Quantifying Plastics in the Environment: Trends, Challenges, and Future Directions.* **Science of the Total Environment**, 798, 2021, 149249. https://doi.org/10.1016/j.scitotenv.2021.149249

19. G. W. Chowdhury, M. Y. Sarker, & M. S. Rahman, *Microplastic Contamination in the Marine Environment: A Review of the Global Scenario, Sources, Fate, Impacts and Potential Solutions.* **Environmental Challenges**, 5, 2021, 100312. https://doi.org/10.1016/j.envc.2021.100312

20. A. A. Koelmans, *Microplastics in Freshwaters and Drinking Water: A Review and Assessment of Environmental Exposure*, **Science of the Total Environment**, 566, 2019, 489–498.

21. A. L. Lusher, *Microplastic Sampling: The Effectiveness of Different Techniques in Capturing Microplastics in Environmental Matrices*, **Environmental Pollution**, 227, 2017, 188–197.

# Chapter Four

## Results and Discussion of Findings

### 4.1 Results of Findings

This chapter presents the results obtained from the determination of morphometric characteristics, heavy metals, microplastic concentration, Phthalate esters (PAEs) and seasonal variations in microplastic concentration from fish and water samples from the Lagos and Epe lagoons. The results of this comprehensive analysis will provide a deeper understanding of the relationship between microplastic and phthalate esters pollution and the overall health of fish in the marine environment. The subsequent sections will detail the findings and variations that were deduced from the data.

#### 4.1.1 Results of Morphometric Characterization of Fishes

The results of morphometric characteristics of *Oreochromis niloticus* and *Clarias gariepinus* fishes from the Lagos Lagoon (Makoko and Ikorodu axis) and the Epe lagoon are presented in tables 4.1.

**Table 4.1:** Morphometric Characteristics of *Oreochromis niloticus* and *Clarias gariepinus* Across Four Seasonal Cycles in Lagos and Epe Lagoons

| **Standard Length (cm)** | | | | | |
|---|---|---|---|---|---|
| **Species** | **Location** | **March** | **May** | **August** | **November** |
| *Oreochromis* | Makoko | 12.40 ± 1.23[a] | 12.50 ± 1.34[a] | 12.90 ± 1.56[a] | 13.10 ± 1.67[a] |
| *Niloticus* | Ikorodu | 12.80 ± 1.34[a] | 13.00 ± 1.45[a] | 13.30 ± 1.68[a] | 13.50 ± 1.79[a] |
| | Epe | 13.00 ± 1.12[a] | 13.20 ± 1.23[a] | 13.60 ± 1.45[a] | 13.80 ± 1.56[a] |
| *Clarias* | Makoko | 25.20 ± 1.67[a] | 25.50 ± 1.90[a] | 26.20 ± 2.12[a] | 26.70 ± 2.24[a] |
| *Gariepinus* | Ikorodu | 25.80 ± 1.57[a] | 26.00 ± 1.68[a] | 26.80 ± 2.01[a] | 27.50 ± 2.35[a] |
| | Epe | 26.00 ± 1.90[a] | 26.50 ± 2.01[a] | 27.20 ± 2.24[a] | 28.00 ± 2.57[a] |
| **Total Length (cm)** | | | | | |
| *Oreochromis* | Makoko | 15.60 ± 1.34[a] | 15.80 ± 1.45[a] | 16.20 ± 1.68[a] | 16.50 ± 1.79[a] |
| *Niloticus* | Ikorodu | 16.10 ± 1.45[a] | 16.30 ± 1.56[a] | 16.80 ± 1.79[a] | 17.10 ± 2.01[a] |
| | Epe | 16.50 ± 1.23[a] | 16.80 ± 1.34[a] | 17.20 ± 1.57[a] | 17.50 ± 1.68[a] |
| *Clarias* | Makoko | 30.50 ± 2.24[a] | 31.00 ± 2.35[a] | 32.00 ± 2.80[a] | 33.20 ± 3.02[a] |
| *Gariepinus* | Ikorodu | 31.00 ± 2.01[a] | 31.80 ± 2.13[a] | 33.00 ± 2.57[a] | 34.00 ± 3.13[a] |
| | Epe | 32.00 ± 2.35[a] | 32.50 ± 2.46[a] | 33.50 ± 3.02[a] | 35.00 ± 3.35[a] |
| **Head Length (cm)** | | | | | |
| *Oreochromis* | Makoko | 3.50 ± 0.45[a] | 3.60 ± 0.56[a] | 3.80 ± 0.67[a] | 4.00 ± 0.79[a] |
| *niloticus* | Ikorodu | 3.70 ± 0.33[a] | 3.80 ± 0.45[a] | 4.00 ± 0.56[a] | 4.30 ± 0.67[a] |
| | Epe | 3.90 ± 0.56[a] | 4.00 ± 0.67[a] | 4.20 ± 0.78[a] | 4.50 ± 0.89[a] |
| *Clarias* | Makoko | 6.80 ± 0.56[a] | 7.00 ± 0.67[a] | 7.30 ± 0.78[a] | 7.50 ± 0.89[a] |
| *gariepinus* | Ikorodu | 7.20 ± 0.45[a] | 7.40 ± 0.56[a] | 7.70 ± 0.67[a] | 7.90 ± 0.78[a] |
| | Epe | 7.50 ± 0.67[a] | 7.60 ± 0.78[a] | 7.90 ± 0.90[a] | 8.10 ± 1.01[a] |
| **Pre-dorsal Length (cm)** | | | | | |
| *Oreochromis* | Makoko | 5.20 ± 0.34[a] | 5.30 ± 0.45[a] | 5.50 ± 0.56[a] | 5.70 ± 0.67[a] |
| *Niloticus* | Ikorodu | 5.50 ± 0.45[a] | 5.60 ± 0.56[a] | 5.70 ± 0.67[a] | 5.90 ± 0.78[a] |
| | Epe | 5.80 ± 0.56[a] | 5.90 ± 0.67[a] | 6.00 ± 0.78[a] | 6.20 ± 0.89[a] |
| *Clarias* | Makoko | 12.00 ± 1.12[a] | 12.30 ± 1.23[a] | 12.90 ± 1.34[a] | 13.00 ± 1.45[a] |
| *Gariepinus* | Ikorodu | 12.50 ± 0.89[a] | 12.80 ± 1.01[a] | 13.30 ± 1.23[a] | 13.50 ± 1.34[a] |
| | Epe | 13.00 ± 1.01[a] | 13.20 ± 1.12[a] | 13.60 ± 1.34[a] | 13.90 ± 1.56[a] |
| **Pre-anal Length (cm)** | | | | | |
| *Oreochromis* | Makoko | 6.10 ± 0.56[a] | 6.20 ± 0.67[a] | 6.40 ± 0.79[a] | 6.60 ± 0.89[a] |
| *Niloticus* | Ikorodu | 6.30 ± 0.45[a] | 6.40 ± 0.56[a] | 6.60 ± 0.67[a] | 6.80 ± 0.79[a] |
| | Epe | 6.50 ± 0.33[a] | 6.70 ± 0.44[a] | 6.90 ± 0.56[a] | 7.20 ± 0.67[a] |
| *Clarias* | Makoko | 14.00 ± 1.34[a] | 14.30 ± 1.45[a] | 14.70 ± 1.57[a] | 15.00 ± 1.90[a] |
| *Gariepinus* | Ikorodu | 14.50 ± 1.12[a] | 14.80 ± 1.34[a] | 15.10 ± 1.68[a] | 15.50 ± 2.01[a] |
| | Epe | 15.00 ± 1.23[a] | 15.20 ± 1.57[a] | 15.50 ± 1.79[a] | 16.00 ± 2.12[a] |
| **Body depth (cm)** | | | | | |
| *Oreochromis* | Makoko | 4.30 ± 0.45[a] | 4.40 ± 0.56[a] | 4.60 ± 0.56[a] | 4.90 ± 0.67[a] |
| *Niloticus* | Ikorodu | 4.50 ± 0.34[a] | 4.60 ± 0.45[a] | 4.80 ± 0.56[a] | 5.10 ± 0.79[a] |
| | Epe | 4.70 ± 0.45[a] | 4.80 ± 0.56[a] | 5.00 ± 0.67[a] | 5.40 ± 0.89[a] |
| *Clarias* | Makoko | 6.80 ± 0.67[a] | 6.90 ± 0.78[a] | 7.10 ± 0.89[a] | 7.30 ± 1.01[a] |
| *Gariepinus* | Ikorodu | 7.00 ± 0.56[a] | 7.10 ± 0.67[a] | 7.40 ± 0.78[a] | 7.60 ± 1.01[a] |
| | Epe | 7.20 ± 0.56[a] | 7.30 ± 0.67[a] | 7.60 ± 0.90[a] | 7.80 ± 1.12[a] |

Values are expressed as Mean ± Standard Deviation (SD).
Superscript letters (a, b, ab) indicate results of Tukey's post-hoc multiple comparison test.
Means within the same row or column that share the same letter are not significantly different ($p < 0.05$), while means with different letters differ significantly

**Source**: Author´s Field work, 2025

### 4.1.2 Results of Analysis of Physicochemical Characteristics of Surface and Subsurface Waters

The results of the physicochemical characteristics of surface and subsurface water collected from the Lagos and Epe lagoons are presented in tables 4.2, 4.3, 4.4 and 4.5.

**Table 4.2:** Mean Values of Physicochemical Parameters of Surface and Subsurface Water (March Cycle)

| Parameters | Water Type | Makoko | Ikorodu | Epe |
|---|---|---|---|---|
| **pH** | Surface | 8.0 | 7.7 | 6.5 |
| | Subsurface | 7.1 | 6.8 | 6.6 |
| **Temperature($^0$C)** | Surface | 29.5 | 28.1 | 26.5 |
| | Subsurface | 25.9 | 24.3 | 24.2 |
| **Dissolved Oxygen(mg/L)** | Surface | 6.7 | 6.4 | 5.5 |
| | Subsurface | 3.7 | 2.9 | 2.3 |
| **Total dissolved solids(mg/L)** | Surface | 650 | 623 | 597 |
| | Subsurface | 500 | 490 | 489 |
| **Electrical conductivity(µS/cm)** | Surface | 1300 | 1250 | 960 |
| | Subsurface | 990 | 960 | 920 |

Values are presented as mean of measured parameters.

**Source:** Author's Field Work, 2025

**Table 4.3:** Mean Values of Physicochemical Parameters of Surface and Subsurface Water (May Cycle)

| Parameters | Water Type | Makoko | Ikorodu | Epe |
|---|---|---|---|---|
| **pH** | Surface | 8.9 | 7.65 | 6.9 |
| | Subsurface | 7.56 | 7.1 | 6.4 |
| **Temperature($^0$C)** | Surface | 29.6 | 28.8 | 27.2 |
| | Subsurface | 25.6 | 25.4 | 24.9 |
| **Dissolved Oxygen(mg/L)** | Surface | 6.9 | 6.3 | 5.5 |
| | Subsurface | 4.3 | 4.0 | 3.6 |
| **Total Dissolved Solids(mg/L)** | Surface | 649 | 644 | 605 |
| | Subsurface | 599 | 557 | 475 |
| **Electrical Conductivity(µS/cm)** | Surface | 1071 | 995 | 906 |
| | Subsurface | 1110 | 1009 | 859 |

Values are presented as mean of measured parameters.

**Source:** Author's Field Work, 2025

**Table 4.4:** Mean Values of Physicochemical Parameters of Surface and Subsurface Water (August Cycle)

| Parameters | Water Type | Makoko | Ikorodu | Epe |
|---|---|---|---|---|
| **pH** | Surface | 8.5 | 8.2 | 7.9 |
| | Subsurface | 7.9 | 7.7 | 6.8 |
| **Temperature($^0$C)** | Surface | 28.4 | 28.1 | 27.2 |
| | Subsurface | 24.8 | 24.6 | 24 |
| **Dissolved Oxygen (mg/L)** | Surface | 7.0 | 6.4 | 6.1 |
| | Subsurface | 4.9 | 4.7 | 4.7 |
| **Total Dissolved Solids(mg/L)** | Surface | 670 | 659 | 650 |
| | Subsurface | 600 | 586 | 500 |
| **Electrical Conductivity(µS/cm)** | Surface | 1200 | 1110 | 1000 |
| | Subsurface | 1189 | 1030 | 906 |

Values are presented as mean of measured parameters.

**Source:** Author's Field Work, 2025

**Table 4.5:** Mean Values of Physicochemical Parameters of Surface and Subsurface Waters (November Cycle)

| Parameters | Water Type | Makoko | Ikorodu | Epe |
|---|---|---|---|---|
| **pH** | Surface | 8.6 | 8.5 | 8.0 |
| | Subsurface | 7.9 | 7.8 | 7.0 |
| **Temperature($^0$C)** | Surface | 26 | 26 | 25 |
| | Subsurface | 23.4 | 23.2 | 23 |
| **Dissolved Oxygen(mg/L)** | Surface | 7.2 | 7.0 | 6.8 |
| | Subsurface | 5.0 | 4.8 | 4.6 |
| **Total Dissolved Solids(mg/L)** | Surface | 679 | 661 | 654 |
| | Subsurface | 611 | 603 | 563 |
| **Electrical Conductivity (µS/cm)** | Surface | 1250 | 1190 | 1110 |
| | Subsurface | 1200 | 1130 | 960 |

Values are presented as mean for measured parameters.

**Source:** Author's Field Work, 2025

### 4.1.3 Results of Heavy Metals Analysis

The concentration of heavy metals in surface and subsurface waters collected from the Lagos and Epe lagoons are presented in Tables 4.6, 4.7, 4.8 and 4.9.

**Table 4.6:** Mean Concentration of Heavy Metals in Surface and Subsurface Water (March Cycle)

| Heavy Metals | Water Type | Makoko | Ikorodu | Epe | WHO Limit for Heavy Metals |
|---|---|---|---|---|---|
| **Lead** | Surface | 2.01 ± 0.10 | 1.64 ± 0.12 | 1.47 ± 0.09 | 0.01 |
| | Subsurface | 5.42 ± 0.12 | 5.31 ± 0.04 | 4.86 ± 0.07 | |
| **Copper** | Surface | 2.71 ± 0.20 | 2.23 ± 0.00 | 1.96 ± 0.12 | 0.05 |
| | Subsurface | 8.10 ± 0.20 | 7.49 ± 0.15 | 6.94 ± 0.10 | |
| **Nickel** | Surface | 0.89 ± 0.04 | 0.65 ± 0.03 | 0.57 ± 0.05 | 0.07 |
| | Subsurface | 12.55 ± 0.26 | 12.13 ± 0.30 | 10.17 ± 0.19 | |
| **Cadmium** | Surface | 0.09 ± 0.01 | 0.07 ± 0.01 | 0.04 ± 0.01 | 0.003 |
| | Subsurface | 0.94 ± 0.04 | 0.91 ± 0.03 | 0.07 ± 0.05 | |
| **Chromium** | Surface | 0.09 ± 0.00 | 0.09 ± 0.05 | 0.08 ± 0.01 | 0.05 |
| | Subsurface | 3.47 ± 0.02 | 3.30 ± 0.01 | 3.22 ± 0.05 | |

Values are expressed as Mean ± Standard Deviation (SD), P>0.05

**Source:** Author's Field Work, 2025

**Table 4.7:** Mean Concentration of Heavy Metals in Surface and Subsurface Water (May Cycle)

| Heavy Metals | Water Type | Makoko | Ikorodu | Epe | WHO Limit for Heavy Metals |
|---|---|---|---|---|---|
| **Lead** | Surface | 2.08 ± 0.12 | 1.53 ± 0.12 | 1.41 ± 0.05 | 0.01 |
| | Subsurface | 5.31 ± 0.12 | 5.11 ± 0.10 | 4.22 ± 0.11 | |
| **Copper** | Surface | 2.67 ± 0.03 | 2.18 ± 0.17 | 1.70 ± 0.15 | 0.05 |
| | Subsurface | 8.12 ± 0.21 | 7.32 ± 0.19 | 5.10 ± 0.10 | |
| **Nickel** | Surface | 0.76 ± 0.02 | 0.64 ± 0.05 | 0.48 ± 0.05 | 0.07 |
| | Subsurface | 12.64 ± 0.32 | 11.10 ± 0.29 | 9.77 ± 0.16 | |
| **Cadmium** | Surface | 0.09 ± 0.00 | 0.06 ± 0.01 | 0.03 ± 0.01 | 0.003 |
| | Subsurface | 0.87 ± 0.03 | 0.88 ± 0.02 | 0.67 ± 0.04 | |
| **Chromium** | Surface | 0.09 ± 0.00 | 0.07 ± 0.04 | 0.06 ± 0.01 | 0.05 |
| | Subsurface | 3.40 ± 0.01 | 3.29 ± 0.00 | 3.10 ± 0.05 | |

Values are expressed as Mean ± Standard Deviation (SD), P>0.05

**Source:** Author's Field Work, 2025

**Table 4.8:** Mean Concentration of Heavy Metals in Surface and Subsurface Water (August Cycle)

| Heavy Metals | Water Type | Makoko | Ikorodu | Epe | WHO Limit for Heavy Metals |
|---|---|---|---|---|---|
| **Lead** | Surface | 3.11 ± 0.11 | 1.68 ± 0.14 | 1.58 ± 0.05 | 0.01 |
| | Subsurface | 5.92 ± 0.05 | 5.66 ± 0.12 | 4.67 ± 0.10 | |
| **Copper** | Surface | 2.87 ± 0.02 | 2.23 ± 0.13 | 1.98 ± 0.16 | 0.05 |
| | Subsurface | 9.10 ± 0.19 | 8.66 ± 0.05 | 7.19 ± 0.04 | |
| **Nickel** | Surface | 0.91 ± 0.21 | 0.86 ± 0.10 | 0.59 ± 0.05 | 0.07 |
| | Subsurface | 13.98 ± 0.34 | 12.77 ± 0.25 | 10.94 ± 0.17 | |
| **Cadmium** | Surface | 0.09 ± 0.00 | 0.08 ± 0.03 | 0.05 ± 0.01 | 0.003 |
| | Subsurface | 0.94 ± 0.01 | 0.89 ± 0.03 | 0.77 ± 0.05 | |
| **Chromium** | Surface | 0.09 ± 0.00 | 0.09 ± 0.06 | 0.07 ± 0.01 | 0.005 |
| | Subsurface | 4.11 ± 0.01 | 3.99 ± 0.00 | 3.79 ± 0.04 | |

Values are expressed as Mean ± Standard Deviation (SD), P>0.05

**Source:** Author's Field Work, 2025

**Table 4.9:** Mean Concentration of Heavy Metals in Surface and Subsurface Water (November Cycle)

| Heavy Metals | Water Type | Makoko | Ikorodu | Epe | WHO Limit for Heavy Metals |
|---|---|---|---|---|---|
| **Lead** | Surface | 3.56 ± 0.20 | 1.87 ± 0.18 | 1.75 ± 0.10 | 0.01 |
| | Subsurface | 6.77 ± 0.10 | 6.54 ± 0.09 | 6.32 ± 0.05 | |
| **Copper** | Surface | 3.11 ± 0.03 | 2.95 ± 0.12 | 2.48 ± 0.15 | 0.05 |
| | Subsurface | 11.56 ± 0.21 | 10.54 ± 0.10 | 9.34 ± 0.07 | |
| **Nickel** | Surface | 1.09 ± 0.30 | 1.02 ± 0.21 | 0.97 ± 0.10 | 0.07 |
| | Subsurface | 14.66 ± 0.44 | 13.77 ± 0.20 | 12.10 ± 0.05 | |
| **Cadmium** | Surface | 0.12 ± 0.03 | 0.10 ± 0.05 | 0.09 ± 0.03 | 0.003 |
| | Subsurface | 1.33 ± 0.00 | 1.09 ± 0.05 | 0.96 ± 0.05 | |
| **Chromium** | Surface | 0.12 ± 0.06 | 0.10 ± 0.01 | 0.09 ± 0.04 | 0.05 |
| | Subsurface | 5.62 ± 0.01 | 5.18 ± 0.01 | 4.91 ± 0.04 | |

Values are expressed as Mean ± Standard Deviation (SD), P>0.05

**Source:** Author's Field Work, 2025

**Table 4.10**: Degree of Contamination **(DC)** and Pollution Load Index **(PLI)** of Surface and Subsurface Waters of Lagos and Epe Lagoons (March Cycle)

| Water Type | Site | Pb | Cu | Ni | Cd | Cr | DC | PLI |
|---|---|---|---|---|---|---|---|---|
| **Surface** | Makoko | 201.00 | 54.22 | 12.73 | 30.00 | 1.98 | 299.93 | 24.17 |
| **Surface** | Ikorodu | 164.30 | 44.56 | 9.30 | 24.33 | 1.70 | 244.19 | 19.50 |
| **Surface** | Epe | 146.60 | 39.26 | 8.10 | 13.00 | 1.52 | 208.48 | 15.59 |
| **Subsurface** | Makoko | 542.00 | 162.00 | 179.29 | 313.33 | 69.40 | 1266.02 | 202.72 |
| **Subsurface** | Ikorodu | 531.00 | 149.80 | 173.29 | 303.33 | 66.00 | 1223.42 | 194.16 |
| **Subsurface** | Epe | 486.00 | 138.80 | 145.29 | 230.00 | 64.40 | 1064.49 | 170.76 |

**Source:** Author's Field Work, 2025

**Table 4.11**: Degree of Contamination (DC) and Pollution Load Index (PLI) of Surface and Benthic Waters of Lagos and Epe Lagoons (May Cycle)

| Water Type | Site | Pb | Cu | Ni | Cd | Cr | DC | PLI |
|---|---|---|---|---|---|---|---|---|
| **Surface** | Makoko | 208.10 | 53.52 | 10.86 | 28.33 | 1.74 | 302.55 | 23.36 |
| **Surface** | Ikorodu | 153.50 | 43.50 | 9.17 | 20.33 | 1.38 | 227.88 | 18.15 |
| **Surface** | Epe | 141.10 | 34.08 | 6.81 | 9.00 | 1.22 | 192.21 | 14.11 |
| **Subsurface** | Makoko | 531.00 | 162.40 | 180.57 | 290.00 | 68.00 | 1231.97 | 197.26 |
| **Subsurface** | Ikorodu | 511.00 | 146.40 | 158.57 | 293.33 | 65.80 | 1174.93 | 186.50 |
| **Subsurface** | Epe | 422.00 | 102.00 | 139.57 | 223.33 | 62.00 | 948.90 | 155.27 |

**Source:** Author's Field Work, 2025

**Table 4.12:** Degree of Contamination (DC) and Pollution Load Index (PLI) of Surface and Benthic Waters of Lagos and Epe Lagoons (August Cycle)

| Water Type | Site | Pb | Cu | Ni | Cd | Cr | DC | PLI |
|---|---|---|---|---|---|---|---|---|
| **Surface** | Makoko | 311.00 | 57.30 | 13.06 | 31.33 | 1.82 | 414.51 | 27.54 |
| **Surface** | Ikorodu | 167.80 | 44.50 | 12.24 | 28.33 | 1.76 | 254.63 | 22.52 |
| **Surface** | Epe | 158.40 | 39.60 | 8.56 | 18.67 | 1.58 | 226.81 | 18.21 |
| **Subsurface** | Makoko | 592.00 | 182.00 | 199.71 | 313.33 | 82.20 | 1369.24 | 218.63 |
| **Subsurface** | Ikorodu | 566.00 | 173.20 | 182.43 | 296.67 | 79.80 | 1298.10 | 208.57 |
| **Subsurface** | Epe | 467.00 | 143.80 | 156.29 | 256.67 | 75.80 | 1099.56 | 179.21 |

**Source:** Author's Field Work, 2025

**Table 4.13:** Degree of Contamination (DC) and Pollution Load Index (PLI) of Surface and Benthic Waters of Lagos and Epe Lagoons (November Cycle)

| Water Type | Site | Pb | Cu | Ni | Cd | Cr | DC | PLI |
|---|---|---|---|---|---|---|---|---|
| **Surface** | Makoko | 355.90 | 62.22 | 15.67 | 40.00 | 2.40 | 476.19 | 33.94 |
| **Surface** | Ikorodu | 187.60 | 59.00 | 14.59 | 33.33 | 2.00 | 296.52 | 27.20 |
| **Surface** | Epe | 175.30 | 49.60 | 13.94 | 32.67 | 1.96 | 273.47 | 25.66 |
| **Subsurface** | Makoko | 677.00 | 231.20 | 209.43 | 443.33 | 112.40 | 1673.36 | 277.09 |
| **Subsurface** | Ikorodu | 654.00 | 210.80 | 196.71 | 363.33 | 103.60 | 1528.44 | 252.22 |
| **Subsurface** | Epe | 632.00 | 186.80 | 172.86 | 320.00 | 98.20 | 1409.86 | 229.83 |

**Source:** Author's Field Work, 2025

### 4.1.4 Results of Microplastic Abundance in Fish Samples from Lagos and Epe Lagoon

The results of microplastics abundance in *Oreochromis niloticus* and *Clarias gariepinus* fish samples from the Lagos and Epe lagoons are presented in tables 4.14, 4.15, 4.16 and 4.17.

**Table 4.14:** Microplastic Abundance in Fish Samples (March Cycle)

| Sample Type | Sample Location | Number of Samples | Average MP Count | Dominant Type | Size Range (µm) | Polymer Composition | Remarks |
|---|---|---|---|---|---|---|---|
| ***Oreochromis niloticus*** | **Makoko** | 5 | 15.2 ± 3.1 | Fibers | 100-500 | Polypropylene, Polyethylene | More fiber content than fragments |
| | **Ikorodu** | 5 | 14.6 ± 2.7 | Fragments | 50-300 | Polyester, Polystyrene | Mostly found in gills |
| | **Epe** | 5 | 12.7 ± 2.9 | Fibers | 50-300 | Polyethylene, Polyamide | Predominant in digestive tract |
| ***Clarias gariepinus*** | **Makoko** | 5 | 16.1 ± 3.0 | Fibers | 150-500 | Polyester, Polystyrene | Predominant in gills |
| | **Ikorodu** | 5 | 14.9 ± 2.5 | Fragments | 50-300 | Polyvinyl Chloride | Predominant in digestive tract |
| | **Epe** | 5 | 14.6 ± 3.3 | Fibers | 50-300 | Polypropylene | Lower fiber content |

Values are expressed as Mean ± Standard Deviation (SD).

**Source:** Author's Field Work, 2024

**Table 4.15:** Microplastic Abundance in Fish Samples (May Cycle)

| Sample Type | Sample Location | Number of Samples | Average MP Count | Dominant Type | Size Range (µm) | Polymer Composition | Remarks |
|---|---|---|---|---|---|---|---|
| ***Oreochromis niloticus*** | **Makoko** | 5 | 16.3 ± 3.2 | Fibers | 100-500 | PET, Polyamide | More fibers than fragments |
| | **Ikorodu** | 5 | 15.4 ± 3.1 | Fibers | 50-300 | PET, PS | Fibers dominant |
| | **Epe** | 5 | 13.9 ± 2.1 | Fragments | 100-400 | PE, PP | Fragments in digestive tract |
| ***Clarias gariepinus*** | **Makoko** | 5 | 16. 9 ± 3.0 | Fragments | 100-500 | PVC, PE | Fragments concentrated in gills |
| | **Ikorodu** | 5 | 14.3 ± 2.4 | Fibers | 50-300 | PE, Polyester | Fibers likely from textile waste |
| | **Epe** | 5 | 12.7 ± 2.6 | Fragments | 30-250 | PS | Fragments concentrated in gills |

Values are expressed as Mean ± Standard Deviation (SD), P>0.05

**Source:** Author's Field Work, 2025

**Table 4.16:** Microplastic Abundance in Fish Samples (August Cycle)

| Sample Type | Sample Location | Number of Samples | Average MP Count | Dominant Type | Size Range (µm) | Polymer Composition | Remarks |
|---|---|---|---|---|---|---|---|
| ***Oreochromis niloticus*** | **Makoko** | 5 | 19.6 ± 4.0 | Fibers | 100-500 | PE, PS | More fibers detected than fragments |
| | **Ikorodu** | 5 | 17.8 ± 3.6 | Fragments | 100-300 | PP, PVC | More fragments than fibers |
| | **Epe** | 5 | 17.1 ± 3.5 | Fibers | 50-300 | PE, Polyamide | Lower fiber detected |
| ***Clarias gariepinus*** | **Makoko** | 5 | 19.2 ± 4.1 | Fragments | 100-500 | Polyester, Polystyrene | Predominant in gills |
| | **Ikorodu** | 5 | 18.2 ± 3.9 | Fibers | 20- 200 | PE, PS | Less fiber detected |
| | **Epe** | 5 | 16.9 ± 3.8 | Fragments | 10-150 | Polyester, Polypropylene | Found mostly in digestive tract |

Values are expressed as Mean ± Standard Deviation (SD), P>0.05

**Source:** Author's Field Work, 2025

**Table 4.17:** Microplastic Abundance in Fish Samples (November Cycle)

| Sample Type | Sample Location | Number of Samples | Average MP Count | Dominant Type | Size Range (µm) | Polymer Composition | Remarks |
|---|---|---|---|---|---|---|---|
| **Oreochromis niloticus** | **Makoko** | 5 | 22.3 ± 5.7 | Fibers | 100-500 | PET, PP | More fiber during this cycle |
| | **Ikorodu** | 5 | 20.6 ± 4.6 | Fragments | 50-300 | PP, PE | More fragments detected |
| | **Epe** | 5 | 20.4 ± 4.3 | Fibers | 50-300 | PP, PS | More fibers detected |
| ***Clarias gariepinus*** | **Makoko** | 5 | 19.4 ± 4.8 | Fragments | 30-250 | PP, PS | Fragments predominant in the gills |
| | **Ikorodu** | 5 | 19.3 ± 4.5 | Fibers | 20-200 | PS, PVC | Fibers predominant in stomach |
| | **Epe** | 5 | 18.5 ± 4.2 | Fibers | 10-150 | PS, PP | More than previous cycles |

Values are expressed as Mean ± Standard Deviation (SD), P>0.05

**Source:** Author's Field Work, 2025

### 4.1.5 Results of Analysis of Phthalate Esters in Fish Samples

The concentrations of Phthalate Esters in *Oreochromis niloticus* and *Clarias gariepinus* fish samples collected from the Lagos and Epe Lagoons are presented in Tables 4.18, 4.19, 4.20 and 4.21.

**Table 4.18:** Phthalate Ester Concentrations (mg/kg) in Fish Samples (March Cycle)

| Sample Type | Sample Location | Dibutyl Phthalate | Diethyl Phthalate | Di(2-ethylhexylphthalate |
|---|---|---|---|---|
| ***Oreochromis niloticus*** | **Makoko** | 45.0 ± 4.8 | 22.0 ± 2.9 | 14.0 ± 2.0 |
| | **Epe** | 35.0 ± 4.2 | 26.0 ± 3.5 | 16.0 ± 2.2 |
| | **Ikorodu** | 50.0 ± 5.1 | 24.0 ± 3.2 | 15.0 ± 2.1 |
| ***Clarias gariepinus*** | **Makoko** | 50.0 ± 5.1 | 24.0 ± 3.2 | 15.0 ± 2.1 |
| | **Epe** | 40.0 ± 4.5 | 28.0 ± 3.7 | 17.5 ± 2.3 |
| | **Ikorodu** | 55.0 ± 5.4 | 26.0 ± 3.4 | 16.5 ± 2.2 |

Values are expressed as Mean ± Standard Deviation (SD), P>0.05

**Source:** Author's Field Work, 2025

**Table 4:19:** Phthalate Ester Concentrations (mg/kg) in Fish Samples (May Cycle)

| Sample Type | Sample Location | Dibutyl Phthalate | Diethyl Phthalate | Di(2-ethylhexylphthalate |
|---|---|---|---|---|
| ***Oreochromis niloticus*** | **Makoko** | 38.0 ± 3.5 | 17.0 ± 2.5 | 11.5 ± 1.7 |
| | **Epe Lagoon** | 28.0 ± 3.5 | 22.0 ±3.0 | 13.0 ± 1.8 |
| | **Ikorodu** | 42.0 ± 4.6 | 19.0 ± 2.8 | 12.0 ± 1.9 |
| ***Clarias gariepinus*** | **Makoko** | 42.0 ± 4.5 | 20.0 ± 2.8 | 13.0 ±1.9 |
| | **Epe Lagoon** | 32.0 ± 3.8 | 24.0 ± 3.2 | 15.0 ± 2.0 |
| | **Ikorodu** | 46.0 ± 4.8 | 22.0 ± 3.0 | 14.0 ± 2.0 |

Values are expressed as Mean ± Standard Deviation (SD), P>0.05

**Source:** Author's Field Work, 2025

**Table 4.20:** Phthalate Ester Concentrations (mg/kg) in Fish Samples (August Cycle)

| Sample Type | Sample Location | Dibutyl Phthalate | Diethyl Phthalate | Di(2-ethylhexylphthalate |
|---|---|---|---|---|
| ***Oreochromis niloticus*** | **Makoko** | 30.0 ± 3.8 | 14.0 ± 2.3 | 9.5 ± 1.5 |
| | **Epe Lagoon** | 20.0 ± 3.0 | 18.0 ± 2.7 | 11.0 ± 1.6 |
| | **Ikorodu** | 34.0 ± 3.9 | 16.0 ± 2.5 | 10.5 ± 1.6 |
| ***Clarias gariepinus*** | **Makoko** | 34.0 ± 4.0 | 17.0 ± 2.6 | 11.5 ± 1.7 |
| | **Epe Lagoon** | 25.0 ± 3.5 | 20.0 ± 2.9 | 13.0 ± 1.8 |
| | **Ikorodu** | 38.0 ± 4.3 | 18.0 ± 2.7 | 12.5 ± 1.8 |

Values are expressed as Mean ± Standard Deviation (SD), P>0.05

**Source:** Author's Field Work, 2024

**Table 4.21:** Phthalate Ester Concentrations (mg/kg) in Fish Samples (November Cycle)

| Sample Type | Sample Location | Dibutyl Phthalate | Diethyl Phthalate | Di(2-ethylhexylphthalate |
|---|---|---|---|---|
| ***Oreochromis niloticus*** | **Makoko** | 42.0 ± 4.5 | 18.0 ± 2.6 | 12.5 ± 1.8 |
| | **Epe Lagoon** | 32.0 ± 3.8 | 23.0 ± 3.1 | 14.0 ± 1.9 |
| | **Ikorodu** | 46.0 ± 4.8 | 21.0 ± 2.9 | 13.0 ± 1.9 |
| ***Clarias gariepinus*** | **Makoko** | 46.0 ± 4.9 | 22.0 ± 3.0 | 14.0 ± 2.0 |
| | **Epe Lagoon** | 35.0 ± 4.1 | 26.0 ± 3.5 | 16.5 ± 2.2 |
| | **Ikorodu** | 50.0 ± 5.2 | 24.0 ± 3.2 | 15.0 ± 2.1 |

Values are expressed as Mean ± Standard Deviation (SD), P>0.05

**Source:** Author's Field Work, 2025

## 4.2 Discussion of Findings

### 4.2.1 Discussion on Morphometric Characteristics of *Oreochromis niloticus* and *Clarias gariepinus*

The morphometric assessment of *Oreochromis niloticus* and *Clarias gariepinus* across four sampling months (March, May, August, and November) and three distinct locations (Makoko, Ikorodu, and Epe) revealed notable spatial and temporal (periodic/seasonal) growth patterns.

#### 4.2.1.1 Standard Length of *Oreochromis niloticus* and *Clarias gariepinus*

For *O. niloticus*, Epe consistently recorded the highest mean standard lengths across all months, ranging from 13.00±1.12 cm in March to 13.80±1.56 cm in November, suggesting optimal environmental conditions or feeding efficiency in this area (Figure 4.1). Ikorodu followed closely, while Makoko reported the lowest standard lengths value for *O. niloticus*. Similarly, *C. gariepinus* from Epe showed the greatest standard lengths value throughout the study, reaching 28.00±2.57 cm by November. The observed spatial trend (Epe > Ikorodu > Makoko) suggests varying habitat quality. Comparable range of standard lengths can be found in a study on *Clarias gariepinus* from Ero Reservoir, Ekiti State, which reported standard lengths between 29.1cm and 33.1cm[1]. Although the sampling environments differ—reservoir versus lagoon—the similarity in length magnitudes reinforces the validity of our measurements and suggests comparable growth potential across different freshwater systems in Nigeria.

#### 4.2.1.2 Total Length of *Oreochromis niloticus* and *Clarias gariepinus*

Total length patterns mirrored those of standard length for both species. *O. niloticus* from Epe had the highest values (17.50 ± 1.68 cm in November), while Makoko presented the lowest. *C. gariepinus* showed a consistent increase across months and sites, with Epe fish again having the highest total length, peaking at 35.00 ± 3.35 cm in November. The increasing total length

observed for both fishes across months, in all three locations (Figure 4.2), although not statistically significant, reflects continuous growth, possibly influenced by seasonal changes in water parameters and primary productivity. A study also reported seasonal variations in fish abundance in Lagos lagoon, which was attributed to the physico-chemical variations in the water parameters[1].

#### 4.2.1.3 Head Length of *Oreochromis niloticus* and *Clarias gariepinus*

A similar seasonal and spatial pattern was observed in head length of both species. *O. niloticus* from Epe reached 4.50±0.89 cm, while *C. gariepinus* from the same site recorded up to 8.10±1.01 cm in November (Figure 4.3). The least head length observed in March; 3.50±0.45 cm for *O. niloticus* and 6.80±0.56 cm for *C. gariepinus*. This consistent increase in head size over time suggests active feeding and favourable growth conditions, particularly in Epe and Ikorodu. These findings are consistent with observed morphometric trends in related studies. For example, a study in Asaba, Nigeria reported that female *O. niloticus* exhibited a mean standard length of 12.18 ± 0.41 cm and total length of 14.68 ± 0.87 cm, indicating relatively advanced growth stages during sampling. Although head length was not directly reported, the proportional dimensions support the growth patterns observed in your study, particularly the progressive increase over seasonal cycles[2].

#### 4.2.1.4 Pre-dorsal and Pre-anal Lengths of *Oreochromis niloticus* and *Clarias gariepinus*

Both pre-dorsal and pre-anal lengths showed marginal monthly increases for both species, the shortest lengths observed in March and the longest observed in November. As observed for their standard length, total length and head length, fish from Epe also exhibited larger values compared to other sites, indicating more advanced development. *C. gariepinus* from Epe attained

the highest pre-dorsal (13.90 ± 1.56 cm) and pre-anal (16.00 ± 2.12 cm) measurements. These regions (pre-dorsal and pre-anal) are associated with swimming ability and reproductive maturity; thus, their steady growth may signal readiness for spawning later in the year[2]. The least values for pre-dorsal and pre-anal length were found in March at Makoko for both fish species. Figure 4.4 and figure 4.5 show the different post-dorsal and pre-anal lengths of *O. niloticus* and *C. gariepinus* in the Lagos lagoons.

