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PLANKTON SPECIES AS BIO-INDICATORS OF WATER QUALITY IN TREATED WASTEWATER FOR THE PURPOSE OF AQUACULTURE PRODUCTION.

BY

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DECLARATION

This dissertation is a result of research work undertaken by Ebenezer Nii Martey Botchway in the Department of Marine and Fisheries Sciences, University of Ghana under the supervision of Prof. Samuel Addo. I Ebenezer Nii Martey Botchway, do hereby declare that except for cited references that have been duly acknowledged, this work is the result of my own original research done under the supervision of Prof. Samuel Addo, for the partial fulfillment of the requirements for the award of Master of Science degree in Aquaculture from University of Ghana and that no part of it has been presented elsewhere for another degree or diploma.

5TH November, 2024

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(Supervisor)

DEDICATION

This work is dedicated to my entire family and to all who helped me during the period of my study especially the Principal Supervisor Prof. Samuel Addo and Mr. Emmanuel Klubi, a Principal Research Assistant of the Department of Marine and Fisheries Science, for their immeasurable support and guidance during my research period.

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ABBREVIATIONS

ARI	Animal Research Institute
CSIR	Council for Scientific and Industrial Research
DMFS	Department of Marine and Fisheries Sciences
MTP	Mudor Treatment Plant
TWW	Treated Wastewater
UG	University of Ghana
WRI	Water Research Institute
WWTP	Wastewater Treatment Plant
DO	Dissolve Oxygen
Temp	Temperature
EPA	Environmental Protection Agency
BOD	Biochemical Oxygen Demand
COD	Chemical Oxygen Demand

ABSTRACT

Effluents from the Mudor Treatment Plant (MTP), stored in ponds at the Water Research Institute (WRI) for irrigation by nearby vegetable farmers, were assessed to determine their potential for supporting aquaculture production. Physico-chemical parameters including temperature, pH, and dissolved oxygen (DO) were monitored both in-situ and ex-situ over a 24-hour period for two consecutive months, September and October 2023. Sampling intervals for the 24-hour monitoring were set at 6 am, 9 am, 12 noon, 5 pm, 8 pm, 12 midnight, 2 am, 4 am, and 6 am for the September period, and 6 am, 9 am, 12 noon, 5 pm, 8 pm, 12 midnight, 3 am, and 6 am for October. Temperature values ranged from 25.70 ±0.10°C to 28.23 ±0.31°C in September and 25.06 ±0.26°C to 27.37 ±0.14°C in October. pH levels fluctuated between 7.75 ±0.01 to 10.63 ±0.05 in September and 8.05 ±0.29 to 9.79 ±0.32 in October. Dissolved oxygen concentrations varied from 1.21 ±0.19 mg/L to 9.89 ±2.00 mg/L in September and 0.32 ±0.21 mg/L to 6.40 ±0.92 mg/L in October. Nutrient concentrations, measured in September at 12 noon and 3 am, indicated ammonia levels between 0.50 mg/L and 2.84 mg/L (noon) and 0.56 mg/L to 2.82 mg/L (3 am), while nitrite concentrations ranged from 0.002 mg/L to 0.216 mg/L (noon) and 0.002 mg/L to 0.221 mg/L (3 am). Plankton composition, identified in November, included 16 species; eight phytoplankton species and eight zooplankton species—across the four study ponds. Phytoplankton were represented by four classes: *Cyanobacteria* (37.5%), *Chlorophyceae* (37.5%), *Rotifera* (12.5%), and *Euglenoidea* (12.5%). The zooplankton population, however, consisted primarily of insect larvae, such as *chironomids*, *stoneflies*, *dragonflies*, *Arachnida*, *Dystacta* spp., *ostracods*, and *polychaetes*, with a lack of true zooplankton like copepods or calanoids. Statistical analysis, including Analysis of Variance (ANOVA), was conducted to investigate variations in physicochemical parameters across the ponds. Results showed significant differences in mean temperature ($p < 0.05$) for September ($p=0.012$) and- no significant differences for the month of October ($p= 0.364$). For Ph a significant difference ($p < 0.05$) was obtained for September ($p= 0.002$) while no significant differences ($p < 0.05$) were observed in October ($p= 0.905$). DO showed a significant difference ($p < 0.05$) for September ($p= 0.00043$) and in contrast no significant differences ($p < 0.05$) obtained in October ($p= 0.551$). For ammonia and Nitrite, both showed no significant differences ($p < 0.05$) with P values; ($p=0.960$) and ($p=0.059$) respectively. Diversity indices, including species richness (Margalef), species evenness (Pielou), and the Shannon-Weiner diversity index (H'), were used to assess plankton diversity. Margalef's species richness suggested higher species composition in pond 2, followed by pond 1, pond 4, and pond 3. Pielou's evenness and Shannon-Weiner index also indicated higher diversity in pond 3, followed by pond 2, pond 1, and pond 4. Overall, the results indicated that these ponds' physico-chemical parameters and nutrient levels were not optimal for aquaculture production. Specifically, the presence of elevated nutrient levels and the predominance of insect larvae over zooplankton species suggest that the ponds may require further intervention to improve water quality. This study underscores the need for further treatment of treated wastewater from the Mudor Treatment Plant to reduce nutrient loads and qualify it for aquaculture production.