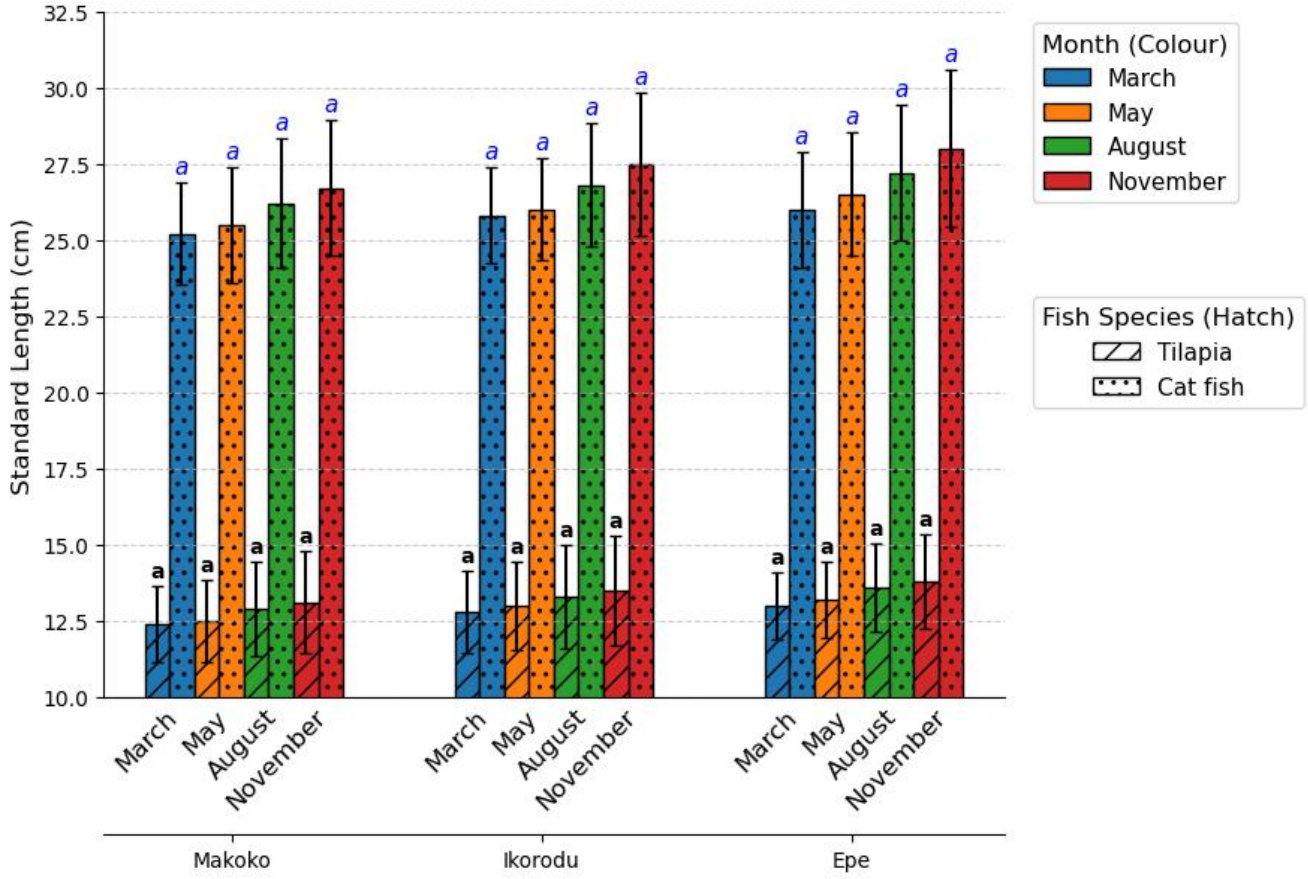


**Figure 4.1:** Comparison of Standard Length of *Oreochromis niloticus* and *Clarias gariepinus* in Lagos (Makoko and Ikorodu axis) and Epe Lagoons

**Source:** Author's Field Work, 2025

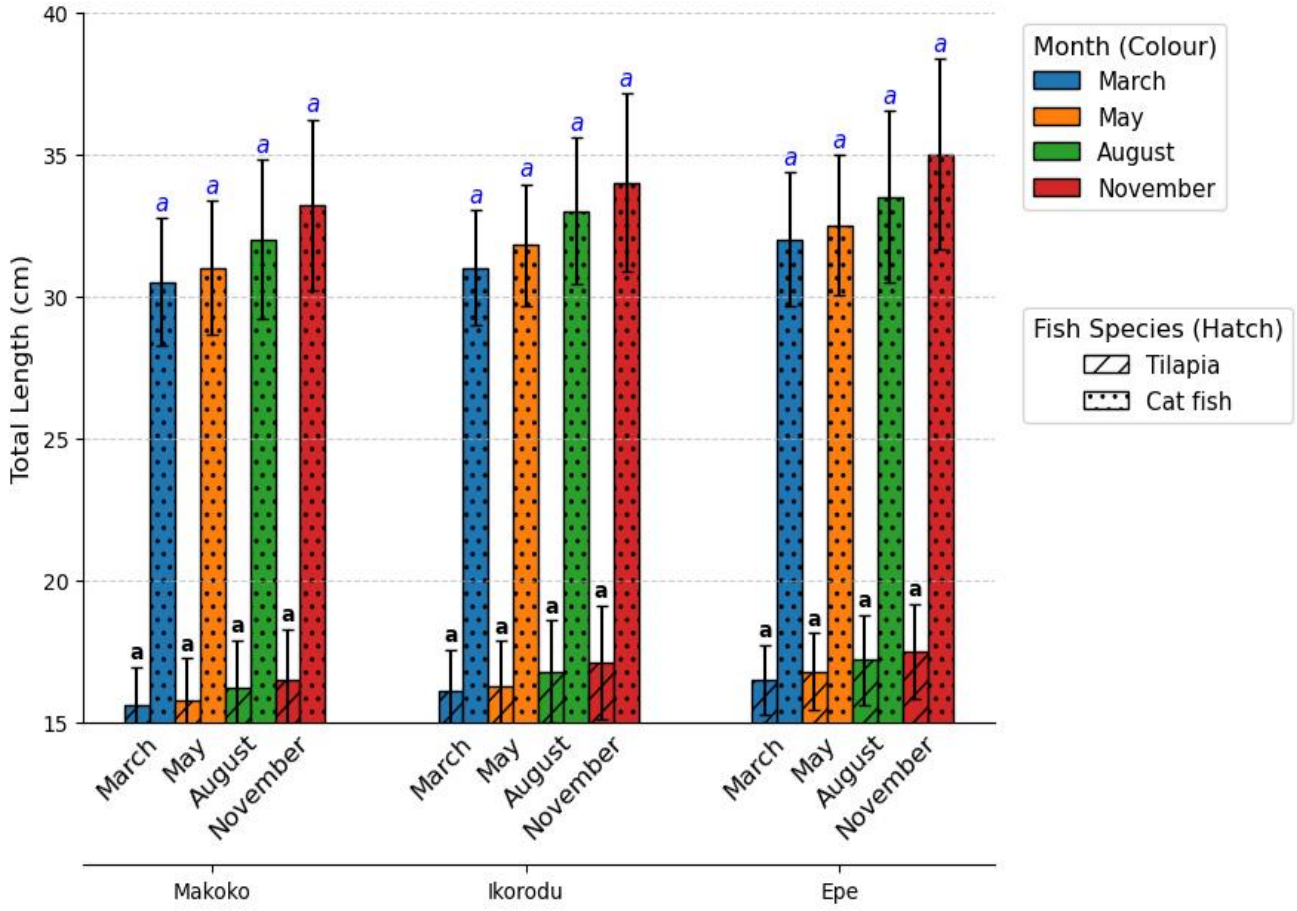


**Figure 4.2:** Comparison of Total Length of *Oreochromis niloticus* and *Clarias gariepinus* in Lagos (Makoko and Ikorodu Axis) and Epe Lagoons

**Source:** Author's Field Work, 2025

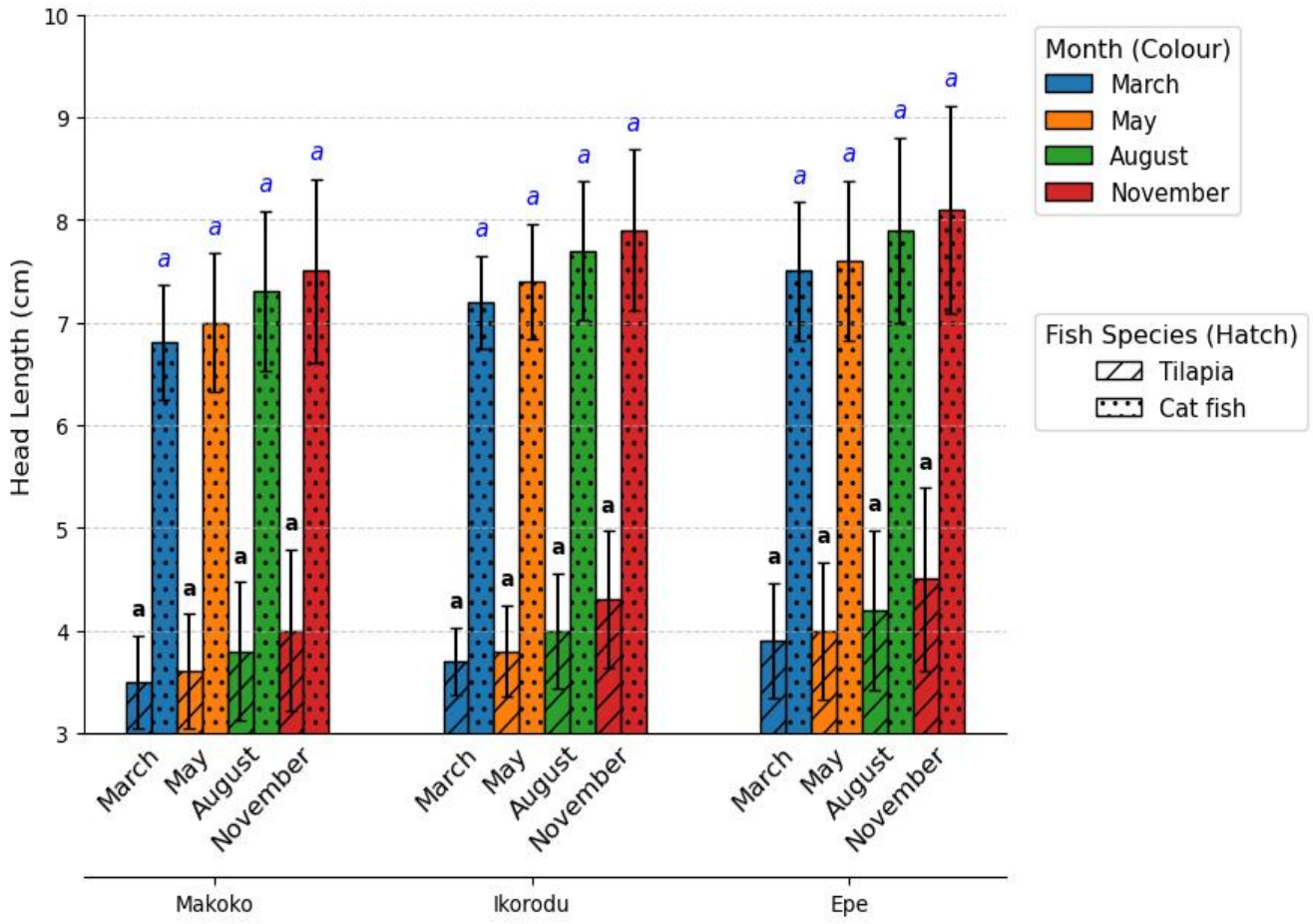


**Figure 4.3:** Comparison of Head Length of *Oreochromis niloticus* and *Clarias gariepinus* in Lagos (Makoko and Ikorodu Axis) and Epe Lagoons

**Source:** Author's Field Work, 2025

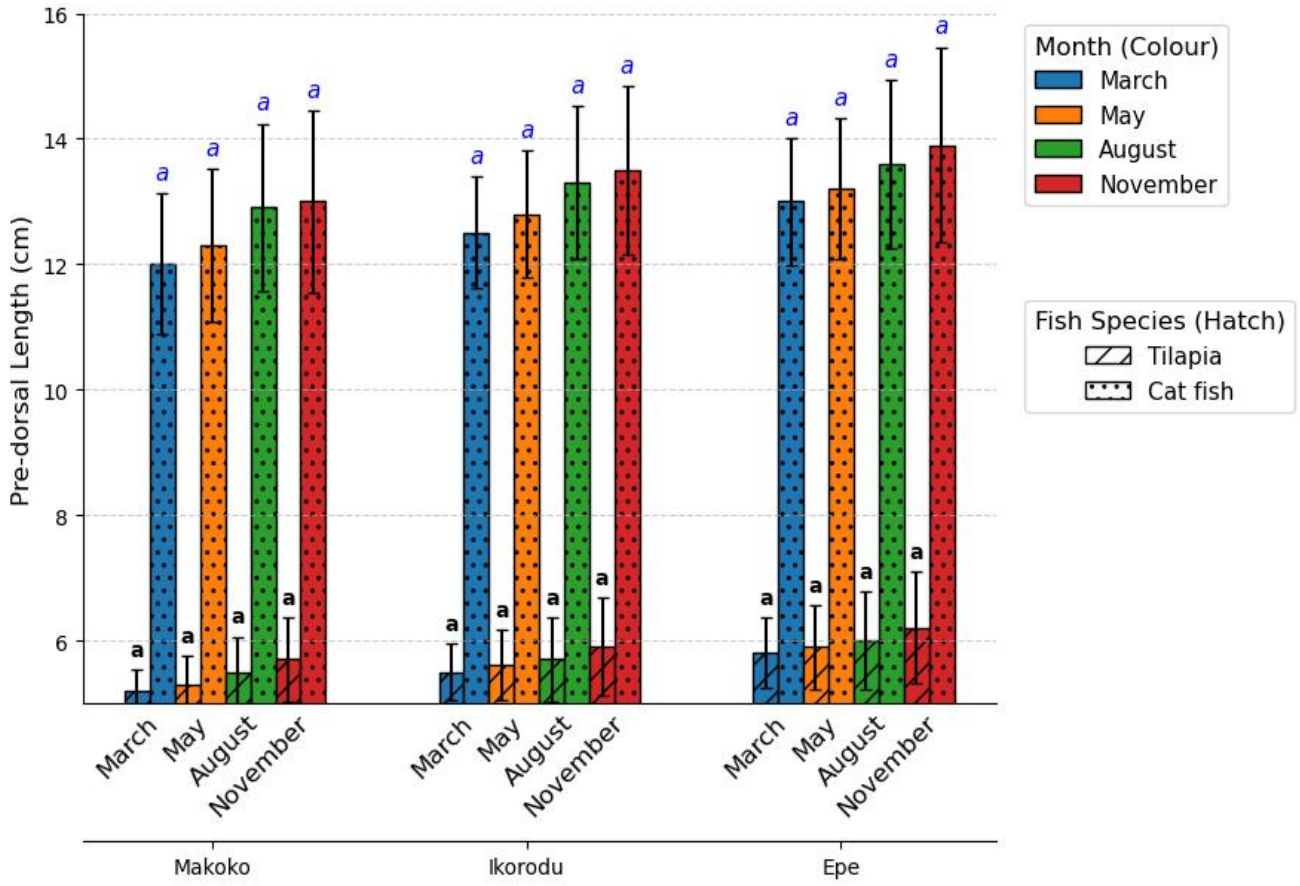


**Figure 4.4:** Comparison of Pre-dorsal Length of *Oreochromis niloticus* and *Clarias gariepinus* in Lagos (Makoko and Ikorodu Axis) and Epe Lagoons

**Source:** Author's Field Work, 2025

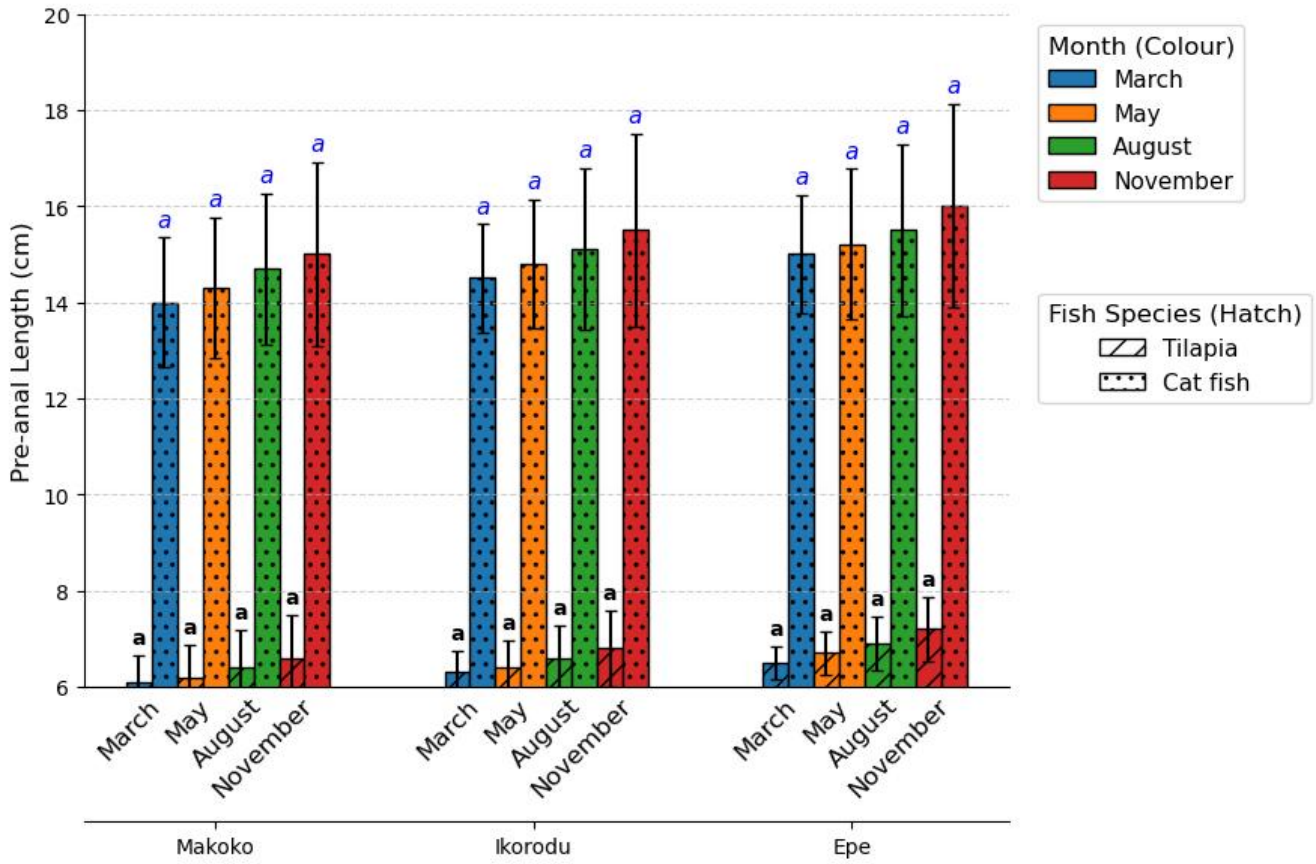


**Figure 4.5:** Comparison of Pre-anal Length of *Oreochromis niloticus* and *Clarias gariepinus* in Lagos (Makoko and Ikorodu Axis) and Epe Lagoons

**Source:** Author's Field Work, 2025

#### 4.2.1.5 Body Depth of *Oreochromis niloticus* and *Clarias gariepinus*

Body depth, an important index for body robustness and energy reserves, followed a consistent upward trend across months, especially in Epe[3]. For *O. niloticus*, Epe recorded a body depth of 5.40 ± 0.89 cm, while *C. gariepinus* reached 7.80 ± 1.12 cm in the same location and month (Figure 4.6). The relatively deeper bodies in fish from Epe suggest healthier body conditions and better nutrient assimilation. Also, the increased body depth of both species towards the end of the year supports follows same pattern as other growth parameters, which implies readiness for spawning in the later season[2]. A comparable morphometric pattern was documented in *O. niloticus* across diverse ecological zones of Nigeria, where specimens from Asejire Lake exhibited significantly greater values across multiple morphometric traits—including body depth—compared with populations from other lakes. This similarity reinforces the hypothesis that favorable environmental conditions can enhance body conformation in tilapia populations[4].

The alignment between our findings and existing literature not only lends greater validity to the robustness of fish from Epe but also points to broader ecological drivers—such as habitat quality and resource availability—that underpin morphometric variation in Nigerian freshwater fish.

#### 4.2.1.6 Weight Distribution of *Oreochromis niloticus* and *Clarias gariepinus*

Across the three sampling locations (Makoko, Ikorodu, and Epe), the mean weight of *O. niloticus* and *C. gariepinus* also showed a progressive increase from March through November, suggesting seasonal influences on growth dynamics, possibly linked to food availability, water quality, and reproductive cycles (Table 4.18).

At Makoko, *O. niloticus* recorded the lowest mean weights across all months, ranging from 120.00 ± 16.77 g in March to 155.00 ± 33.54 g in November. No statistically significant

differences ($p > 0.05$) were observed across the months at this location, indicating relatively stable growth patterns over time. Conversely, in Ikorodu and Epe, *O. niloticus* showed significantly higher weights, particularly in March and May, with values increasing from 130.00 ± 11.18 g to 165.00 ± 27.95 g in Ikorodu and from 140.00 ± 13.41 g to 175.00 ± 31.31 g in Epe. *C. gariepinus* in Makoko also exhibited the lowest, compared to the other two locations. This suggests more favourable environmental or ecological conditions for both species in Ikorodu and Epe.

For *C. gariepinus*, a markedly higher weight range was recorded across all locations compared to *O. niloticus*, reflecting its faster growth potential and larger adult size. At Makoko, *C. gariepinus* showed a steady increase from 280.00 ± 22.36 g in March to 350.00 ± 67.08 g in November. However, its weight values were significantly lower than those recorded in Ikorodu and Epe, especially in March ($p < 0.05$), where the values were 300.00 ± 27.95 g and 320.00 ± 33.54 g, respectively.

Notably, across all months, Epe consistently recorded the highest mean weights for *C. gariepinus*, peaking at 390.00 ± 78.26 g in November. This suggests that the aquatic conditions at Epe may be most conducive for growth, likely due to optimal water quality, prey availability, or habitat structure. The weight of both fish species increased over time across all locations, with Epe generally supporting higher biomass. These seasonal and spatial weight trends align with findings from River Donga, Nigeria, where both *O. niloticus* and *C. gariepinus* demonstrated negative allometric growth ($b < 3$), and mean condition factor (K) values ranged between 0.481–0.803 for *C. gariepinus* and 1.298–2.460 for *O. niloticus*[5]. Seasonal variations in these growth parameters were attributed to ecological stressors and resource availability, similar to the patterns observed in our lagoon systems. The statistically significant variations observed suggest

spatial and periodical differences in ecological productivity and/or anthropogenic influence across the sampling sites. Together, these results underscore spatial and temporal heterogeneity in fish growth performance, with Epe and Ikorodu presenting more conducive environments for fish biomass accrual. The observed differences in mean weights are indicative of variations in ecological productivity and anthropogenic impact across the sampling sites. The seasonal progression of weight—visually summarized in Figure 4.7—reinforces these insights.

**Table 4.22:** Mean Body Weight of *Oreochromis niloticus* and *Clarias gariepinus* Across Cycles from the Three Sample Locations

| Species | Location | March (g) | May (g) | August (g) | November (g) |
|---|---|---|---|---|---|
| *O. Niloticus* | Makoko | $120.00 \pm 16.77^{a}$ | $125.00 \pm 22.36^{a}$ | $140.00 \pm 27.95^{a}$ | $155.00 \pm 33.54^{a}$ |
| | Ikorodu | $130.00 \pm 11.18^{b}$ | $135.00 \pm 16.77^{b}$ | $150.00 \pm 22.36^{ab}$ | $165.00 \pm 27.95^{a}$ |
| | Epe | $140.00 \pm 13.41^{b}$ | $145.00 \pm 20.12^{ab}$ | $160.00 \pm 24.60^{ab}$ | $175.00 \pm 31.31^{a}$ |
| *C. Gariepinus* | Makoko | $280.00 \pm 22.36^{b}$ | $290.00 \pm 33.54^{ab}$ | $320.00 \pm 50.31^{ab}$ | $350.00 \pm 67.08^{a}$ |
| | Ikorodu | $300.00 \pm 27.95^{a}$ | $310.00 \pm 39.13^{a}$ | $340.00 \pm 55.90^{a}$ | $370.00 \pm 72.67^{a}$ |
| | Epe | $320.00 \pm 33.54^{a}$ | $330.00 \pm 44.72^{a}$ | $360.00 \pm 61.49^{a}$ | $390.00 \pm 78.26^{a}$ |

Values are expressed as Mean ± Standard Deviation (SD).
Superscript letters (a, b, ab) indicate results of Tukey's post-hoc multiple comparison test.
Means within the same row or column that share the same letter are not significantly different ($p < 0.05$), while means with different letters differ significantly.

**Source:** Author's Field Work, 2025

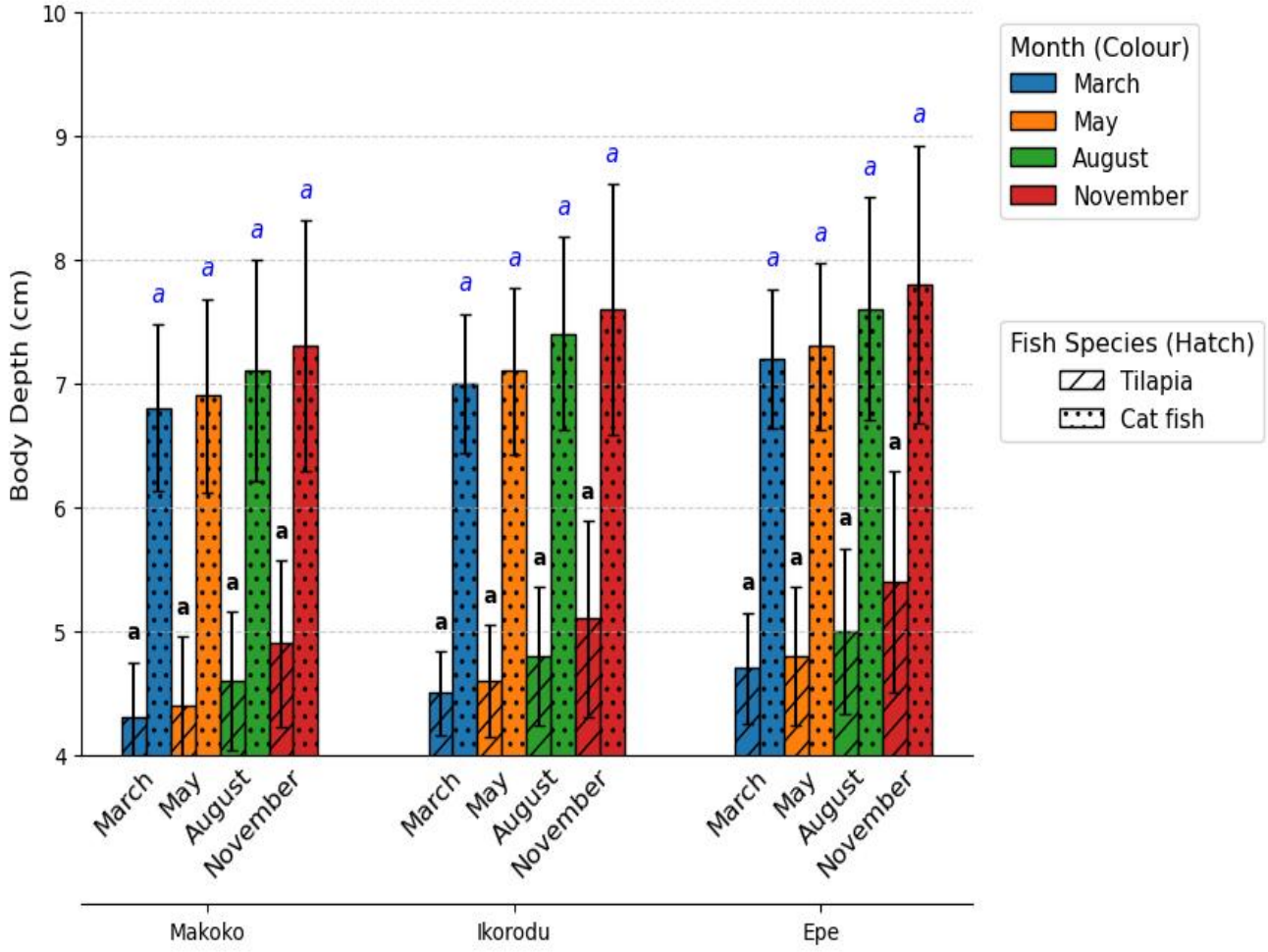


**Figure 4.6:** Comparison of Body Depth of *Oreochromis niloticus* and *Clarias gariepinus* in Lagos (Makoko and Ikorodu Axis) and Epe Lagoons

**Source:** Author's Field Work, 2025

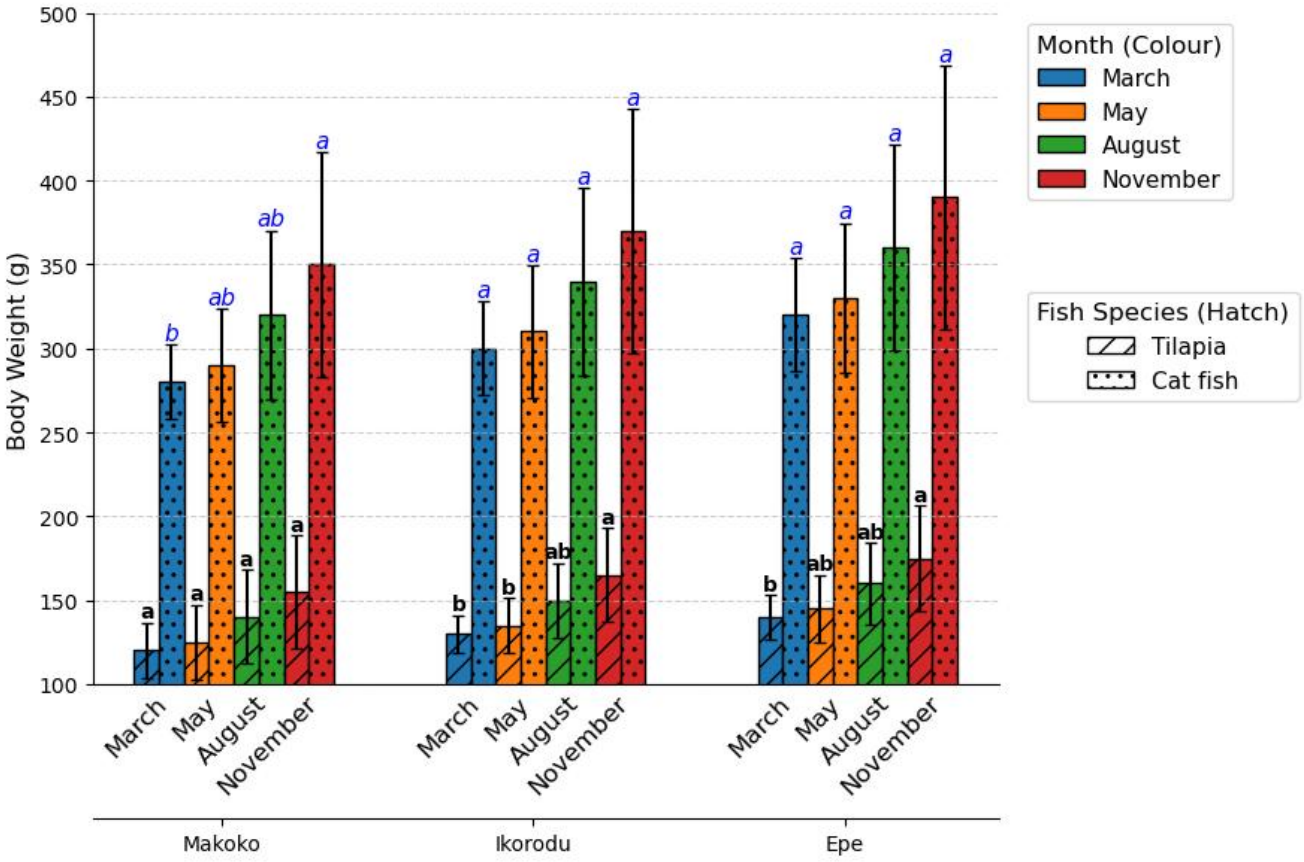


**Figure 4.7:** Comparison of Body Weight of *Oreochromis niloticus* and *Clarias gariepinus* in Lagos (Makoko and Ikorodu Axis) and Epe Lagoons

**Source:** Author's Field Work, 2025

#### 4.2.1.7 Condition Factor (K) of *Oreochromis niloticus* and *Clarias gariepinus*

The condition factor (K), a widely accepted index to evaluate the "well-being" or fatness of fish, varied across species, locations, and months, with notable interspecific differences (Table 4.23). Across all months and locations, *O. niloticus* consistently recorded significantly higher K values than *C. gariepinus* ($p < 0.01$), indicating a generally better body condition. For instance, at Makoko, *O. niloticus* had a K value of $3.23 \pm 0.72$ in March, rising slightly to $3.55 \pm 1.01$ in November. In contrast, *C. gariepinus* maintained relatively stable K values across months, ranging narrowly from $1.01 \pm 0.24$ to $0.99 \pm 0.31$, reflecting a leaner body morphology typical of this more streamlined species (Figure 4.8).

Similar patterns were observed in Ikorodu and Epe, where *O. niloticus* showed slightly increasing K values toward November, while *C. gariepinus* values remained below 1.05 throughout. The persistent and statistically significant difference between the two species (as shown by the t-test values < 0.01 across all months and locations) underscores inherent physiological differences, where *O. niloticus* tends to accumulate more body reserves compared to the generally more muscular *C. gariepinus* (Figure 4.8).

Notably, *O. niloticus* condition factor values remained above the threshold of 3.0 across all locations and months, suggesting healthy fish populations and favourable environmental conditions. This follows the recommended K value range (2.9-4.8) for matured fresh water fish[4,5]. In contrast, *C. gariepinus* values, which lingered around 1.0, fall outside acceptable ranges. A study reported even lower K values for *C. gariepinus* (0.52 – 0.83) and *O. niloticus* (1.53 – 1.81) in Wudil River, Kano, Nigeria[5]. Another study reported slightly higher K values ranging from 1.24 -1.80 for *C. gariepinus* and 1.5 - 3.7 for *O. niloticus* (which falls in range with that of the present study) in Orashi River, Nigeria[6].

**Table 4.23:** Condition Factor (K) of *Oreochromis niloticus* and *Clarias gariepinus* Fishes Across Four Cycles from Lagos (Makoko and Ikorodu Axis) and Epe Lagoons

| Location | Species | March | May | August | November |
|---|---|---|---|---|---|
| Makoko | *O. niloticus* | 3.23 ± 0.72[a] | 3.24 ± 0.78[a] | 3.39 ± 0.92[a] | 3.55 ± 1.01[a] |
| | *C. gariepinus* | 1.01 ± 0.24[a] | 1.00 ± 0.25[a] | 1.01 ± 0.30[a] | 0.99 ± 0.31[a] |
| | Sig. (t-test) | <0.01 | <0.01 | <0.01 | <0.01 |
| Ikorodu | *O. niloticus* | 3.21 ± 0.74[a] | 3.21 ± 0.77[a] | 3.27 ± 0.86[a] | 3.44 ± 0.98[a] |
| | *C. gariepinus* | 1.02 ± 0.18[a] | 0.97 ± 0.17[a] | 0.96 ± 0.19[a] | 0.96 ± 0.22[a] |
| | Sig. (t-test) | <0.01 | <0.01 | <0.01 | <0.01 |
| Epe | *O. niloticus* | 3.18 ± 0.61[a] | 3.12 ± 0.67[a] | 3.22 ± 0.78[a] | 3.35 ± 0.88[a] |
| | *C. gariepinus* | 1.00 ± 0.26[a] | 0.99 ± 0.28[a] | 1.00 ± 0.34[a] | 0.96 ± 0.36[a] |
| | Sig. (t-test) | <0.01 | <0.01 | <0.01 | <0.01 |

Values are expressed as Mean ± Standard Deviation (SD).
Superscript letters (a, b, ab) indicate results of Tukey's post-hoc multiple comparison test.
Means within the same row or column that share the same letter are not significantly different ($p < 0.05$), while means with different letters differ significantly.

**Source:** Author's Field Work, 2025

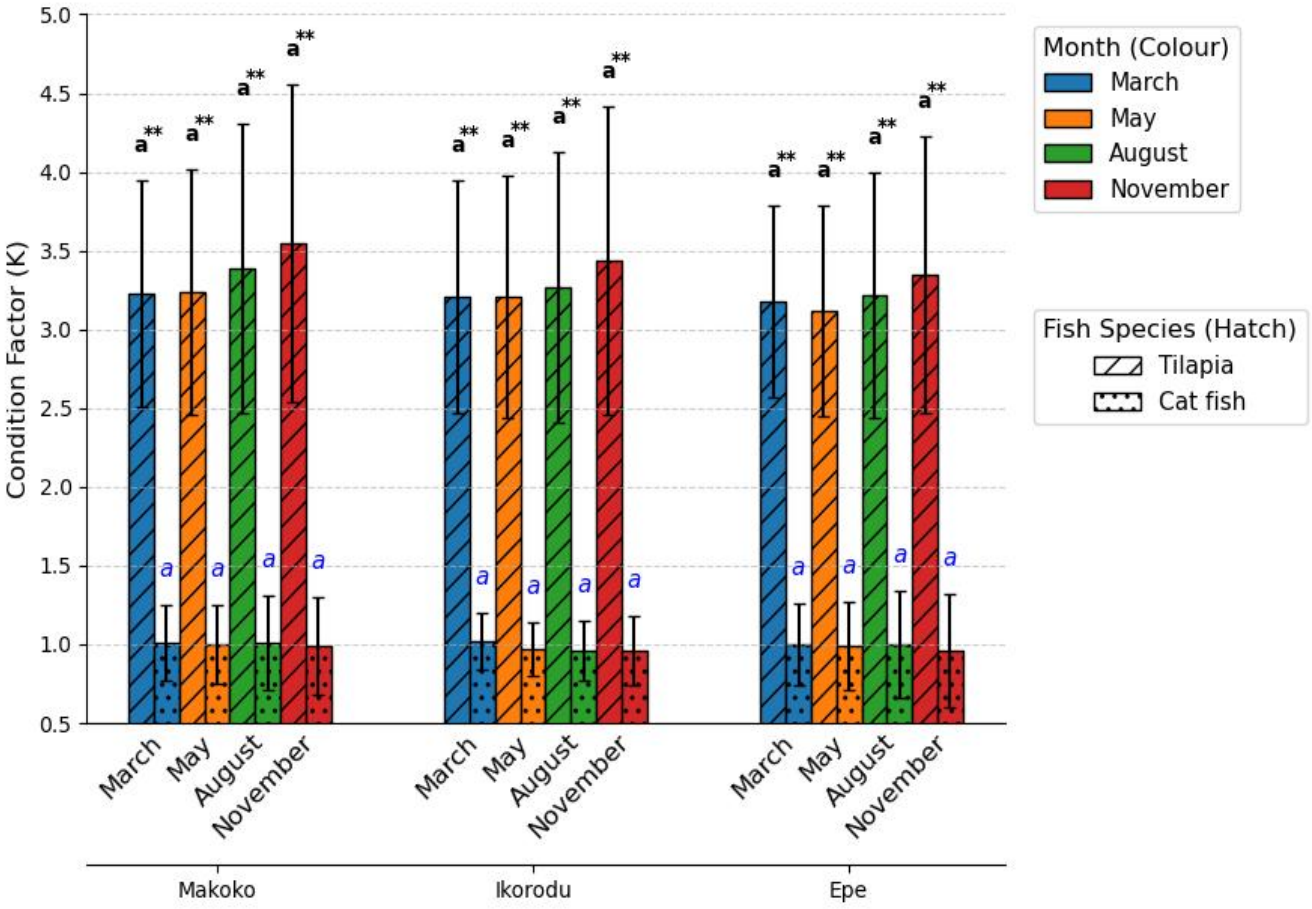


**Figure 4.8:** Condition factor (K) of *Oreochromis niloticus* and *Clarias gariepinus* in Lagos (Makoko and Epe Axis) and Epe Lagoons

**Source:** Author's Field Work, 2025

#### 4.2.1.8 Comparative and Ecological Implications of Morphometric Characteristics of *Clarias gariepinus and Oreochromis niloticus* in Lagos Lagoons

Across all morphometric parameters, Epe consistently supported the highest growth indices for both fish species, followed by Ikorodu, with Makoko trailing. This gradient may signify differences in trophic status, pollution load, or hydrological conditions across the Lagos lagoons. For instance, the growth-limiting potential of Makoko lagoon—reflected by the relatively low growth indices of *O. niloticus* and *C. gariepinus*, irrespective of spatial and seasonal variations—may be attributed to higher anthropogenic pressure and poorer water quality. A study highlighted the increasingly fragile ecosystem in Makoko lagoon, which resulted from wetland expansion, flooding, and erosion[7]. Also, the urban growth experienced in this region has led to slum conditions, characterized by poor infrastructure and environmental sanitation, which further degrades the water systems[8]. Another study further emphasized the negative anthropogenic impacts on the lagoon's water chemistry, which has considerably reduced the diversity and abundance of aquatic organisms, particularly the benthic macroinvertebrates[9].

Epe Lagoon has also been reported with elevated levels of pollutants such as radionuclides and heavy metals[10]. Nevertheless, this lagoon provides a conducive environment for continuous fish development as revealed by this study and also supported by a study, which reported good health conditions of *Parachanna obscura* in Epe lagoon[11]. Conversely, findings revealed contrasting results for *Leuciscus niloticus* that showed poor adaptation with low condition factors in the lagoon[12]. This collectively implies relatively better growth-influencing conditions in Epe Lagoon, compared to Makoko and Ikorodu Lagoons. However, these conditions appear to be species-specific; they may have varying effects on different species as revealed in previous studies, even though this study revealed a similar growth trend for *O. niloticus* and *C. gariepinus*.

It is noteworthy that while differences detected in the fish lengths are marginal ($p > 0.05$), the biological significance of the trend that shows a spatial and seasonal growth progression remains evident. This warrants attention concerning fisheries productivity and resource management. It is also notable that while statistically significant differences ($p < 0.05$) were detected in weight across locations for certain months, the condition factor (K) of *O. niloticus* remained consistently and significantly higher than that of *C. gariepinus* ($p < 0.01$) at all locations and time points. This suggests *O. niloticus* has better potential for converting available resources into body condition or energy reserves. Despite this, *C. gariepinus* exhibited higher absolute weights.

This seemingly contradictory result could be explained by comparing their nutritional compositions. Recent findings revealed that *C. gariepinus* has lower moisture and carbohydrate contents, but higher crude fat and protein content, compared to *O. niloticus*[13]. The higher protein and fat contents of *C. gariepinus* contribute to a denser body mass (hence greater weight), while its lower moisture and carbohydrate content accounts for its reduced plumpness or overall volume relative to its length, which leads to a lower condition factor, compared to *O. niloticus*. In other words, while *C. gariepinus* gains more mass, it's not necessarily more robust than *O. niloticus* when compared to its body size.

### 4.2.2 Discussion of Physicochemical Properties of Surface and Benthic Waters

The analysis of surface and benthic water samples collected in March from Makoko, Ikorodu, and Epe showed notable variations across the different locations and water layers (Table 4.2).

The pH values in surface waters ranged between 6.5 at Epe, 7.7 in Ikorodu and 8.0 at Makoko, indicating neutral to slightly alkaline conditions. In contrast, pH levels in benthic waters were slightly lower, varying between 6.6 in Epe, 6.8 in Ikorodu and 7.1 in Makoko. This slight decline in pH at the bottom layers may be linked to organic matter degradation, which can produce

acidic by-products, and the limited influence of photosynthesis near the sediments. Similar observations have been reported by a study, which noted that decomposition processes of organic matter tend to lower pH in bottom waters[14].

Temperature readings revealed that surface waters were generally warmer, with values between 26.5°C in Makoko, 28.1$^0$C in Ikorodu and 29.5°C in Epe, while benthic waters recorded lower temperatures of 24.2°C in Epe, 24.3$^0$C in Ikorodu and 25.9°C in Makoko. This difference can be attributed to solar radiation heating the upper layers, while deeper layers remain cooler due to reduced light penetration and thermal stratification. As pointed out by a study, such thermal gradients can influence the vertical transport and distribution of suspended particles, including microplastics[15].

Dissolved Oxygen (DO) concentrations were higher in surface waters, ranging from 5.5mg/L in Epe, 6.4mg/L in Ikorodu and 6.7 mg/L. Lower levels were found in benthic waters - 2.3mg/L in Epe, 2.9mg/L in Ikorodu and 3.7 mg/L in Makoko. The lowest level of dissolved oxygen in both surface and benthic waters was recorded in Epe. These reduced DO contents at the benthic zone likely result from oxygen consumption by microbial decomposition of organic matter and reduced mixture with oxygen-rich surface water. A recent study noted that oxygen depletion is common in benthic waters of polluted or eutrophic systems[16].

Total Dissolved Solids were higher at the surface in Makoko and Ikorodu (Table 4.2), with Epe having the lowest at 597mg/L compared to the benthic zone with 489mg/L. Makoko and Ikorodu have higher TDS values at 650mg/L and 623mg/L, 500mg/L and 490mg/L for surface and benthic waters respectively. The elevated surface TDS could be linked to inputs from domestic

and industrial effluents, tidal mixing, and evaporation. Elevated TDS levels can pose stress on aquatic organisms by affecting osmoregulation, as observed in similar studies[17].

Electrical Conductivity followed a similar pattern, with surface values of 960 in Epe, 1250 µS/cm in Ikorodu and 1300 µS/cm in Makoko, and benthic values slightly lower (920–990 µS/cm). Makoko recorded the highest conductivity levels, which could reflect increased anthropogenic inputs. Conductivity is directly related to ionic concentration and serves as an indicator of water quality and salinity, both of which can influence the distribution and behavior of microplastic particles in the aquatic environment[18].

The results of the physiochemical parameters for this cycle shows that surface waters are more influenced by atmospheric conditions and pollution from surface runoff, while benthic waters reflect ongoing organic matter decomposition and reduced oxygen availability. These conditions not only impact aquatic life but also influence the fate and persistence of contaminants like microplastics and heavy metals.

The May cycle results across the three study sites (Makoko, Ikorodu, and Epe) reveal seasonal shifts in physicochemical parameters that reflect both natural processes and anthropogenic inputs as shown in Table 4.3.

The pH values of surface water ranged from 6.9 in Epe, 7.65 in Ikorodu to 8.9 in Makoko, showing a shift towards alkaline conditions at Makoko. Benthic pH values were slightly lower, ranging from 6.4 (Epe), 7.1(Ikorodu) to 7.56 (Makoko). This decrease in pH at the benthic zone can be attributed to organic matter degradation and reduced photosynthetic activity, like trends

noted in the March cycle. These findings support observations by a study, which indicated that decomposition near the sediment tends to lower pH values in benthic environments[14].

Temperature in surface water were consistently higher, with Makoko recording the highest at 29.6°C, and Epe the lowest at 27.2°C. Benthic waters showed cooler temperatures (24.9°C to 25.6°C), reflecting reduced solar influence. This stratification is common in tropical aquatic systems and has implications for contaminant movement. Warmer surface layers tend to hold lighter particles such as microplastics in suspension, while cooler bottom layers may encourage settling of denser particles[15].

Dissolved Oxygen levels in surface waters remained relatively stable (5.5–6.9 mg/L), while benthic waters exhibited increased oxygenation compared to March, with values ranging from 3.6 to 4.3 mg/L. The slightly improved DO concentrations at the bottom may result from increased mixing during the rainy season or changes in biological activity. Nevertheless, these values still reflect oxygen stress in the benthic environment, which can inhibit aerobic decomposition and encourage the persistence of microplastics[16].

Total Dissolved Solids in surface waters had values between 605mg/L and 649 mg/L, while in benthic waters, it ranged from 475mg/L to 599 mg/L. The higher TDS in surface waters, especially in Makoko and Ikorodu, may stem from increased runoff and effluent discharge during the early rainy season. Elevated TDS levels contribute to increased ionic strength in water, which can influence contaminant interactions with microplastics[17].

Electrical Conductivity values showed an interesting pattern. Surface waters recorded slightly lower conductivity than benthic waters at Makoko (1071 µS/cm surface vs. 1110 µS/cm benthic), suggesting that some solute concentration may be settling or accumulating in bottom layers.

Conversely, at Epe, the surface conductivity was 906 µS/cm compared to 859 µS/cm in the benthic zone. These spatial differences may reflect localized discharges, sediment resuspension, and hydrodynamic conditions. According to a study, such changes in ionic balance influence microplastic distribution by affecting particle density and buoyancy[18].

Overall, the May cycle results suggest an increased influence of seasonal runoff and early rainy season dynamics. The changes in physicochemical conditions—especially temperature, conductivity, and DO—play a critical role in shaping the vertical and horizontal movement of microplastics and associated pollutants.

**The August cycle** results represent the peak of the rainy season and reveals distinctive shifts in water quality parameters across the three sampling locations—Makoko, Ikorodu, and Epe—highlighting the influence of both seasonal hydrological patterns and site-specific anthropogenic activities (Table 4.4).