CHAPTER ONE

INTRODUCTION

1.1 Background of the Study

Aquaculture has emerged as one of the fastest-growing food production sectors globally, which is vital in enhancing food security, generating employment, and reducing pressure on wild fish stocks (FAO, 2022). However, the sustainability of aquaculture is highly dependent on water availability and quality according to (Boyd, 2019). Freshwater resources, which are increasingly scarce due to urbanization, industrialization, and agriculture, have led to the exploration of alternative aquaculture water sources, including treated wastewaters (Tom *et al.*,2021). While treated wastewater holds potential as an alternative water source for aquaculture, its suitability needs to be rigorously evaluated to ensure that it does not pose risks to fish health or human consumers.

The use of treated wastewater in aquaculture requires an in-depth understanding of the water quality parameters and biological communities, such as plankton, that could serve as bioindicators of environmental health and water safety. Water quality is a critical determinant of aquaculture productivity, influencing both the health of aquatic organisms and the overall ecosystem balance (Boyd, 2019). Poor water quality can lead to stress in fish, reduced growth rates, and increased disease susceptibility, making it essential to ensure that treated wastewater meets the necessary standards for aquaculture. Plankton communities, comprising both phytoplankton and zooplankton, play a crucial role in aquatic ecosystems by serving as primary producers and food sources for fish. Plankton diversity and abundance are often used as indicators of water quality, as they respond rapidly to changes in environmental conditions (Díez-Montero *et al.*, 2020).

In recent years, treated wastewater has gained attention as a potential resource for aquaculture, especially in regions facing freshwater shortages (Zaibel *et al*., 2022). Studies have shown that when properly treated, wastewater can meet the water quality requirements for aquaculture. However, the quality of treated wastewater can vary widely depending on the treatment processes employed and the sources of the wastewater. Parameters such as dissolved oxygen, pH, ammonia, nitrites, and heavy metals need to be monitored closely to ensure they fall within acceptable ranges for fish production (Adino,2024). Moreover, the presence of harmful microorganisms, residual pharmaceuticals, and other contaminants in treated wastewater can compromise its suitability for aquaculture, necessitating continuous monitoring and rigorous testing.

Despite these challenges, some countries have begun to explore the feasibility of using treated wastewater in aquaculture. For instance, in Israel and the United States, treated wastewater has been used in limited aquaculture operations under controlled conditions (Zaibel *et al*., 2022). However, much of the research has focused on the physicochemical aspects of water quality, with relatively few studies examining the role of biological indicators, such as plankton communities, in assessing the suitability of treated wastewater for aquaculture.

1.2 Problem Statement

The growing global demand for water resources has placed immense pressure on freshwater availability, particularly in industrialized regions or urbanized areas such as Accra, Ghana. In response, there is an increasing interest in the reuse of treated wastewater as an alternative water source for various sectors, including aquaculture. However, despite its potential, there are significant concerns regarding the suitability of treated wastewater for aquaculture production due to the variability in water quality and the presence of residual contaminants. Most existing studies have primarily focused on the physicochemical characteristics of treated wastewater, but there is a need for comprehensive assessments that also consider biological indicators, particularly plankton communities, to ensure the long-term sustainability of aquaculture systems.