Surface water pH in August was highest at Makoko (8.5), followed by Ikorodu (8.2), and lowest at Epe (7.9). This indicates a slightly alkaline condition at all sites, likely due to dilution from rainfall and continued anthropogenic input. Benthic waters had consistently lower pH levels: 7.9 at Makoko, 7.7 at Ikorodu, and 6.8 at Epe. The drop in pH with depth, particularly at Epe, is suggestive of organic matter breakdown and limited buffering capacity in the bottom sediments. This pattern aligns with a study, which observed that microbial respiration and decay processes typically acidify benthic layers during the wet season[14].

Surface water temperatures were relatively uniform across sites, ranging from 27.2°C at Epe to 28.4°C at Makoko. Ikorodu recorded 28.1°C. Benthic waters were notably cooler: 24.8°C at Makoko, 24.6°C at Ikorodu, and 24.0°C at Epe. These differences reflect thermal stratification,

with surface waters absorbing more solar energy. Stratification impacts oxygen mixing and contaminant dispersion. As noted by a recent study, such temperature gradients influence the buoyancy and vertical distribution of microplastics, with denser particles tending to settle in cooler, less turbulent benthic zones[15].

Dissolved Oxygen levels were comparatively higher during this cycle than in earlier months. Surface DO was highest at Makoko at 7.0mg/L, followed by Ikorodu at 6.4mg/L and Epe at 6.1mg/L. These elevated values indicate increased aeration from rainfall and surface runoff. Benthic DO, while improved from previous cycles, remained lower, ranging from 4.7mg/L at Ikorodu and Epe to 4.9mg/L at Makoko. While still deficient for many benthic organisms, the elevated values may indicate increased mixing during storm events. According to a study, such fluctuations can affect organic decomposition rates and the degradation behavior of plastics in sediments[16].

Surface TDS concentrations were highest at Makoko (670 mg/L), followed by Ikorodu (659 mg/L) and Epe (650 mg/L). In benthic waters, TDS was lower, with values of 600 mg/L at Makoko, 586 mg/L at Ikorodu, and 500 mg/L at Epe. These patterns suggest increased solute inputs in surface waters due to runoff, effluents, and tidal actions, particularly in urbanized Makoko. The benthic zone, though receiving less direct input, still accumulates materials over time. Elevated TDS affects water density and ionic strength, which may influence microplastic behavior and contaminant adsorption[17].

Electrical Conductivity values in this cycle correspond closely with TDS levels. Surface water conductivity was highest at Makoko (1200 µS/cm), moderate at Ikorodu (1110 µS/cm), and lowest at Epe (1000 µS/cm). In the benthic zone, Makoko again recorded high values (1189

µS/cm), followed by Ikorodu (1030 µS/cm) and Epe (906 µS/cm). This consistency reinforces the view that Makoko experiences heavier pollutant loading. Conductivity is a measure of ionic content, and elevated levels, especially in benthic zones, may enhance the interaction between microplastics and dissolved metals[18].

During this cycle, Makoko consistently exhibited the highest values for conductivity, TDS, and surface DO, reflecting its urban setting and high pollutant input. Despite these influences, DO levels remained relatively high in surface water, likely aided by rainfall and mixing. Ikorodu showed intermediate values for most parameters, suggesting moderate anthropogenic impact, while Epe, typically considered less polluted, recorded the lowest pH and conductivity, especially in the benthic zone. However, the reduced DO in bottom waters likely points to organic loading and limited oxygen penetration, potentially linked to sediment deposition during the rainy season.

The **November cycle** results, representing the post-rainy season, reveal significant site-specific and vertical differences in the physicochemical quality of surface and benthic waters across Makoko, Ikorodu, and Epe (table 4.5). These differences reflect the effects of seasonal transitions, runoff tapering, and sediment–water interactions.

Surface water pH ranged from 8.0 at Epe to 8.6 at Makoko, with Ikorodu at 8.5. These values indicate slightly alkaline surface conditions, likely influenced by reduced rainfall, organic decay, and localized effluent input. In contrast, benthic pH values were lower across the board, with 7.9 in Makoko, 7.8 in Ikorodu, and 7.0 in Epe. The pH drop with depth, especially at Epe, likely results from microbial decomposition of organic matter, which releases acidic by-products. Such depth-based pH gradients are known to influence the solubility of metals and interaction with

microplastics[14].

Temperature measurements reveal that surface waters had cooled slightly compared to previous cycles, recording 26°C in both Makoko and Ikorodu, and 25°C at Epe. Benthic temperatures were lower still, with 23.4°C at Makoko, 23.2°C at Ikorodu, and 23.0°C at Epe. This temperature stratification, though narrow, suggests that solar heating still influences surface layers more than benthic zones. According to a study, such gradients can determine the vertical mobility and aggregation behavior of microplastics and suspended matter in the water column[15].

Dissolved oxygen levels were at their highest during the November cycle across both surface and bottom layers. Surface DO values ranged from 6.8 mg/L at Epe to 7.2 mg/L at Makoko. The benthic DO values also increased compared to earlier cycles, measuring 5.0 mg/L at Makoko, 4.8 mg/L at Ikorodu, and 4.6 mg/L at Epe. This improvement may result from lower water temperatures increasing oxygen solubility and reduced organic load following the rainy season. Nevertheless, benthic zones remain partially oxygen-depleted, a concern for sediment-dwelling fauna and microplastic degradation rates[16].

Surface water TDS levels were highest at Makoko (679 mg/L), followed by Ikorodu (661 mg/L) and Epe (654 mg/L). In benthic waters, TDS was slightly reduced, measuring 611 mg/L at Makoko, 603 mg/L at Ikorodu, and 563 mg/L at Epe. These elevated surface values likely reflect residual runoff and urban discharge in Makoko and Ikorodu. The higher ionic concentration at the surface contributes to elevated conductivity and affects the suspension behavior of fine particles and contaminants[17].

Electrical conductivity mirrored the TDS trend, with surface readings of 1250 µS/cm (Makoko), 1190 µS/cm (Ikorodu), and 1110 µS/cm (Epe). Benthic values followed closely: 1200 µS/cm at

Makoko, 1130 µS/cm at Ikorodu, and 960 µS/cm at Epe. This slight elevation in benthic conductivity, particularly in Makoko, may be due to the accumulation of denser ions in sediments or limited vertical mixing post-rainfall. A study noted that such conditions can enhance the retention of microplastics in benthic layers by altering their density and electrostatic interactions with particles[18].

During this cycle, Makoko recorded the highest values in nearly all parameters, indicating strong urban and domestic pollution influence. The minimal difference between surface and benthic TDS and conductivity suggests persistent vertical mixing and possible resuspension of sediments. In Ikorodu, there was intermediate readings across parameters, consistent with moderate anthropogenic impact and proximity to semi-urban developments. While Epe, though relatively less impacted, it recorded the lowest benthic pH and conductivity, indicating more acidic and less saline benthic conditions. This could be associated with slower turnover rates and reduced anthropogenic discharge.

The result from November depicts a post-rainfall stabilization phase where surface inputs are reduced, but prior pollutant loads still influence water quality. The improved DO levels suggest partial recovery, though bottom layers remain less favorable. Horizontal and vertical stratification establishes pronounced density gradients that shape the vertical profile of physicochemical conditions, thereby influencing the transport, settling behavior, and persistence of microplastics and other contaminants in aquatic systems (e.g., through buoyancy changes and trapping at pycnoclines)[19].

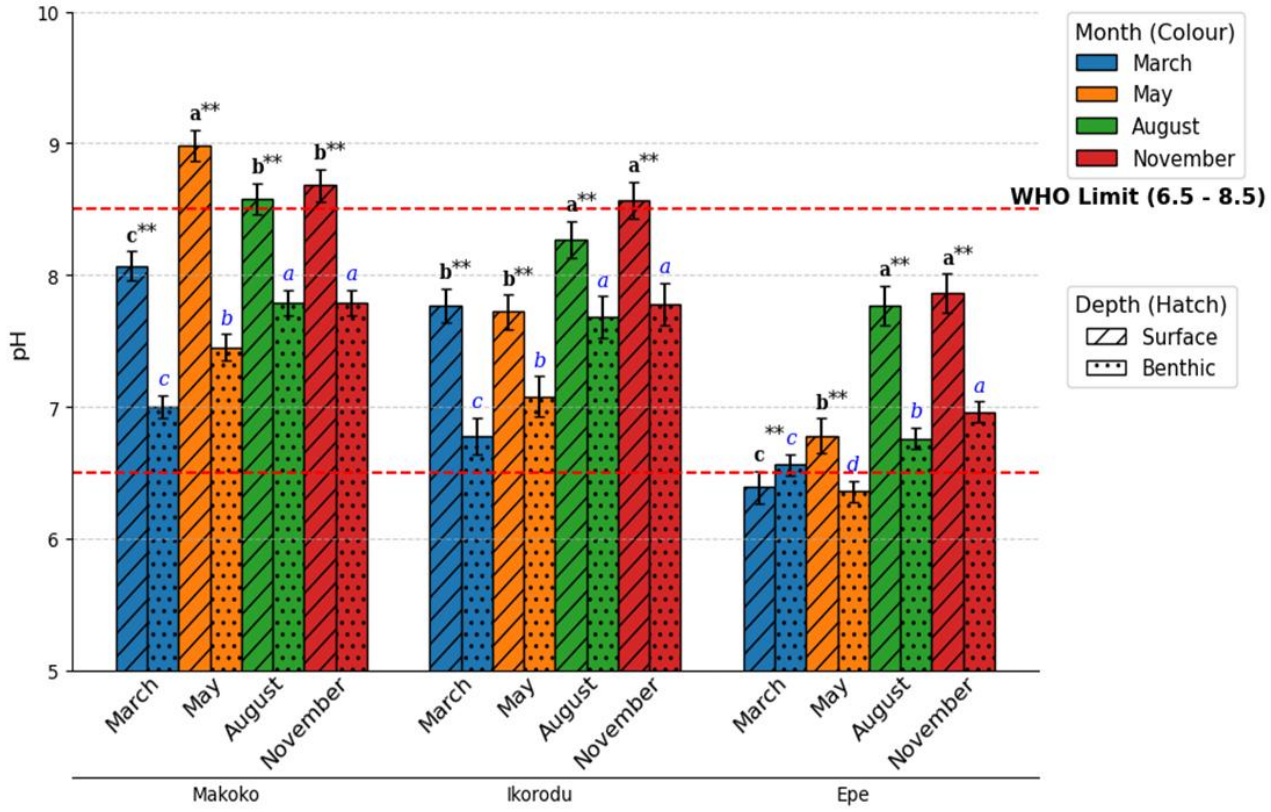


**Figure 4.9:** Seasonal Variation in pH of Surface and Benthic Waters from Lagos (Makoko and Ikorodu Axis) and Epe Lagoons

**Source:** Author's Field Work, 2025

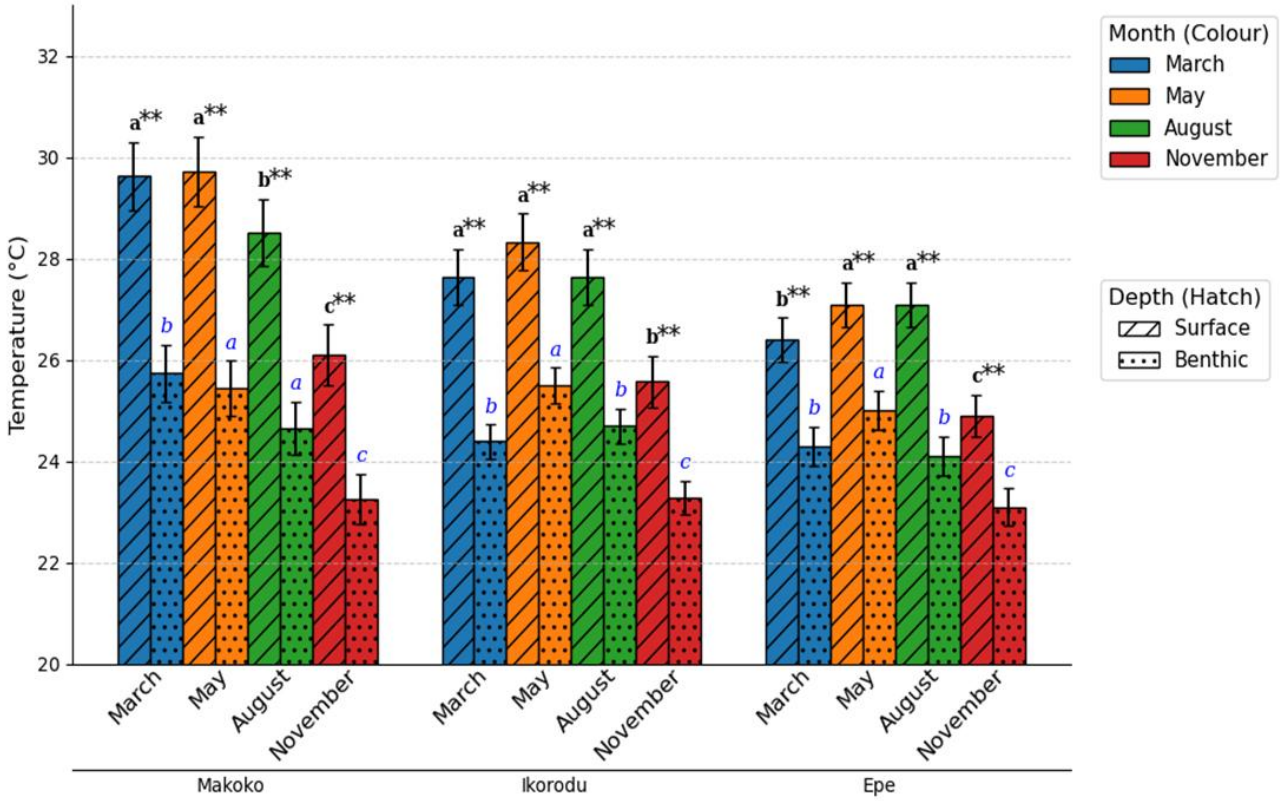


**Figure 4.10:** Seasonal Variation in Temperature of Surface and Benthic Waters from Lagos (Makoko and Ikorodu Axis) and Epe Lagoons

**Source:** Author's Field Work, 2025

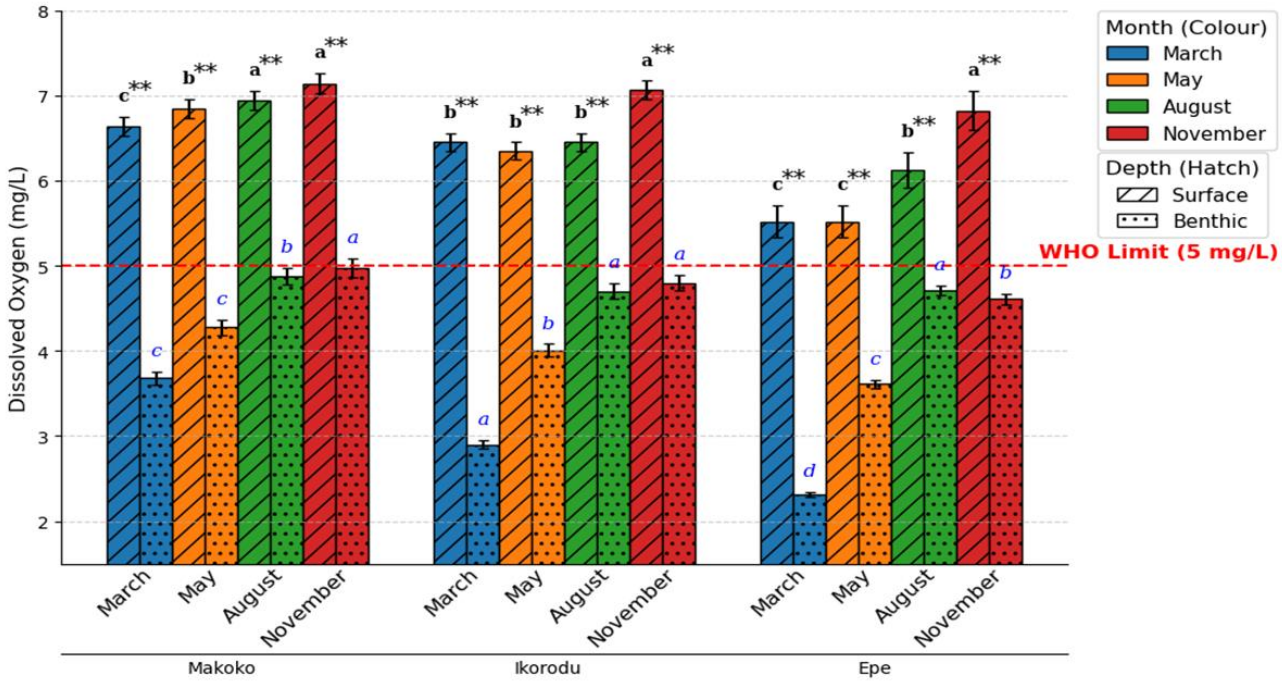


**Figure 4.11:** Seasonal Variation in DO of Surface and Benthic Waters from Lagos (Makoko and Ikorodu Axis) and Epe Lagoons

**Source:** Author's Field Work, 2025

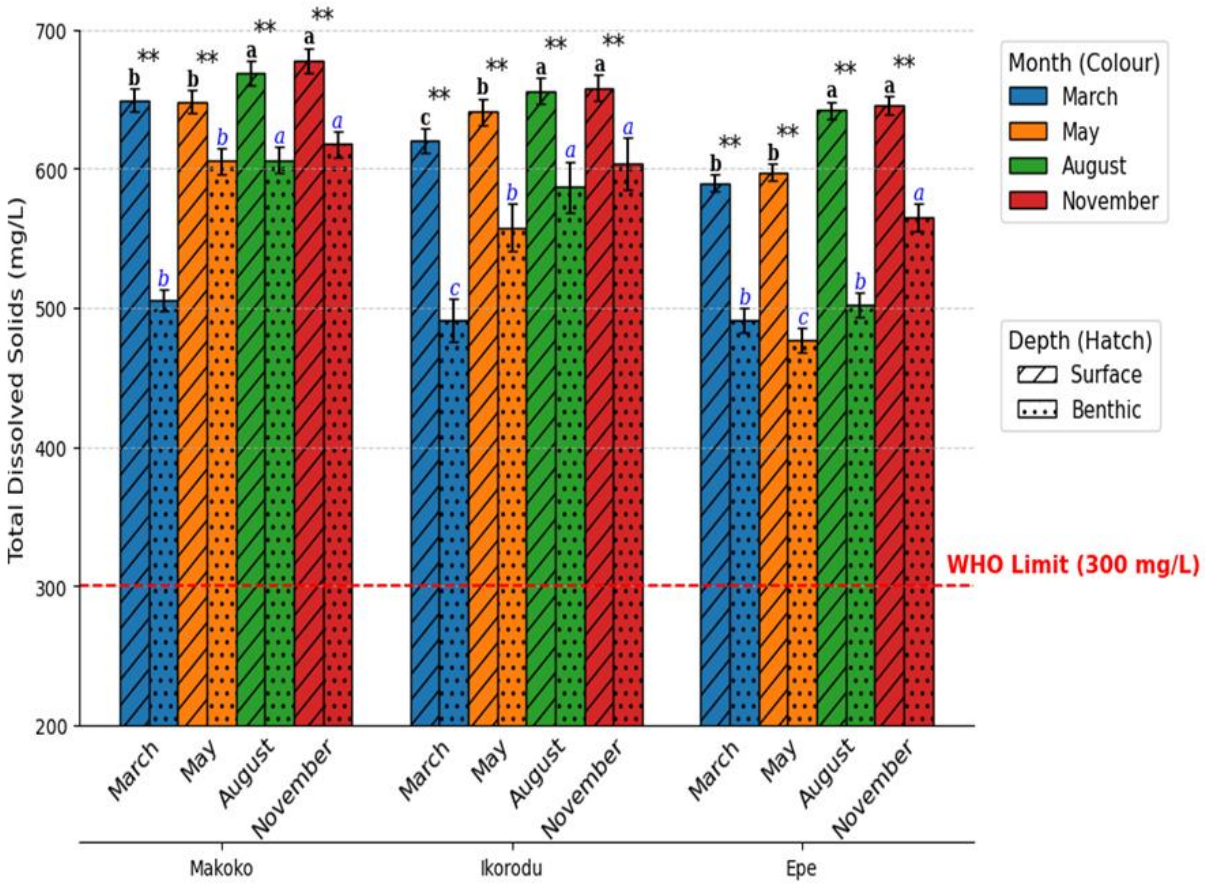


**Figure 4.12:** Seasonal Variation in TDS of Surface and Benthic Waters from Lagos (Makoko and Ikorodu Axis) and Epe Lagoons

**Source:** Author's Field Work, 2025

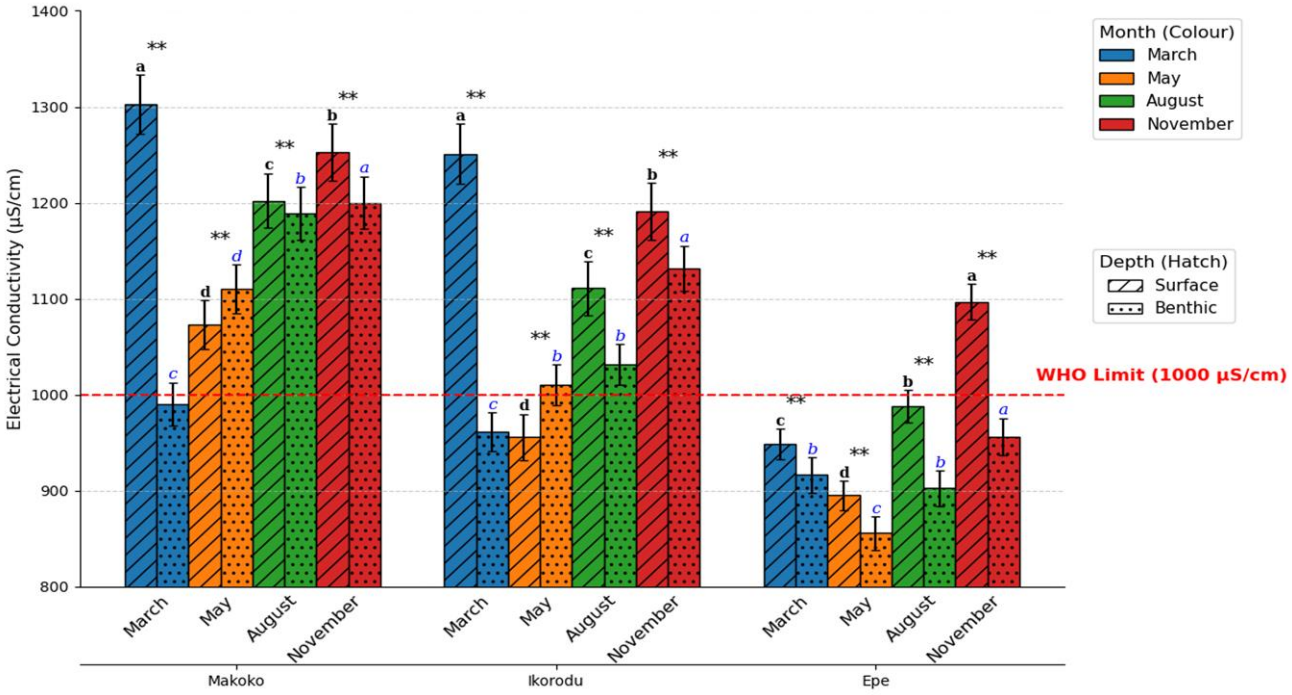


**Figure 4.13:** Seasonal Variation in EC of Surface and Benthic Waters from Lagos (Makoko and Ikorodu Axis) and Epe Lagoons

**Source: Author's Field Work, 2025**

#### 4.2.2.1 Seasonal Variations in Physicochemical Properties of Surface and Benthic Waters Across Sampling Cycles

The physicochemical dynamics of surface and benthic waters in Lagos and Epe Lagoons exhibited clear seasonal trends shaped by climatic conditions, rainfall patterns, anthropogenic inputs, and hydrological processes. Analysis across the four sampling cycles—March (early dry season), May (onset of rainy season), August (peak rainy season), and November (post-rainy season)—showed notable fluctuations in temperature, pH, dissolved oxygen (DO), total dissolved solids (TDS), and electrical conductivity. These parameters are interlinked and collectively influence the ecological quality of aquatic ecosystems as well as the behavior of pollutants such as microplastics and heavy metals.

Across all cycles, surface waters were consistently warmer than benthic waters, due to solar heating and atmospheric exposure as shown in **Figure 4.10**. The highest surface temperatures were recorded in March (up to 29.5$^0$C) at Makoko, signaling the tail end of the dry season when solar intensity is typically highest. In contrast, August and November showed slightly lower surface temperatures (26$^0$C - 28$^0$C), attributable to increased cloud cover and rainfall. Benthic temperatures ranged from 23.0$^0$C (Epe, November) to 25.9$^0$C (Makoko, March), remaining relatively stable compared to surface layers. The temperature differential between surface and bottom layers was most pronounced in the dry season and slightly narrowed during the rainy season due to water column mixing and storm-induced turbulence[14,15].

These variations are ecologically significant, as thermal stratification influences oxygen dynamics, nutrient cycling, and the vertical distribution of microplastics, which tend to float at higher temperatures and settle as temperatures decrease[18].

The pH values ranged from 6.4 to 8.9, with surface waters showing higher (more alkaline) values than benthic waters across all sampling periods. The highest surface pH was observed in May (8.9 at Makoko), possibly due to increased photosynthetic activity, decreased $CO_2$ concentrations, and dilution of acidic effluents during the early rainy season. Conversely, the lowest pH was observed in Epe's benthic zone in March (6.5) and again in November (7.0), where organic matter decay and $CO_2$ accumulation likely lowered pH values at the sediment-water interface. Seasonal variation in pH as shown in **figure 4.9**, reflects changes in biological productivity, organic matter breakdown, and input of acidic/alkaline substances from runoff and effluents[14]. These fluctuations also impact the speciation and bioavailability of contaminants, including heavy metals and plastic-associated pollutants[19].

Dissolved oxygen levels are strongly influenced by temperature, as higher temperatures reduce oxygen solubility; organic load, which consumes oxygen during decomposition; and mixing dynamics, which facilitate aeration—all of which are modulated by rainfall-driven hydrodynamics[20]. Across the board, surface waters recorded higher DO levels than benthic layers, consistent with oxygenation from the atmosphere and aquatic photosynthesis. The lowest DO levels were recorded in March benthic waters (2.3 - 3.7 mg/L), indicating oxygen depletion due to thermal stratification and active microbial decomposition of organic matter **(figure 4.11)**. In contrast, DO levels peaked in November, with surface DO ranging from 6.8 - 7.2 mg/L and benthic DO improving to 4.6 - 5.0 mg/L. This increase likely stems from post-rainfall aeration, cooler water temperatures that enhance oxygen solubility, and reduced organic load following the peak runoff season[16].

Persistent hypoxia in benthic zones, particularly in Makoko and Ikorodu during March and May, may stress aquatic fauna and limit the degradation of pollutants like microplastics, which tend to persist longer under low-oxygen conditions[19].

Total dissolved solids levels, which indicate the concentration of inorganic and organic dissolved substances, varied from 475 to 679 mg/L across sites and seasons. Makoko consistently showed the highest TDS levels, reflecting urban runoff, domestic discharge, and industrial effluents, particularly in surface waters during March and November (**figure 4.12**). TDS was generally higher during the dry season (March) and post-rainy season (November), likely due to evapo-concentration and legacy pollution, while slightly lower during peak rainy months (May and August) due to dilution by rainfall. Higher TDS values correlate with increased ionic strength, which affects water density and influences microplastic behavior by altering aggregation, flotation, and adsorption[17]. Excessive TDS also poses physiological stress to aquatic species, particularly those with limited osmoregulatory capacities[20].

Electrical conductivity ranged from 859 µS/cm (Epe benthic, May) to 1250 µS/cm (Makoko surface, November). The highest values were recorded during November and March, highlighting accumulated salts, nutrients, and pollutants from both natural and anthropogenic sources (figure 4.13). Rain-induced dilution effects were evident during the May and August cycles, with lower conductivity values due to the influx of freshwater from surface runoff. The difference between surface and benthic conductivity was generally more pronounced during the dry season, narrowing during the wet season due to mixing.

Elevated conductivity, especially in Makoko and Ikorodu, underscores the pollution burden in these urbanized areas. According to a study, such environments may promote the adsorption of

heavy metals and organic contaminants onto microplastics, thus increasing their ecotoxicological significance[18].

Seasonal fluctuations in physicochemical parameters strongly influence the mobility, distribution, and persistence of microplastics and associated contaminants[21]. Warmer, low-oxygen benthic conditions, common in March and May, may limit microbial degradation and enhance the accumulation of microplastics in sediments. In contrast, higher DO and lower pH in the rainy and post-rainy months may influence chemical interactions and bioavailability of pollutants.

Furthermore, stratification and conductivity gradients across seasons dictate the vertical flux of microplastics, whether they remain suspended, sink to the bottom, or interact with other particles and organisms in the water column[14,18].

The dry season (March) marked the peak of stratification, with pronounced temperature and DO differences between surface and benthic waters, higher TDS, and potential for pollutant concentration at the surface.

The onset of rains in May initiated vertical mixing and partial oxygenation of deeper layers, though organic load still suppressed DO at some benthic sites. At the height of the rainy season in August, there was an improvement in DO, reduced TDS, and relatively stable temperatures, but also potential for resuspension of sediment-bound contaminants.

During the post-rainy season in November, the best water quality indicators in terms of oxygenation, though legacy effects from runoff led to high conductivity and TDS, especially in Makoko.

### 4.2.3 Discussion on Heavy Metals in Surface and Benthic Waters of the Lagos (Makoko and Ikorodu Axis) and Epe Lagoons

The concentrations of key heavy metals—lead (Pb), copper (Cu), nickel (Ni), cadmium (Cd), and chromium (Cr)— analyzed in surface and benthic waters from the sample locations: Makoko, Ikorodu, and Epe are shown in table 4.6 to 4.9. These values revealed significant seasonal and spatial variability, which is further reiterated by their corresponding Pollution Indices (PIs) in Table 4.10 to 4.13. All heavy metal concentrations of the Lagos Lagoon across the three locations significantly exceeded the World Health Organization (WHO) guideline limits for surface waters, indicating ecological and human health risks.

**Lead** levels showed a steady increasing pattern from March to November in all locations and depths. Notably, benthic waters consistently recorded much higher Pb concentrations than surface waters, with a Pairwise t-test confirming this difference as highly significant ($p < 0.01$). At Makoko, Pb levels in surface water started at 2.01±0.11 mg/L in March and increased significantly to 3.56±0.22 mg/L in November. In the same period, Pb levels in benthic water rises from 5.42±0.13 mg/L to 6.77±0.11 mg/L. Ikorodu and Epe showed a similar trend of increase in both surface and benthic waters. in November, Pb concentrations in surface water at Ikorodu and Epe peaked at 1.88±0.20 mg/L and 1.75±0.11 mg/L, respectively, and Pb concentrations in benthic water reached 6.54±0.10 mg/L and 6.32±0.06 mg/L, respectively, as shown in Figure 4.14.

The Pollution Index (PI) for Pb supports the findings of the Pb concentration pattern. In November, benthic pollution indices at Ikorodu and Epe reached alarmingly high values of 677 and 632, respectively, signifying severe contamination. Surface waters also presented elevated PIs, with Makoko hitting the highest value at 355.9 in November (Figure 4.15). The significantly

elevated PIs reflect the direct correlation between Pb concentrations and pollution severity, with benthic zones exhibiting the highest contamination burden. These high values highlight the extent of lead pollution in the Lagos Lagoon, particularly in sediments, likely due to runoff, industrial waste, and sediment accumulation over time.

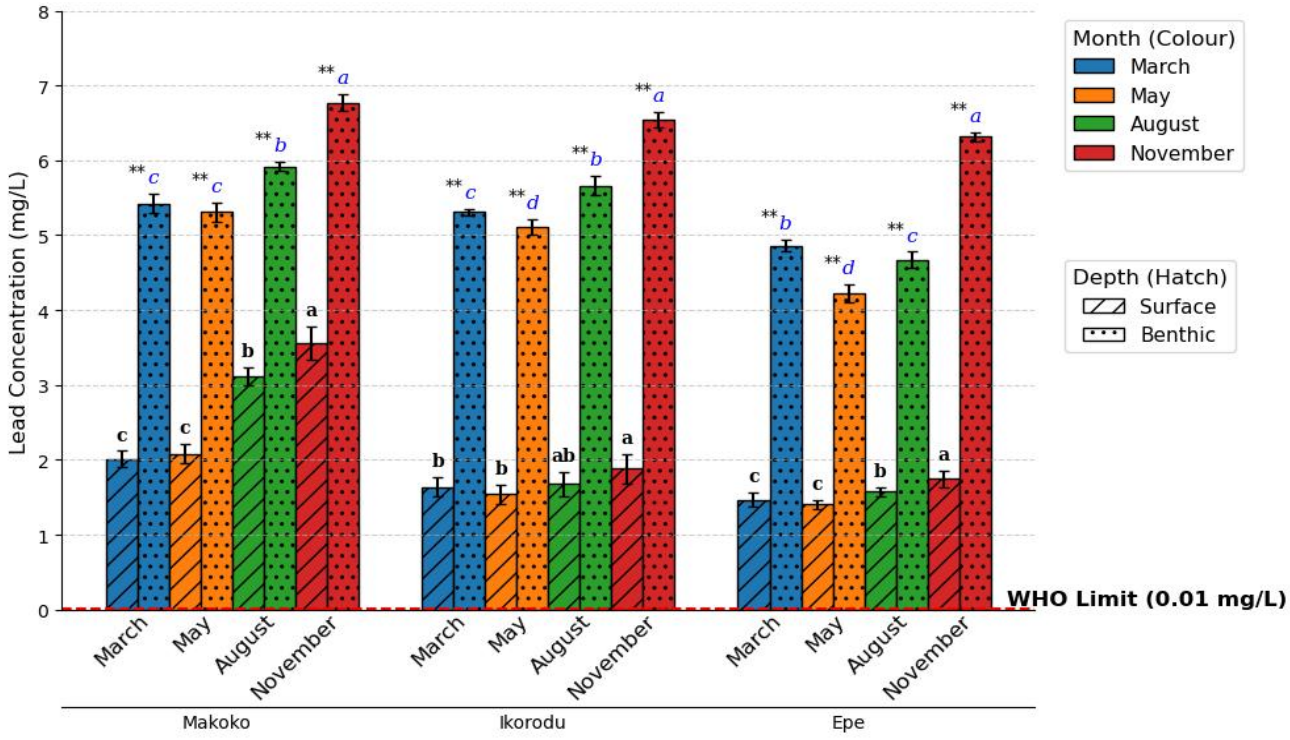


**Figure 4.14:** Variations in Lead Concentrations in Surface and Benthic Waters from Lagos (Makoko and Ikorodu Axis) and Epe Lagoons

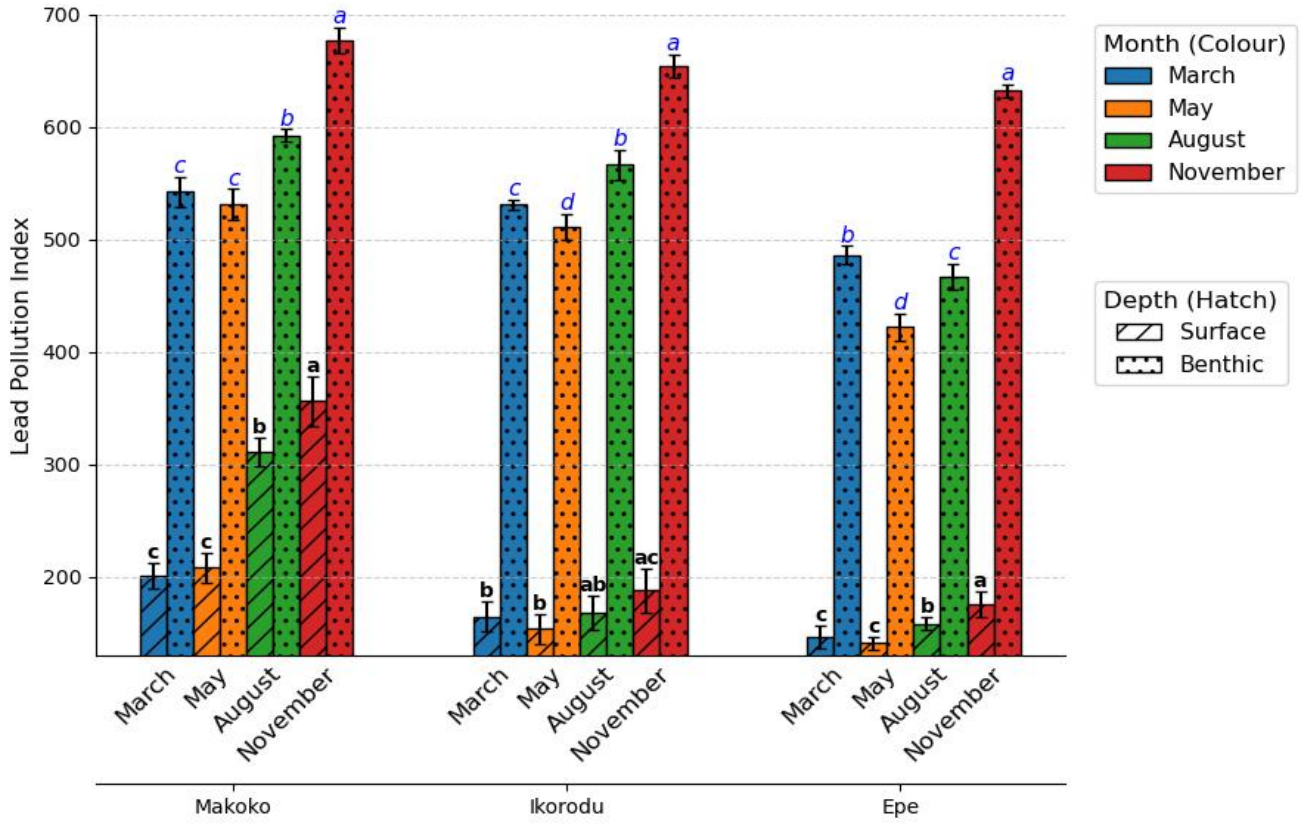


**Figure 4.15:** Seasonal Variation in Lead Pollution Indices of Surface and Benthic Waters from Makoko, Ikorodu and Epe

**Source:** Author's Field Work, 2025

**Copper** concentrations followed a similar temporal and spatial pattern to lead. In Makoko, surface Cu level in surface water rise from 2.71±0.22 mg/L in March to 3.11±0.03 mg/L in November ($p < 0.05$), while benthic waters showed much higher values of copper, reaching 11.56±0.23 mg/L by November. Ikorodu showed a slight increase of Cu levels in surface water, with November cycle values of 2.95±0.13 mg/L, and benthic Cu levels peaking at 10.54±0.11 mg/L. In Epe, copper concentration in surface water increased to 2.48±0.17 mg/L in November, with Cu level in benthic water reaching 9.34±0.08 mg/L. These values substantially exceed the WHO guideline of 0.05 mg/L, as displayed in Figure 4.16, signifying an urgency for targeted remediation.

The Cu Pollution Index (presented in Figure 4.17) simulates these concentration trends. Notably, benthic waters across all locations exhibited very high Cu PIs, with Makoko and Ikorodu surpassing 11, indicating extreme contamination. Although surface water indices remained moderate, they are still elevated (around 2.5 to 3.1). This shows the lagoon's critical Cu pollution status, likely from domestic waste, industrial discharge, and leaching from marine plants.

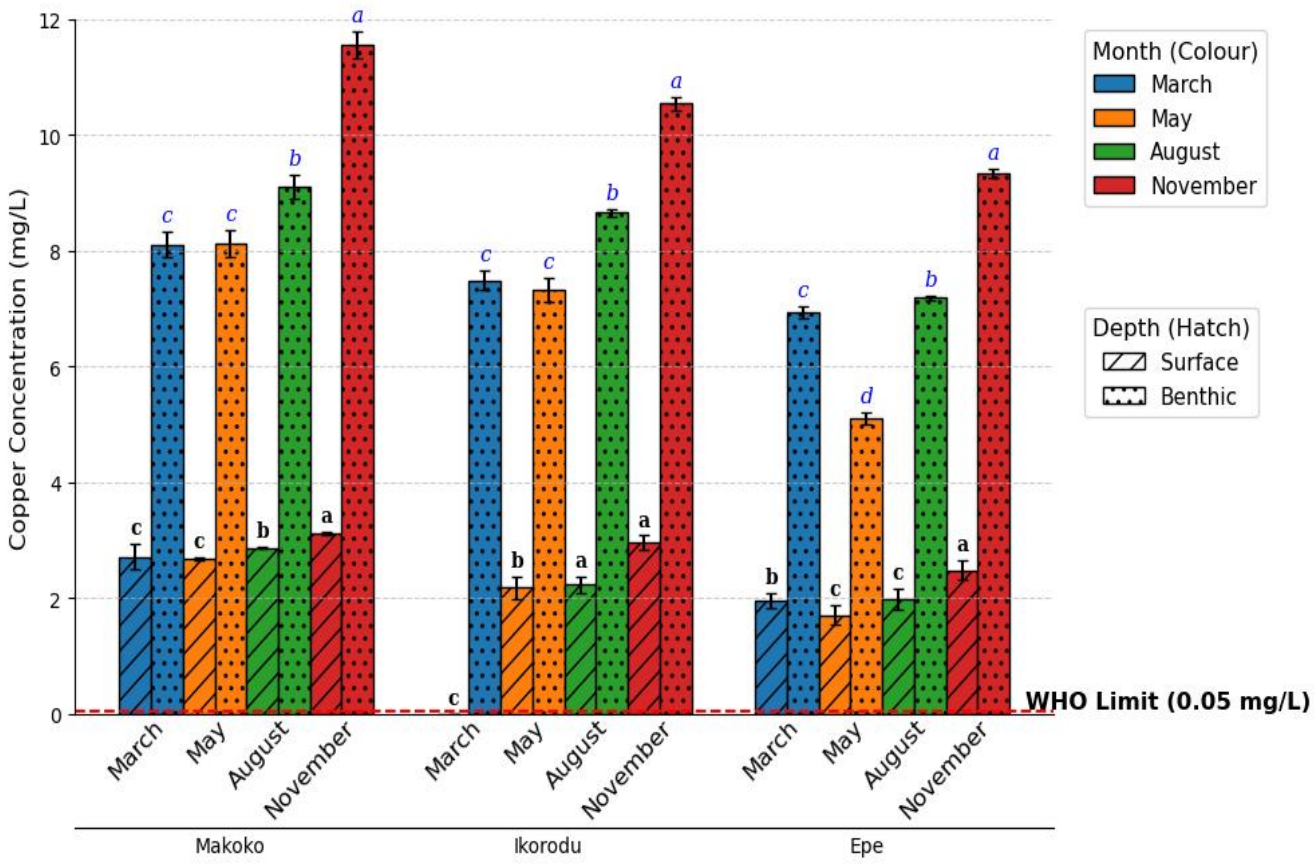


**Figure 4.16:** Seasonal Variation in Copper Concentrations in Surface and Benthic Waters from Lagos (Makoko and Ikorodu Axis) and Epe Lagoons

**Source:** Author's Field Work, 2025

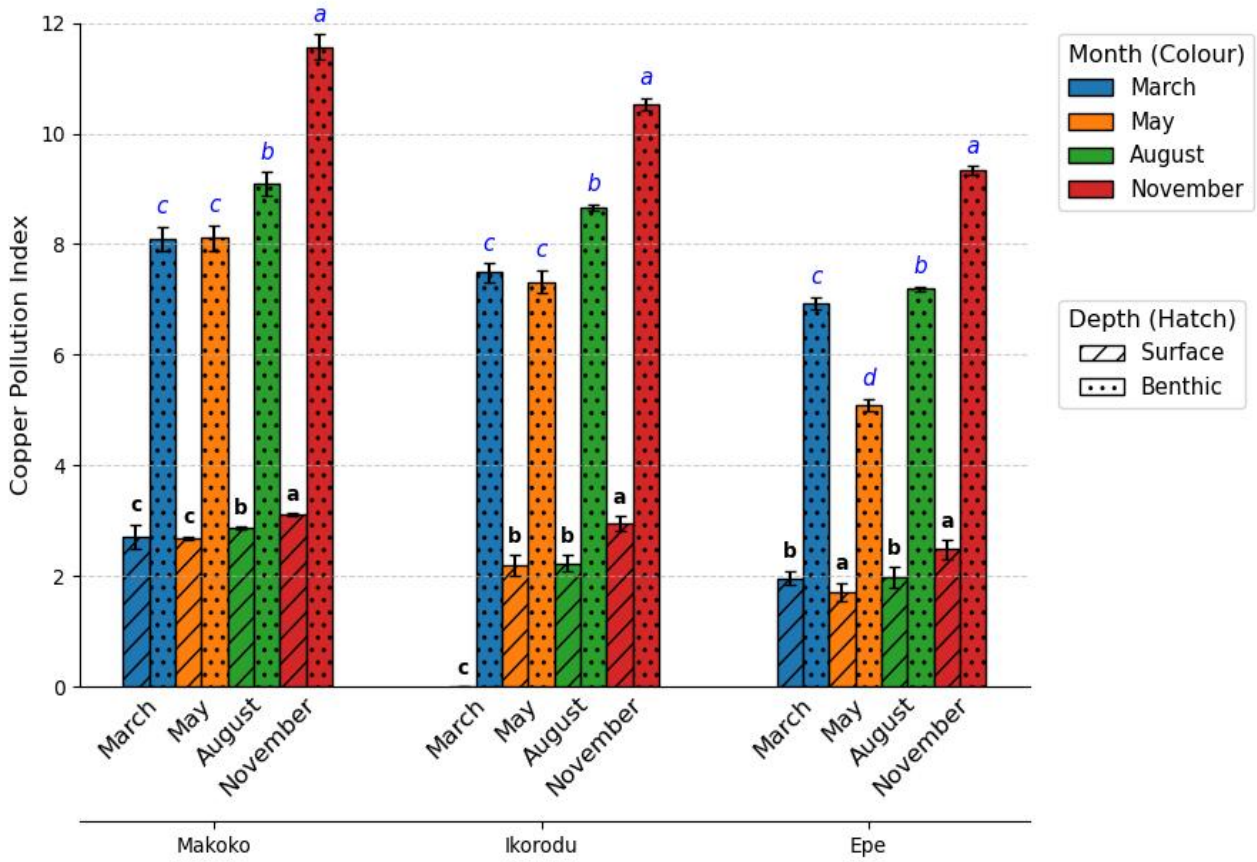


**Figure 4.17:** Seasonal Variation in Copper Concentrations in Surface and Benthic Waters from Lagos (Makoko and Ikorodu Axis) and Epe Lagoons

**Source:** Author's Field Work, 2025

As observed in lead and copper concentrations, **Nickel** concentrations in surface waters were also relatively low compared to benthic water concentrations and showed a significant seasonal rise as shown in figure 4.18. Nickel level in Makoko surface water ranged from 0.76±0.02 mg/L in May to 1.10±0.34 mg/L in November, whereas Ni level in benthic water increases from 12.55±0.29 mg/L in March to 14.66±0.49 mg/L in November. Ikorodu and Epe benthic waters similarly showed elevated Ni levels, peaking at 13.77±0.22 mg/L and 12.10±0.06 mg/L, respectively, in November. Surface waters at Ikorodu and Epe had lower concentrations but still showed increasing trends toward November.

The Pollution Index for Ni reflects these high benthic concentrations, with PI values exceeding 12 at both Ikorodu and Epe benthic sites in November, indicating extreme pollution levels (Figure 4.19). Surface PIs remained under 1.1 but increased over the months, suggesting accumulating contamination. The marked difference between the levels of Ni in surface and benthic water is consistent with Ni's typical tendency to settle in sediments. Two separate studies explained that the typical behaviour of Ni, accumulating in sediments, is associated with organic matter from plants and dead animals that settle in the benthic region. Nickel has a high affinity for organic matter, so it sinks with this debris as it settles in sediments[20,21].

**Cadmium** (Cd) levels were notably high across the lagoon at all times and in all locations, surpassing the WHO limit of 0.003 mg/L by wide margins, as shown in Figure 4.20. Surface waters at Makoko and Ikorodu exhibited relatively stable Cd concentrations around 0.09–0.12 mg/L, while Epe surface waters increased from 0.04±0.01 mg/L in March to 0.10±0.03 mg/L in November. Cadmium concentrations in benthic water were consistently higher across all sites compared to surface water. At Makoko, benthic Cd levels increased from 0.94 ± 0.04 mg/L in March to 1.33 ± 0.00 mg/L in November. Similarly, Ikorodu recorded an increase from 0.91 ±

0.03 mg/L to 1.09 ± 0.06 mg/L over the same period, while Epe showed a rise from 0.67 ± 0.04 mg/L to 0.96 ± 0.06 mg/L.

The Cd Pollution Indices, as presented in Figure 4.21, reveal extreme contamination in benthic zones, with Makoko and Ikorodu benthic PIs exceeding 1.0 in November—an indicator of severe pollution given the low WHO permissible limit. Surface water PIs were lower but still signify contamination, with a seasonal increase in all locations. The high Cd levels most probably result from battery waste, domestic sewage, and industrial and urban runoff accumulating in the sediments, which is typical of Lagos Lagoon[9]. The accumulation of Cd in the benthic region is also attributed to organic matter settling in the zone[22].

**Chromium** concentrations also exhibited significant seasonal and spatial variation (Figure 4.22). Surface waters in Makoko rose sharply from non-detectable levels in March to 0.12±0.07 mg/L in November. The concentration of chromium in benthic waters at Makoko increased from 3.47±0.02 mg/L in March to 5.62±0.01 mg/L in November. Similar patterns were seen at Ikorodu and Epe, with Cr concentrations in benthic water peaking at 5.18±0.01 mg/L and 4.91±0.04 mg/L, respectively, in November. All Cr levels exceeded the WHO limit of 0.03 mg/L throughout the year, underscoring the present Cr polluted state of the Lagos Lagoon.

Corresponding Cr Pollution Indices showed benthic sites in all locations to be highly contaminated (Figure 4.23). Cr PIs rose significantly over the seasons, reflecting the metal's bioaccumulative nature and persistence in sediments. Surface water PIs were lower than the benthic level but also increased consistently throughout the year.

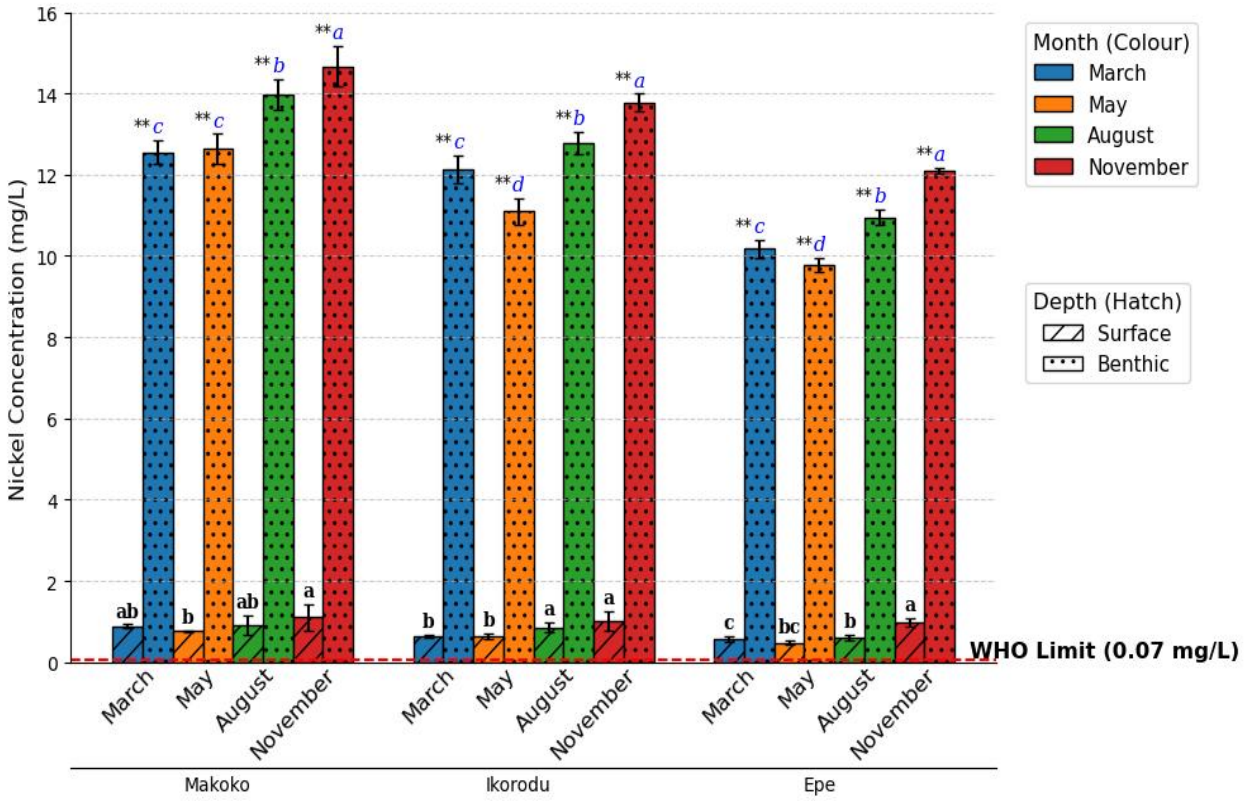


**Figure 4.18:** Seasonal Variation in Nickel Concentrations in Surface and Benthic Waters from Lagos (Makoko and Ikorodu Axis) and Epe Lagoons

**Source:** Author's Field Work, 2025

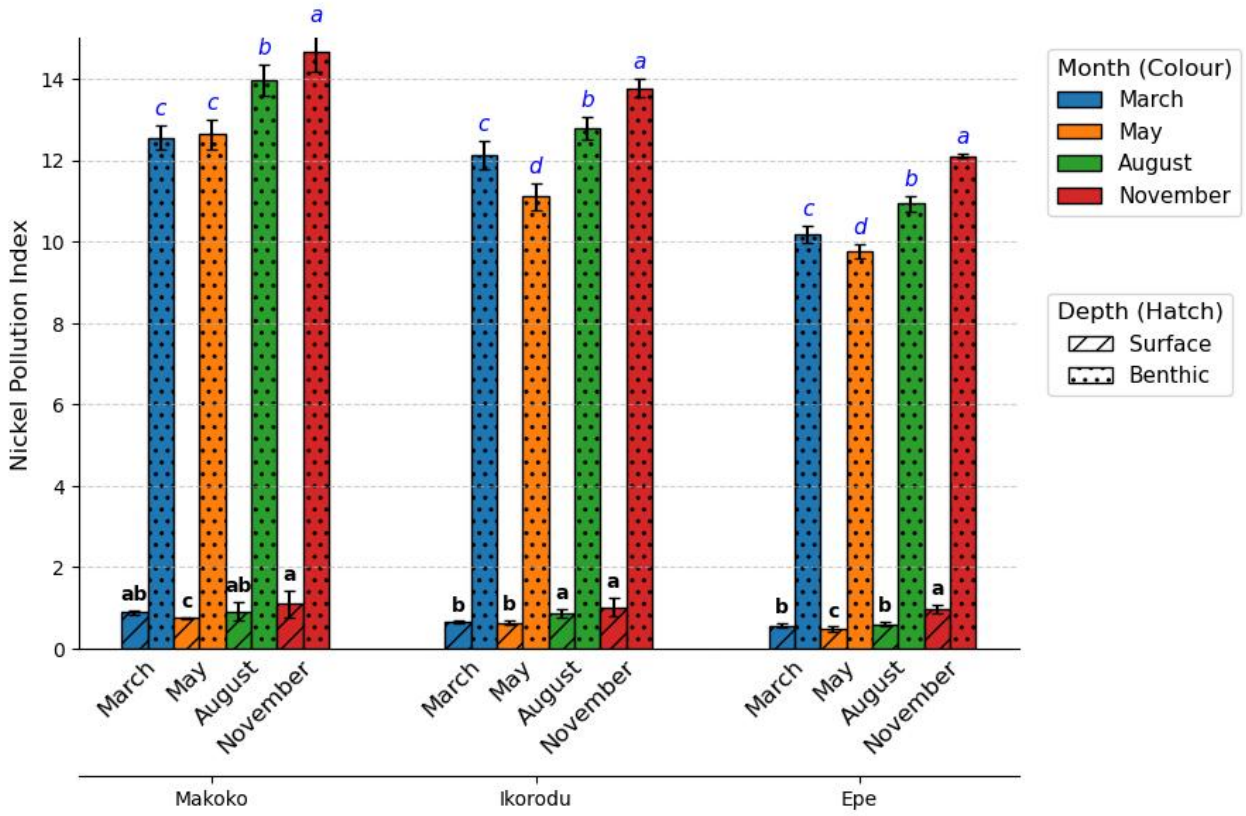


**Figure 4.19:** Seasonal Variation in Nickel Pollution indices in Surface and Benthic Waters from Lagos (Makoko and Ikorodu Axis) and Epe

**Source:** Author's Field Work, 2025

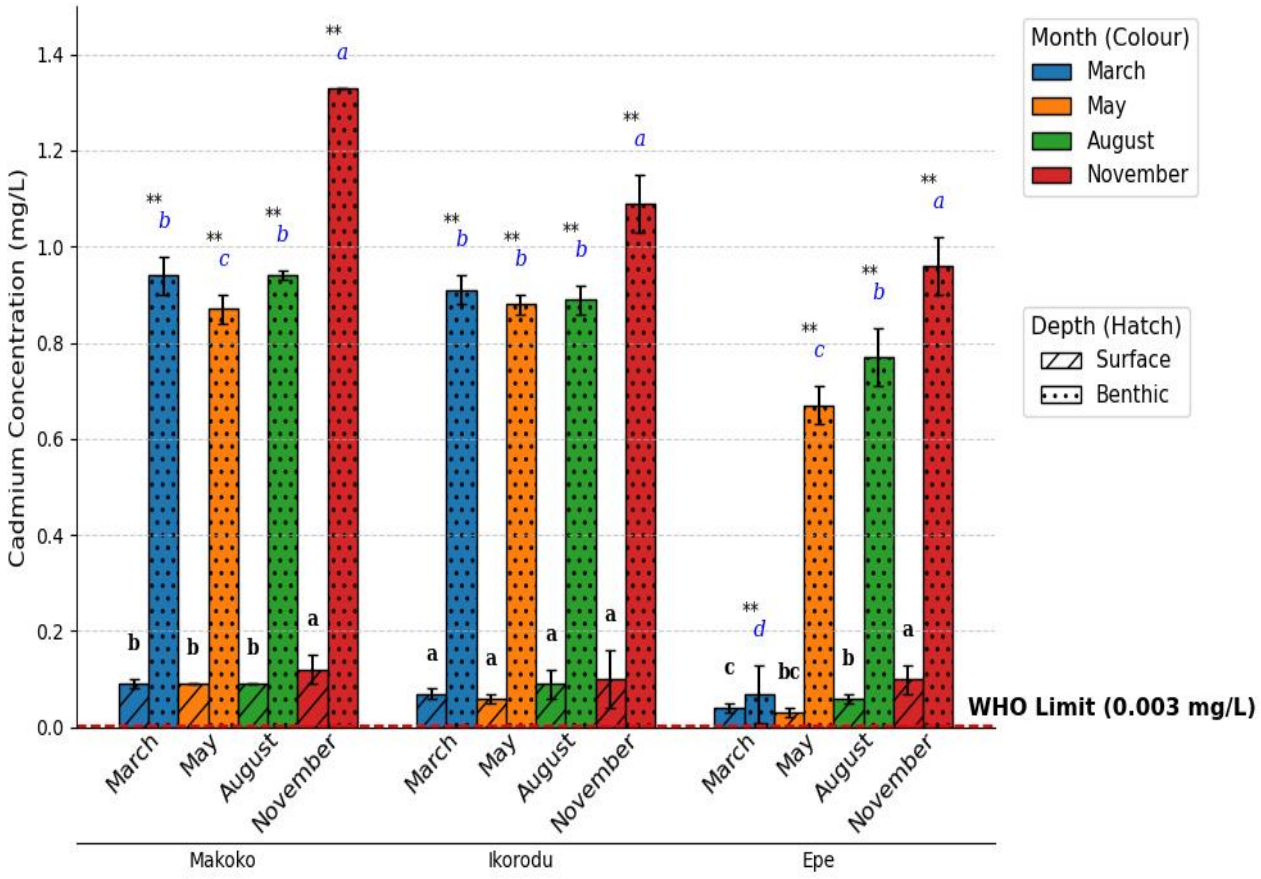


**Figure 4.20:** Seasonal Variation in Cadmium Concentrations in Surface and Benthic Waters from Lagos (Makoko and Ikorodu Axis) and Epe Lagoons

**Source:** Author's Field Work, 2025

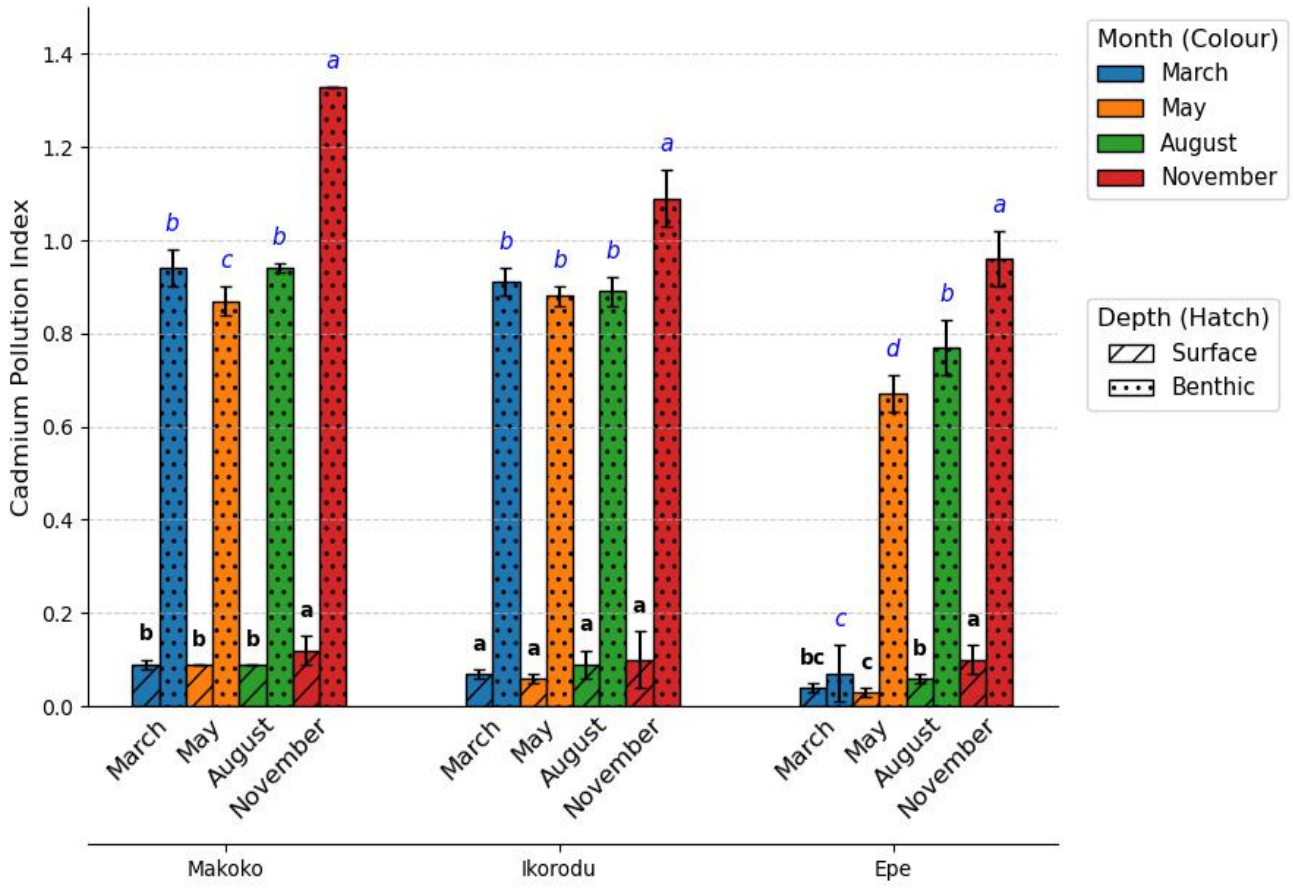


**Figure 4.21:** Seasonal Variation in Cadmium Pollution indices in Surface and Benthic Waters from Lagos (Makoko, Ikorodu Axis) and Epe Lagoons

**Source:** Author's Field Work, 2025

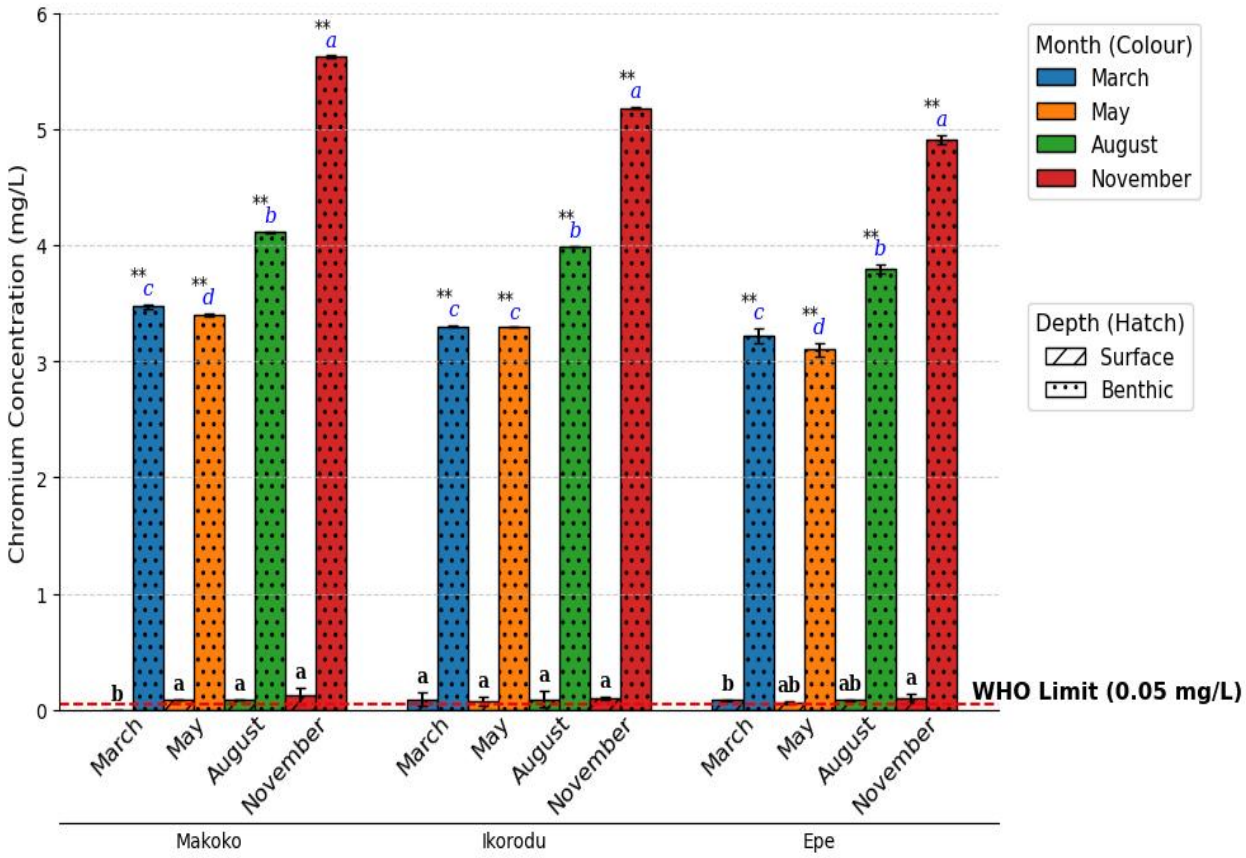


**Figure 4.22:** Seasonal Variation in Chromium Concentrations in Surface and Benthic Waters from Lagos (Makoko and Ikorodu Axis) and Epe Lagoons

**Source:** Author's Field Work, 2025

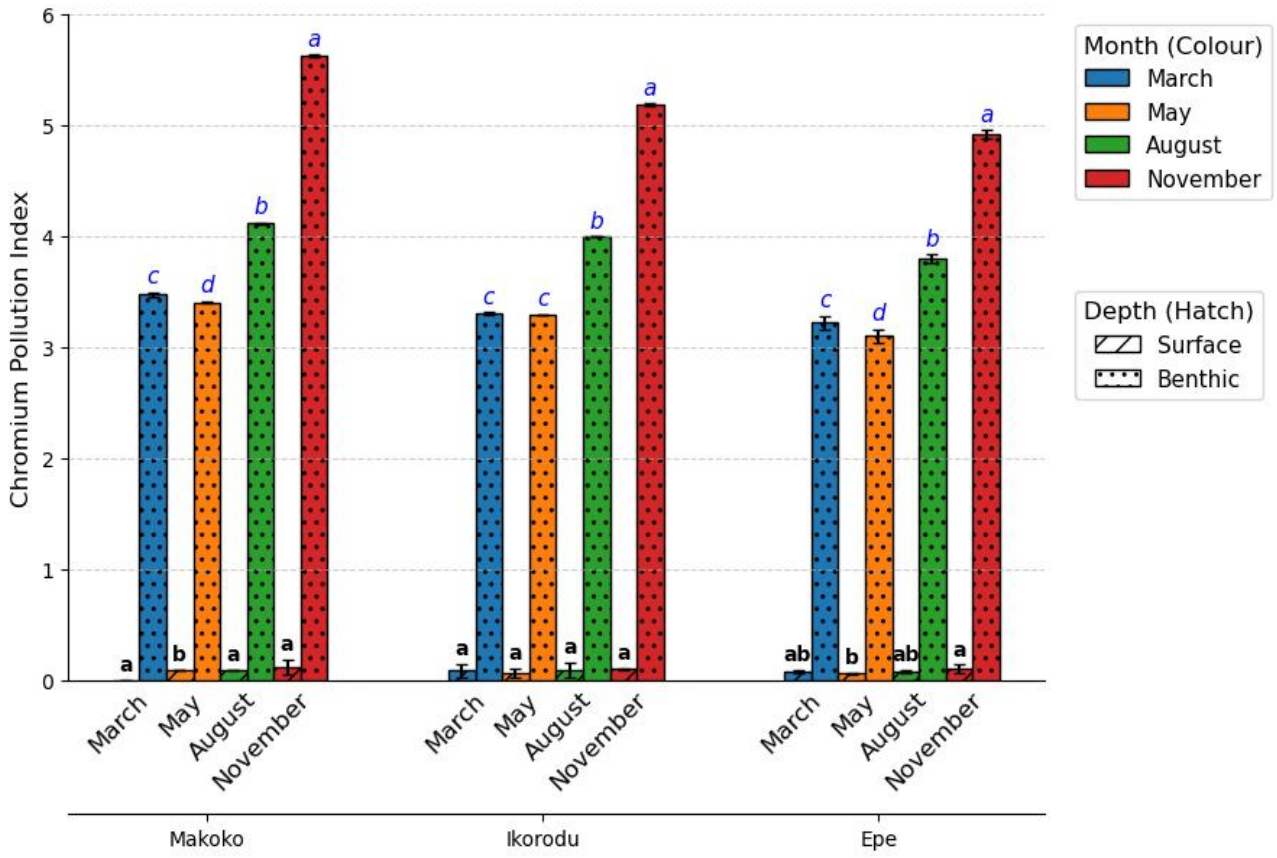


**Figure 4.23:** Seasonal Variation in Chromium Pollution indices in Surface and Benthic Waters from Lagos (Makoko and Ikorodu Axis) and Epe Lagoons

**Source:** Author's Field Work, 2025

The elevated heavy metal load in the Lagos Lagoon has been long persisting, reports that date back over a decade have identified this issue. A study reported fairly high concentrations of Cu, Cr at levels just below the WHO marine water guidelines, and Pb, Cd and Ni levels were just exceeding the WHO limits[23]. Another study also reported elevated levels of heavy metals, particularly in the surface water of Lagos Lagoon. At the time, they reported concentrations of Pb and Ni in surface water higher than WHO/FEPA limits, while Cd, Cu, Cr and were found below the permissible limit for freshwater[24]. The report of another study also showed higher concentrations of cadmium, copper, and lead in the lagoon[25]. More recently, a study reported high concentrations of Pb, Cu, Cd and Cr in the Lagos Lagoon that exceed the permissible limits[26]. However, the levels of heavy metals reported in these previous studies do not compare with those found in the present study. This indicates a long-standing, widespread metal pollution through the lagoon, significantly and continuously accumulating over the years. This worsening trend in contamination is possibly due to intensified urbanization in these areas, industrial discharge, and insufficient enforcement of environmental safety and waste regulatory policies over time.

The Lagos Lagoon's benthic waters at Makoko, Ikorodu, and Epe exhibited the highest heavy metal concentrations across all metals studied, highlighting sediments as critical reservoirs of pollutants. Several previous reports also showed higher levels of heavy metals in sediments and biota (benthic) compared to surface water[23,24,25,26]. This aligns with the findings of this study, reflected by the steady and significant elevation of heavy metal levels in benthic waters, relative to surface water.

The general trend of abundance of heavy metals in both surface and benthic water in the Lagos Lagoon is Ni > Cd > Cr > Pb > Cu, from the highest to the lowest. This trend does not follow the

pattern observed by another study (Pb > Cu > Cr > Cd) in the lagoon. This may be as a result of a significant change in metal waste-producing systems that affect the type and abundance of metals that are released into the lagoon water.

More so, the overall sequence of metal pollution severity follows the trend: Makoko > Ikorodu > Epe. Makoko showed the highest overall level of metal pollution, followed by Ikorodu, then Epe. This trend follows the same pattern as the severity of physicochemical degradation, which collectively (by observing the same trend) impacts the size and condition of the inhabiting fish in the same spatial pattern: Makoko > Ikorodu > Epe.

Temporally, heavy metal concentrations generally peaked in November, which marks the end of the rainy season and the beginning of the dry season. This period is characterized by reduced water flow, accumulation of runoff pollutants, and intensified anthropogenic activities. Among the metals, Pb and Ni showed especially high pollution indices, with values sometimes exceeding 600 in benthic waters, indicating extremely high contamination levels posing ecological risks and potential biomagnification through aquatic food webs. The heavy metal concentrations far exceed the WHO recommended limits for surface waters, confirming a severe pollution status that demands urgent monitoring and mitigation efforts.

#### 4.2.3.1 Contamination Factor, Degree of Contamination and Pollution Load Index

The calculated CF values revealed that all metals, across all sites and sampling cycles, exceeded the WHO permissible limits (CF > 1), with the exception of occasional Cr values in surface waters, which were comparatively lower. Notably, Pb and Cd consistently recorded extremely high CF values, particularly in benthic waters where values often exceeded several hundred, indicating severe enrichment and anthropogenic influence. The DC values confirmed this trend, with benthic waters in all sites and cycles falling within the “very high contamination” category

(DC ≥ 32), in contrast to surface waters which, although generally lower, still showed substantial contamination. PLI values for all sites and cycles were far greater than 1, indicating a pervasive deterioration of water quality throughout the Lagos and Epe Lagoons. Seasonal variation was also evident, with November and August cycles typically exhibiting the highest DC and PLI values, suggesting possible cumulative pollutant loading during or after the rainy season. Spatially, Makoko benthic waters consistently recorded the highest CF, DC, and PLI values, reflecting its proximity to intense human activity, waste discharge, and shoreline industrial operations. The contamination patterns observed in this study are consistent with findings from other Nigerian lagoon systems and international water bodies subject to high anthropogenic pressures. For example, a study reported similarly elevated CF values for Pb and Cd in Lagos Lagoon sediments, attributing these to direct industrial effluent discharge, leaching from corroded roofing sheets, and the use of leaded paints[21]. Another related study found PLI values exceeding 2 in the Ogun River estuary, indicative of significant multi-metal contamination from upstream agricultural runoff and urban waste[22]. Comparable trends have been documented in other developing countries; a study observed DC values above 500 in the lower reaches of the Niger Delta creeks, while another reported PLI values of 3–5 in the Pearl River Delta, China,linked to rapid urbanisation and industrialisation[23,24]. The benthic enrichment observed in our study mirrors global patterns where metals preferentially bind to particulates and accumulate in sediments, leading to higher CF, DC, and PLI values compared to the water column[25]. However, the magnitude of contamination in this present study, especially for Pb and Cd, exceeds most reported values in comparable ecosystems, highlighting the severity of pollution stress in Lagos and Epe Lagoons. The contamination factor, degree of contamination, and pollution load index analyses provide a clear and quantitative confirmation of the heavy metal pollution burden in the Lagos and Epe Lagoons. The consistently elevated values, particularly in

benthic waters, point to sustained and possibly increasing pollutant inputs from both point and non-point sources. These results establish a strong baseline for assessing ecological risk and justify the integration of complementary pollutant metrics, such as microplastic and phthalate abundance, to obtain a holistic understanding of environmental quality in the study area.

### 4.2.4 Discussion on Microplastic Occurence in *Oreochromis niloticus* and *Clarias gariepinus* Fishes from the Lagos (Makoko and Ikorodu Axis) and Epe Lagoons

Microplastic occurence, like the heavy metals, showed temporal and spatial variation in *Oreochromis niloticus* and *Clarias gariepinus* across the three study locations, with microplastic counts increasing steadily from March to November. Fish samples from Makoko consistently exhibited the highest microplastic loads, followed by Ikorodu and then Epe. This pattern aligns with the level of urbanization and pollution pressure in each location, with Makoko receiving heavy inputs from domestic and industrial waste, including poorly managed plastic debris.

The predominant microplastic types included fibers and fragments, with size ranges between 10–500 µm, and polymers such as polypropylene (PP), polyethylene (PE), polystyrene (PS), polyester, and polyamide dominating the composition. These microplastic types are all associated with packaging, textiles, and household waste, which highlights the contribution of poor plastic waste management practices in Lagos to aquatic pollution. The trend of microplastic abundance in the locations follows the pattern: Makoko > Ikorodu > Epe, in order of decreasing abundance. The microplastic counts of *O. niloticus* and *C. gariepinus* across these locations are shown in Figure 4.24.

In March, which marks the late dry season in Lagos, the highest concentrations of microplastics were recorded in both fish species. Fish collected from Makoko contained the greatest number of microplastic particles per individual, followed by samples from Ikorodu and Epe. This pattern

aligns with the period's limited water flow and low dilution capacity, allowing pollutants like plastics and their fragments to accumulate in the water and sediment[27]. The lack of rainfall means less flushing of waste, while high human activity in urban areas contributes to persistent inputs of plastic debris. Similar seasonal peaks were observed in a study of the Yangtze River in China, where microplastic abundance in aquatic organisms was highest during low-flow, high-residency periods[28].

By May, with the onset of the rainy season, there was a modest decrease in microplastic levels in fish tissues. This can be attributed to increased rainfall and runoff, which may initially mobilize more plastic particles from land but also enhance water turnover and dilution in the lagoon system. However, levels remained significant, especially in *Clarias gariepinus*, likely due to its benthic feeding habits, which increase exposure to sediment-bound microplastics[29]. Studies from Ghana's urban lagoons have also reported similar persistence of microplastics in benthic feeders during early rainy seasons, despite water column dilution[30].

In August, typically the peak of the rainy season, microplastic concentrations were at their lowest across all study areas. This period is characterized by high-intensity rainfall, water turbulence, and stronger hydrodynamic flushing, which may transport microplastics out of the lagoons into adjacent estuarine or marine systems[31]. Additionally, sediment burial and water mixing during storms can temporarily reduce the bioavailability of microplastic particles. This trend is consistent with findings by an investigation, which observed the lowest levels of microplastic ingestion in estuarine fish during peak flood events in Southeast Asia[32].

In November, as the rains began to subside, microplastic concentrations increased again, though not to the levels observed in March. This post-rainy period is characterized by reduced water

volumes and slower flow rates, creating conditions that allow previously buried microplastics in sediments to become resuspended[33]. The rise in microplastic levels may also reflect the re-concentration of particles due to lower dilution. Notably, fish from Makoko and Ikorodu again showed higher contamination, indicating continued influence of urban runoff and poor waste management. Similar post-rain increases have been reported in studies from Brazil's coastal lagoons, where particle re-entry into the food web was associated with sediment disturbance and lower water renewal rates[34].

A comparative assessment of the results shows that *Clarias gariepinus* had higher MP concentration than *Oreochromis niloticus* in most cycles. This may be as a result of the ecological niches which they both occupy. *Clarias gariepinus* is a benthic fish which makes it more exposed to MPs accumulated in sediments, while *Oreochromis niloticus* is a pelagic feeder and interacts mostly with MPs in surface water.

Coincidentally, August and November had peak levels of TDS which suggests a link between suspended materials and microplastic availability in the water column. Fiber-shaped microplastics were predominant in *O. niloticus* samples, particularly in Makoko and Epe, and were mostly identified as polyester or polyethylene terephthalate (PET). This points to textile effluent and synthetic clothing wash-off as key sources. In contrast, *C. gariepinus* showed more plastic fragments, suggesting ingestion of degraded packaging or solid waste materials that settle in sediments. These findings are consistent with prior research, which reported similar spatial and seasonal trends in Lagos waters, and also emphasized the co-occurrence of microplastics and heavy metals in Nigerian coastal ecosystems[35,36].

#### 4.2.4.1 Seasonal Variation of Microplastic Occurrence in Makoko

In *O. niloticus*, microplastic counts increased significantly from 15.20±3.47 in March to 22.30±6.37 in November ($p < 0.05$, ANOVA). *C. gariepinus* showed slightly higher baseline microplastic counts (16.10±3.36 in March), but exhibited a slower rate of increase, ending at 19.40±5.37 in November. A study also reported seasonal variations in microplastic ingestion of *O. niloticus* and *C. gariepinus*, with higher frequencies during wet seasons[37]. This aligns with the findings of the present study as November marks the end of the rainy season, and the high microplastic counts in both species were recorded in this period.

Although the actual plastic load at each sampling site was not quantified, the result provides insight into the relative abundance of plastic materials at different parts of the Lagos Lagoon during the rainy season. High microplastic ingestion by fish is associated with the abundance of plastics on the surface of water bodies, which most probably results from rain-initiated runoffs that pull plastics from nearby terrestrial systems into the water bodies. Therefore, the more plastic waste is released into a terrestrial system, the more the likelihood of microplastic ingestion by nearby aquatic animals, especially during the rainy season. Hence, locations that reported high accumulation of microplastics in their inhabiting fish most likely generate more plastic waste. Coupled with the densely populated shoreline communities of Makoko, where domestic wastes are directly discharged into nearby water channels that link to the water bodies, the microplastic count in this area is understandably high[38]. Furthermore, this area is characterized by high boating and fishing activity, which contributes synthetic fibers and gear residues.

Furthermore, the pairwise *t*-test revealed significant differences between species in March ($p < 0.05$) and November ($p = 0.05$), indicating seasonal accumulation is more pronounced in *O.*

*niloticus*, possibly due to feeding behaviour and vertical habitat use. That is, *O. niloticus* is more likely to be exposed to microplastics because it tends to feed or spend more time in water layers where microplastics are more concentrated, likely the surface or upper layers, where floating plastic particles tend to accumulate. On the other hand, *C. gariepinus* is typically a bottom-dweller and might encounter fewer microplastics if those are less abundant in deeper or benthic zones. This finding is supported by a report, which discovered higher microplastic occurrence in *O. niloticus* compared to *C. gariepinus* in a Nile canal[39].

Dominant microplastic types in Makoko included fibers (March–August), primarily polyester, polyethylene, and polyethylene terephthalate (PET) in *O. niloticus*, and fragments in *C. gariepinus*, especially in May and August. Notably, these MPs were frequently found in the gills and digestive tracts, suggesting both inhalation and ingestion pathways. Seasonal increase in fiber presence correlated with elevated textile activity and rainfall-driven runoff, especially in August and November.

#### 4.2.4.2 Seasonal Variation of Microplastic Contamination in Ikorodu

Microplastic counts in *O. niloticus* rose from 14.60±3.02 in March to 20.60±5.14 in November, while *C. gariepinus* showed a similar trend (14.90±2.80 to 19.30±5.03). Despite higher counts in *O. niloticus*, differences were not statistically significant across most months (*t*-test $p > 0.05$), except for a marginal difference in November ($p = 0.05$). This may be as a result of less abundant plastic materials with passing time, when compared to that of Makoko; *O. niloticus* in Makoko had higher counts of microplastics across the seasons.

Fragment types were dominant in *O. niloticus* (March, August, November), composed of polyester, polystyrene (PS), and polyvinyl chloride (PVC), particularly accumulating in the gills.

In contrast, *C. gariepinus* showed a higher frequency of fibers during the dry seasons (May, November), especially from textile-related polymers like PE and polyester, hinting at anthropogenic textile effluent inputs from local industries. It is also noteworthy that lower MP counts in May correlated with relatively reduced rainfall and runoff events.

**4.2.4.3 Seasonal Variation of Microplastic Contamination in Epe**

Epe recorded the lowest MP counts in March, with *O. niloticus* at 12.70±3.24 and *C. gariepinus* at 14.60±3.69, but these values rose sharply by November to 20.40±4.81 and 18.50±4.70, respectively. With respect to the above discussions, Epe appears to be the least polluted among the three locations. This sharp rise in microplastic counts in November could be due to tides of plastic material washed down from faraway sources, initiated by heavy downpours. Consequently, the fish assimilate the microplastics from these plastics, thus elevating their MP counts.

While interspecies differences were not statistically significant across months, *O. niloticus* consistently accumulated more microplastics than *C. gariepinus* in August and November, which is likely due to their feeding behavior as mentioned earlier. Fibers were dominant in *O. niloticus* throughout the year, especially in November, when polypropylene (PP) and polystyrene (PS) were frequently observed. In *C. gariepinus*, fragments (especially PS and polyester) were more common, with notable deposition in the digestive tract and gills. The smaller microplastic size ranges observed in Epe (as low as 10 µm) indicate higher microplastic fragmentation in this region, likely a consequence of prolonged photodegradation in shallower waters.