The plankton community plays an integral role in the aquatic food web, and its composition and abundance are highly sensitive to changes in water quality (Reynolds, 2006; Moss, 2008). As such, plankton can serve as valuable bioindicators for evaluating the health of aquatic ecosystems and determining whether treated wastewater is suitable for fish farming (Van Hulten and Van der Velde, 2009; Barbier and Grandjean, 2014). However, research on the use of plankton communities as indicators for wastewater suitability in aquaculture remains limited. This gap in knowledge highlights the need for further investigation into the relationship between water quality, plankton diversity, and aquaculture productivity when using treated wastewater.

1.3 Main objective

The primary objective of this thesis is to evaluate the water quality and plankton community structure of treated wastewater from the Mudor Treatment Plant, Accra, Ghana to determine its suitability for tilapia production. By assessing both physico-chemical parameters and biological indicators, this study could provide a more comprehensive understanding of how treated wastewater could be used in tilapia culture.

The specific objectives were to assess:

1. Water quality in the maturation ponds and its suitability for aquaculture.
2. Plankton diversity and abundance in the ponds as bioindicators of water quality.

1.4 Research Questions

1. What are the key water quality parameters of treated wastewater from the Mudor Treatment Plant that influence aquaculture production, and how do they compare with the requirements for optimal aquaculture?
2. What is the plankton community structure, including species diversity and abundance in this treated wastewater?
3. Can plankton communities be used as reliable bioindicators to assess the suitability of treated wastewater for sustainable aquaculture?

1.5 Significance of the Study

This study seeks to contribute to the growing wealth of knowledge on the use of alternative water sources for aquaculture by providing insights into both the physico-chemical and biological factors that determine the suitability of treated wastewater for fish production. By focusing on the plankton community as a bioindicator, this research will offer a novel approach to water quality assessment in aquaculture, which could be applied in regions facing freshwater scarcity. Moreover, the findings from this study could help inform policy decisions regarding the safe and sustainable use of treated wastewater in aquaculture, ultimately contributing to more resilient food production systems.

CHAPTER TWO

LITERATURE REVIEW

2.1 Global Waste Water Treatment and General Usage

Wastewater treatment is essential for environmental protection and public health, ensuring the removal of pollutants from domestic, industrial, and agricultural wastewater before it is released back into the environment or reused. With urbanization and water scarcity on the rise, efficient wastewater treatment and water reuse are central to achieving sustainable development goals (United Nations, 2020). However, globally only about 52% of wastewater is treated, with significant regional disparities, high-income countries treat 74%, while low-income countries treat only 8% (UNESCO, 2020). Untreated wastewater is often discharged into natural water bodies in many developing regions, causing environmental damage and waterborne diseases (Mateo-Sagasta *et al*., 2015).

Conventional wastewater treatment includes primary, secondary, and tertiary stages. Primary treatment removes solids, secondary treatment uses biological processes to degrade organic pollutants and tertiary treatment removes nutrients and pathogens (Tchobanoglous *et al*., 2014; Spellman, 2017). Advanced technologies, such as membrane bioreactors (MBRs) and Moving Bed Biofilm Reactors (MBBRs), are being adopted for more efficient treatment (Judd, 2011; Ødegaard, 2006). However, these technologies are often costly and energy-intensive, limiting their use in low-income countries (Kümmerer *et al*., 2018).

Treated wastewater is increasingly seen as a resource for agricultural, industrial, and even potable reuse (Asano *et al*., 2007), with countries like Israel and Singapore leading large-scale reuse programs (Friedler & Hadari, 2006). Wastewater treatment plants are also evolving to recover resources like biogas and nutrients (Guest *et al*., 2009).

Despite technological advances, challenges persist, including financial constraints, insufficient infrastructure, and public resistance to water reuse, particularly for drinking water (Po *et al*., 2003; WHO/UNICEF, 2021). Future strategies should focus on decentralized treatment systems, integrated water resource management, and incentivizing water reuse through policies and smart technologies like real-time monitoring (Rizzo *et al*., 2019).