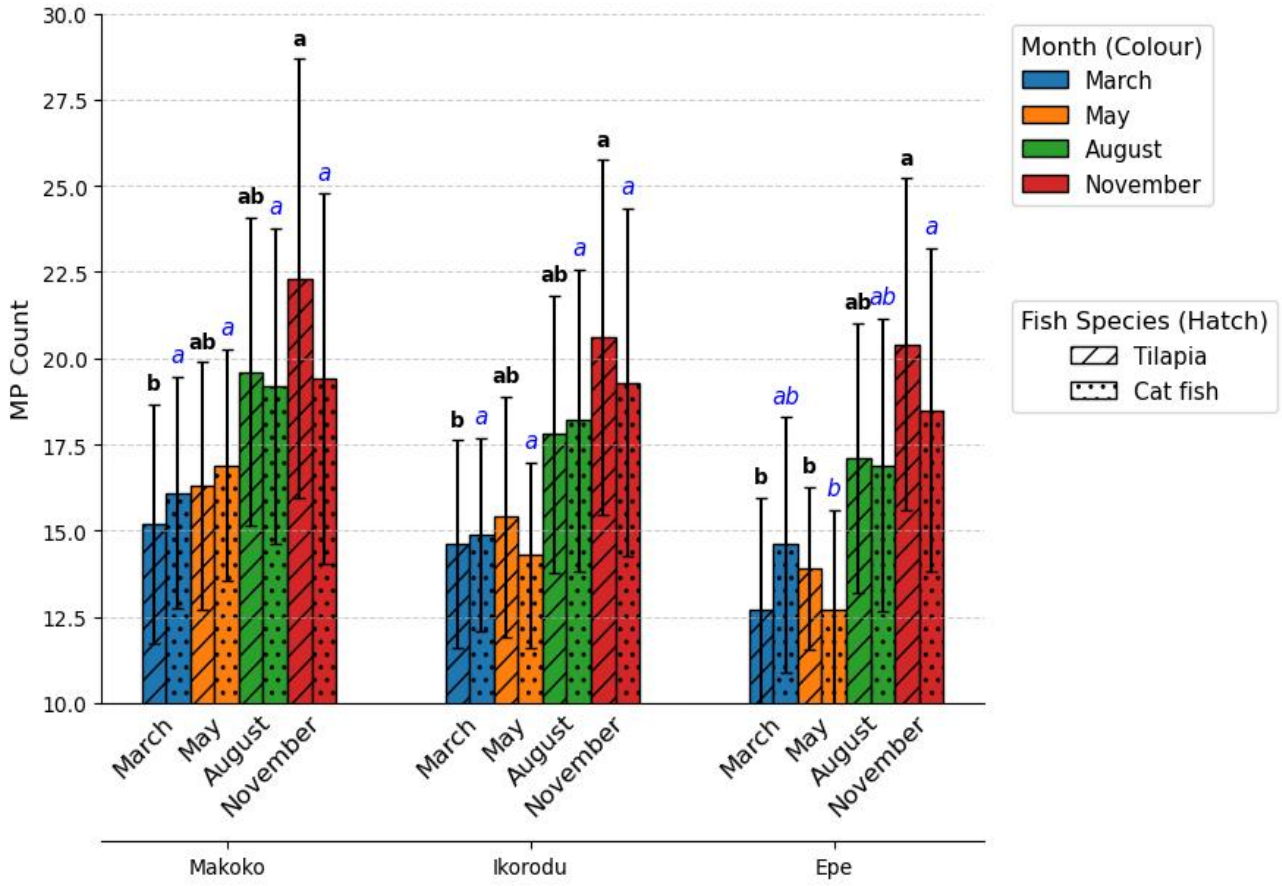


**Figure 4.24:** Seasonal Variation in Microplastic Count in *Oreochromis niloticus* and *Clarias gariepinus* Fishes from the Lagos (Makoko and Ikorodu) and Epe Lagoons

**Source:** Author's Field Work, 2025

### 4.2.5 Discussion on Phthalate Esters Contamination in *Oreochromis niloticus* and *Clarias gariepinus* Fishes from Lagos (Makoko and Ikorodu Axis) and Epe Lagoons

Diethyl phthalate (DEP), Dibutyl phthalate (DBP) and Di-2ethylhexyl phthalate (DEHP) were identified in the tissues of *Oreochromis niloticus* and *Clarias gariepinus* fishes across the three sample locations.

During the dry season in March, the highest concentrations of all three phthalates were recorded across the three sampling sites. In Makoko, DEP reached 22.0±3.24 mg/kg, DBP was 18.5±2.96 mg/kg, and DEHP peaked at 15.3±2.65 mg/kg. Similar trends were observed in Ikorodu and Epe, though at slightly lower concentrations. The elevated levels are likely due to minimal rainfall and dilution, leading to the accumulation of phthalates from untreated domestic sewage, plastic debris, and industrial effluents. The dry conditions promote reduced water flow, resulting in a higher residence time of contaminants in the lagoon systems. Additionally, increased human activity during this period, especially around urban hubs like Makoko, may contribute to greater input of plastic waste and associated chemicals.

In May, with the onset of the rainy season, a general decline in phthalate concentrations was observed. DEP levels dropped to 17.1±2.82 mg/kg in Makoko, DBP to 14.2±2.37 mg/kg, and DEHP to 11.4±2.01 mg/kg. Similar reductions were noted in Ikorodu and Epe. The declining trend is attributed to dilution effects resulting from increased rainfall, which lowers contaminant concentrations in the water and reduces bioaccumulation in aquatic organisms. However, the presence of measurable levels still suggests continued influx of phthalate-laden runoff from land-based sources. DBP and DEHP, being more hydrophobic than DEP, may also partition more into sediments or organic matter, affecting their immediate bioavailability during early rains.

August marked the peak of the rainy season and corresponded with the lowest concentrations of all three phthalates across the study areas. In Makoko, DEP reduced further to 13.6±2.11 mg/kg, DBP to 10.3±1.84 mg/kg, and DEHP to 8.5±1.67 mg/kg. Ikorodu and Epe showed similar declines. This sharp reduction is due to intense rainfall and frequent flushing of the lagoon systems, which likely transport suspended pollutants into adjoining water bodies or coastal zones. In addition, stormwater may resuspend sediment-bound phthalates, but rapid water exchange and sediment burial reduce prolonged exposure to fish. The significant drop in DEHP, which has a higher molecular weight and stronger hydrophobicity, supports the hypothesis that it tends to sorb more to sediments and be less bioavailable in high-flow conditions.

With the cessation of rains in November, phthalate concentrations began to rise again across all sites. Makoko recorded 19.5±2.89 mg/kg for DEP, 16.8±2.64 mg/kg for DBP, and 13.2±2.35 mg/kg for DEHP. This pattern was similarly observed in Ikorodu and Epe. The post-rainy season is marked by lower water levels and slower flow, promoting the reconcentration of pollutants and re-exposure of aquatic organisms to contaminants. The resuspension of sediments and organic matter, possibly due to wind and tidal activity, may also contribute to elevated levels of DBP and DEHP. The increase in DEHP concentration at this time is particularly noteworthy, as it suggests seasonal remobilization of sediment-bound pollutants, increasing bioavailability to bottom-dwelling and omnivorous species such as *Clarias gariepinus*.

Across all four cycles, a consistent pattern emerged: phthalate ester concentrations peaked in the dry season (March), declined during the rainy season (May and August), and increased again post-rain (November). This seasonal cycle highlights the influence of water dynamics on phthalate transport, dilution, and partitioning. DEP, being more water-soluble, showed relatively

higher concentrations throughout, while DBP and DEHP, with greater hydrophobicity, displayed sharper fluctuations likely driven by sediment interactions and stormwater events.

Furthermore, Makoko consistently exhibited the highest concentrations of all three phthalates, followed by Ikorodu and then Epe. This spatial trend reflects the intensity of urbanization, population density, and waste discharge in each location. Makoko's informal settlements and poor waste infrastructure likely contribute significantly to environmental loading of phthalate esters.

The elevated concentrations of DBP and DEHP in *Clarias gariepinus*, especially during the March and November cycles, may be linked to its benthic feeding habits, making it more prone to ingesting sediment-bound contaminants. *Oreochromis niloticus*, a more pelagic feeder, showed lower phthalate accumulation in most cases, supporting the role of ecological behavior in exposure levels. Due to their lipophilic nature, PAEs can accumulate in the fatty tissues of aquatic species, with higher concentrations often found in predatory or bottom-dwelling fish such as *Clarias gariepinus*[29].

The concentrations of diethyl phthalate (DEP), dibutyl phthalate (DBP), and di(2-ethylhexyl) phthalate (DEHP) recorded in *Oreochromis niloticus* and *Clarias gariepinus* from Makoko, Ikorodu, and Epe Lagoons were significantly higher than those reported in urban freshwater ecosystems in China. In fish tissues from urban rivers such as the Pearl River, Yangtze River, and Haihe River, average levels reported were 0.65 – 5.1 mg/kg(DEP), 1.2 – 6.8 mg/kg(DBP) and 0.9 – 7.4 mg/kg(DEHP)[40]. Across all three phthalate types, the levels found in this study exceed the upper thresholds reported in Chinese urban rivers. DEP levels in Makoko were more than 4 times higher than peak levels in Chinese studies. DBP levels in Lagos samples exceeded

those in the Pearl and Yangtze Rivers by over 2–3 fold. DEHP, a known endocrine-disrupting compound, reached 15.3 mg/kg in Makoko, more than double the highest values recorded in urban rivers of China. These elevated concentrations highlight the severe contamination of the Lagos Lagoon system, particularly in Makoko and Ikorodu, where poor waste management, plastic pollution, and proximity to industrial and domestic discharges are prevalent. By contrast, although heavily urbanized, Chinese cities have implemented more rigorous wastewater treatment and industrial regulation, which may partly account for the relatively lower phthalate levels.

During the dry season (March), limited rainfall leads to reduced water volume and flow in lagoon systems, which allows contaminants to accumulate due to low dilution capacity and minimal flushing[27]. The higher concentrations of DEP, DBP, and DEHP recorded during this period, especially in Makoko and Ikorodu, are also driven by intense domestic and industrial activities, including open dumping of plastic waste and untreated sewage discharge common in densely populated urban areas[40,41].

Environmental factors such as temperature, pH, and salinity may influence the chemical behavior of phthalates in water. Higher temperatures has been reported to affect chemical degradation rates, while shifts in pH and salinity alter phthalate solubility and partitioning between water and sediments, affecting their mobility and uptake by aquatic organisms[42,43].

#### 4.2.5.1 Seasonal Variation of Diethyl Phthalate (DEP) Contamination in *Oreochromis niloticus* and *Clarias gariepinus* Fishes from the Lagos and Epe Lagoons

In Makoko, *Oreochromis niloticus* exhibited the highest DEP concentration in March (22.00±3.24 mg/kg), with a significant decline ($p < 0.05$) in May and August, followed by a

moderate rebound in November. A similar pattern was observed in *C. gariepinus*, where March (24.00 ±3.58 mg/kg) and November (22.00±3.35 mg/kg) presented significantly higher concentrations than August (17.00±2.91 mg/kg). The peak concentrations of DEP in March, is likely due to increased land runoff from dry season accumulation and initial rains. Significant interspecies differences were observed at all sites and periods, with *C. gariepinus* typically accumulating more DEP. This could be as a result of the fish being a benthic-feeder, as it ingests DEP accumulated in sediments.

Both species in Ikorodu recorded the highest DEP levels in March (24.00 mg/kg in *O. niloticus* and 26.00 mg/kg in *C. gariepinus*), followed by a significant decline through August, with November showing some recovery. More so, *C. gariepinus* showed significantly higher values in November compared to *O. niloticus* (24.00 vs. 21.00 mg/kg, t-test $p < 0.01$). This trend can be viewed in Figure 4.25.

In Epe, the highest DEP levels were also found in March for both species (26.00 mg/kg in *O. niloticus*, 28.00 mg/kg in *C. gariepinus*), which declined steadily through August. November levels (23.00–26.00 mg/kg) were still high but slightly below the March highest values. These variations were statistically significant ($p < 0.05$). The interspecies differences were pronounced in May and November (< 0.01). Epe recorded the lowest overall contamination, especially for DBP and DEP, most likely due to the reduced urban influence compared to Makoko and Ikorodu.

#### 4.2.5.2 Seasonal Variations in Dibutyl Phthalate (DBP) Contamination in *Oreochromis niloticus* and *Clarias gariepinus* Fishes from the Lagos and Epe Lagoons

In Makoko, *O. niloticus* showed significantly higher DBP concentrations in March (45.00±5.37 mg/kg), which declined through August and slightly increased by November. *C. gariepinus*

followed the same trend, peaking at 50.00±5.70 mg/kg in March and rebounding to 46.00±5.48 mg/kg by November. All seasonal differences were significant ($p < 0.05$), with *C. gariepinus* consistently more contaminated ($p \leq 0.012$).

Ikorodu recorded the highest DBP concentrations for both species among all locations. *O. niloticus* reached 50.00 mg/kg in March, while *C. gariepinus* peaked at 55.00 mg/kg. A similar decreasing pattern was observed through August (34.00–38.00 mg/kg), followed by partial recovery in November. Seasonal changes were significant, and vertical differences were also statistically significant ($p \leq 0.012$). These patterns can be observed in Figure 4.26.

Lower DBP levels were observed at Epe. *O. niloticus*, recording the highest value (35.00 mg/kg) in March) which dropped to 20.00 mg/kg in August, and then increased in November (32.00 mg/kg). *C. gariepinus* followed a similar trajectory but consistently had higher concentrations. Significant seasonal variations and vertical differences (May–November, $p < 0.05$) were observed.

#### 4.2.5.3 Seasonal Variations in Di(2-ethylhexyl) Phthalate (DEHP) Contamination in *Oreochromis niloticus* and *Clarias gariepinus* Fishes from the Lagos and Epe Lagoons

Di(2-ethylhexyl) Phthalate (DEHP) concentrations were generally lower than DEP and DBP, likely due to its reduced solubility and different partitioning behavior in aquatic systems. Nevertheless, its consistent detection across sites and seasons underscores its persistence. In Makoko, *O. niloticus* showed DEHP levels peaking in March (14.00±2.24 mg/kg), with significant decreases in May and August, and a slight rebound in November. *C. gariepinus* displayed higher concentrations in all months, reaching highest at 15.00±2.35 mg/kg in March.

While seasonal differences were less dramatic than for DEP and DBP, the superscripts indicate some statistical significance (e.g., March–August). However, t-tests did not reveal significant interspecies differences ($p > 0.1$), suggesting more uniform distribution (Figure 4.27).

Values were slightly higher in Ikorodu than in Makoko, with *O. niloticus* ranging from 10.50 to 15.00 mg/kg and *C. gariepinus* from 12.50 to 16.50 mg/kg. The pattern followed that of other sites—highest in March, lowest in August, with partial recovery by November. ANOVA revealed significant seasonal variation, but vertical differences remained statistically nonsignificant ($p > 0.1$).

In Epe, DEHP levels were the highest among all sites. *O. niloticus* ranged from 11.00 to 16.00 mg/kg and *C. gariepinus* from 13.00 to 17.50 mg/kg. Like other sites, values dropped significantly during the rainy season (May–August) before increasing again in November. However, variations between species were not statistically significant.

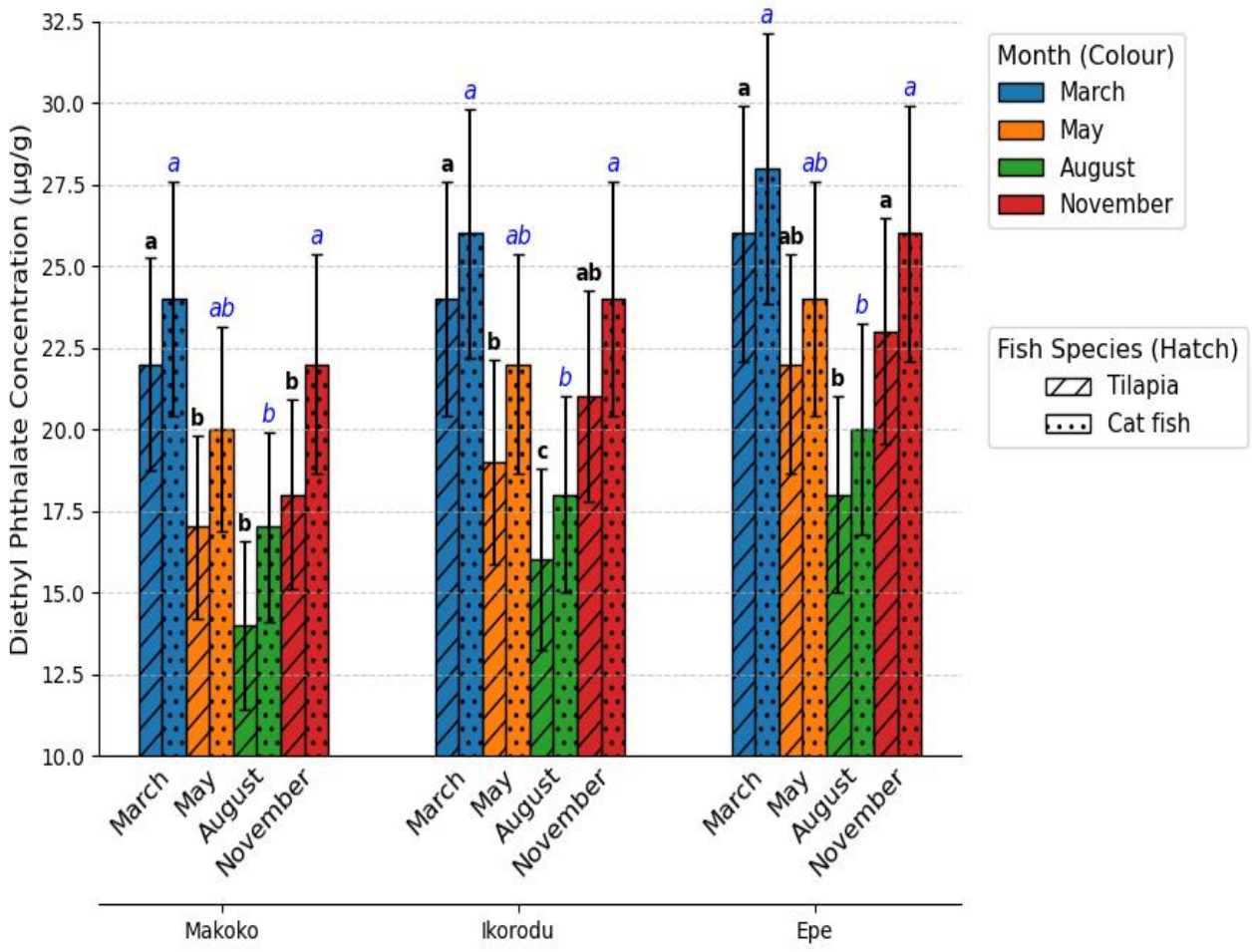


**Figure 4.25:** Seasonal Variation in Diethyl Phthalate Concentration in *Oreochromis niloticus* and *Clarias gariepinus* from the Lagos (Makoko and Ikorodu Axis) and Epe Lagoons

**Source:** Author's Field Work, 2025

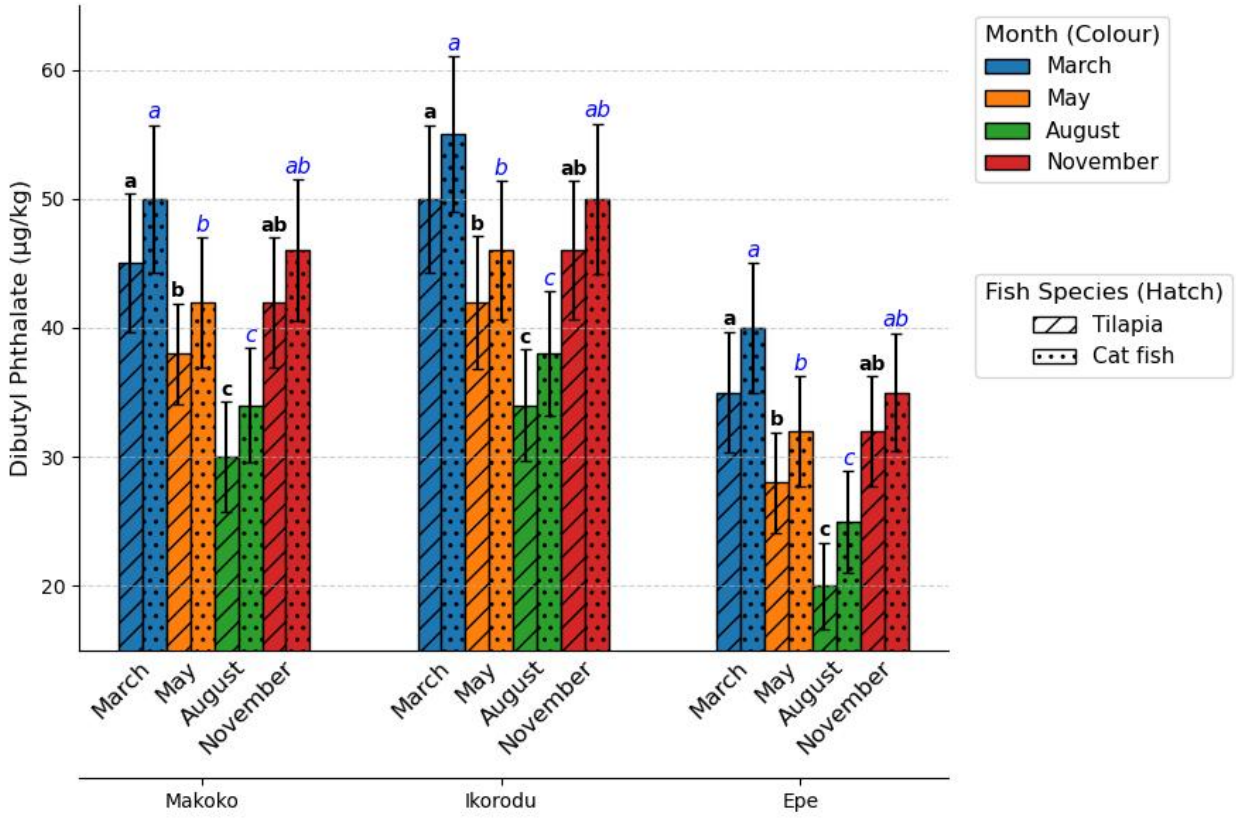


**Figure 4.26:** Seasonal Variation in Dibutyl Phthalate Concentration in *Oreochromis niloticus* and *Clarias gariepinus* from the Lagos (Makoko and Ikorodu Axis) and Epe Lagoons

**Source:** Author's Field Work, 2025

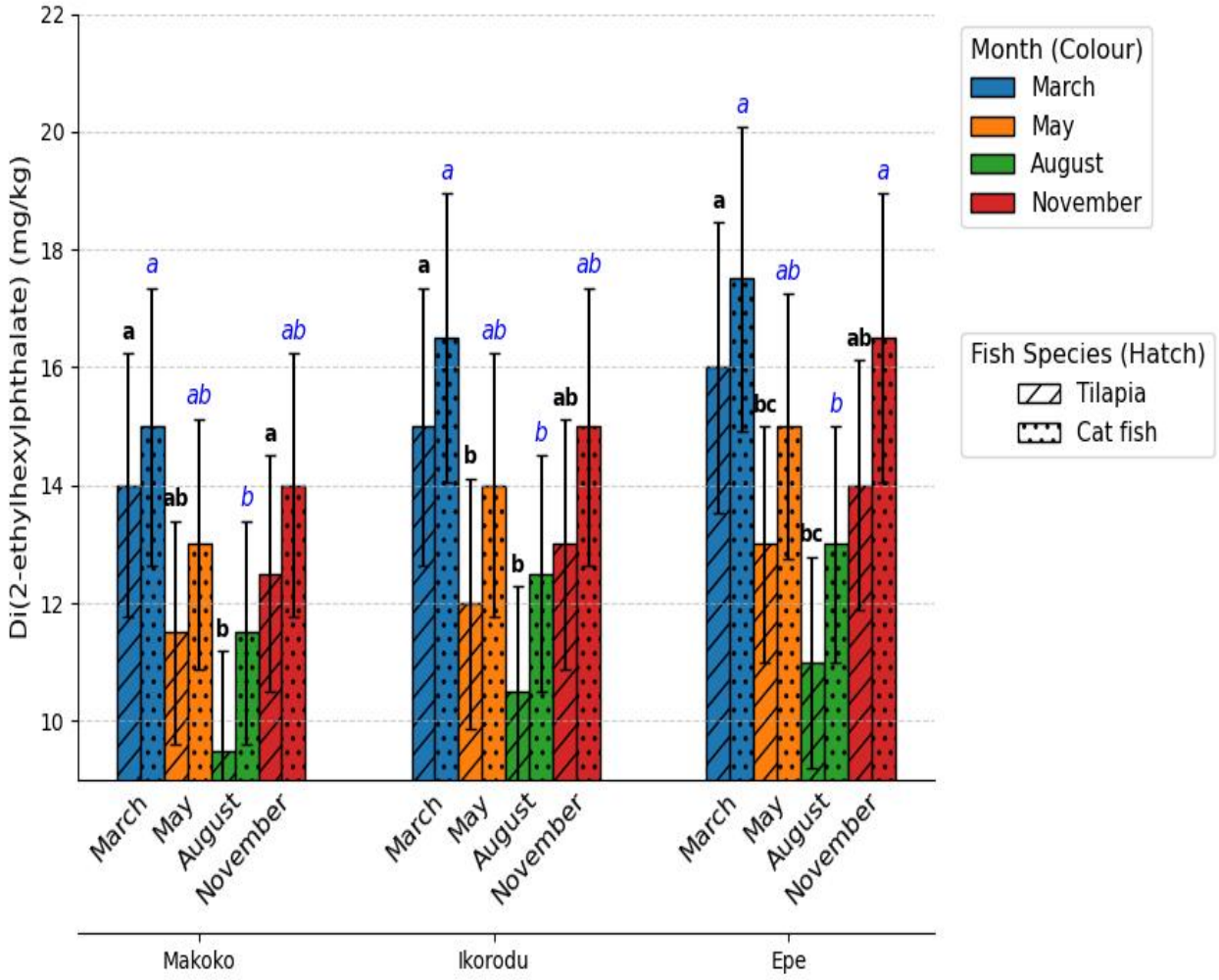


**Figure 4.27:** Seasonal Variation in Di(2-ethylhexylphthalate) Concentration in *Oreochromis niloticus* and *Clarias gariepinus* from the Lagos (Makoko and Ikorodu Axis) and Epe Lagoons

**Source:** Author's Field Work, 2025

**Table 4.24**: Total Phthalate Esters Concentrations in Fish Samples from Lagos and Epe Lagoons

| Cycle | Species | Location | DBP | DEP | DEHP | ΣPAEs |
|---|---|---|---|---|---|---|
| **March** | *Oreochromis niloticus* | Makoko | 45.0 | 22.0 | 14.0 | 81.0 |
| | *Oreochromis niloticus* | Epe | 35.0 | 26.0 | 16.0 | 77.0 |
| | *Oreochromis niloticus* | Ikorodu | 50.0 | 24.0 | 15.0 | 89.0 |
| | *Clarias gariepinus* | Makoko | 50.0 | 24.0 | 15.0 | 89.0 |
| | *Clarias gariepinus* | Epe | 40.0 | 28.0 | 17.5 | 85.5 |
| | *Clarias gariepinus* | Ikorodu | 55.0 | 26.0 | 16.5 | 97.5 |
| **May** | *Oreochromis niloticus* | Makoko | 38.0 | 17.0 | 11.5 | 66.5 |
| | *Oreochromis niloticus* | Epe | 28.0 | 22.0 | 13.0 | 63.0 |
| | *Oreochromis niloticus* | Ikorodu | 42.0 | 19.0 | 12.0 | 73.0 |
| | *Clarias gariepinus* | Makoko | 42.0 | 20.0 | 13.0 | 75.0 |
| | *Clarias gariepinus* | Epe | 32.0 | 24.0 | 15.0 | 71.0 |
| | *Clarias gariepinus* | Ikorodu | 46.0 | 22.0 | 14.0 | 82.0 |
| **August** | *Oreochromis niloticus* | Makoko | 30.0 | 14.0 | 9.5 | 53.5 |
| | *Oreochromis niloticus* | Epe | 20.0 | 18.0 | 11.0 | 49.0 |
| | *Oreochromis niloticus* | Ikorodu | 34.0 | 16.0 | 10.5 | 60.5 |
| | *Clarias gariepinus* | Makoko | 34.0 | 17.0 | 11.5 | 62.5 |
| | *Clarias gariepinus* | Epe | 25.0 | 20.0 | 13.0 | 58.0 |
| | *Clarias gariepinus* | Ikorodu | 38.0 | 18.0 | 12.5 | 68.5 |
| **November** | *Oreochromis niloticus* | Makoko | 42.0 | 18.0 | 12.5 | 72.5 |
| | *Oreochromis niloticus* | Epe | 32.0 | 23.0 | 14.0 | 69.0 |
| | *Oreochromis niloticus* | Ikorodu | 46.0 | 21.0 | 13.0 | 80.0 |
| | *Clarias gariepinus* | Makoko | 46.0 | 22.0 | 14.0 | 82.0 |
| | *Clarias gariepinus* | Epe | 35.0 | 26.0 | 16.5 | 77.5 |
| | *Clarias gariepinus* | Ikorodu | 50.0 | 24.0 | 15.0 | 89.0 |

Values are Mean ± SD; ΣPAEs = sum of the three quantified phthalates.

**Source**: Author's Field Work, 2025

The summed phthalate ester concentrations (ΣPAEs) in *Oreochromis niloticus* and *Clarias gariepinus* across the Lagos and Epe Lagoons ranged from 49.0 mg/kg (*O. niloticus*, Epe, August) to 97.5 mg/kg (*C. gariepinus*, Ikorodu, March). Notably, *C. gariepinus* exhibited consistently higher ΣPAEs than *O. niloticus*, likely influenced by its benthic foraging and greater sediment exposure. ΣPAEs peaked during March (onset of the dry season), declined through May, reached their lowest point during August (peak rainy season), and rose again by November. These trends support a seasonal dilution hypothesis—during the rainy season, increased water flow dilutes contaminant levels, while drier months concentrate pollutants. Location such as Makoko and Ikorodu recorded consistently elevated ΣPAEs relative to Epe Lagoon. This pattern likely reflects intensified phthalate contributions from urban and industrial activities—sources such as plastic manufacturing, domestic waste, and sewage—entering the more developed zones of the Lagos system.

Similar patterns of elevated DBP and DEHP in Nigerian aquatic biota have been documented. For instance, studies in the Ogun River catchments (Ketu, Lagos) reported DEP, DBP, and DEHP concentrations in fish ranging from 320–810 µg/kg, 380–1080 µg/kg, and 40–150 µg/kg, respectively[46]. Likewise, investigations of Epe and Lagos Lagoons revealed DBP as predominant in fish tissues, indicating widespread local phthalate bioaccumulation[47]. In Poyang Lake (China), edible fish ΣPAEs averaged between 119 and 820 µg/kg (wet weight), equating to 0.12–0.82 mg/kg, with DEHP and DBP contributing over 90% of the total. Although these figures are lower than your observations, they affirm a similar contaminant profile dominated by high-molecular-weight phthalates[48]. Phthalates such as DBP and DEHP are well-recognized endocrine disruptors with documented adverse effects across aquatic species—including fish reproductive and developmental toxicity[48]. The significantly elevated ΣPAEs in fish tissues—well into the

tens of mg/kg—highlight potential risks to local human consumers, particularly via dietary exposure.

### 4.2.6 Relationship Between Microplastics Occurrence, Phthalate Ester Contamination, Heavy Metals, Physicochemical Parameters and Morphometric Parameters of *O. niloticus* and *C. gariepinus*

Phthalate-related morphometric effects were influenced by co-occurring heavy metals and environmental parameters. Lead (Pb) and Copper (Cu) displayed consistently positive correlations with head length (HL), pre-anal length (PAL), and contamination factor (CF) across both water types, suggesting possible hypertrophy or compensatory growth responses due to oxidative stress. Nickel (Ni), Cadmium (Cd), and Chromium (Cr) exhibited variable associations, though Cr significantly correlated with total length (TL) ($r = 0.30$, $p < 0.05$) and body depth (BD) ($r = 0.27$, $p < 0.05$) in surface waters, possibly reflecting a role in metabolic disruption. Temperature had strong negative correlations with multiple morphometrics, especially body weight (BW) ($r = -0.47$, $p < 0.01$) and BD ($r = -0.53$, $p < 0.01$), suggesting heat may worsen the impacts of contaminants on fish linear growth. Conversely, Dissolved Oxygen (DO) and Total Dissolved Solids (TDS) were positively correlated with head length (HL) and pre-anal length (PAL), implying that better oxygenation could buffer toxicant effects.

Phthalates such as dibutyl phthalate (DBP), diethyl phthalate (DEP), and di(2-ethylhexyl) phthalate (DEHP) showed generally negative correlations with the morphometric parameters of *C. gariepinus*, suggesting possible endocrine-disrupting or growth-inhibitory effects:

In surface water, dibutyl phthalate (DBP) was negatively correlated with head length (HL) ($r = -0.35$, $p < 0.01$), standard length, and body depth. Diethyl phthalate (DEP) significantly reduced pre-dorsal length ($r = -0.54$, $p < 0.01$), total length ($r = -0.29$, $p < 0.05$), and standard length ($r = -0.29$, $p < 0.05$), while surprisingly showing a *positive* correlation with CF ($r = 0.39$, $p < 0.01$), possibly indicating compensatory mass accumulation or bloating not reflective of healthy growth.

Di(2-ethylhexyl) phthalate (DEHP) was significantly negatively associated with body weight (BW) ($r = -0.29$, $p < 0.05$) and TL ($r = -0.28$, $p < 0.05$), suggesting systemic growth retardation. These results reinforce phthalates' inhibitory impact, especially from sediment-associated exposure routes. The overall trend implies that prolonged exposure to phthalates in both water layers is associated with reduced growth metrics in *Clarias gariepinus*, potentially through hormonal interference or metabolic disruption. This is in support of the findings of a study that Di(2-ethylhexyl) phthalate (DEHP) significantly affects the growth of *C. gariepinus*[31].

Unlike phthalates, many heavy metals exhibited positive and statistically significant correlations with morphometric parameters, particularly in benthic water. Lead (Pb) showed strong positive correlations with total length (TL) ($r = 0.39$, $p < 0.01$, surface; 0.37, $p < 0.01$, benthic) and pre-dorsal length (PDL) ($r = 0.54$, $p < 0.01$, surface; 0.48, $p < 0.01$, benthic), suggesting possible stimulatory or adaptive hypertrophy in response to stress. Copper (Cu) and Nickel (Ni) had consistently positive associations across most metrics: Cu: total length (TL) ($r = 0.38$, $p < 0.01$, surface, 0.44, $p < 0.01$, benthic); pre-dorsal length (PDL) ($r = 0.37$, $p < 0.01$, surface, 0.55, $p < 0.01$, benthic); Ni: total length (TL) ($r = 0.36$, $p < 0.01$, surface, 0.37, $p < 0.01$, benthic); pre-dorsal length (PDL) ($r = 0.42$, $p < 0.01$, surface, 0.65, $p < 0.01$, benthic); Cadmium (Cd): PDL ($r = 0.55$, $p < 0.01$, surface, 0.53, $p < 0.01$, benthic) and total length (TL) ($r = 0.40$, $p < 0.01$, both surface and benthic), showing potentially dose-dependent effects.

While these correlations might reflect metal-induced compensatory growth, they may also indicate bioaccumulation-linked hypertrophy, which should not be misinterpreted as indicative of healthy development. A study also highlighted that the complex interplay between metals and body growth, as reflected by their correlations, can often be misleading[32]. Therefore, care must

be taken, and robust investigations to better characterize these parameters are required for a proper understanding.

Microplastic count demonstrated generally weak or non-significant correlations with fish morphometry. In surface waters, microplastic count had mild positive correlations with total length (TL), standard length (SL), and pre-dorsal length (PDL) but none were statistically significant. In benthic waters, microplastic count was positively correlated with head length (HL) ($r = 0.28$, $p < 0.05$) and standard length (SL) ($r = 0.21$), suggesting minimal or variable influence. These findings indicate that while microplastics may be ingested, they might not exert as pronounced morphometric impacts as phthalates or metals in *C. gariepinus* within the studied exposure scope.

Several water quality variables were significantly associated with morphometry, suggesting that ambient water conditions play a regulatory role in fish development. Dissolved Oxygen (DO) and Total Dissolved Solids (TDS) exhibited consistently positive correlations with total length (TL), pre-dorsal length (PDL), and pre-anal length (PAL), indicating favourable oxygenation and nutrient conditions enhancing growth. pH and Electrical Conductivity (EC) showed similar positive trends, especially with pre-dorsal length (PDL) ($r = 0.61$, $p < 0.01$, pH surface; 0.59, $p < 0.01$, pH benthic). Temperature was negatively correlated with head length (HL) ($r = -0.33$, $p < 0.05$), body weight (BW) ($r = -0.36$, $p < 0.01$), and SL ($r = -0.31$, $p < 0.05$) in surface waters, suggesting thermal stress leading to suppressed development. These results reiterate the relevance of stable oxygenation, moderate salinity (EC), and optimal temperature ranges in maintaining robust growth in *Clarias gariepinus*, as also highlighted by a study[33].

Contamination factor (CF) showed notable negative correlations with environmental pollutants: DEP ($r = 0.39$, $p < 0.01$) in surface water appeared to increase contamination factor (CF). This is

possibly due to swelling, fat accumulation, or organomegaly rather than genuine health. In contrast, heavy metals such as Pb ($r = -0.43$, $p < 0.01$), Ni ($r = -0.34$, $p < 0.01$), and Cd ($r = -0.28$, $p < 0.05$) were negatively correlated with contamination factor (CF), especially in surface waters, implying reduced condition and fitness under metal stress. Dissolved oxygen (DO) and total dissolved solid (TDS) were also negatively associated with contamination factor (CF) ($r = -0.42$, $p < 0.01$, and $-0.33$, $p < 0.01$), indicating that metabolic stress may lead to imbalances in weight-to-length ratios.

Generally, *C. gariepinus* in Lagos Lagoon exhibited varied morphometric responses to chemical stressors. Phthalates, particularly Diethyl phthalate (DEP) and di(2-ethylhexylphthalate) (DEHP), were strongly associated with suppressed growth, especially in length dimensions and head morphology. Heavy metals showed paradoxically positive correlations, possibly indicating stress-induced hypertrophy or differential metabolic adaptation. Water quality parameters such as dissolved oxygen (DO), pH, and total dissolved solid (TDS) supported growth, whereas elevated temperature and heavy contamination generally impaired condition factor and select morphometric indices. The combined stress from microplastics, phthalates, and metals signals a complex, multilayered ecological burden on aquatic life in Lagos Lagoon.

## Endnotes


1. S. O. Ayoola & M. P. Kuton, *Seasonal Variation in Fish Abundance and Physico-chemical Parameters of Lagos Lagoon, Nigeria*. **Advances in Aquaculture and Fisheries Management**, 5(5), 2017, 1–9.

2. B. Xu, D. Li, K. Wei, X. Zhu, J. Xu & B. Ma, *Allometric Growth Patterns and Ontogenetic Development During Early Larval Stages of Schizothorax waltoni Regan and Percocypris retrodorslis in Southwest China*. **Water**, 15(4), 2023, 824. https://doi.org/10.3390/w15040824

3. S. Lukanov & I. Atanasova, *Correlation Between Individual Body Condition and Seasonal Activity in Buresch's Crested Newt, Triturus ivanbureschi*. **Diversity**, 17(5), 2025, 350. https://doi.org/10.3390/d17050350

4. T. B. Bagenal & F. W. Tesch, *Methods of Assessment of Fish Production in Fresh Waters*. **IBP Handbook No. 3**, 3rd ed., Oxford Blackwell Scientific Publication, London, 1978, pp. 101–136.

5. B. U. Getso, J. M. Abdullahi & I. A. Yola, *Length-Weight Relationship and Condition Factor of Clarias gariepinus and Oreochromis niloticus of Wudil River, Kano, Nigeria*. **Journal of Tropical Agriculture, Food, Environment and Extension**, 16(1), 2017, 1–4.

6. N. E. Okwodu, P. U. Okorie & B. E. B. Nwoke, *Length-Weight Relationship and Condition Factor of African Mud Catfish (Clarias gariepinus (Burchell 1822) and Tilapia (Oreochromis nilotica Linnaeus 1758) in Orashi River of Ogba/Egbema/Ndoni LGA of Rivers State, Nigeria*. **Asian Journal of Fisheries and Aquatic Research**, 16(2), 2022, 16–31. https://doi.org/10.9734/ajfar/2022/v16i230368

7. S. Odunuga, S. Udofia, O. E. Osho & O. Adegun, *Environmental Degradation in the Ikorodu Sub-urban Lagos - Lagoon Coastal Environment, Nigeria*. **Open Environmental Sciences**, 10(1), 2018, 16–33. https://doi.org/10.2174/1876325101810010016

8. M. A. Oyinloye, I. O. Olamiju & O. O. Popoola, *Urban Renewal Strategies in Developing Nations: A Focus on Makoko, Lagos State, Nigeria*. **Journal of Geography and Regional Planning**, 10(8), 2017, 229–241. https://doi.org/10.5897/JGRP2017.0631

9. O. F. Fasuyi, W. A. Yusuf & J. A. Nkwoji, *Impacts of Anthropogenic Stressors on Hydrochemistry, Benthic Macroinvertebrates and Sediment Characteristics of the Lagos Lagoon, Nigeria*. **Journal of Applied Sciences and Environmental Management**, 25(8), 2021, 1349–1356.

10. T. O. Olurin, *Assessment of Naturally Occurring Radionuclides and Heavy Metals Level and Health Risks in Commonly Consumed African Catfish, White Catfish and Nile Tilapia Fish Species From Epe Waterside Region of Lagos, Nigeria*. **Journal of Applied Sciences and Environmental Management**, 28(2), 2024, 449–457.

11. O. C. Bakare, *Length-Weight Relationships and Condition Factor of Parachanna obscura Günther, 1861 in Epe Lagoon, Lagos, Nigeria*. **Pan African Journal of Life Sciences**, 5(2), 2021, 299–305. https://doi.org/10.36108/pajols/1202.50.0280

12. O. A. Olopade, I. O. Taiwo & A. E. Ogunbanwo, *Length-Weight Relationship and Condition Factor of Leuciscus niloticus (De Joannis, 1853) from Epe Lagoon, Lagos State, Nigeria*. **Ege Journal of Fisheries and Aquatic Sciences**, 32(3), 2015, 165–168. https://doi.org/10.12714/egejfas.2015.32.3.07

13. C. J. O. Anarado, E. Ekolie, C. G. Iziga, C. E. Anarado & A. N. Eboatu, *A Comparative Study of the Nutritional Composition, Fatty Acid Profiles and Organoleptic Evaluation of Clarias gariepinus (African catfish) and Oreochromis nilotius (Nile tilapia) Raised Under Wild and Pond-Cultured Systems*. **European Journal of Nutrition & Food Safety**, 15(9), 2023, 118–126. https://doi.org/10.9734/ejnfs/2023/v15i91340

14. H. T. Nguyen, T. T. Pham, & L. H. Tran, *Seasonal Changes in Water Quality and Their Impacts on Aquatic Ecosystems in Urban River Systems*. **Environmental Monitoring and Assessment**, 194(3), 2022, 185.

15. F. Galgani, G. Hanke, S. Werner, & L. De Vrees, *Marine Litter Within the European Marine Strategy Framework Directive*. **ICES Journal of Marine Science**, 78(1), 2021, 156–165. https://doi.org/10.1093/icesjms/fsaa200

16. Y. Zhou, C. Wu, & P. Xie, *Hypoxia in Freshwater Ecosystems: Causes, Consequences, and Management Strategies*. **Water Research**, 185, 2020, 116231

17. M. Kumar, R. Z. Gaur, & S. K. Sharma, *Water Quality and Human Health: Impact of TDS and Conductivity in Drinking Water*. **Environmental Research**, 185, 2020, 109348.

18. A. A. Koelmans, E. Besseling, & W. J. Shim, *Nanoplastics in the Aquatic Environment. Critical Review*. **Marine Anthropogenic Litter**, 2020, 325–340. https://doi.org/10.1007/978-3-319-16510-3_12

19. W. Wang, J. Ge, & X. Yu, *Bioavailability and Toxicity of Microplastics to Aquatic Organisms: A Review*. **Ecotoxicology and Environmental Safety**, 209, 2021, 111885.

20. E. R. Ciscato, T. R. R. Bontognali & D. Vance, *Nickel and its Isotopes in Organic-Rich Sediments: Implications for Oceanic Budgets and a Potential Record of Ancient Seawater*. **Earth and Planetary Science Letters**, 494, 2018, 239–250. https://doi.org/10.1016/j.epsl.2018.04.061

21. P. Böning, T. Shaw, K. Pahnke & H. J. Brumsack, *Nickel as Indicator of Fresh Organic Matter in Upwelling Sediments*. **Geochimica et Cosmochimica Acta**, 162, 2015, 99–108. https://doi.org/10.1016/j.gca.2015.04.027

22. W. K. Lenstra, N. A. G. M. van Helmond, O. M. Żygadłowska, R. van Zummeren, R. Witbaard & C. P. Slomp, *Sediments as a Source of Iron, Manganese, Cobalt and Nickel to Continental Shelf Waters (Louisiana, Gulf of Mexico)*. **Frontiers in Marine Science**, 9, 2022, 811953. https://doi.org/10.3389/fmars.2022.811953

23. G. O. Mekuleyi, M. A. Anetekhai, O. J. Aderinola, & A. Adu, *Environmental Health Status of Some Aquatic Ecosystems in Badagry Division, Lagos, Nigeria*. **International Journal of Ecotoxicology and Ecobiology**, 4(4), 2019, 93–102. https://doi.org/10.11648/j.ijee.20190404.13

24. I. A. Ladigbolu, K. J. Balogun, & E. D. Olaji, *Hydrochemistry of a Tropical Harbor: Influence of Industrial and Municipal Inputs*. **Journal of Applied Sciences and Environmental Management**, 15(4), 2011, 575–581

25. A. O. Oyeyiola, C. M. Davidson, K. O. Olayinka, & B. I. Alo, *Fractionation and Ecotoxicological Implication of Potentially Toxic Metals in Sediments of Three Urban Rivers and the Lagos Lagoon, Nigeria, West Africa*. **Environmental Monitoring and Assessment**, 186, 2014, 7321–7333. https://doi.org/10.1007/s10661-014-3929-9

26. K. J. Balogun & E. K. Ajani, *Assessment of Zooplankton Community in an Anthropogenic-Disturbance Coastal Creek, Southwest Nigeria*. **Archives of Agriculture and Environmental Science**, 6(2), 2021, 160–169. https://doi.org/10.26832/24566632.2021.060207

27. I. B. Ogundele, A. Adebayo, & S. K. Ojo, *Microplastic Pollution in Nigerian Aquatic Environments: A Review.* **Environmental Science and Pollution Research**, 28(9), 2021, 11544–11557.

28. X. Zhang, X. Kang, X. Liu, & J. Li, *Microplastics in River Sediments: Patterns and Pathways*. **Water Research**, 198, 2021, 117104.

29. Barboza, L. G. A., A. D. Vethaak, B. R. B. O. Lavorante, A. K. Lundebye,& L. Guilhermino, *Microplastics in Marine Organisms and Human Health Impacts*. **Marine Pollution Bulletin**, 133, 2020, 336–348.

30. J. Osei, E. M. Boakye, & P. K. Ofori-Danson, *Microplastic Contamination in Urban Lagoons of Accra*. **African Journal of Aquatic Science**, 47(1), 2022, 15–26.

31. W. Lau, Y. Shiran, R. M. Bailey, E. Cook, M. R. Stuchtey, J. Koskella, C. A. Velis, L. Godfrey, J. Boucher, M. B. Murphy, R. C. Thompson, E. Jankowska, A. T. D. Castillo Castillo, B. Pilditch, L. Dixon, E. Koerselman, E. Kosior, E. Favoino, J. Gutberlet, D. Hogg, M. Linder, A. Mead, & J. R. Jambeck, *Evaluating Scenarios Toward Zero Plastic Pollution.* **Science**, 369(6510), 2020, 1455–1461.

32. J. Chen, J. Li, H. Zhang, H. Shi, G. Peng, K. Li, X. Wang, & J. Ding, *Microplastic Occurrence in Estuarine Fish during Seasonal Rainfall Variation*. **Environmental Pollution**, 273, 2021, 116405.

33. J. C. Prata, J. P. da Costa, I. Lopes, A. C. Duarte, & T. Rocha-Santos, *Environmental Impacts of Microplastics and Their Interactions with Sediments*. **Science of the Total Environment**, 702, 2020, 134455.

34. G. V. B. Ferreira, A. K. S. Justino, L. N. Eduardo, N. Schmidt, J. R. Martins, F. Ménard, V. Fauvelle, M. M. Mincarone, F. Lucena-Frédou, *Influencing Factors for Microplastic Intake in Abundant Deep-sea Lantern Fishes (Myctophidae).* **Science of The Total Environment**, 867, 2023, 161478. DOI: 10.1016/j.scitotenv.2023.161478

35. A. A. Ajiboye, D. A. Olabode, & M. M. Hassan, *Seasonal Distribution of Microplastics in Urban Lagos Waters*. **Nigerian Journal of Environmental Science**, 38(1), 2024, 22–34.

36. O. A. Olowu, O. T. Ayoola, & A. O. Ogunyemi, *Heavy Metal and Microplastic Interactions in Nigerian Coastal Waters*. **Environmental Monitoring and Assessment**, 195(12), 2023, 1074.

37. L. B. Merga, P. E. Redondo-Hasselerharm, P. J. Van den Brink & A. A. Koelmans, *Distribution of Microplastic and Small Macroplastic Particles Across Four Fish Species and Sediment in an African Lake*. **Science of The Total Environment**, 2020, 140527. https://doi.org/10.1016/j.scitotenv.2020.140527

38. O. M. Olarinmoye, F. Stock, N. Scherf, O. Whenu, C. Asenime & S. Ganzallo, *Microplastic Presence in Sediment and Water of a Lagoon Bordering the Urban Agglomeration of Lagos, Southwest Nigeria*. **Geosciences**, 10(12), 2020, 494. https://doi.org/10.3390/geosciences10120494

39. E. Khallaf, A. Alne-na-ei, M. Authman & R. Saqr, *Plastic Pollution in Fish (O. niloticus and C. gariepinus) in a Nile canal, Delta of Egypt*. **Egyptian Journal of Aquatic Biology & Fisheries**, 27(4), 2023, 781–802. https://www.ejabf.journals.ekb.eg/article_300429.html

40. W. K. Yang, L. F. Chiang, S. W. Tan & P. J. Chen, *Environmentally Relevant Concentrations of Di(2-ethylhexyl)Phthalate Exposure Alter Larval Growth and Locomotion in Medaka Fish via Multiple Pathways*. **Science of The Total Environment**, 640–641, 2018, 512–522. https://doi.org/10.1016/j.scitotenv.2018.05.312

41. A. O. Adeogun, O. R. Ibor, M. E. Imiuwa, E. D. Omogbemi, A. V. Chukwuka, R. A. Omiwole & A. Arukwe, *Endocrine Disruptor Responses in African Sharptooth Catfish (Clarias gariepinus) Exposed to Di-(2-ethylhexyl)-Phthalate*. **Comparative Biochemistry and Physiology Part C: Toxicology & Pharmacology**, 213, 2018, 7–18. https://doi.org/10.1016/j.cbpc.2018.07.001

42. A. Keswani, D. M. Oliver, T. Gutierrez & R. S. Quilliam, *Microbial Hitchhikers on Marine Plastic Debris*. **Marine Environmental Research**, 146, 2019, 1–10.

43. D. He, Y. Luo, S. Lu, M. Liu, Y. Song, & L. Lei, *Weathering of Microplastics and Phthalate Migration*. **Sustainability**, 12(20), 2020, 8739.

44. C. D. Pollman & D. M. Axelrad, *Mercury Bioaccumulation Factors and Spurious Correlations*. **Science of The Total Environment**, 496, 2014, vi–xii. https://doi.org/10.1016/j.scitotenv.2014.07.050

45. B. Nchegang, T. M. Enow, G. O. Nkongho & P. V. Tan, *Impact of Water Quality on the Growth Performance of Clarias gariepinus in Fish Farms Within Fako Division, Cameroon*. **Asian Journal of Fisheries and Aquatic Research**, 26(7), 2024, 98–107. https://doi.org/10.9734/ajfar/2024/v26i7787

46. A. O. Adeogun & O. O. Fafioye, *Occurrence and Levels of Phthalate Esters in Water, Sediment and Fish Samples from Ogun River Catchments, Ketu, Lagos, Nigeria*. **Environmental Monitoring and Assessment**, *172*(1–4), 2011, 561–569. https://doi.org/10.1007/s10661-010-1355-2

47. A. S. Aderinto, A. O. Adeogun, & A. V. Chukwuka, *Bioaccumulation of Phthalate Esters in Fish from Epe and Lagos Lagoons, Nigeria*. **Environmental Science and Pollution Research**, *22*(13), 2015, 10085–10093. https://doi.org/10.1007/s11356-015-4226-2

48. X. Chen, Y. Wang, L. Qiu, H. Chen & Y. He, *Occurrence, Distribution, and Ecological Risks of Phthalate Esters in Water and Fish from Poyang Lake*, *China*. **Environmental Sciences Europe**, *34*(14), 1–12. https://doi.org/10.1186/s12302-022-00702-3

# Chapter Five

# Conclusion

## 5.1 Summary of Findings

This research explored the extent of microplastic pollution in *Oreochromis niloticus* and *Clarias gariepinus*, as well as heavy metal pollution in benthic and surface waters across three highly populated areas within the Lagos and Epe Lagoons. Seasonal sampling was conducted in March, May, August, and November. In both species, fish from Epe Lagoon consistently recorded the highest values for both total and standard lengths across all sampling cycles. For instance, *O. niloticus* in Epe reached a total length of 32.5 cm and standard length of 28.3 cm in November, compared to Makoko where the maximum total length was 22.4 cm and standard length was 18.1 cm. Similarly, *C. gariepinus* had a total length of 36.4 cm and standard length of 32.1 cm in Epe in November, while the lowest was from Makoko at 25.6 cm and 21.8 cm respectively. This suggests that the Epe Lagoon provides a more conducive habitat for growth, possibly due to lower pollution levels and better water quality.The head length of both species followed a similar spatial trend, with Epe showing the highest values. In the November cycle, *C. gariepinus* from Epe had a head length of 10.5 cm, whereas in Makoko, the maximum head length was only 7.8 cm. These values indicate a larger feeding apparatus in fish from cleaner, less stressed environments, which may correlate with better foraging opportunities.

*C. gariepinus* in Epe had the highest pre-dorsal and pre-anal lengths of 13.9 cm and 16.0 cm respectively in November, compared to Makoko where maximum values were 10.3 cm (pre-dorsal) and 12.2 cm (pre-anal). These values indicate enhanced physiological development in less degraded habitats.The body depth, an important parameter indicating robustness and muscle mass, was notably higher in fish from Epe Lagoon. *O. niloticus* from Epe recorded a body depth

of 9.6 cm in November, while those from Makoko had a maximum of 6.4 cm. This suggests better physical conditioning and possibly higher energy reserves in Epe fish populations.

There was a significant increase in body weight over time and across locations. *C. gariepinus* exhibited the heaviest weights, especially in Epe, with individuals weighing up to 390 g in November. In contrast, Makoko fish of the same species and period weighed around 245 g. *O. niloticus* followed a similar pattern with a top weight of 310 g in Epe and just 198 g in Makoko. The differences are strongly indicative of environmental influence on biomass accumulation.

The condition factor (K), which assesses the "well-being" of fish based on the relationship between length and weight, varied significantly between species and locations. *O. niloticus* had K values consistently above 3.0 across all sites and seasons, signifying healthy body conditions. Conversely, *C. gariepinus* had relatively lower K values, around 1.0–1.2, indicating a more slender build that is characteristic of the species but also reflective of lower fat storage or nutritional intake, especially in more polluted sites like Makoko.

The analysis of surface and benthic waters across the four seasonal cycles (March, May, August, and November) revealed significant spatial and temporal variations in parameters including pH, temperature, dissolved oxygen (DO), total dissolved solids (TDS), and electrical conductivity (EC).

pH values ranged from 6.4 to 8.9, with surface waters being more alkaline than benthic ones. The highest surface pH was 8.9 at Makoko in May, suggesting high photosynthetic activity, while the lowest was 6.4 in Epe benthic waters, likely due to organic matter decomposition.

Temperature followed a clear vertical gradient, with surface waters warmer than benthic layers. The highest surface temperature was 29.6°C (Makoko, May) and the lowest benthic temperature

was 23.0°C (Epe, November). This stratification affects oxygen solubility and contaminant dynamics. Dissolved Oxygen (DO) was generally higher in surface waters (6.1–7.2 mg/L) than benthic waters (2.3–5.0 mg/L). November recorded the highest DO across both layers, attributed to post-rainy season mixing and lower biological oxygen demand. Total Dissolved Solids (TDS) and Electrical Conductivity (EC) were consistently highest at Makoko, indicating intense pollution from runoff and effluents. In November, Makoko surface waters recorded TDS of 679 mg/L and EC of 1250 µS/cm, while Epe had the lowest (TDS: 654 mg/L; EC: 1110 µS/cm). These findings confirm that Makoko exhibited the most degraded water quality, followed by Ikorodu, with Epe showing the best physicochemical conditions. Seasonal shifts were evident, with August (rainy season) showing the most improvement in DO and reduced TDS, while March (dry season) marked the peak of stratification and pollution concentration.

Five heavy metals—Lead (Pb), Copper (Cu), Nickel (Ni), Cadmium (Cd), and Chromium (Cr)—were assessed across the same sampling cycles and locations. Pb concentrations consistently exceeded the WHO limit of 0.01 mg/L. The highest surface Pb was 3.56 ± 0.22 mg/L (Makoko, November), and the highest benthic Pb was 6.77 ± 0.11 mg/L (Makoko, November). Pollution Index (PI) values were also alarming, with Makoko surface PI at 355.9 and benthic PI as high as 677 (Ikorodu). Cu also showed elevated levels, particularly in the benthic zone. Benthic Cu reached 11.56 ± 0.23 mg/L (Makoko, November), while surface levels ranged from 2.48 to 3.11 mg/L, all above the WHO limit of 0.05 mg/L. Cu PIs exceeded 11 in benthic waters. Ni levels were highest among all metals, with benthic Ni at 14.66 ± 0.49 mg/L (Makoko, November) and surface Ni peaking at 1.10 ± 0.34 mg/L. PI values surpassed 12 in benthic waters, underscoring extreme sediment pollution. Cd was persistently high across all locations and exceeded the WHO guideline of 0.003 mg/L. Benthic Cd peaked at 1.33 ± 0.00 mg/L (Makoko, November), with

surface levels also high, especially in Makoko and Ikorodu. Benthic Cr rose to 5.62 ± 0.01 mg/L (Makoko, November), while surface Cr was lower (e.g., 0.12 ± 0.07 mg/L). Cr PIs in benthic waters highlighted its persistent accumulation in sediments and ecological risk.

Microplastics were detected in all fish samples, with varying concentrations depending on the location, species, and season: Makoko consistently recorded the highest microplastic counts per individual fish, followed by Ikorodu and then Epe, aligning with levels of urbanization and pollution inputs. *Clarias gariepinus* generally had higher microplastic concentrations than *O. niloticus* across most sampling cycles due to its benthic feeding habits, which increase contact with sediment-bound microplastics. March had the highest microplastic count observed in Makoko in *O. niloticus* (MP count: 15.2±3.1) and *C. gariepinus* (MP count: 16.1±3.0). This was due to low water volume and reduced flow which likely caused the accumulation of floating and sedimented microplastics. In May, there was a slight decline in MP levels due to increase in runoff and dilution. However, *C. gariepinus* still had microplastic levels of 16.9±3.0 in Makoko. August recorded the lowest microplastic count due to intense rainfall and water flushing, which diluted or exported microplastic out of the lagoons. *O. niloticus* recorded 19.6±4.0 while *C. gariepinus* recorded 19.2±4.1, both in Makoko. In November there was a resurgence of microplastic contamination due to reduced flow and remobilization of sediment-buried plastics, (*O. niloticus*: 22.3 ± 5.7 *C. gariepinus*: 19.4 ± 4.8).

Microplastics were predominantly classified into fibers and fragments ranging from 10–500 µm, with *O. niloticus* exhibiting more fibers, especially in Makoko and Epe. *C. gariepinus* showed more fragments, likely due to sediment feeding and interaction with degraded waste. In *O. niloticus*, microplastic were commonly found in the digestive tract and gills while in *C. gariepinus*, fragments were frequently concentrated in the gills, indicating respiratory exposure

to sediment-suspended MPs. A positive correlation was observed between microplastic levels and total dissolved solids (TDS). Higher TDS often coincided with increased microplastics, suggesting shared sources (e.g., wastewater runoff). Surface waters with more organic activity and plastic debris exhibited increased microplastic loads, particularly in dry and post-rainy periods.

The highest concentrations of all three phthalates were observed in March due to low water levels, minimal dilution, and higher contaminant retention. In *Makoko*, *O. niloticus* showed diethyl phthalate (DEP) (22.0±3.24 mg/kg), dibutyl phthalate (DBP) (18.5±2.96 mg/kg) and di(2-ethylhexylphthalate (DEHP) (15.3±2.65 mg/kg). A general decline in phthalate levels was recorded in May, attributed to rainfall-induced dilution and water flushing. Makoko fish had reduced diethyl phthalate (DEP) (17.1±2.82 mg/kg), dibutyl phthalate (DBP) (14.2±2.37 mg/kg), and di(2-ethylhexylphthalate (DEHP) (11.4±2.01 mg/kg) concentrations. The lowest phthalate levels occurred in August, corresponding with the most intense rainfall and highwater exchange. *O. niloticus* in Makoko had diethyl phthalate (DEP) (13.6±2.11 mg/kg), dibutyl phthalate (DBP) (10.3 ± 1.84 mg/kg) and di(2-ethylhexylphthalate (DEHP) (8.5±1.67 mg/kg). In November, a rebound in contamination was observed, likely due to sediment resuspension and reduced flushing. *C. gariepinus* in Makoko recorded diethyl phthalate (DEP) (22.0±3.0 mg/kg), dibutyl phthalate (DBP) (16.8±2.64 mg/kg) and di(2-ethylhexylphthalate (DEHP) (14.0 ±2.0 mg/kg).

## 5.2 Conclusion

This study identified and confirmed microplastics and phthalate esters in *Oreochromis niloticus* and *Clarias gariepinus*, as well as heavy metals in the surface and benthic waters of Lagos and Epe Lagoons across different seasons and locations. The exhibited morphometric traits of the fishes suggest the need for a healthier aquatic environment. The presence of these pollutants in edible fish tissues signifies a serious risk of bioaccumulation and biomagnification, which can have cascading effects on aquatic food webs and human health. The co-occurrence of microplastics and heavy metals or phthalates further compounds the toxicity risks.

This research underscores the urgent need for policy intervention, environmental monitoring, waste management reforms, and community education to safeguard the integrity of aquatic ecosystems and the health of fish consumers in Lagos and beyond. The observed correlations between pollutants and growth parameters reflect the complex relationship between environmental stress and metabolic responses. These findings emphasize the escalating threat of plastic, phthalate and metal pollution to aquatic biodiversity, food security, and public health in Nigeria's coastal ecosystems.

## 5.3 Recommendations

Based on the findings of this study, the following actions are recommended to mitigate pollution and protect aquatic life and human health:

i. Regulatory agencies such as NESREA and LASEPA should intensify enforcement of laws on waste disposal, effluent discharge, and plastic usage.

ii. Grassroots educational campaigns should be implemented to inform communities about the impacts of plastic pollution and encourage behavioural change.

iii. Government and private sector collaboration is vital to support alternatives to single-use plastics, such as biodegradable packaging.

iv. Long-term monitoring of microplastics and associated pollutants should be institutionalized to track trends and inform policies.

v. Periodic testing of commercially important fish species for pollutants should be conducted to protect consumers.

vi. Investment in modernized solid waste management, particularly in informal and waterfront settlements, is urgently needed.

## 5.4 Contribution to Knowledge

i. This research provides a detailed seasonal and spatial profile of microplastic contamination in Lagos and Epe Lagoons.

ii. It delivers one of the first systematic multi-season datasets on microplastics in Nigerian lagoon ecosystems.

iii. It elucidates the interaction between fish morphometrics and water quality parameters, heavy metals, phthalate esters, and microplastic abundance.

iv. It establishes comparative pollution burdens across three key fishery zones. It has provided novel and region-specific data on microplastic pollution, heavy metal toxicity and phthalate esters accumulation.

v. It informs environmental health and policy by providing empirical evidence relevant to sub-Saharan Africa.

vi. It contributes valuable scientific knowledge to the field of environmental toxicology, aquatic ecology and public health, especially within the context of Nigeria where data is scarce.

## 5.5 Suggested Areas for Further Research

i. **Multi-Matrix Environmental Assessment:** Simultaneously analyze water, sediment, biota, and atmospheric deposition samples to develop a complete contaminant pathway model for Lagos and Epe Lagoons.

ii. **Trophic Transfer and Food Safety Investigations**: Investigate contaminant transfer across the lagoon food web, quantify biomagnification factors, and conduct human dietary exposure risk assessments based on local consumption patterns.

iii. **Broader Contaminant Profiling:** Extend the scope beyond phthalate esters to include other classes of organic pollutants (e.g., polycyclic aromatic hydrocarbons, polychlorinated biphenyls, bisphenol A, per- and polyfluoroalkyl substances) to obtain a more comprehensive picture of aquatic contamination in Nigerian lagoons.

iv. **Geospatial and Modelling Approaches:** Integrate remote sensing, GIS mapping, and hydrodynamic models to visualize pollutant distribution patterns and predict future hotspots under different land-use and climate scenarios.

v. **Community-Based and Policy-Oriented Research:** Incorporate socio-economic surveys to link environmental contamination data with local livelihood impacts, and assess the effectiveness of existing environmental laws and policies.

## Bibliography

### Journal


Adada, O. A., & Adeogun, A. O., *Microplastics in Carnivorous Fish Species, Water and Sediments of a Nigerian Lagoon Ecosystem.* **Environmental Science and Pollution Research**, 30(3), 2023, 2045–2058. https://doi.org/10.1007/s11356-023-26410-w

Adeniyi, O. A., & Yusuf, K. A., *Microplastic Contamination in Lagos Lagoon, Nigeria: TheRole of Urbanization and Waste Management*. **Science of the Total Environment**,685,2019,577583. https://doi.org/10.1016/j.scitotenv.2019.05.303

Adeogun A.O., Ibor O.R., Imiuwa M.E., Omogbemi E.D., Chukwuka A.V., Omiwole R.A., & Arukwe A., *Endocrine Disruptor Responses in African Sharptooth Catfish (Clarias gariepinus) Exposed to Di-(2-ethylhexyl)-Phthalate.* **Comparative Biochemistry and Physiology Part C: Toxicology & Pharmacology**, 213, 2018, 7–18. https://doi.org/10.1016/j.cbpc.2018.07.001.

Adeogun A.O., Ibor O.R., Khan E. A., Chukwuka A.V., Omogbemi E.D., & Arukwe A., *Detection and Occurrence of Microplastics in the Stomach of Commercial Fish Species from a Municipal Water Supply Lake in Southwestern Nigeria*. **Environmental Science and Pollution Research**, 27, 2020, 31035–31045. https://doi.org/10.1007/s11356-020-09031-5.

Adeogun A.O., Ogunola O.S., & Akindele E.O., *Microplastic Contamination in Sediment and Water of a Tropical Lagoon*. **Marine Pollution Bulletin**, 184(2), 2022, 1009–1022.

Afolayan A.O., *Accumulation of Heavy Metals from Battery Waste Topsoil Surface Water, and Garden Grown Maize at Omilende Area, Olodo*, *Nigeria*. **Global Challenges**, 2, 2018, 1–12.

Akagha, C. O., Edegbene, A. O., & Igbehinse, A. E., *Pollution Assessment in Surface Waters of Lagos Lagoon using Biotic Indices*. **Nigerian Journal of Fisheries and Aquatic Sciences**, 17(1), 2019, 37–44.

Akan, J. C., Abdulrahman, F. I., Dimari, G. A., & Ogugbuaja, V. O., *Heavy Metal Contamination in Urban and Peri-urban Aquatic Ecosystems: A Review of Sources, Pathways, and Effects.* **Journal of Environmental Sciences**, 98, 2020, 141–159.

Akintan, O. B., Olusola, J. A., & Azeez, A. M., *Heavy Metals Loads in Soil, Farmlands and Plant Crop at Open Dumpsite*. **Global Journal of Environmental Science and Management**, 5(4), 2019, 449–460.

Alava, J., *Modeling the Bioaccumulation and Biomagnification Potential of Microplastics in a Cetacean Foodweb of the Northeastern Pacific: A Prospective Tool to Assess the Risk Exposure to Plastic Particles*. **Frontiers in Marine Science**, 7, 2020, 566101.

Ali, M. M., Ali, M. L., Rakib, M. R. J., Islam, M. S., Bhuyan, M. S., Senapathi, V., Chung, S. Y., Roy, P. D., Sekar, S., Md, T. I., *Seasonal Behavior and Accumulation of Some Toxic Metals in Commercial Fishes from Kirtankhola Tidal River of Bangladesh – a Health Risk Estimation.* **Chemosphere**, 301, 2022, 134660. doi**:**10.1016/j.chemosphere.2022.134660

Ali, S. S., Elsamahy, T., Koutra, E., Kornaros, M., & El-Naggar, M. E., *Environmental Impact of Bioplastics: A Review*. **Science of The Total Environment**, 806, 2021, 150998.

Ali, S., Naqvi, S. R., Naqvi, M., & Islam, M., *Global Plastic Waste Management: Challenges and Opportunities*. **Marine Pollution Bulletin**, 173, 2021, 112998.

Alimi, O. S., Budarz, J. F., Hernandez, L. M., & Tufenkji, N., *Microplastics and Nanoplastics in Aquatic Environments: Aggregation, Deposition, and Enhanced Contaminant Transport*. **Environmental Science & Technology**, 52(4), 2018, 1704–1724. DOI: 10.1021/acs.est.7b05559

Amaral-Zettler A., Zettler E.R., & Mincer T.J., *Ecology of the Plastisphere*. **Nature Reviews Microbiology**, 18(3), 2020, 139–151.

Anarado C. J. O., Ekolie E., Iziga C. G., Anarado C. E., & Eboatu A. N., *A Comparative Study of the Nutritional Composition, Fatty Acid Profiles and Organoleptic Evaluation of Clarias gariepinus (African catfish) and Oreochromis nilotius (Nile tilapia) Raised Under Wild and Pond-Cultured Systems*. **European Journal of Nutrition & Food Safety**, 15(9), 2023, 118–126. https://doi.org/10.9734/ejnfs/2023/v15i91340.

Andrady A., *Persistence of Plastic Litter in the Oceans*. **Marine Anthropogenic Litter**, 2015, 57–72.

Andrady A.L., *The Plastic in Microplastics: A Review*. **Marine Pollution Bulletin**, 119(1), 2017, 12–22. https://doi.org/10.1016/j.marpolbul.2017.01.082.

Anik, M., Arefin, A., Rahman, M. M., Hossain, K., & Islam, M. N., *Role of Reactive Oxygen Species in Aging and Age-Related Diseases: A Review*. **ACS Applied Biomaterials,** 2022. https://doi.org/10.1021/acsabm.2c00411

Aransiola, S. A., Victor-Ekwebelem, M. O., Ajiboye, A. E., Leh-Togi Zobeashia, S. S., Ijah, U. J. J., & Oyedele, O. J., *Microplastics and Nanoplastics in Soil: Sources, Impacts, and Solutions for Soil Health and Environmental Sustainability*. **Journal of Environmental Quality**, *53*(6), 2024, 1048–1072.

Auta, H. S., Chijioke, E., & Shahu, H., *Distribution and Importance of Microplastics in the Marine Environment: A Review of the Sources, Fate, Effects, and Potential Solutions.* **Environment International**, 102, 2017, 165–176.

Ayoola, S. O., & Kuton, M. P., *Seasonal Variation in Fish Abundance and Physico-chemical Parameters of Lagos Lagoon, Nigeria*. **Advances in Aquaculture and Fisheries Management**, 5(5), 2017, 1–9.

Bacha A., Nabi I., & Zhang L., *Mechanisms and the Engineering Approaches for the Degradation of Microplastics*. **ACS ES&T Engineering**, 1(11), 2021, 1481–1501.

Bagenal, T. B., & Tesch, F. W., *Methods of Assessment of Fish Production in Fresh Waters. IBP Handbook No. 3, 3rd ed*., **Oxford Blackwell Scientific Publication, London**, 1978, pp. 101–136.

Bakare, O. C., *Length-Weight Relationships and Condition Factor of Parachanna obscura Günther, 1861 in Epe Lagoon, Lagos, Nigeria*. **Pan African Journal of Life Sciences**, 5(2), 2021, 299–305. https://doi.org/10.36108/pajols/1202.50.0280

Balogun, K. J., & Ajani, E. K., *Assessment of Zooplankton Community in an Anthropogenic-Disturbance Coastal Creek, Southwest Nigeria*. **Archives of Agriculture and Environmental Science**, 6(2), 2021, 160–169. https://doi.org/10.26832/24566632.2021.060207

Barboza A.M., Vethaak A.D., Lavorante B.R.B., Lundebye A.K., & Guilhermino L., *Marine Microplastic Debris: An Emerging Issue for Food Security, Food Safety, and Human Health*. **Marine Pollution Bulletin**, 133, 2020, 336–348.

Barrett, H., Cheeseman, J., & Hardesty, B. D., *Quantifying Plastics in the Environment: Trends, Challenges, and Future Directions*. **Science of the Total Environment**, 798, 2021, 149249. https://doi.org/10.1016/j.scitotenv.2021.149249

Besseling, E., Redondo-Hasselerharm, P. E., Foekema, E. M., & Koelmans, A. A., *Quantifying Ecological Risks of Aquatic Micro- and Nanoplastic*. **Critical Reviews in Environmental Science and Technology**, 49(1), 2019, 32–80.

Bhuyan, M. S., Bakar, M. A., Akhtar, A., Hossain, M. B., Ali, M. M., & Islam, M. S., *Distribution of Heavy Metals in Surface Sediments of the Buriganga River and Ecological Risk Assessment*. **Chemosphere**, 287, 2022, 132147. https://doi.org/10.1016/j.chemosphere.2021.132147

Bhuyan, S., *Effects of Microplastics on Fish and in Human Health*. **Frontiers in Environmental Science**, 10, 2022, 827289.

Birnie-Gauvin, K., Costantini, D., Cooke, S. J., & Willmore, W. G., *A Comparative and Evolutionary Approach to Oxidative Stress in Fish: A Review*. **Fish and Fisheries**, 18(5), 2017, 928–942.

Böning, P., Shaw, T., Pahnke, K., & Brumsack, H. J., *Nickel as Indicator of Fresh Organic Matter in Upwelling Sediments*. **Geochimica et Cosmochimica Acta**, 162, 2015, 99–108. https://doi.org/10.1016/j.gca.2015.04.027

Bornehag C.G., Carlstedt F., Jönsson B.A., Lindh C.H., Jensen T.K., Bodin A., & Jonsson C., *Prenatal Phthalate Exposures and Anogenital Distance in Swedish Boys*. **Environmental Health Perspectives**, 123(1), 2015, 101-107. doi:10.1289/ehp.1408163

Brander, S. M., Renick, V. C., Foley, M. M., Steele, C., Woo, M., Lusher, A., & Carr, S., *Microplastics Impact on Animal Behavior and Physiology: A Call for Integrated Research on Social-Ecological Systems.* **Environmental Toxicology and Chemistry**, 39(5), 2020, 1117–1130.

Campanale C., Massarelli C., Savino I., Locaputo V., & Uricchio V.F., *A Detailed Review Study on Potential Effects of Microplastics and Additives of Concern on Human Health*. **International Journal of Environmental Research and Public Health**, 17(4), 2020, 1212.

Chen, J., Mubeen, B., Hasnain, A., Iqbal, S., Kamran, M., El-Sabrout, A. M., *Microplastic Occurrence in Estuarine Fish during Seasonal Rainfall Variation.* **Environmental Pollution**, 273, 2021, 116405.

Chen, J., Liu, J., Wei, X., Zhang, P., & Zhuang, G., *How to Build a Microplastics-Free Environment: Strategies for Microplastics Degradation and Plastics Recycling*. **Advanced Science**, 9(6), 2022, 2103764.

Chen, M., Zhang, L., Wang, J., & Zheng, L., *Phthalate Bioaccumulation in Aquatic Food Webs and Human Health Risks*. **Environmental Pollution**, 276, 2021, 116661.

Chowdhury, G. W., Sarker, M. Y., & Rahman, M. S., *Microplastic Contamination in the Marine Environment: A Review of the Global Scenario, Sources, Fate, Impacts and Potential Solutions*. **Environmental Challenges**, 5, 2021, 100312. https://doi.org/10.1016/j.envc.2021.100312

Ciscato, E. R., Bontognali, T. R. R., & Vance, D., *Nickel and its Isotopes in Organic-Rich Sediments: Implications for Oceanic Budgets and a Potential Record of Ancient Seawater*. **Earth and Planetary Science Letters**, 494, 2018, 239–250. https://doi.org/10.1016/j.epsl.2018.04.061

d'Ambrières, W., *Plastics Recycling Worldwide: Current Overview and Desirable Changes*. **Field Actions Science Reports**, 19, 2019, 12–21.

Davidson, K., & Dudas, S. E., *Microplastic Ingestion by Wild and Cultured Manila Clams (Venerupis philippinarum) from Baynes Sound, British Columbia*. **Archives of Environmental Contamination and Toxicology**, 71, 2016, 147–156.

Davidson, T., *Boring Crustaceans Damage Polystyrene Floats Under Docks Polluting Marine Waters with Microplastic*. **Marine Pollution Bulletin**, 64(9), 2012, 1821–1828.

Deng, Y., Zhang, Y., Lemos, B., & Ren, H., *Tissue Accumulation of Microplastics in Mice and Biomarker Responses Suggest Widespread Health Risks of Exposure*. **Scientific Reports**, 7(1), 2017, 46687.

Duan, J., Li, Y., Gao, J., Cao, R., Shang, E., & Zhang, W., *ROS-mediated Photoaging Pathways of Nano- and Micro-plastic Particles Under UV Irradiation*. **Water Research**, 216, 2022, 118320.

Elgarahy A.M., Akhdhar A., & Elwakeel K.Z., *Microplastics Prevalence, Interactions, and Remediation in the Aquatic Environment: A Critical Review*. **Journal of Environmental Chemical Engineering**, 9(5), 2021, 106224.

Fasuyi, O. F., Yusuf, W. A., & Nkwoji, J. A., *Impacts of Anthropogenic Stressors on Hydrochemistry, Benthic Macroinvertebrates and Sediment Characteristics of the Lagos Lagoon, Nigeria*. **Journal of Applied Sciences and Environmental Management**, 25(8), 2021, 1349–1356.

Fatoki, I. S., Bornman, M., & Ncube, E. J., *Phthalates in Coastal Water Environments: Sources, Risks, and Management*. **Marine Pollution Bulletin**, 159, 2020, 111489.

Fendall, L. S., & Sewell, M. A., *Contributing to Marine Pollution by Washing your Face: Microplastics in Facial Cleansers*. **Marine Pollution Bulletin**, 58(8), 2009, 1225–1228.

Ferreira, G. V. B., Justino, A.K.S., Eduardo, L.N., Schmidt, N., Martins, J.R., Ménard, F., Fauvelle, V., Mincarone, M.M., Lucena-Frédou, F., *Influencing Factors for Microplastic Intake in Abundant Deep-sea Lantern Fishes (Myctophidae).* **Science of The Total Environment**, 867, 2023, 161478. DOI: 10.1016/j.scitotenv.2023.161478

Frederiksen, H., Aksglaede, L., Sørensen, K., Skakkebæk, N.E., Juul, A., Andersson, A. M., *Urinary Excretion of Phthalate Metabolites in 129 Healthy Danish Children and Adolescents: Estimation of Daily Phthalate Intake.* **Environmental Research**, 111,2011, 656–663

Galgani, F., Hanke, G., Werner, S., & De Vrees, L., *Marine Litter Within the European Marine Strategy Framework Directive*. **ICES Journal of Marine Science**, 78(1), 2021, 156–165. https://doi.org/10.1093/icesjms/fsaa200

Galloway, T. S., Cole, M., & Lewis, C., *Interactions of Microplastic Debris Throughout the Marine Ecosystem*. **Nature Reviews Earth & Environment**, 2(3), 2017, 105–118. https://doi.org/10.1038/s41559-017-0116

Gao C., Wang Y., Zhang X., & Zhang Q., *Phthalate Esters in Riverine and Estuarine Sediments: Distribution, Risk Assessment, and Management Strategies*. **Science of the Total Environment**, 769, 2021, 145186.

Gao D., Li Z., Wang H., Liang H., *An Overview of Phthalate Acid Ester Pollution in China over the Last Decade: Environmental Occurrence and Human Exposure.* **Science of The Total Environment**, 645, 2018, 1400–1409. doi: 10.1016/j.scitotenv.2018.07.093

Gedik, K., & Eryaşar, A. R., *Microplastic Pollution Profile of Mediterranean Mussels (Mytilus galloprovincialis) Collected along the Turkish Coasts*. **Chemosphere**, 260, 2020, 127570.

Getso B.U., Abdullahi J.M., & Yola I.A., *Length-Weight Relationship and Condition Factor of Clarias gariepinus and Oreochromis niloticus of Wudil River, Kano, Nigeria*. **Journal of Tropical Agriculture, Food, Environment and Extension**, 16(1), 2017, 1–4.

Geyer, R., Jambeck, J. R., & Law, K. L., *Production, Use, and Fate of all Plastics Ever Made*. **Science Advances**, 3(7), 2017, e1700782. https://doi.org/10.1126/sciadv.1700782

Giani, D., Baini, L., Galli, E., Casini, L., & Fossi, M., *Microplastics Occurrence in Edible Fish Species (Mullus barbatus and Merluccius merluccius) Collected in Three Different Geographical Sub-areas of the Mediterranean Sea*. **Marine Pollution Bulletin**, 140, 2019, 129–137. https://doi.org/10.1016/j.marpolbul.2019.01.005

Gupta, S., Singh, R. K., Kumar, A., Kumar, S., & Sinha, R., *Plastic Pollution in the Marine Environment: A Review of its Impacts, Sources, and Mitigation Strategies*. **Journal of Environmental Management**, 302, 2022, 114908. https://doi.org/10.1016/j.jenvman.2021.114908

Hale, R. C., Seeley, M. E., La Guardia, M. J., Mai, L., & Zeng, E. Y., *A Global Perspective on Microplastics*. **Journal of Geophysical Research: Oceans**, 125(1), 2020, e2018JC014719. https://doi.org/10.1029/2018JC014719

He, D., Luo, Y., Lu, S., Liu, M., Song, Y., & Lei, L., *Microplastics in Terrestrial Ecosystems: A Scientometric Analysis*. **Sustainability**, 12(20), 2020, 8739.

He, D., Luo, Y., Lu, S., Liu, M., Song, Y., & Lei, L., *Weathering of Microplastics and Phthalate Migration*. **Sustainability**, 12(20), 2020, 8739.

Horton A.A., Svendsen C., Williams R.J., Spurgeon D.J., & Lahive E., *The Influence of Exposure and Physiology on Microplastic Ingestion by the Freshwater Fish Rutilus rutilus (roach) in the River Thames, UK*. **Environmental Pollution**, 236, 2018, 188–194.

Ibrahim A.T., Odulate D.O., & Olayinka K.O., *Distribution of Heavy Metals in Water and Sediments from Lagos Lagoon, Nigeria*. **Journal of Health and Pollution**, 9(24), 2019, 191211. https://doi.org/10.5696/2156-9614-9.24.191211.

Jang, M., Shim, W. J., Han, G. M., Rani, M., Song, Y. K., & Hong, S. H., *Styrofoam Debris as a Source of Hazardous Additives for Marine Organisms*. **Environmental Science & Technology**, 50(10), 2016, 4951–4960. https://doi.org/10.1021/acs.est.6b02602

Jiang A., Yin L., Li L., Wen X., Luo X., & Wang J., *Microplastics as Vectors of Toxic Chemicals: Insights into Their Mechanisms and Environmental Impacts*. **Journal of Hazardous Materials**, 426, 2022, 128112. https://doi.org/10.1016/j.jhazmat.2021.128112.

Kannan, K., & Krishnamoorthi, V., *A Review of Human Exposure to Microplastics and Insights into Microplastics as Obesogens*. **Frontiers in Endocrinology**, 12, 2021, 724989.

Keswani A., Oliver D.M., Gutierrez T., & Quilliam R.S., *Microbial Hitchhikers on Marine Plastic Debris: Human Exposure Risks at Bathing Waters and Beach Environments*. **Marine Environmental Research**, 146, 2019, 1–10.

Khallaf, E., Alne-na-ei, A., Authman, M., & Saqr, R., *Plastic Pollution in Fish (O. niloticus and C. gariepinus) in a Nile canal, Delta of Egypt*. **Egyptian Journal of Aquatic Biology & Fisheries**, 27(4), 2023, 781–802. https://www.ejabf.journals.ekb.eg/article_300429.html

Kiendrebeogo, M., Ceccato, L., Letten, B., De Girolamo, A., Teychene, B., & De Lary, L., *Treatment of Microplastics in Water by Anodic Oxidation: A Case Study for Polystyrene*. **Environmental Pollution**, 269, 2021, 116168.

Kim, S., Choi, Y., Kim, Y., & Kim, Y., *Endocrine Disrupting Effects of DEHP on Reproduction in Aquatic Organisms*. **Environmental Toxicology and Pharmacology**, 77, 2020, 103356.

Koelmans A.A., Besseling E., & Foekema E.M., *Leaching of Plastic Additives to Marine Organisms.* **Environmental Pollution**, 212, 2016, 238–249. https://doi.org/10.1016/j.envpol.2016.01.012.

Koelmans A.A., Besseling E., & Shim W.J., *Nanoplastics in the Aquatic Environment. Critical Review*. **Marine Anthropogenic Litter**, 2015, 325–340. https://doi.org/10.1007/978-3-319-16510-3_12.

Koelmans A.A., *Microplastics in Freshwaters and Drinking Water: A Review and Assessment of Environmental Exposure*. **Science of the Total Environment**, 566, 2019, 489–498.

Kumar, M., Xiong, X., He, M., Tsang, D. C. W., Gupta, J., Khan, E., & Harrad, S., *Microplastics as Pollutants in Aquatic Ecosystems: Determination, Toxicological Implications, and Remediation*. **Journal of Hazardous Materials**, 403, 2021, 123910. https://doi.org/10.1016/j.jhazmat.2020.123910

Kumar, R. Z., Gaur, S. K., & Sharma, S. K., *Water Quality and Human Health: Impact of TDS and Conductivity in Drinking Water*. **Environmental Research**, 185, 2020, 109348.

Kusemiju, K., *A Study of the Catfish (Clarias lazera) in the Lekki Lagoon, Lagos, Nigeria*. **Journal of Fish Biology**, 5(5), 1973, 473–478.

Ladigbolu, I. A., Balogun, K. J., & Olaji, E. D., *Hydrochemistry of a Tropical Harbor: Influence of Industrial and Municipal Inputs*. **Journal of Applied Sciences and Environmental Management**, 15(4), 2011, 575–581.

Landon-Lane, M., *Corporate Social Responsibility in Marine Plastic Debris Governance.* **Marine Pollution Bulletin**, 127, 2018, 310–319.

Lau, W., *Evaluating Scenarios Toward Zero Plastic Pollution*. ***Science***, 369(6510), 2020, 1455–1461.

Lau, W., Shiran, Y., Bailey, R. M., Cook, E., Stuchtey, M. R., Koskella, J., Velis, C. A., Godfrey, L., Boucher, J., Murphy, M. B., Thompson, R. C., Jankowska, E., Castillo, A. C., Pilditch, T. D., Dixon, B., Koerselman, L., Kosior, E., Favoino, E. J., Gutberlet, J., & Palardy, J. E., *Evaluating Scenarios Toward Zero Plastic Pollution*. **Science**, 369(6510), 2020, 1455–1461.

Lavers, J., & Bond, A. L., *Exceptional and Rapid Accumulation of Anthropogenic Debris on One of the World's Most Remote and Pristine Islands*. **Proceedings of the National Academy of Sciences**, 114(23), 2017, 6052–6055.

Le Guern C., *When The Mermaids Cry: The Great Plastic Tide*. **Accessed January 13, 2024**. https://plasticpollution.org/.

Lebreton, L., Slat, B., Ferrari, F., Sainte-Rose, B., Aitken, J., Marthouse, R., Hajbane, S., Cunsolo, S., Schwarz, A., Levivier, A., Noble, K., Debeljak, P., Maral, H., Schoeneich-Argent, R., Brambini, R., & Reisser, J., *Evidence that the Great Pacific Garbage Patch is Rapidly Accumulating Plastic*. **Scientific Reports**, 8(1), 2018, 4666.

Lee, Y., Cho, J., Sohn, J., & Kim, C., *Health Effects of Microplastic Exposures: Current Issues and Perspectives in South Korea*. **Yonsei Medical Journal**, 64(5), 2023, 301.

Lenstra, W. K., van Helmond, N. A. G. M., Żygadłowska, O. M., van Zummeren, R., Witbaard, R., & Slomp, C. P., *Sediments as a Source of Iron, Manganese, Cobalt and Nickel to Continental Shelf Waters (Louisiana, Gulf of Mexico)*. **Frontiers in Marine Science**, 9, 2022, 811953. https://doi.org/10.3389/fmars.2022.811953

Li Z., Zhu J., Wang T., Huang X., & Wang S., *Is Microplastic an Oxidative Stressor? Evidence from a Meta-analysis on Bivalves*. **Journal of Hazardous Materials**, 423, 2022, 127211. https://doi.org/10.1016/j.jhazmat.2021.127211

Li, I., Liu, H., & Chen, J. P., *Microplastics in Freshwater Systems: A Review on Occurrence, Environmental Effects, and Methods for Microplastics Detection*. **Water Research**, 137, 2018, 362–374.