2.2 Wastewater as a Source of Water for Aquaculture Production

Wastewater used in aquaculture is generally categorized into two types: treated and untreated. Treated wastewater has undergone various purification processes to remove contaminants, making it safer for use in aquaculture, while untreated wastewater comes from municipal, industrial, or agricultural runoff and may contain a range of pollutants (Liu *et al*., 2020). While treated wastewater has been successfully used in aquaculture systems without harming species like tilapia and shrimp (Guitart *et al*., 2015; Cui et al., 2019), untreated wastewater often contains high levels of nutrients and organic matter, which can complicate water quality management (Li *et al*., 2019). One significant advantage of using wastewater is nutrient recycling. Wastewater is rich in nitrogen, phosphorus, and organic carbon, which are essential for the growth of aquatic organisms, reducing the need for external inputs like fertilizers and feed, leading to cost savings in aquaculture operations (Liu *et al*., 2020; He *et al*., 2021). Wastewater also supports aquatic plants like algae that help maintain water quality by absorbing excess nutrients (Bergheim *et al*., 2018). Furthermore, it can provide organic matter beneficial to species that thrive in nutrient-rich environments, such as bivalves and crustaceans (Shen *et al*., 2021). Studies have shown that the use of treated wastewater in closed-loop aquaculture systems promotes water conservation and enhances system productivity (Mbuya *et al*., 2016).

Despite these benefits, the use of wastewater in aquaculture raises concerns regarding water quality and the health of aquaculture species. Untreated wastewater may contain harmful substances such as heavy metals, pathogens, and chemicals that can adversely affect aquatic organisms (Guitart *et al*., 2015). Even treated wastewater may still contain trace contaminants, including pharmaceuticals and endocrine-disrupting chemicals, posing potential risks to aquatic life (Duan *et al*., 2019). Research has shown that wastewater can lead to fluctuations in ammonia, nitrite, and dissolved oxygen levels, which can stress fish and increase the likelihood of disease outbreaks (Cui *et al*., 2019). Additionally, the bioaccumulation of toxic substances in aquaculture species is a concern, as pollutants may accumulate in edible tissues, posing health risks to consumers (Li *et al*., 2019).

To address these risks, various treatment and management strategies have been developed. These include advanced filtration systems, biological treatments, and the use of constructed wetlands to further purify wastewater before introducing it into aquaculture systems (Mbuya *et al*., 2016). Pretreatment methods, such as sedimentation, chlorination, or adsorption, have also been found to improve water quality and reduce pathogen loads (He *et al*., 2021). Regular monitoring of water quality parameters is essential, with automated water quality monitoring systems and real-time data analytics enabling producers to adjust management strategies to maintain a safe environment for aquaculture species (Shen *et al*., 2021).

The use of wastewater in aquaculture offers notable environmental and economic benefits. It helps reduce the environmental footprint of aquaculture by minimizing freshwater use, which is particularly important in regions facing water scarcity (He *et al*., 2021). Additionally, using wastewater reduces operational costs, making aquaculture more economically viable, especially for small-scale farmers or in areas with limited access to clean water (Bergheim *et al*., 2018).

Furthermore, wastewater treatment and reuse contribute to sustainable development goals by promoting circular economy practices, reducing waste, lowering pollution, and conserving water resources (Mbuya *et al*., 2016).

2.3 Wastewater Treatment and its Utilization in Aquaculture Production

Wastewater, depending on its source, may contain pollutants such as organic matter, heavy metals, nutrients, and pathogens, which can negatively impact the health of aquatic organisms and water quality in aquaculture systems. To safely reuse wastewater, it must undergo various treatment processes, including physical, chemical, and biological treatments. Physical treatments like sedimentation and filtration remove suspended solids, while chemical treatments such as chlorination and coagulation target dissolved contaminants like heavy metals and pathogens (He *et al*., 2021). Biological treatments, such as activated sludge systems and constructed wetlands, rely on microorganisms to break down organic matter and nutrients like nitrogen and phosphorus (Li *et al*., 2020). Advanced technologies, including membrane filtration and reverse osmosis are increasingly used in aquaculture to meet stringent water quality standards, effectively removing pathogens, nutrients, and chemicals (Mbuya *et al*., 2016)

Treated wastewater has garnered attention as a potential solution to reduce pressure on freshwater resources. When properly managed, it can provide a nutrient-rich environment beneficial for the growth of aquaculture species like fish, shrimp, and bivalves. Wastewater's high concentrations of nitrogen, phosphorus, and organic carbon are essential for aquatic organisms' growth (Liu *et al*., 2020), and its use reduces the need for external nutrients, lowering production costs (He *et al*., 2021). Additionally, aquatic plants such as algae, which thrive in nutrient-rich environments, help maintain water quality by absorbing excess nutrients and preventing eutrophication (Bergheim *et al*., 2018). The use of treated wastewater has proven effective in both open and closed-loop systems like recirculating aquaculture systems (RAS), improving water conservation and species productivity, such as tilapia and shrimp (Guitart *et al*., 2015; Cui *et al*., 2019).