Li, J., Green, C., Reynolds, A., Shi, H., & Rotchell, J. M., *Microplastics in Mussels Sampled from Coastal Waters and Supermarkets in the United Kingdom*. **Environmental Pollution**, 241, 2018, 35–44. https://doi.org/10.1016/j.envpol.2018.05.038

Li, J., Zhang, K., & Zhang, H., *Adsorption of Antibiotics on Microplastics*. **Environmental Pollution**, 237, 2018, 460–467.

Liu, F., Olesen, K. B., Borregaard, A. R., & Vollertsen, J., *Microplastics in Urban and Highway Stormwater Retention Ponds*. **Science of the Total Environment**, 707, 2020, 136010. https://doi.org/10.1016/j.scitotenv.2019.136010

Liu, S., Liu, Y., He, Y., & Zhao, F., *Inhaled Microplastics Induce Systemic Immune and Metabolic Disruption in Mice*. **Journal of Hazardous Materials**, 408, 2021, 124840.

Lomonaco, T., La Nasa, J., Corti, A., Manco, E., & Ghimenti, S., *Release of Harmful Volatile Organic Compounds (VOCs) from Photo-degraded Plastic Debris: A Neglected Source of Environmental Pollution*. **Journal of Hazardous Materials**, 394, 2020, 122596.

Lukanov, S., & Atanasova, I., *Correlation Between Individual Body Condition and Seasonal Activity in Buresch's Crested Newt, Triturus ivanbureschi*. **Diversity**, 17(5), 2025, 350. https://doi.org/10.3390/d17050350

Luo, H., Zhang, Y., He, D., Li, Y., & Zhao, Y., *Aging of Microplastics Affects Their Surface Properties, Thermal Decomposition, Additives Leaching and Interactions in Simulated Fluids*. **Science of The Total Environment**, 714, 2020, 136862.

Lusher A.L., *Microplastic Sampling: The Effectiveness of Different Techniques in Capturing Microplastics in Environmental Matrices*. **Environmental Pollution**, 227, 2017, 188–197.

Lusher, A.L., Welden, N.A., Sobral, P., & Cole, M., *Sampling, Isolating and Identifying Microplastics Ingested by Fish and Invertebrates.* **Analytical Methods**, 9, 2017, 1346–1360. DOI: **10.1039/C6AY0241**

Mattsson, K., Johnson, E. V., Malmendal, A., Linse, S., Hansson, L. A., & Cedervall, T., *Brain Damage and Behavioural Disorders in Fish Induced by Plastic Nanoparticles Delivered Through the Food Chain*. **Scientific Reports**, 7(1), 2017, 11452.

Mekuleyi, G. O., Anetekhai, M. A., Aderinola, O. J., & Adu, A., *Environmental Health Status of Some Aquatic Ecosystems in Badagry Division, Lagos, Nigeria.* **International Journal of Ecotoxicology and Ecobiology**, 4(4), 2019, 93–102. https://doi.org/10.11648/j.ijee.20190404.13

Mengen, K., Li, Y., Wang, H., Yu, L., & Zhang, W., *Physiological Toxicity and Antioxidant Mechanism of Photoaging Microplastics on Pisum sativum L. Seedlings*. **Toxics**, 11(3), 2023, 242.

Merga, L. B., Redondo-Hasselerharm, P. E., Van den Brink, P. J., & Koelmans, A. A., *Distribution of Microplastic and Small Macroplastic Particles Across Four Fish Species and Sediment in an African Lake*. **Science of The Total Environment**, 2020, 140527. https://doi.org/10.1016/j.scitotenv.2020.140527

Miller, I., Hamann, M., & Kroon, F. J., *Bioaccumulation and Biomagnification of Microplastics in Marine Organisms: A Review and Meta-analysis of Current Data*. **PloS One**, 15(10), 2020, e0240792.

Mukherjee, S., Chatterjee, S., Kumar, M., & Basu, P., *Microplastic Pollution in the Environment and its Implications for Human Health*. **Environmental Science & Pollution Research**, 28(16), 2021, 20544–20556. https://doi.org/10.1007/s11356-021-12778-1

Naji A., Esmaili Z., & Khan F.R., *Plastic Debris and Microplastics Along the Beaches of the Strait of Hormuz, Persian Gulf*. **Marine Pollution Bulletin**, 114(4), 2017, 1057–1062. https://doi.org/10.1016/j.marpolbul.2016.10.001.

Napper, I. E., Bakir, A., Rowland, S. J., & Thompson, R. C., *Characterization, Quantity, and Sorptive Properties of Microplastics Extracted from Cosmetics*. **Marine Pollution Bulletin**, 99(1-2), 2015, 178–185.

Nchegang B., Enow T.M., Nkongho G.O., & Tan P.V., *Impact of Water Quality on the Growth Performance of Clarias gariepinus in Fish Farms Within Fako Division, Cameroon*. **Asian Journal of Fisheries and Aquatic Research**, 26(7), 2024, 98–107. https://doi.org/10.9734/ajfar/2024/v26i7787.

Nguyen, H. T., Pham, T. T., & Tran, L. H., *Seasonal Changes in Water Quality and Their Impacts on Aquatic Ecosystems in Urban River Systems*. **Environmental Monitoring and Assessment**, 194(3), 2022, 185.

Nubi, O. A., Osibote, O. A., & Amuda, O. S., *Microplastic Pollution in Lagos Lagoon: Abundance, Characteristics, and Sources*. **Environmental Monitoring and Assessment**, 192(5), 2020, 315. https://doi.org/10.1007/s10661-020-8225-8

Nwankwo, D. I., *Euglenoids of Some Polluted Storm-water Channels in Lagos, Nigeria*. **Tropical Freshwater Biology**, 4, 1995, 29–39.

Odunuga, S., Udofia, S., Osho, O. E., & Adegun, O., *Environmental Degradation in the Ikorodu Sub-urban Lagos - Lagoon Coastal Environment, Nigeria*. *Open* **Environmental Sciences**, 10(1), 2018, 16–33. https://doi.org/10.2174/1876325101810010016

Ogunbiyi, O. J., *Impacts of Microplastics on Lagos Lagoon Ecosystems*. *Journal of* **Environmental Studies**, 15(4), 2021, 234–245. https://doi.org/10.1016/j.envst.2021.234

Ogundele, D. T., Ojo, A. O., & Adeyemi, O. A., *Microplastics in Lagos Lagoon: An Emerging Threat to the Aquatic Ecosystem*. **Journal of Environmental Management**, 255(2), 2021, 109834.

Ogundele, I. B., Adebayo, A., & Ojo, S. K., *Microplastic Pollution in Nigerian Aquatic Environments: A Review of the Current State and Implications for Future Research*. **Journal of Environmental Science and Pollution Research**, 28(9), 2021, 11544–11557.

Ogunola, I. S., Adeogun, A. O., & Akindele, E. O., *Microplastics in Tropical Aquatic Environments: The Case of Epe Lagoon, Nigeria*. **Ecotoxicology and Environmental Safety**, 244(1), 2022, 113443.

Ogunola, O. S., Onada, O. A., & Falaye, A. E., *Impacts of Microplastics on Fish and Aquatic Organisms: The Nigerian Perspective*. **Environmental Science and Pollution Research**, 27(22), 2020, 27661–27673. https://doi.org/10.1007/s11356-020-09218-3

Ogunola, O. S., Onada, O. A., & Falaye, A. E., *Microplastics in Marine Environment: Current Trends in Environmental Pollution and Mechanisms of Toxicological Profile*. **Environmental Science and Pollution Research**, 25(9), 2018, 9293–9304. https://doi.org/10.1007/s11356-018-1232-5

Ojo A.O., Akande F.A., & Ogundele D.T., *Distribution and Characterization of Microplastics in Surface Waters of Lagos Lagoon*. **Science of the Total Environment**, 859(4), 2023, 160341.

Ojo, T., Akinyemi, I. O., & Ojo, A. A., *Microplastic Contamination in Lagos Lagoon, Nigeria: Sources, Distribution, and Ecological Implications*. **Environmental Pollution**, 307, 2023, 119580.

Okwodu, N. E., Okorie, P. U., & Nwoke, B. E. B., *Length-Weight Relationship and Condition Factor of African Mud Catfish (Clarias gariepinus (Burchell 1822)) and Tilapia (Oreochromis nilotica Linnaeus 1758) in Orashi River of Ogba/Egbema/Ndoni LGA of Rivers State, Nigeria*. **Asian Journal of Fisheries and Aquatic Research**, 16(2), 2022, 16–31. https://doi.org/10.9734/ajfar/2022/v16i230368

Olarinmoye, O. M., Stock, F., Scherf, N., Whenu, O., Asenime, C., & Ganzallo, S., *Microplastic Presence in Sediment and Water of a Lagoon Bordering the Urban Agglomeration of Lagos, Southwest Nigeria*. **Geosciences**, 10(12), 2020, 494. https://doi.org/10.3390/geosciences10120494

Olatunde A.A., *Some Aspects of the Biology of Synodontis Species in Jebba Lake, Nigeria*. **Journal of Aquatic Sciences**, 4, 1989, 49–54.

Olawusi-Peters, O. O., & Solomon, J. R., *Environmental Parameters and Fish Assemblages of the Badagry-Lagos Coastal Lagoon System, Nigeria*. **West African Journal of Applied Ecology**, 29(2), 2021, 1–14. https://doi.org/10.4314/wajae.v29i2.1

Olopade, O. A., Taiwo, I. O., & Ogunbanwo, A. E., *Length-Weight Relationship and Condition Factor of Leuciscus niloticus (De Joannis, 1853) from Epe Lagoon, Lagos State, Nigeria*. **Ege Journal of Fisheries and Aquatic Sciences**, 32(3), 2015, 165–168. https://doi.org/10.12714/egejfas.2015.32.3.07

Olowu, O. A., Ayoola, O. T., & Ogunyemi, A. O., *Heavy Metal and Microplastic Interactions in Nigerian Coastal Waters*. **Environmental Monitoring and Assessment**, 195(12), 2023, 1074.

Olurin, T. O., *Assessment of Naturally Occurring Radionuclides and Heavy Metals Level and Health Risks in Commonly Consumed African Catfish, White Catfish and Nile Tilapia Fish Species From Epe Waterside Region of Lagos, Nigeria*. **Journal of Applied Sciences and Environmental Management**, 28(2), 2024, 449–457.

Omoyajowo, K., Raimi, M. O., Waleola, T. O., Odipe, O. E., & Ogunyebi, A. L., *Public Awareness, Knowledge, Attitude and Perception on Microplastics Pollution around Lagos Lagoon*. **Research Square**, 2021. https://doi.org/10.21203/rs.3.rs-506361/v1

Onyia, L. U., Okoh, F. A., & Ayotunde, E. O., *Ecological Characterization and Abundance of Clarias gariepinus in Cross River System, Nigeria*. **Egyptian Journal of Aquatic Biology & Fisheries**, 26(1), 2022, 217–234. https://doi.org/10.21608/ejabf.2022.215341

Osei, J., Boakye, E. M., & Ofori-Danson, P. K., *Microplastic Contamination in Urban Lagoons of Accra*. **African Journal of Aquatic Science**, 47(1), 2022, 15–26.

Otitoloju A.A., Olagoke A.O., & Oladipo O., *Heavy Metal Contamination and Its Ecological Risk Assessment in Sediments of the Lagos Lagoon, Nigeria*. **Environmental Monitoring and Assessment**, 191(5), 2019, 305. https://doi.org/10.1007/s10661-019-7445-8.

Oyekunle, J. A. O., Olatunji, O. S., Fatoki, O. S., & Ogunfowokan, A. O., *Occurrence and Distribution of Microplastics in Surface Waters and Sediments of Lagos Lagoon, Nigeria*. **Environmental Science and Pollution Research**, 28, 2021, 41067–41080. https://doi.org/10.1007/s11356-021-13691-3

Oyeyiola A.O., Davidson C.M., Olayinka K.O., & Alo B.I., *Fractionation and Ecotoxicological Implication of Potentially Toxic Metals in Sediments of Three Urban Rivers and the Lagos Lagoon, Nigeria, West Africa.* **Environmental Monitoring and Assessment**, 186, 2014, 7321–7333. https://doi.org/10.1007/s10661-014-3929-9.

Oyinloye, M. A., Olamiju, I. O., & Popoola, O. O., *Urban Renewal Strategies in Developing Nations: A Focus on Makoko, Lagos State, Nigeria*. **Journal of Geography and Regional Planning**, 10(8), 2017, 229–241. https://doi.org/10.5897/JGRP2017.0631

Pellini, G., Gomiero, A., Fortibuoni, T., Ferra, C., Grati, F., Tassetti, A. N., Polidori, P., & Fabi, G., *Characterization of Microplastic Litter in the Gastrointestinal Tract of Solea solea from the Adriatic Sea*. **Environmental Pollution**, 234, 2018, 943–952. https://doi.org/10.1016/j.envpol.2017.12.038

Phuong, N. N., Poirier, L., Pham, Q. T., Lagarde, F., & Zalouk-Vergnoux, A., *Quantification and Characterization of Microplastics in Blue Mussels (Mytilus edulis): Protocol Setup and Preliminary Data on the Contamination of the French Atlantic Coast*. **Environmental Science and Pollution Research**, 25, 2018, 6135–6144. https://doi.org/10.1007/s11356-017-8862-3

Plastics Europe, *Plastics - the Facts 2020: An Analysis of European Plastics Production, Demand and Waste Data*. **PlasticsEurope**. https://www.plasticseurope.org/en/resources/publications/4312-plastics-facts-2020

Pollman C.D., & Axelrad D.M., *Mercury Bioaccumulation Factors and Spurious Correlations*. **Science of The Total Environment**, 496, 2014, vi–xii. https://doi.org/10.1016/j.scitotenv.2014.07.050.

Prata, J. C., da Costa, J. P., Lopes, I., Duarte, A. C., & Rocha-Santos, T., *Environmental Exposure to Microplastics: An Overview on Possible Human Health Effects*. **Science of The Total Environment**, 702, 2020, 134455.

Prata, J. C., da Costa, J. P., Lopes, I., Duarte, A. C., & Rocha-Santos, T., *Environmental Impacts of Microplastics and Their Interactions with Sediments*. **Science of The Total Environment**, 702, 2020, 134455.

Provenza, F., Russo, G., Falco, M., & Maffioli, G., *Chemical Pollution and Ecotoxicological Effects of High-density Polyethylene Microplastics in Mytilus galloprovincialis from Two Italian Lagoon Ecosystems*. **Environmental Toxicology and Pharmacology**, 98, 2023, 104075. https://doi.org/10.1016/j.etap.2023.104075

Raamsdonk, L. V., van der Zande, M., Koelmans, A. A., Hoogenboom, R. L. A. P., Peters, R. J. B., Groot, M. J., & Peijnenburg, A. A. C., *Current Insights into Monitoring, Bioaccumulation, and Potential Health Effects of Microplastics Present in the Food Chain*. **Foods**, 9(1), 2020, 72.

Rao, I., *Microplastics in the Aquatic Environment: Implications for Post-harvest Fish Quality*. **Indian Journal of Fisheries**, 66(10), 2019, 142–152.

Renzi, I., Guerranti, C., & Blašković, A., *Marine Litter in Stomach Content of Small Pelagic Fishes from the Adriatic Sea: Sardines (Sardina pilchardus) and Anchovies (Engraulis encrasicolus)*. **Environmental Science and Pollution Research**, 26, 2019, 2771–2781. https://doi.org/10.1007/s11356-018-3886-8

Rochman A.M., Tahir A., Williams S.L., Baxa D.V., Lam R., Miller J.T., Teh F.C., Werorilangi S., & Teh S.J., *Anthropogenic Debris in Seafood: Plastic Debris and Fibers from Textiles in Fish and Bivalves Sold for Human Consumption.* **Scientific Reports**, 5, 2019, 14340. https://doi.org/10.1038/srep14340.

Rochman C.M., Hoh E., Kurobe T., & Teh S.J., *Ingested Plastic Transfers Hazardous Chemicals to Fish and Induces Hepatic Stress*. **Scientific Reports**, 3, 2015, 3263. https://doi.org/10.1038/srep03263.

Rochman C.M., Kurobe T., Flores I., & Teh S.J., *Early Warning Signs of Endocrine Disruption in Adult Fish from the Ingestion of Polyethylene With and Without Sorbed Chemical Pollutants From the Marine Environment*. **Science of the Total Environment**, 493, 2020, 656–661.

Rochman, C. M., Browne, M. A., Underwood, A. J., van Franeker, J. A., Thompson, R. C., & Amaral-Zettler, L. A., *The Ecological Impacts of Marine Debris: Unraveling the Demonstrated Evidence From What is Perceived*. **Ecology**, 94(3), 2021, 248–264.

Rodríguez-Seijo A., Santos J., da Silva T., Cachada S., Pereira S., Carvalho P., Duarte A., Moreira F.F.R., & Duarte A.C., *Oxidative Stress, Energy Metabolism and Molecular Responses of Earthworms (Eisenia fetida) Exposed to Low-density Polyethylene Microplastics*. **Environmental Science and Pollution Research**, 25, 2018, 33599–33610. https://doi.org/10.1007/s11356-018-3255-3.

Šaravanja A., Pusic T., & Dekanic T., *Microplastics in Wastewater by Washing Polyester Fabrics*. **Materials (Basel, Switzerland)**, 15(7), 2022, 2683.

Schimdt, C., Krauth, T., & Wagner, S., *Export of Plastic Debris by Rivers into the Sea*. **Environmental Science & Technology**, 51(21), 2021, 12246–12253.

Serrano, S. E., Braun, J., Trasande, L., Dills, R., & Sathyanarayana, S., *Phthalates and Diet: A Review of the Food Monitoring and Epidemiology Data*. **Environmental Health**, 13(1), 2014, 43.

Stafford, R., & Jones, P. S., *Viewpoint – Ocean Plastic Pollution: A Convenient but Distracting Truth*. **Marine Policy**, 103, 2019, 187–191.

Sun Y., Liu Y., & Huang Z., *Microplastic Contamination in the Surface Waters of the Mediterranean Sea*. **Science of The Total Environment**, 725, 2020, 138594. https://doi.org/10.1016/j.scitotenv.2020.138594

Sun, L., Lu, M., Zhu, X., & Wu, L., *Exposure to Phthalates and Human Reproductive Health: Insights from Epidemiology and Experimental Studies*. **Environmental Pollution**, 297, 2022, 118753.

Thushari, G., & Senevirathna, J. D. M., *Plastic Pollution in the Marine Environment*. **Heliyon**, 6, 2020, e04709. https://doi.org/10.1016/j.heliyon.2020.e04709

Tiwari, M., Rathore, A., & Pandey, S. K., *Microplastics in the Environment: Occurrence, Detection, and Remediation Techniques*. **Environmental Science and Pollution Research**, 28(15), 2021, 18725–18740. https://doi.org/10.1007/s11356-021-13215-7

Trotter Bailey B.E., & Wingfield J.C., *Reproductive Success and Embryonic Development are Impacted by Exposure to Microplastics in Aquatic Species*. **Environmental Science and Pollution Research**, 26(7), 2019, 6534–6543.

van der Hal, N. D., Angel, & Ronen, Z., *Dynamics of Heavy Metals and Persistent Organic Pollutants in Plastic Debris*. **Marine Pollution Bulletin**, 146, 2020, 423–432.

Vanapalli, K. R., Sharma, H. B., Ranjan, V. P., Samal, B., Bhattacharya, J., Dubey, B. K., & Goel, S., *Challenges and Strategies for Effective Plastic Waste Management During*

*and Post COVID-19 Pandemic*. **Science of the Total Environment**, 750, 2021, 141514. https://doi.org/10.1016/j.scitotenv.2020.141514

Vo, H. C., & Pham, M. H., *Ecotoxicological Effects of Microplastics on Aquatic Organisms: A Review*. **Environmental Science and Pollution Research**, 28, 2021, 44716–44725.

Wakkaf, T., Dhib, A., El Zrelli, R., Zouari, I., Rabaoui, L., & Beyrem, H., *Microplastics in Edible Mussels from a Southern Mediterranean Lagoon: Preliminary Results on Seawater-Mussel Transfer and Implications for Environmental Protection and Seafood Safety*. **Marine Pollution Bulletin**, 158, 2020, 111355. https://doi.org/10.1016/j.marpolbul.2020.111355

Wang A., Liu X., Li Y., Powell T., Wang X., Wang G., & Zhang P., *Microplastics as Contaminants in the Soil Environment: A Mini Review*. **Science of the Total Environment**, 785, 2022, 147403.

Wang, L., Zhang, X., Zhang, Y., & Wang, Q., *Occurrence of Phthalate Esters in River Sediments: Ecological Risks and Human Exposure*. **Journal of Hazardous Materials**, 408, 2021, 124982.

Wang, W., Ge, J., & Yu, X., *Bioavailability and Toxicity of Microplastics to Fish Species: A Review*. **Ecotoxicology and Environmental Safety**, 173, 2019, 110–117.

Wang, W., Ge, J., & Yu, X., *Bioavailability and Toxicity of Microplastics to Aquatic Organisms: A Review*. **Ecotoxicology and Environmental Safety**, 209, 2021, 111885.

Wright, S. L., & Kelly, F. J., *Plastic and Human Health: A Micro Issue?* **Environmental Science & Technology**, 51(12), 2020, 6634–6647. https://doi.org/10.1021/acs.est.7b00423

Xu B., Li D., Wei K., Zhu X., Xu J., & Ma B., *Allometric Growth Patterns and Ontogenetic Development During Early Larval Stages of Schizothorax waltoni Regan and Percocypris retrodorslis in Southwest China.* **Water**, 15(4), 2023, 824. https://doi.org/10.3390/w15040824.

Xu, X., Liu, W., Wei, W., & Lu, Z., *Microplastics in the Freshwater Environment: Occurrence, Distribution, and Risks*. **Science of the Total Environment**, 536, 2016, 739–744. https://doi.org/10.1016/j.scitotenv.2015.12.060

Yang, L., Zhang, Y., Kang, S., Wang, Z., & Wu, C., *Microplastics in the Marine Environment: Sources, Distribution, and Pathways*. **Environmental Science: Processes & Impacts**, 23(2), 2021, 167–184.

Yang, W. K., Chiang, L. F., Tan, S. W., & Chen, P. J., *Environmentally Relevant Concentrations of Di(2-ethylhexyl)Phthalate Exposure Alter Larval Growth and Locomotion in Medaka Fish via Multiple Pathways*. **Science of The Total Environment**, 640–641, 2018, 512–522. https://doi.org/10.1016/j.scitotenv.2018.05.312

Yang, X., Wang, Z., Zhang, Y., Li, J., Wu, Y., & Zhao, X., *Microplastics in Freshwater Sediments: Current Status and Knowledge Gaps*. **Water Research**, 198, 2021, 117104.

Yin, L., Chen, B., Xia, B., Shi, X., Qu, K., & Zhang, T., *Impacts of Microplastics on Fish: What we Know and What we Need to Know*. **Environmental Science & Technology**, 55(4), 2021, 2189–2206.

Zarean, M., Poursafa, P., Tajadini, M. H., Ghaffari, M., & Amin, M. M., *Phthalates Exposure, Human Health Risk Assessment, and Their Reproductive Health Toxicity*. **Environmental Toxicology and Pharmacology**, 89, 2022, 103742.

Zeng, Q., Chen, G., & Xu, Y., *Toxic Effects of Phthalate Esters on Freshwater Invertebrates: Oxidative Stress and Bioaccumulation*. **Environmental Toxicology**, 37(3), 2022, 456–468.

Zhang Y., Kang S., Allen S., Allen D., Gao T., & Sillanpää M., *Atmospheric Microplastics: A Review on Current Status and Perspectives*. **Earth-Science Reviews**, 203, 2020, 103118. https://doi.org/10.1016/j.earscirev.2020.103118

Zhang Y., Kang X., Liu X., & Li J., *Sources, Distribution, and Fate of Microplastics in River Sediments: A Global Review*. **Environmental Research**, 220, 2023, 115179. https://doi.org/10.1016/j.envres.2023.115179

Zhang, C., Ding, J., Liu, Y., Wang, L., Zhang, X., & Christie, P., *Plastic Particles Reduce Microbial Activity and Carbon Utilization Efficiency in Agricultural Soils*. **Environmental Pollution**, 284, 2022, 117152. https://doi.org/10.1016/j.envpol.2021.117152

Zhang, K., Shi, H., Peng, J., Wang, Y., Xiong, X., Wu, C., & Lam, P. K. S., *Microplastic Pollution in China's Inland Water Systems: A Review of Findings, Methods, Characteristics, Effects, and Management*. **Science of the Total Environment**, 579, 2020, 1484–1495. https://doi.org/10.1016/j.scitotenv.2016.11.137

Zhang, X., Kang, X., Liu, X., & Li, J., *Microplastics in River Sediments: Patterns and Pathways*. **Water Research**, 198, 2021, 117104.

Zhang, X., Kang, X., Liu, X., & Li, J., *Sources, Distribution, and Fate of Microplastics in River Sediments: A Global Review*. **Environmental Research**, 220, 2023, 115179.

Zhao, F., Wang, X., Liang, Z., & Yuan, D., *Phthalate Removal from Wastewater: Advances in Treatment Technologies*. **Water Research**, 204, 2022, 117573.

Zheng, M., *Adsorption/Desorption Behavior of Ciprofloxacin on Aged Biodegradable Plastic PLA Under Different Exposure Conditions*. **Journal of Environmental Chemical Engineering**, 11(1), 2023, 109256.

Zhou Y., Wu C., & Xie P., *Hypoxia in Freshwater Ecosystems: Causes, Consequences, and Management Strategies*. **Water Research**, 185, 2020, 116231. https://doi.org/10.1016/j.watres.2020.116231

**Periodical Article**

G7, *Ocean Plastics Charter*. **G7 Summit**, 2018. https://www.canada.ca/en/environment-climate-change/services/managing-reducing-waste/international-commitments/ocean-plastics-charter.html.

National Emergency Management Agency (NEMA), *Annual Report on Environmental Hazards and Pollution Control in Nigeria*. **NEMA**, 2020. https://www.nema.gov.ng.

Plastics Europe, *Plastics - The Facts 2020: An Analysis of European Plastics Production, Demand and Waste Data*. **PlasticsEurope**. https://www.plasticseurope.org/en/resources/publications/4312-plastics-facts-2020.

Plastics Europe, *Plastics – The Facts 2022: An Analysis of European Plastics Production, Demand and Waste Data*. **Plastics Europe**, 2022.

United States Environmental Protection Agency (US EPA), *Ambient Water Quality Criteria for Dissolved Oxygen. Method 360.1*, *EPA Report*, **EPA** 440/5-86-003, 1984.

United States Environmental Protection Agency (US EPA), De*termination of Trace Elements by Stabilized Temperature Graphite Furnace Atomic Absorption. Method 200.9, EPA Report, Revision 2.2,* **EPA**/600/R-94/111, 1994.

United States Environmental Protection Agency (US EPA), *Microwave Assisted Acid Digestion of Sediments, Soils, and Oils. Method 3051A*, **EPA Report**, 1997.

United States Environmental Protection Agency (US EPA), *Semi-Volatile Organic Compounds by Gas Chromatography/Mass Spectrometry (GC/MS). Method 8270D*. **EPA Office of Solid Waste**, Washington, DC, 2007.

**Textbook**

Ajao E.A., Oyewo E.O., & Unyimadu J.P., A *Review of the Pollution of Coastal Waters in Nigeria*. **NIOMR Technical Paper**, Nigerian Institute for Oceanography and Marine Research, Lagos, Nigeria, 1996, 107.

**Website**

European Commission, *A European Strategy for Plastics in a Circular Economy*. **European Union**, 2018. https://ec.europa.eu/environment/circular-economy/pdf/plastics-strategy-brochure.pdf

European Commission, *Reducing Marine Litter: Action on Single-Use Plastics and Fishing Gear*, **European Union**, 2019.

https://ec.europa.eu/environment/pdf/waste/plastics/reducing_marine_litter_brochure.pdf.

European Commission, *Reducing Marine Litter: Action on Single-Use Plastics and Fishing Gear*. **European Union**, 2019. https://ec.europa.eu/environment/pdf/waste/plastics/reducing_marine_litter_brochure.pdf.

International Maritime Organization (IMO), *Action Plan to Address Marine Plastic Litter From Ships*. **IMO**, 2019. https://www.imo.org/en/MediaCentre/PressBriefings/Pages/24-marine-litter-action-plan-.aspx.

United Nations Environment Programme (UNEP), *Plastic Pollution in Marine Environments: Status and Trends*. **UNEP**, 2019. https://www.unep.org

United States Environmental Protection Agency (US EPA), *Clean Water Act Analytical Methods. Method 1669*, **EPA**, 2023. Available at: https://www.epa.gov.

United States Environmental Protection Agency (US EPA), *Clean Water Act Analytical Methods. Method 1669*, **EPA**, 2023. Available at: https://www.epa.gov.

# Appendices

## Appendix I

## Ethical Approval


**Lead City University (LCU)**

*Motto: Knowledge for Self-reliance*

*Lagos - Ibadan Expressway, Toll Gate Area, Ibadan, Oyo State, Nigeria*

*Email: lcu.hrec@lcu.edu.ng*

**University Research Ethics Committee**

NHREC


**PROJECT TITLE: MICROPLASTIC POLLUTION IN OREOCHROMIS NILOTICUS AND CLARIAS GARIEPINUS FISHES, AND BENTHIC AND SURFACE WATERS FROM THREE HIGH POPULATION DENSITY ZONES OF LAGOS (MAKOKO AND IKORODU AXIS) AND EPE LAGOONS.**

**PROJECT NUMBER:** **LCU-REC/25/0042**

**APP ROVAL DATE:** **12/05/2025**

**EXPIRY DATE:** **12/05/2026**

**APPROVAL LETTER**

The above-named proposal has been adequately reviewed; the protocol and safety guidelines satisfy the conditions of *LCU-REC* policies regarding experiments that use human subjects. Therefore, the study under its reviewed state is hereby approved by the LCU-Research Ethics Committee.

**Prof. Olusola Ladokun**

*Name of LCU-REC Chairman* .....

LCU RESEARCH ETHICS COMMITTEE APPROVAL Sign. Date

**Dr. Posi Aduroja**

*Name of LCU-REC Secretary* .....

**This approval is given with the investigator's Declaration as stated below;**

**By signing below, I agree/certify that:**

1. I have reviewed this protocol submission in its entirety and that I am fully cognizant of, and in agreement with all submitted statements.

2. I will conduct this research study in strict accordance with all submitted statements except where a change may be necessary to eliminate apparent immediate hazard to a given research subject.
   - I will notify the LCU-REC promptly of any change in research procedures necessitated in the interest of the safety of a given research subject.

## Appendix I Continued

## Ethical Approval

- I will request and obtain LCU-REC approval of any proposed modification to the research protocol or informed consent document(s) prior to implementing such modifications.

3. I will ensure that all co-investigator and other personnel assisting in the conduct of this research study have been provided a copy of the entire current version of the research protocol and are fully informed of the current (a) study procedures (including procedure modifications); (b) informed consent requirements and process; (c) potential risks associated with the study participation and the steps required to be taken to prevent or minimize these potential risks; (d) adverse events reporting requirements; (e) data and record-keeping; and (f) the current REC approval status of the research study.

4. I will respond promptly to all requests for information or materials solicited by the REC or REC Office.

5. I will submit the research study in a timely manner for the REC renewal approval.

6. I will not enroll any individual into this research study until such time I obtain his/her written informed consent, or if applicable, the written informed consent of his/her authorized representative (i.e. unless the REC has granted a waiver of the requirement to obtain informed consent).

7. I will employ and oversee an informed consent process that ensures that potential research subjects understand fully the purpose of the research study, the nature of the research procedures they are being asked to undergo, the potential risks of these research procedures, and their rights as a research study volunteer.

8. I will ensure that the research subjects are kept fully informed of any new information that may affect their willingness to continue to participate in the research study.

9. I will maintain adequate, current, and accurate records of research data, outcomes, and adverse events to permit an ongoing assessment of the risks/benefits ratio of research study participation.

10. I am cognizant of, and will comply with, current federal regulations and REC requirements governing human subject research including adverse event reporting requirements.

11. I will make a reasonable effort to ensure that subjects who have suffered adverse event associated with research participation receive adequate care to correct or alleviate the consequences of the adverse event in the extent possible.

12. I will ensure that the conduct of this research study adheres to Good Clinical Practice guidelines.

**Ms. OTU-EKUMA NKECHINYERE** .....

Principal Investigator's Name Principal Investigator's Signature and Date

## Appendix II

## Raw Result of Morphology of *Oreochromis niloticus* and *Clarias gariepinus* from the Lagos and Epe Lagoons

| March | Sample Site | Head Length (cm) | Body Weight (g) | Total Length (cm) | Standard Length (cm) | Pre-dorsal Length (cm) | Pre-anal Length (cm) | Body Depth (cm) |
|---|---|---|---|---|---|---|---|---|
| **Oreochromis niloticus** | Makoko | 3.52 | 106.29 | 16.19 | 12.69 | 5.71 | 6.49 | 3.99 |
| **Oreochromis niloticus** | Makoko | 3.12 | 143.33 | 16.18 | 11.84 | 5.17 | 5.32 | 4.94 |
| **Oreochromis niloticus** | Makoko | 3.62 | 126.75 | 17.21 | 14.35 | 5.26 | 6.74 | 4.22 |
| **Oreochromis niloticus** | Makoko | 4.17 | 101.48 | 14.07 | 12.04 | 4.79 | 5.83 | 3.82 |
| **Oreochromis niloticus** | Makoko | 3.06 | 122.15 | 14.35 | 11.08 | 5.07 | 6.12 | 4.54 |
| ***Oreochromis niloticus*** | Ikorodu | 3.56 | 142.8 | 15.54 | 13.07 | 5.9 | 6.27 | 4.9 |
| ***Oreochromis niloticus*** | Ikorodu | 4.05 | 135.04 | 15.89 | 11.72 | 4.9 | 6.43 | 4.3 |
| ***Oreochromis niloticus*** | Ikorodu | 3.31 | 131.11 | 17.95 | 11.17 | 5.2 | 5.93 | 4.75 |
| ***Oreochromis niloticus*** | Ikorodu | 3.53 | 128.58 | 16.98 | 13.62 | 5.56 | 5.88 | 4.48 |
| ***Oreochromis niloticus*** | Ikorodu | 4.05 | 112.47 | 14.13 | 14.42 | 5.93 | 6.98 | 4.06 |
| ***Oreochromis niloticus*** | Epe | 3.97 | 153.8 | 16.12 | 11.87 | 5.95 | 5.93 | 3.99 |
| ***Oreochromis niloticus*** | Epe | 4.35 | 144.4 | 16.91 | 14.35 | 6.7 | 6.74 | 4.89 |
| ***Oreochromis niloticus*** | Epe | 3.84 | 139.46 | 18.43 | 13.32 | 5.26 | 6.73 | 4.96 |
| ***Oreochromis niloticus*** | Epe | 4.36 | 144.46 | 15.72 | 11.82 | 5.58 | 6.61 | 4.54 |
| ***Oreochromis niloticus*** | Epe | 2.99 | 117.87 | 15.32 | 13.64 | 5.52 | 6.5 | 5.12 |
| ***Clarias gariepinus*** | Makoko | 6.71 | 251.43 | 30.76 | 26.18 | 13.9 | 12.48 | 7.7 |
| ***Clarias gariepinus*** | Makoko | 7.77 | 278.52 | 29.91 | 23.79 | 11.08 | 15.07 | 6.34 |
| ***Clarias gariepinus*** | Makoko | 6.54 | 279.76 | 27.05 | 27.04 | 11.46 | 13.26 | 6.61 |
| ***Clarias gariepinus*** | Makoko | 6.6 | 314.16 | 32.89 | 23.1 | 12.05 | 15.71 | 7.27 |
| ***Clarias gariepinus*** | Makoko | 6.37 | 276.14 | 31.9 | 25.89 | 11.51 | 13.49 | 6.08 |
| ***Clarias gariepinus*** | Ikorodu | 7.27 | 326.99 | 31.76 | 24.59 | 11.46 | 14.48 | 6.38 |
| ***Clarias gariepinus*** | Ikorodu | 7.72 | 271 | 32.2 | 27.94 | 12.99 | 14.93 | 6.45 |
| ***Clarias gariepinus*** | Ikorodu | 6.48 | 268.68 | 27.42 | 26.13 | 13.31 | 16.11 | 7.15 |
| ***Clarias gariepinus*** | Ikorodu | 7.25 | 319.78 | 31.67 | 23.97 | 11.6 | 13.76 | 7.55 |
| ***Clarias gariepinus*** | Ikorodu | 7.28 | 313.54 | 31.95 | 26.37 | 13.14 | 13.21 | 7.48 |
| ***Clarias gariepinus*** | Epe | 7.43 | 356.6 | 34.44 | 25.15 | 13.99 | 15.03 | 6.62 |
| ***Clarias gariepinus*** | Epe | 6.98 | 291.55 | 33.83 | 28.27 | 12.12 | 15.27 | 7.08 |
| ***Clarias gariepinus*** | Epe | 7.78 | 282.17 | 32.45 | 26.73 | 13.65 | 15.91 | 7.08 |
| ***Clarias gariepinus*** | Epe | 6.82 | 350.32 | 30.49 | 23.23 | 13.51 | 15.88 | 7.08 |
| ***Clarias gariepinus*** | Epe | 8.49 | 319.35 | 28.8 | 26.63 | 11.73 | 12.91 | 8.13 |

**Source:** Author's Field Work, 2025

## Appendix II Continued

## Raw Result of Morphology of *Oreochromis niloticus* and *Clarias gariepinus* from the Lagos and Epe Lagoons

| May | Sample Site | Head Length (cm) | Body Weight (g) | Total Length (cm) | Standard Length (cm) | Pre-dorsal Length (cm) | Pre-anal Length (cm) | Body Depth (cm) |
|---|---|---|---|---|---|---|---|---|
| ***Oreochromis niloticus*** | Makoko | 3.63 | 106.72 | 16.43 | 12.82 | 5.98 | 6.67 | 4.01 |
| ***Oreochromis niloticus*** | Makoko | 3.13 | 156.1 | 16.43 | 11.89 | 5.26 | 5.26 | 5.2 |
| ***Oreochromis niloticus*** | Makoko | 3.75 | 133.99 | 17.55 | 14.62 | 5.38 | 6.97 | 4.3 |
| ***Oreochromis niloticus*** | Makoko | 4.44 | 100.31 | 14.14 | 12.1 | 4.75 | 5.88 | 3.79 |
| ***Oreochromis niloticus*** | Makoko | 3.05 | 127.87 | 14.44 | 11.07 | 5.13 | 6.22 | 4.7 |
| ***Oreochromis niloticus*** | Ikorodu | 3.62 | 154.2 | 15.7 | 13.3 | 6.11 | 6.37 | 5.13 |
| ***Oreochromis niloticus*** | Ikorodu | 4.27 | 142.56 | 16.08 | 11.83 | 4.85 | 6.57 | 4.34 |
| ***Oreochromis niloticus*** | Ikorodu | 3.28 | 136.67 | 18.29 | 11.23 | 5.23 | 5.94 | 4.93 |
| ***Oreochromis niloticus*** | Ikorodu | 3.57 | 132.87 | 17.25 | 13.89 | 5.68 | 5.87 | 4.58 |
| ***Oreochromis niloticus*** | Ikorodu | 4.27 | 108.7 | 14.18 | 14.75 | 6.14 | 7.25 | 4.02 |
| ***Oreochromis niloticus*** | Epe | 4.08 | 165.7 | 16.39 | 11.96 | 6.07 | 5.94 | 3.92 |
| ***Oreochromis niloticus*** | Epe | 4.54 | 151.6 | 17.24 | 14.68 | 6.98 | 7.02 | 5.03 |
| ***Oreochromis niloticus*** | Epe | 3.92 | 144.2 | 18.91 | 13.55 | 5.25 | 7 | 5.12 |
| ***Oreochromis niloticus*** | Epe | 4.55 | 151.69 | 15.94 | 11.9 | 5.63 | 6.84 | 4.6 |
| ***Oreochromis niloticus*** | Epe | 2.91 | 111.8 | 15.51 | 13.91 | 5.57 | 6.7 | 5.32 |
| ***Clarias gariepinus*** | Makoko | 6.89 | 247.14 | 31.27 | 26.61 | 14.39 | 12.65 | 7.95 |
| ***Clarias gariepinus*** | Makoko | 8.17 | 287.77 | 30.38 | 23.9 | 11.29 | 15.45 | 6.36 |
| ***Clarias gariepinus*** | Makoko | 6.69 | 289.64 | 27.37 | 27.59 | 11.7 | 13.5 | 6.68 |
| ***Clarias gariepinus*** | Makoko | 6.77 | 341.23 | 33.51 | 23.12 | 12.35 | 16.16 | 7.45 |
| ***Clarias gariepinus*** | Makoko | 6.48 | 284.21 | 32.47 | 26.29 | 11.76 | 13.74 | 6.06 |
| ***Clarias gariepinus*** | Ikorodu | 7.48 | 347.79 | 32.6 | 24.7 | 11.63 | 14.78 | 6.35 |
| ***Clarias gariepinus*** | Ikorodu | 8.06 | 269.4 | 33.07 | 28.29 | 13.35 | 15.32 | 6.44 |
| ***Clarias gariepinus*** | Ikorodu | 6.51 | 266.16 | 28.02 | 26.36 | 13.71 | 16.73 | 7.28 |
| ***Clarias gariepinus*** | Ikorodu | 7.46 | 337.7 | 32.51 | 24.04 | 11.78 | 13.92 | 7.75 |
| ***Clarias gariepinus*** | Ikorodu | 7.5 | 328.96 | 32.81 | 26.61 | 13.52 | 13.25 | 7.68 |
| ***Clarias gariepinus*** | Epe | 7.52 | 378.81 | 35.06 | 25.6 | 14.3 | 15.23 | 6.61 |
| ***Clarias gariepinus*** | Epe | 6.99 | 292.07 | 34.42 | 28.9 | 12.22 | 15.55 | 7.16 |
| ***Clarias gariepinus*** | Epe | 7.93 | 279.56 | 32.97 | 27.27 | 13.92 | 16.36 | 7.16 |
| ***Clarias gariepinus*** | Epe | 6.81 | 370.43 | 30.92 | 23.57 | 13.77 | 16.32 | 7.16 |
| ***Clarias gariepinus*** | Epe | 8.76 | 329.13 | 29.14 | 27.17 | 11.79 | 12.54 | 8.42 |

**Source:** Author's Field Work, 2025

## Appendix II Continued

## Raw Result of Morphology of *Oreochromis niloticus* and *Clarias gariepinus* from the Lagos and Epe Lagoons

| August | Sample Site | Head Length (cm) | Body Weight (g) | Total Length (cm) | Standard Length (cm) | Pre-dorsal Length (cm) | Pre-anal Length (cm) | Body Depth (cm) |
|---|---|---|---|---|---|---|---|---|
| ***Oreochromis niloticus*** | Makoko | 3.84 | 117.15 | 16.93 | 13.27 | 6.35 | 6.95 | 4.21 |
| ***Oreochromis niloticus*** | Makoko | 3.23 | 178.88 | 16.93 | 12.19 | 5.45 | 5.3 | 5.4 |
| ***Oreochromis niloticus*** | Makoko | 3.98 | 151.24 | 18.22 | 15.38 | 5.61 | 7.3 | 4.5 |
| ***Oreochromis niloticus*** | Makoko | 4.81 | 109.14 | 14.29 | 12.44 | 4.81 | 6.02 | 3.99 |
| ***Oreochromis niloticus*** | Makoko | 3.14 | 143.59 | 14.63 | 11.23 | 5.28 | 6.43 | 4.9 |
| ***Oreochromis niloticus*** | Ikorodu | 3.77 | 175.6 | 16.11 | 13.64 | 6.31 | 6.56 | 5.46 |
| ***Oreochromis niloticus*** | Ikorodu | 4.59 | 160.08 | 16.55 | 11.95 | 4.8 | 6.8 | 4.47 |
| ***Oreochromis niloticus*** | Ikorodu | 3.35 | 152.23 | 19.08 | 11.26 | 5.25 | 6.04 | 5.22 |
| ***Oreochromis niloticus*** | Ikorodu | 3.71 | 147.15 | 17.89 | 14.33 | 5.8 | 5.97 | 4.77 |
| ***Oreochromis niloticus*** | Ikorodu | 4.58 | 114.93 | 14.37 | 15.32 | 6.34 | 7.62 | 4.07 |
| ***Oreochromis niloticus*** | Epe | 4.29 | 185.3 | 16.72 | 12.13 | 6.2 | 5.94 | 3.94 |
| ***Oreochromis niloticus*** | Epe | 4.83 | 168.07 | 17.72 | 15.35 | 7.26 | 7.3 | 5.28 |
| ***Oreochromis niloticus*** | Epe | 4.11 | 159.02 | 19.66 | 14.02 | 5.24 | 7.28 | 5.38 |
| ***Oreochromis niloticus*** | Epe | 4.84 | 168.18 | 16.2 | 12.07 | 5.69 | 7.08 | 4.77 |
| ***Oreochromis niloticus*** | Epe | 2.93 | 119.42 | 15.7 | 14.44 | 5.61 | 6.89 | 5.63 |
| ***Clarias gariepinus*** | Makoko | 7.17 | 255.72 | 32.33 | 27.44 | 15.18 | 12.92 | 8.3 |
| ***Clarias gariepinus*** | Makoko | 8.66 | 316.66 | 31.26 | 24.41 | 11.8 | 15.94 | 6.49 |
| ***Clarias gariepinus*** | Makoko | 6.94 | 319.45 | 27.68 | 28.53 | 12.25 | 13.83 | 6.85 |
| ***Clarias gariepinus*** | Makoko | 7.03 | 396.85 | 34.98 | 23.54 | 12.96 | 16.7 | 7.73 |
| ***Clarias gariepinus*** | Makoko | 6.69 | 311.32 | 33.75 | 27.08 | 12.32 | 14.1 | 6.14 |
| ***Clarias gariepinus*** | Ikorodu | 7.8 | 393.98 | 33.96 | 25.25 | 11.87 | 15.08 | 6.53 |
| ***Clarias gariepinus*** | Ikorodu | 8.49 | 282 | 34.54 | 29.55 | 13.97 | 15.75 | 6.62 |
| ***Clarias gariepinus*** | Ikorodu | 6.63 | 277.37 | 28.42 | 27.23 | 14.41 | 17.52 | 7.61 |
| ***Clarias gariepinus*** | Ikorodu | 7.77 | 379.57 | 33.86 | 24.45 | 12.06 | 14 | 8.16 |
| ***Clarias gariepinus*** | Ikorodu | 7.82 | 367.09 | 34.22 | 27.53 | 14.18 | 13.16 | 8.08 |
| ***Clarias gariepinus*** | Epe | 7.81 | 427.11 | 36.64 | 26.19 | 14.92 | 15.54 | 6.68 |
| ***Clarias gariepinus*** | Epe | 7.2 | 307.84 | 35.85 | 29.87 | 12.42 | 15.9 | 7.41 |
| ***Clarias gariepinus*** | Epe | 8.28 | 290.65 | 34.07 | 28.06 | 14.47 | 16.83 | 7.41 |
| ***Clarias gariepinus*** | Epe | 6.99 | 415.59 | 31.56 | 23.94 | 14.28 | 16.78 | 7.41 |
| ***Clarias gariepinus*** | Epe | 9.22 | 358.81 | 29.38 | 27.94 | 11.91 | 12.46 | 9.1 |

**Source:** Author's Field Work, 2025

## Appendix II Continued

## Raw Result of Morphology of *Oreochromis niloticus* and *Clarias gariepinus* from the Lagos and Epe Lagoons

| November | Sample Site | Head Length (cm) | Body Weight (g) | Total Length (cm) | Standard Length (cm) | Pre-dorsal Length (cm) | Pre-anal Length (cm) | Body Depth (cm) |
|---|---|---|---|---|---|---|---|---|
| ***Oreochromis niloticus*** | Makoko | 4.04 | 127.58 | 17.28 | 13.5 | 6.72 | 7.23 | 4.44 |
| ***Oreochromis niloticus*** | Makoko | 3.34 | 201.65 | 17.28 | 12.34 | 5.64 | 5.35 | 5.85 |
| ***Oreochromis niloticus*** | Makoko | 4.21 | 168.49 | 18.65 | 15.75 | 5.83 | 7.62 | 4.78 |
| ***Oreochromis niloticus*** | Makoko | 5.18 | 117.97 | 14.46 | 12.61 | 4.88 | 6.17 | 4.17 |
| ***Oreochromis niloticus*** | Makoko | 3.23 | 159.31 | 14.83 | 11.31 | 5.44 | 6.63 | 5.26 |
| ***Oreochromis niloticus*** | Ikorodu | 4.02 | 197 | 16.33 | 13.86 | 6.61 | 6.75 | 6.03 |
| ***Oreochromis niloticus*** | Ikorodu | 5.01 | 177.6 | 16.81 | 12.06 | 4.85 | 7.04 | 4.64 |
| ***Oreochromis niloticus*** | Ikorodu | 3.52 | 167.79 | 19.66 | 11.32 | 5.38 | 6.15 | 5.69 |
| ***Oreochromis niloticus*** | Ikorodu | 3.95 | 161.44 | 18.32 | 14.59 | 6.01 | 6.06 | 5.06 |
| ***Oreochromis niloticus*** | Ikorodu | 5 | 121.16 | 14.37 | 15.66 | 6.65 | 8 | 4.08 |
| ***Oreochromis niloticus*** | Epe | 4.6 | 207.21 | 16.99 | 12.22 | 6.43 | 6.05 | 3.99 |
| ***Oreochromis niloticus*** | Epe | 5.22 | 185.27 | 18.06 | 15.68 | 7.64 | 7.69 | 5.77 |
| ***Oreochromis niloticus*** | Epe | 4.4 | 173.75 | 20.14 | 14.25 | 5.33 | 7.65 | 5.91 |
| ***Oreochromis niloticus*** | Epe | 5.23 | 185.41 | 16.43 | 12.15 | 5.85 | 7.42 | 5.09 |
| ***Oreochromis niloticus*** | Epe | 3.04 | 123.35 | 15.89 | 14.7 | 5.76 | 7.19 | 6.24 |
| ***Clarias gariepinus*** | Makoko | 7.36 | 264.29 | 33.55 | 28.01 | 15.47 | 12.84 | 8.65 |
| ***Clarias gariepinus*** | Makoko | 9.06 | 345.55 | 32.4 | 24.82 | 11.8 | 16.51 | 6.61 |
| ***Clarias gariepinus*** | Makoko | 7.09 | 349.27 | 28.54 | 29.15 | 12.29 | 13.95 | 7.01 |
| ***Clarias gariepinus*** | Makoko | 7.19 | 452.47 | 36.42 | 23.89 | 13.06 | 17.43 | 8.01 |
| ***Clarias gariepinus*** | Makoko | 6.81 | 338.43 | 35.09 | 27.62 | 12.37 | 14.27 | 6.22 |
| ***Clarias gariepinus*** | Ikorodu | 8.01 | 440.18 | 35.17 | 25.69 | 11.94 | 15.47 | 6.48 |
| ***Clarias gariepinus*** | Ikorodu | 8.82 | 294.6 | 35.87 | 30.7 | 14.23 | 16.28 | 6.6 |
| ***Clarias gariepinus*** | Ikorodu | 6.65 | 288.58 | 28.43 | 28 | 14.72 | 18.4 | 7.87 |
| ***Clarias gariepinus*** | Ikorodu | 7.98 | 421.43 | 35.04 | 24.76 | 12.14 | 14.17 | 8.58 |
| ***Clarias gariepinus*** | Ikorodu | 8.04 | 405.21 | 35.48 | 28.35 | 14.46 | 13.17 | 8.47 |
| ***Clarias gariepinus*** | Epe | 8 | 475.41 | 38.48 | 26.84 | 15.44 | 16.05 | 6.64 |
| ***Clarias gariepinus*** | Epe | 7.31 | 323.62 | 37.61 | 31.07 | 12.53 | 16.47 | 7.56 |
| ***Clarias gariepinus*** | Epe | 8.52 | 301.74 | 35.64 | 28.98 | 14.91 | 17.58 | 7.56 |
| ***Clarias gariepinus*** | Epe | 7.08 | 460.75 | 32.84 | 24.25 | 14.69 | 17.51 | 7.56 |
| ***Clarias gariepinus*** | Epe | 9.59 | 388.49 | 30.42 | 28.85 | 11.93 | 12.39 | 9.67 |

**Source:** Author's Field Work, 2025

## Appendix III

### Raw pH values of the Surface and Benthic Waters from the Lagos and Epe Lagoons

| March | Water Type | Sample Site | Value | May | Water Type | Sample Site | Value |
|---|---|---|---|---|---|---|---|
| **pH** | Surface | Makoko | 8.08 | pH | Surface | Makoko | 8.99 |
| **pH** | Surface | Ikorodu | 7.66 | pH | Surface | Ikorodu | 7.61 |
| **pH** | Surface | Epe | 6.44 | pH | Surface | Epe | 6.84 |
| **pH** | Surface | Makoko | 7.98 | pH | Surface | Makoko | 8.88 |
| **pH** | Surface | Ikorodu | 7.94 | pH | Surface | Ikorodu | 7.89 |
| **pH** | Surface | Epe | 6.44 | pH | Surface | Epe | 6.84 |
| **pH** | Surface | Makoko | 8.1 | pH | Surface | Makoko | 9.02 |
| **pH** | Surface | Ikorodu | 7.82 | pH | Surface | Ikorodu | 7.77 |
| **pH** | Surface | Epe | 6.53 | pH | Surface | Epe | 6.93 |
| **pH** | Surface | Makoko | 8.24 | pH | Surface | Makoko | 9.17 |
| **pH** | Surface | Ikorodu | 7.63 | pH | Surface | Ikorodu | 7.58 |
| **pH** | Surface | Epe | 6.25 | pH | Surface | Epe | 6.64 |
| **pH** | Surface | Makoko | 7.96 | pH | Surface | Makoko | 8.86 |
| **pH** | Surface | Ikorodu | 7.78 | pH | Surface | Ikorodu | 7.73 |
| **pH** | Surface | Epe | 6.28 | pH | Surface | Epe | 6.66 |
| **pH** | Benthic | Makoko | 7.02 | pH | Benthic | Makoko | 7.47 |
| **pH** | Benthic | Ikorodu | 7 | pH | Benthic | Ikorodu | 7.31 |
| **pH** | Benthic | Epe | 6.61 | pH | Benthic | Epe | 6.41 |
| **pH** | Benthic | Makoko | 6.96 | pH | Benthic | Makoko | 7.41 |
| **pH** | Benthic | Ikorodu | 6.77 | pH | Benthic | Ikorodu | 7.07 |
| **pH** | Benthic | Epe | 6.45 | pH | Benthic | Epe | 6.25 |
| **pH** | Benthic | Makoko | 7.14 | pH | Benthic | Makoko | 7.61 |
| **pH** | Benthic | Ikorodu | 6.81 | pH | Benthic | Ikorodu | 7.11 |
| **pH** | Benthic | Epe | 6.65 | pH | Benthic | Epe | 6.45 |
| **pH** | Benthic | Makoko | 6.97 | pH | Benthic | Makoko | 7.42 |
| **pH** | Benthic | Ikorodu | 6.61 | pH | Benthic | Ikorodu | 6.9 |
| **pH** | Benthic | Epe | 6.52 | pH | Benthic | Epe | 6.32 |
| **pH** | Benthic | Makoko | 6.9 | pH | Benthic | Makoko | 7.35 |
| **pH** | Benthic | Ikorodu | 6.73 | pH | Benthic | Ikorodu | 7.02 |
| **pH** | Benthic | Epe | 6.56 | pH | Benthic | Epe | 6.36 |

**Source:** Author's Field Work, 2025

## Appendix III Continued

### Raw pH values of the Surface and Benthic Waters from the Lagos and Epe Lagoons

| August | Water Type | Sample Site | Value | November | Water Type | Sample Site | Value |
|---|---|---|---|---|---|---|---|
| **pH** | Surface | Makoko | 8.58 | pH | Surface | Makoko | 8.69 |
| **pH** | Surface | Ikorodu | 8.16 | pH | Surface | Ikorodu | 8.46 |
| **pH** | Surface | Epe | 7.83 | pH | Surface | Epe | 7.93 |
| **pH** | Surface | Makoko | 8.48 | pH | Surface | Makoko | 8.58 |
| **pH** | Surface | Ikorodu | 8.46 | pH | Surface | Ikorodu | 8.77 |
| **pH** | Surface | Epe | 7.83 | pH | Surface | Epe | 7.93 |
| **pH** | Surface | Makoko | 8.61 | pH | Surface | Makoko | 8.71 |
| **pH** | Surface | Ikorodu | 8.33 | pH | Surface | Ikorodu | 8.63 |
| **pH** | Surface | Epe | 7.94 | pH | Surface | Epe | 8.04 |
| **pH** | Surface | Makoko | 8.76 | pH | Surface | Makoko | 8.86 |
| **pH** | Surface | Ikorodu | 8.12 | pH | Surface | Ikorodu | 8.42 |
| **pH** | Surface | Epe | 7.6 | pH | Surface | Epe | 7.69 |
| **pH** | Surface | Makoko | 8.46 | pH | Surface | Makoko | 8.56 |
| **pH** | Surface | Ikorodu | 8.29 | pH | Surface | Ikorodu | 8.59 |
| **pH** | Surface | Epe | 7.63 | pH | Surface | Epe | 7.72 |
| **pH** | Benthic | Makoko | 7.81 | pH | Benthic | Makoko | 7.81 |
| **pH** | Benthic | Ikorodu | 7.93 | pH | Benthic | Ikorodu | 8.03 |
| **pH** | Benthic | Epe | 6.82 | pH | Benthic | Epe | 7.02 |
| **pH** | Benthic | Makoko | 7.74 | pH | Benthic | Makoko | 7.74 |
| **pH** | Benthic | Ikorodu | 7.67 | pH | Benthic | Ikorodu | 7.76 |
| **pH** | Benthic | Epe | 6.64 | pH | Benthic | Epe | 6.84 |
| **pH** | Benthic | Makoko | 7.95 | pH | Benthic | Makoko | 7.95 |
| **pH** | Benthic | Ikorodu | 7.71 | pH | Benthic | Ikorodu | 7.81 |
| **pH** | Benthic | Epe | 6.85 | pH | Benthic | Epe | 7.05 |
| **pH** | Benthic | Makoko | 7.76 | pH | Benthic | Makoko | 7.76 |
| **pH** | Benthic | Ikorodu | 7.48 | pH | Benthic | Ikorodu | 7.58 |
| **pH** | Benthic | Epe | 6.72 | pH | Benthic | Epe | 6.92 |
| **pH** | Benthic | Makoko | 7.68 | pH | Benthic | Makoko | 7.68 |
| **pH** | Benthic | Ikorodu | 7.62 | pH | Benthic | Ikorodu | 7.72 |
| **pH** | Benthic | Epe | 6.76 | pH | Benthic | Epe | 6.96 |

**Source:** Author's Field Work, 2025

## Appendix IV

## Raw Temperature values of Surface and Benthic Waters Temperature from the Lagos and Epe Lagoons

| | Water | Location | March | May | August | November |
|---|---|---|---|---|---|---|
| **Temperature (Â°C)** | Surface | Makoko | 29.14 | 29.24 | 28.06 | 25.69 |
| **Temperature (Â°C)** | Surface | Ikorodu | 27.41 | 28.1 | 27.41 | 25.37 |
| **Temperature (Â°C)** | Surface | Epe | 26.89 | 27.6 | 27.6 | 25.37 |
| **Temperature (Â°C)** | Surface | Makoko | 30.59 | 30.7 | 29.45 | 26.96 |
| **Temperature (Â°C)** | Surface | Ikorodu | 28.22 | 28.92 | 28.22 | 26.11 |
| **Temperature (Â°C)** | Surface | Epe | 26.59 | 27.29 | 27.29 | 25.09 |
| **Temperature (Â°C)** | Surface | Makoko | 29.49 | 29.59 | 28.39 | 25.99 |
| **Temperature (Â°C)** | Surface | Ikorodu | 27 | 27.67 | 27 | 24.98 |
| **Temperature (Â°C)** | Surface | Epe | 26.44 | 27.14 | 27.14 | 24.94 |
| **Temperature (Â°C)** | Surface | Makoko | 28.88 | 28.97 | 27.8 | 25.45 |
| **Temperature (Â°C)** | Surface | Ikorodu | 27.35 | 28.03 | 27.35 | 25.31 |
| **Temperature (Â°C)** | Surface | Epe | 26.34 | 27.04 | 27.04 | 24.85 |
| **Temperature (Â°C)** | Surface | Makoko | 29.99 | 30.09 | 28.87 | 26.43 |
| **Temperature (Â°C)** | Surface | Ikorodu | 28.21 | 28.91 | 28.21 | 26.1 |
| **Temperature (Â°C)** | Surface | Epe | 25.72 | 26.4 | 26.4 | 24.26 |
| **Temperature (Â°C)** | Benthic | Makoko | 25.53 | 25.23 | 24.44 | 23.06 |
| **Temperature (Â°C)** | Benthic | Ikorodu | 24.46 | 25.56 | 24.76 | 23.35 |
| **Temperature (Â°C)** | Benthic | Epe | 24.65 | 25.36 | 24.45 | 23.43 |
| **Temperature (Â°C)** | Benthic | Makoko | 25.66 | 25.36 | 24.57 | 23.18 |
| **Temperature (Â°C)** | Benthic | Ikorodu | 24.11 | 25.2 | 24.41 | 23.02 |
| **Temperature (Â°C)** | Benthic | Epe | 23.79 | 24.48 | 23.6 | 22.61 |
| **Temperature (Â°C)** | Benthic | Makoko | 26.45 | 26.14 | 25.32 | 23.89 |
| **Temperature (Â°C)** | Benthic | Ikorodu | 23.97 | 25.06 | 24.27 | 22.89 |
| **Temperature (Â°C)** | Benthic | Epe | 24.05 | 24.75 | 23.85 | 22.86 |
| **Temperature (Â°C)** | Benthic | Makoko | 26.08 | 25.78 | 24.97 | 23.56 |
| **Temperature (Â°C)** | Benthic | Ikorodu | 24.6 | 25.71 | 24.9 | 23.48 |
| **Temperature (Â°C)** | Benthic | Epe | 24.36 | 25.06 | 24.16 | 23.15 |
| **Temperature (Â°C)** | Benthic | Makoko | 24.99 | 24.7 | 23.93 | 22.57 |
| **Temperature (Â°C)** | Benthic | Ikorodu | 24.8 | 25.92 | 25.11 | 23.68 |
| **Temperature (Â°C)** | Benthic | Epe | 24.67 | 25.39 | 24.47 | 23.45 |

**Source:** Author's Field Work, 2025

## Appendix V

## Raw Results of Dissolved Oxygen in Surface and Benthic Waters From the Lagos and Epe Lagoons

| | Water | Location | March | May | August | November |
|---|---|---|---|---|---|---|
| **Dissolved Oxygen (mg/L)** | Surface | Makoko | 6.64 | 6.83 | 6.93 | 7.13 |
| **Dissolved Oxygen (mg/L)** | Surface | Ikorodu | 6.57 | 6.47 | 6.57 | 7.19 |
| **Dissolved Oxygen (mg/L)** | Surface | Epe | 5.54 | 5.54 | 6.14 | 6.85 |
| **Dissolved Oxygen (mg/L)** | Surface | Makoko | 6.68 | 6.87 | 6.97 | 7.17 |
| **Dissolved Oxygen (mg/L)** | Surface | Ikorodu | 6.39 | 6.29 | 6.39 | 6.99 |
| **Dissolved Oxygen (mg/L)** | Surface | Epe | 5.67 | 5.67 | 6.29 | 7.01 |
| **Dissolved Oxygen (mg/L)** | Surface | Makoko | 6.55 | 6.75 | 6.85 | 7.04 |
| **Dissolved Oxygen (mg/L)** | Surface | Ikorodu | 6.53 | 6.43 | 6.53 | 7.14 |
| **Dissolved Oxygen (mg/L)** | Surface | Epe | 5.5 | 5.5 | 6.1 | 6.8 |
| **Dissolved Oxygen (mg/L)** | Surface | Makoko | 6.54 | 6.73 | 6.83 | 7.03 |
| **Dissolved Oxygen (mg/L)** | Surface | Ikorodu | 6.45 | 6.35 | 6.45 | 7.05 |
| **Dissolved Oxygen (mg/L)** | Surface | Epe | 5.67 | 5.67 | 6.29 | 7.01 |
| **Dissolved Oxygen (mg/L)** | Surface | Makoko | 6.81 | 7.01 | 7.11 | 7.32 |
| **Dissolved Oxygen (mg/L)** | Surface | Ikorodu | 6.32 | 6.22 | 6.32 | 6.91 |
| **Dissolved Oxygen (mg/L)** | Surface | Epe | 5.21 | 5.21 | 5.78 | 6.44 |
| **Dissolved Oxygen (mg/L)** | Benthic | Makoko | 3.76 | 4.37 | 4.98 | 5.08 |
| **Dissolved Oxygen (mg/L)** | Benthic | Ikorodu | 2.89 | 3.98 | 4.68 | 4.78 |
| **Dissolved Oxygen (mg/L)** | Benthic | Epe | 2.28 | 3.56 | 4.65 | 4.55 |
| **Dissolved Oxygen (mg/L)** | Benthic | Makoko | 3.71 | 4.31 | 4.91 | 5.01 |
| **Dissolved Oxygen (mg/L)** | Benthic | Ikorodu | 2.92 | 4.03 | 4.73 | 4.83 |
| **Dissolved Oxygen (mg/L)** | Benthic | Epe | 2.34 | 3.67 | 4.79 | 4.68 |
| **Dissolved Oxygen (mg/L)** | Benthic | Makoko | 3.68 | 4.27 | 4.87 | 4.97 |
| **Dissolved Oxygen (mg/L)** | Benthic | Ikorodu | 2.99 | 4.12 | 4.84 | 4.94 |
| **Dissolved Oxygen (mg/L)** | Benthic | Epe | 2.32 | 3.62 | 4.73 | 4.63 |
| **Dissolved Oxygen (mg/L)** | Benthic | Makoko | 3.71 | 4.31 | 4.91 | 5.01 |
| **Dissolved Oxygen (mg/L)** | Benthic | Ikorodu | 2.87 | 3.96 | 4.65 | 4.75 |
| **Dissolved Oxygen (mg/L)** | Benthic | Epe | 2.28 | 3.56 | 4.65 | 4.55 |
| **Dissolved Oxygen (mg/L)** | Benthic | Makoko | 3.55 | 4.13 | 4.71 | 4.8 |
| **Dissolved Oxygen (mg/L)** | Benthic | Ikorodu | 2.85 | 3.94 | 4.62 | 4.72 |
| **Dissolved Oxygen (mg/L)** | Benthic | Epe | 2.32 | 3.64 | 4.75 | 4.65 |

**Source:** Author's Field Work, 2025

## Appendix VI

## Raw Results of Total Dissolved Solid (mg/L) in Surface and Benthic Waters from the Lagos and Epe Lagoons

| | Water | Location | March | May | August | November |
|---|---|---|---|---|---|---|
| **Total Dissolved Solids (mg/L)** | Surface | Makoko | 651.26 | 650.26 | 671.3 | 680.32 |
| **Total Dissolved Solids (mg/L)** | Surface | Ikorodu | 604.76 | 625.15 | 639.71 | 641.65 |
| **Total Dissolved Solids (mg/L)** | Surface | Epe | 580.1 | 587.87 | 631.6 | 635.49 |
| **Total Dissolved Solids (mg/L)** | Surface | Makoko | 662.59 | 661.57 | 682.98 | 692.15 |
| **Total Dissolved Solids (mg/L)** | Surface | Ikorodu | 626.69 | 647.81 | 662.9 | 664.91 |
| **Total Dissolved Solids (mg/L)** | Surface | Epe | 591.98 | 599.91 | 644.53 | 648.5 |
| **Total Dissolved Solids (mg/L)** | Surface | Makoko | 640.87 | 639.89 | 660.59 | 669.47 |
| **Total Dissolved Solids (mg/L)** | Surface | Ikorodu | 626.25 | 647.36 | 662.44 | 664.45 |
| **Total Dissolved Solids (mg/L)** | Surface | Epe | 592.91 | 600.85 | 645.54 | 649.52 |
| **Total Dissolved Solids (mg/L)** | Surface | Makoko | 645.74 | 644.75 | 665.61 | 674.55 |
| **Total Dissolved Solids (mg/L)** | Surface | Ikorodu | 623.06 | 644.07 | 659.07 | 661.07 |
| **Total Dissolved Solids (mg/L)** | Surface | Epe | 587.42 | 595.29 | 639.57 | 643.51 |
| **Total Dissolved Solids (mg/L)** | Surface | Makoko | 644.9 | 643.91 | 664.75 | 673.68 |
| **Total Dissolved Solids (mg/L)** | Surface | Ikorodu | 620.08 | 640.98 | 655.91 | 657.9 |
| **Total Dissolved Solids (mg/L)** | Surface | Epe | 595.07 | 603.05 | 647.9 | 651.89 |
| **Total Dissolved Solids (mg/L)** | Benthic | Makoko | 504.04 | 603.84 | 604.85 | 615.94 |
| **Total Dissolved Solids (mg/L)** | Benthic | Ikorodu | 471.2 | 535.62 | 563.51 | 579.86 |
| **Total Dissolved Solids (mg/L)** | Benthic | Epe | 491.95 | 477.86 | 503.02 | 566.4 |
| **Total Dissolved Solids (mg/L)** | Benthic | Makoko | 518.86 | 621.6 | 622.63 | 634.05 |
| **Total Dissolved Solids (mg/L)** | Benthic | Ikorodu | 489.74 | 556.7 | 585.69 | 602.68 |
| **Total Dissolved Solids (mg/L)** | Benthic | Epe | 488.66 | 474.67 | 499.65 | 562.61 |
| **Total Dissolved Solids (mg/L)** | Benthic | Makoko | 501.75 | 601.09 | 602.09 | 613.13 |
| **Total Dissolved Solids (mg/L)** | Benthic | Ikorodu | 490.59 | 557.67 | 586.71 | 603.73 |
| **Total Dissolved Solids (mg/L)** | Benthic | Epe | 477.57 | 463.9 | 488.31 | 549.84 |
| **Total Dissolved Solids (mg/L)** | Benthic | Makoko | 502.58 | 602.09 | 603.09 | 614.15 |
| **Total Dissolved Solids (mg/L)** | Benthic | Ikorodu | 514.14 | 584.44 | 614.87 | 632.71 |
| **Total Dissolved Solids (mg/L)** | Benthic | Epe | 500.18 | 485.86 | 511.43 | 575.87 |
| **Total Dissolved Solids (mg/L)** | Benthic | Makoko | 499.26 | 598.11 | 599.11 | 610.09 |
| **Total Dissolved Solids (mg/L)** | Benthic | Ikorodu | 488.11 | 554.86 | 583.75 | 600.68 |
| **Total Dissolved Solids (mg/L)** | Benthic | Epe | 496.35 | 482.14 | 507.52 | 571.47 |

**Source:** Author's Field Work, 2025

## Appendix VII

## Raw Results of Electrical Conductivity in Surface and Benthic Waters of the Lagos and Epe Lagoons

| | Water | Location | March | May | August | November |
|---|---|---|---|---|---|---|
| **Electrical Conductivity (ÂµS/cm)** | Surface | Makoko | 1320.57 | 1087.94 | 1218.98 | 1269.78 |
| **Electrical Conductivity (ÂµS/cm)** | Surface | Ikorodu | 1304.76 | 996.84 | 1158.63 | 1242.13 |
| **Electrical Conductivity (ÂµS/cm)** | Surface | Epe | 930.23 | 877.9 | 968.99 | 1075.58 |
| **Electrical Conductivity (ÂµS/cm)** | Surface | Makoko | 1276.36 | 1051.52 | 1178.17 | 1227.27 |
| **Electrical Conductivity (ÂµS/cm)** | Surface | Ikorodu | 1225.24 | 936.08 | 1088.01 | 1166.43 |
| **Electrical Conductivity (ÂµS/cm)** | Surface | Epe | 961.32 | 907.24 | 1001.37 | 1111.52 |
| **Electrical Conductivity (ÂµS/cm)** | Surface | Makoko | 1336.47 | 1101.05 | 1233.67 | 1285.07 |
| **Electrical Conductivity (ÂµS/cm)** | Surface | Ikorodu | 1235.84 | 944.18 | 1097.43 | 1176.52 |
| **Electrical Conductivity (ÂµS/cm)** | Surface | Epe | 939.6 | 886.75 | 978.75 | 1086.42 |
| **Electrical Conductivity (ÂµS/cm)** | Surface | Makoko | 1263.55 | 1040.97 | 1166.36 | 1214.95 |
| **Electrical Conductivity (ÂµS/cm)** | Surface | Ikorodu | 1252.49 | 956.9 | 1112.21 | 1192.37 |
| **Electrical Conductivity (ÂµS/cm)** | Surface | Epe | 969.09 | 914.58 | 1009.47 | 1120.51 |
| **Electrical Conductivity (ÂµS/cm)** | Surface | Makoko | 1315.26 | 1083.57 | 1214.08 | 1264.67 |
| **Electrical Conductivity (ÂµS/cm)** | Surface | Ikorodu | 1237.41 | 945.38 | 1098.82 | 1178.02 |
| **Electrical Conductivity (ÂµS/cm)** | Surface | Epe | 942.35 | 889.34 | 981.61 | 1089.59 |
| **Electrical Conductivity (ÂµS/cm)** | Benthic | Makoko | 1020.69 | 1144.41 | 1225.86 | 1237.2 |
| **Electrical Conductivity (ÂµS/cm)** | Benthic | Ikorodu | 964.37 | 1013.59 | 1034.69 | 1135.14 |
| **Electrical Conductivity (ÂµS/cm)** | Benthic | Epe | 934.39 | 872.43 | 920.17 | 975.01 |
| **Electrical Conductivity (ÂµS/cm)** | Benthic | Makoko | 974.49 | 1092.61 | 1170.37 | 1181.2 |
| **Electrical Conductivity (ÂµS/cm)** | Benthic | Ikorodu | 985.1 | 1035.38 | 1056.93 | 1159.54 |
| **Electrical Conductivity (ÂµS/cm)** | Benthic | Epe | 897.24 | 837.75 | 883.59 | 936.25 |
| **Electrical Conductivity (ÂµS/cm)** | Benthic | Makoko | 983.62 | 1102.85 | 1181.34 | 1192.27 |
| **Electrical Conductivity (ÂµS/cm)** | Benthic | Ikorodu | 929.14 | 976.56 | 996.89 | 1093.67 |
| **Electrical Conductivity (ÂµS/cm)** | Benthic | Epe | 895.7 | 836.31 | 882.07 | 934.65 |
| **Electrical Conductivity (ÂµS/cm)** | Benthic | Makoko | 1006.11 | 1128.06 | 1208.35 | 1219.52 |
| **Electrical Conductivity (ÂµS/cm)** | Benthic | Ikorodu | 963.54 | 1012.73 | 1033.8 | 1134.17 |
| **Electrical Conductivity (ÂµS/cm)** | Benthic | Epe | 929.6 | 867.97 | 915.46 | 970.02 |
| **Electrical Conductivity (ÂµS/cm)** | Benthic | Makoko | 965.63 | 1082.67 | 1159.73 | 1170.46 |
| **Electrical Conductivity (ÂµS/cm)** | Benthic | Ikorodu | 964.99 | 1014.24 | 1035.35 | 1135.87 |
| **Electrical Conductivity (ÂµS/cm)** | Benthic | Epe | 925.46 | 864.1 | 911.38 | 965.7 |

**Source:** Author's Field Work, 2025

## Appendix VIII

## Seasonal Variation of Lead Concentration (mg/L) in Surface and Benthic Waters of the Lagos and Epe Lagoons

| Location | Depth | March (mg/L) | May (mg/L) | August (mg/L) | November (mg/L) |
|---|---|---|---|---|---|
| **Makoko** | Surface | $2.01 \pm 0.11^{c}$ | $2.08 \pm 0.13^{c}$ | $3.11 \pm 0.12^{b}$ | $3.56 \pm 0.22^{a}$ |
| | Benthic | $5.42 \pm 0.13^{c}$ | $5.31 \pm 0.13^{c}$ | $5.92 \pm 0.06^{b}$ | $6.77 \pm 0.11^{a}$ |
| | Sig. (t-test) | <0.01 | <0.01 | <0.01 | <0.01 |
| **Ikorodu** | Surface | $1.64 \pm 0.13^{b}$ | $1.54 \pm 0.13^{b}$ | $1.68 \pm 0.16^{ab}$ | $1.88 \pm 0.20^{a}$ |
| | Benthic | $5.31 \pm 0.04^{c}$ | $5.11 \pm 0.11^{d}$ | $5.66 \pm 0.13^{b}$ | $6.54 \pm 0.10^{a}$ |
| | Sig. (t-test) | <0.01 | <0.01 | <0.01 | <0.01 |
| **Epe** | Surface | $1.47 \pm 0.10^{c}$ | $1.41 \pm 0.06^{c}$ | $1.58 \pm 0.06^{b}$ | $1.75 \pm 0.11^{a}$ |
| | Benthic | $4.86 \pm 0.08^{b}$ | $4.22 \pm 0.12^{d}$ | $4.67 \pm 0.11^{c}$ | $6.32 \pm 0.06^{a}$ |
| | Sig. (t-test) | <0.01 | <0.01 | <0.01 | <0.01 |
| **WHO limit** | | | | | **0.01 mg/L** |

**Source:** Author's Field Work, 2024

## Appendix IX

## Seasonal Variation of Copper Concentration (mg/L) in Surface and Benthic From the Lagos and Epe Lagoons

| Location | Depth | March (mg/L) | May (mg/L) | August (mg/L) | November (mg/L) |
|---|---|---|---|---|---|
| **Makoko** | Surface | $2.71 \pm 0.22^{c}$ | $2.68 \pm 0.03^{c}$ | $2.87 \pm 0.02^{b}$ | $3.11 \pm 0.03^{a}$ |
| | Benthic | $8.10 \pm 0.22^{c}$ | $8.12 \pm 0.23^{c}$ | $9.10 \pm 0.21^{b}$ | $11.56 \pm 0.23^{a}$ |
| | Sig. (t-test) | <0.01 | <0.01 | <0.01 | <0.01 |
| **Ikorodu** | Surface | $0.00 \pm 0.00^{c}$ | $2.18 \pm 0.19^{b}$ | $2.23 \pm 0.15^{a}$ | $2.95 \pm 0.13^{a}$ |
| | Benthic | $7.49 \pm 0.17^{c}$ | $7.32 \pm 0.21^{c}$ | $8.66 \pm 0.06^{b}$ | $10.54 \pm 0.11^{a}$ |
| | Sig. (t-test) | <0.01 | <0.01 | <0.01 | <0.01 |
| **Epe** | Surface | $1.96 \pm 0.13^{b}$ | $1.70 \pm 0.17^{c}$ | $1.98 \pm 0.18^{c}$ | $2.48 \pm 0.17^{a}$ |
| | Benthic | $6.94 \pm 0.11^{c}$ | $5.10 \pm 0.11^{d}$ | $7.19 \pm 0.04^{b}$ | $9.34 \pm 0.08^{a}$ |
| | Sig. (t-test) | <0.01 | <0.01 | <0.01 | <0.01 |
| **WHO limit** | | | | | **0.05 mg/L** |

**Source:** Author's Field Work, 2025

## Appendix X

## Seasonal Variation of Nickel Concentration (mg/L) in Surface and Benthic Waters From the Lagos and Epe Lagoons

| Location | Depth | March (mg/L) | May (mg/L) | August (mg/L) | November (mg/L) |
|---|---|---|---|---|---|
| **Makoko** | Surface | $0.89 \pm 0.04^{ab}$ | $0.76 \pm 0.02^{b}$ | $0.91 \pm 0.23^{ab}$ | $1.10 \pm 0.34^{a}$ |
| | Benthic | $12.55 \pm 0.29^{c}$ | $12.64 \pm 0.36^{c}$ | $13.98 \pm 0.38^{b}$ | $14.66 \pm 0.49^{a}$ |
| | Sig. (t-test) | <0.01 | <0.01 | <0.01 | <0.01 |
| **Ikorodu** | Surface | $0.65 \pm 0.03^{b}$ | $0.64 \pm 0.06^{b}$ | $0.86 \pm 0.11^{a}$ | $1.02 \pm 0.23^{a}$ |
| | Benthic | $12.13 \pm 0.34^{c}$ | $11.10 \pm 0.32^{d}$ | $12.77 \pm 0.28^{b}$ | $13.77 \pm 0.22^{a}$ |
| | Sig. (t-test) | <0.01 | <0.01 | <0.01 | <0.01 |
| **Epe** | Surface | $0.57 \pm 0.06^{c}$ | $0.48 \pm 0.06^{bc}$ | $0.60 \pm 0.06^{b}$ | $0.98 \pm 0.11^{a}$ |
| | Benthic | $10.17 \pm 0.21^{c}$ | $9.77 \pm 0.18^{d}$ | $10.94 \pm 0.19^{b}$ | $12.10 \pm 0.06^{a}$ |
| | Sig. (t-test) | <0.01 | <0.01 | <0.01 | <0.01 |
| **WHO limit** | | | | | **0.07 mg/L** |

**Source:** Author's Field Work, 2025

## Appendix XI

## Seasonal variation of cadmium concentration (mg/L) in Surface and Benthic Waters From the Lagos and Epe Lagoons

| Location | Depth | March (mg/L) | May (mg/L) | August (mg/L) | November (mg/L) |
|---|---|---|---|---|---|
| **Makoko** | Surface | $0.09 \pm 0.01^{b}$ | $0.09 \pm 0.00^{b}$ | $0.09 \pm 0.00^{b}$ | $0.12 \pm 0.03^{a}$ |
| | Benthic | $0.94 \pm 0.04^{b}$ | $0.87 \pm 0.03^{c}$ | $0.94 \pm 0.01^{b}$ | $1.33 \pm 0.00^{a}$ |
| | Sig. (t-test) | <0.01 | <0.01 | <0.01 | <0.01 |
| **Ikorodu** | Surface | $0.07 \pm 0.01^{a}$ | $0.06 \pm 0.01^{a}$ | $0.09 \pm 0.03^{a}$ | $0.10 \pm 0.06^{a}$ |
| | Benthic | $0.91 \pm 0.03^{b}$ | $0.88 \pm 0.02^{b}$ | $0.89 \pm 0.03^{b}$ | $1.09 \pm 0.06^{a}$ |
| | Sig. (t-test) | <0.01 | <0.01 | <0.01 | <0.01 |
| **Epe** | Surface | $0.04 \pm 0.01^{c}$ | $0.03 \pm 0.01^{bc}$ | $0.06 \pm 0.01^{b}$ | $0.10 \pm 0.03^{a}$ |
| | Benthic | $0.07 \pm 0.06^{d}$ | $0.67 \pm 0.04^{c}$ | $0.77 \pm 0.06^{b}$ | $0.96 \pm 0.06^{a}$ |
| | Sig. (t-test) | 0.31 | <0.01 | <0.01 | <0.01 |
| **WHO limit** | | | | | **0.003 mg/L** |

**Source:** Author's Field Work, 2025

## Appendix XII

## Seasonal Variation of Chromium Concentration (mg/L) in Surface and Benthic Waters From the Lagos and Epe Lagoons

| Location | Depth | March (mg/L) | May (mg/L) | August (mg/L) | November (mg/L) |
|---|---|---|---|---|---|
| **Makoko** | Surface | $0.00 \pm 0.00^{b}$ | $0.09 \pm 0.00^{a}$ | $0.09 \pm 0.00^{a}$ | $0.12 \pm 0.07^{a}$ |
| | Benthic | $3.47 \pm 0.02^{c}$ | $3.40 \pm 0.01^{d}$ | $4.11 \pm 0.01^{b}$ | $5.62 \pm 0.01^{a}$ |
| | Sig. (t-test) | <0.01 | <0.01 | <0.01 | <0.01 |
| **Ikorodu** | Surface | $0.09 \pm 0.06^{a}$ | $0.07 \pm 0.04^{a}$ | $0.09 \pm 0.07^{a}$ | $0.10 \pm 0.01^{a}$ |
| | Benthic | $3.30 \pm 0.01^{c}$ | $3.29 \pm 0.00^{c}$ | $3.99 \pm 0.00^{b}$ | $5.18 \pm 0.01^{a}$ |
| | Sig. (t-test) | <0.01 | <0.01 | <0.01 | <0.01 |
| **Epe** | Surface | $0.08 \pm 0.01^{b}$ | $0.06 \pm 0.01^{ab}$ | $0.08 \pm 0.01^{ab}$ | $0.10 \pm 0.04^{a}$ |
| | Benthic | $3.22 \pm 0.06^{c}$ | $3.10 \pm 0.06^{d}$ | $3.79 \pm 0.04^{b}$ | $4.91 \pm 0.04^{a}$ |
| | Sig. (t-test) | <0.01 | <0.01 | <0.01 | <0.01 |
| **WHO limit** | | | | | **0.03 mg/L** |

**Source:** Author's Field Work, 2024

## Bio Data

### A. Personal Data

1. **Full Name:** Nkechinyere Veronica Otu-Ekuma
2. **Home Address:** 28, Farm Road 2, Eliozu, Port Harcourt, Rivers State
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5. **Date and Place of Birth:** 11th November 1992, Navy Town, Ojo, Lagos State
6. **Nationality:** Nigerian
7. **Name and Adddress of Next of Kin:** Ugochi Otu-Ekuma,
   14, Ugbeji-Aki Street, Mende, Maryland, Lagos

### B.Educational Background

| Institutions Attended | Qualifications Obtained | Date |
|---|---|---|
| 1. University of Abuja | BSc, Biological Science | 2017 |
| 2. University of Lagos | MSc, Cell Biology and Genetics (Environmental Biology) | 2021 |
| 3. Lead City University | PhD, Environmental Biology in view | 2025 |

### C. Work Experience with Dates

1. Intern,
FCT-Agricultural Development Projects, FCT, Abuja — May 2015- Dec 2015
2. Environmental Health Officer,
Ministry of Health, Lokoja, Kogi State — May 2017- April 2018
3. Special Assistant,
Ebonyi State Government House, Abakaliki,
Ebonyi State — July 2019- March 2020
4. Environmental Remediation Specialist
Farmside Garden, Lagos — April 2020- Jan 2023
5. Environmental Specialist,
Green Nest Eco-consult, Lagos State — 2023 till date

**D. Awards and Fellowships** (If Any): Nil

**E. Membership of Academic Professional Bodies:**

1. Organisation for Women in Science for the Developing World(OWSD)
2. Society of Environmental Toxicology and Chemistry(SETAC)
3. Society for Conservation Biology, Nigeria
4. Member, Policy Team, Nigeria National Plastic Action Partnership(NPAP)

**F. Publications:**

1. Otu-Ekuma Nkechinyere Veronica, *Assessment of Plant Species and Physicochemical Properties of Three Landfills in Lagos State*, FASCON International Conference 2022.

**G. Major Conferences/Workshops Attended with Dates:**

1. Circular Economy Innovation Partnership(CEIP) - Circular Economy Strategy Week. 25th - 28th March 2024
2. Third International Conference of the Faculty of Natural and Applied Sciences (FASCON 2022) on "Translational Research in Science and Technology for Sustainable Development Circa Covid-19 Era" held at Lead City University, Ibadan, Oyo State, Nigeria. 2nd - 4th November 2022.
3. West African Society of Toxicology(WASOT), 3rd International Conference; Global Understanding of Chemicals in Health, Diseases and Economics II. 19th-22nd February 2014. University of Lagos.

**H. References:**

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**The University Compliance Certification**

This is to certify that, this Thesis written by **Nkechinyere Veronica, Otu-Ekuma,** with Matric No**. LCU/PG/003012,** in the Department of Biological Science, Faculty of Natural and Applied Sciences, Lead City University, Ibadan is in full compliance with the approved University format and style.



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**Signature** **Date**