However, concerns remain regarding water quality and the health of aquatic species. Untreated or inadequately treated wastewater may contain harmful contaminants like heavy metals and pathogens, which can harm aquatic life and pose risks to human health through bioaccumulation in the food chain (Guitart *et al*., 2015; Duan et al., 2019). Even treated wastewater may still contain trace contaminants, such as ammonia and nitrites, which can stress fish and make them more vulnerable to diseases (Cui *et al*., 2019). Bioaccumulation of toxic substances in species consumed by humans is another concern (Li *et al*., 2019). Thus, treatment processes must be carefully designed to ensure the safety of both aquatic species and human consumers.

To mitigate these risks, various treatment and management strategies have been proposed. These include advanced filtration systems, biological treatments and constructed wetlands for further purification (Mbuya *et al*., 2016). Pretreatment processes such as sedimentation, chlorination, or adsorption help reduce pathogen loads and improve water quality (He *et al*., 2021). Continuous monitoring of water quality is essential, using automated systems and real-time data analytics to ensure that parameters like pH, ammonia, nitrate, and dissolved oxygen remain within acceptable ranges for aquatic organisms (Shen *et al*., 2021).

The use of treated wastewater in aquaculture also offers significant environmental and economic benefits. By reducing reliance on freshwater, wastewater reuse helps mitigate the environmental impact of aquaculture, particularly in regions with limited or declining freshwater resources (He *et al*., 2021). It can also reduce operational costs, benefiting small-scale farmers or operations in water-scarce areas (Bergheim *et al*., 2018). Furthermore, wastewater reuse aligns with sustainability goals, promoting a circular economy by minimizing waste and recycling valuable resources such as water and nutrients, which contributes to pollution reduction and conservation of natural resources (Mbuya *et al*., 2016).

2.4 Importance of Water Quality in Aquaculture

Water quality is a critical factor in aquaculture because it directly affects fish health, growth rates, and overall productivity. Parameters such as dissolved oxygen (DO), pH, temperature, turbidity, salinity, ammonia, nitrite, and nitrate concentrations must be kept within optimal ranges to promote healthy fish growth and minimize stress-induced mortality (Sinha *et al*., 2025). In wastewater-treated aquaculture systems, the continuous monitoring of these parameters becomes even more essential due to the potential for contamination and the presence of residual pollutants.

2.5 Water Quality Standards for Aquaculture Production

Various regulatory bodies have established water quality guidelines for aquaculture. For instance, the Food and Agriculture Organization (FAO) provide standards that highlight the acceptable levels of nutrients, heavy metals, and biological contaminants in aquaculture systems. The suitability of treated wastewater for aquaculture must meet these standards. Specifically, acceptable levels of ammonia (<0.02 mg/L), nitrite (<0.1 mg/L), and nitrate (<100 mg/L) are crucial to prevent fish stress and toxicity (FAO and WHO, 2023). Furthermore, DO levels should remain above 5 mg/L, and pH should be within the range of 6.5–8.5 to promote optimal physiological functions in fish (FAO and WHO, 2023). However, individual fish species have their levels of tolerance and acceptable ranges for their survival. For instance, the Nile tilapia is one of the most widely cultured fish species globally due to its adaptability to a range of environmental conditions, rapid growth, and ability to thrive in diverse aquaculture systems. However, maintaining optimal water quality is essential to ensuring the health and productivity of tilapia.

For successful Nile tilapia farming, the following standards have been recommended for the ensuing water quality parameters:

2.5.1 Water Temperature

Temperature plays a critical role in metabolism, feeding, growth, and reproduction of Nile tilapia. As a tropical species, tilapia thrives in warm water environments and requires an optimal range of 25–30°C. the lethal limits are below 12°C and above 38°C. Water temperatures outside the optimal range can reduce feeding efficiency, slow growth, or lead to mortality. Low temperatures can stress the fish and cause diseases, while excessively high temperatures can reduce dissolved oxygen levels and increase metabolic rates, leading to oxygen depletion (El-Sayed, 2006).

2.5.2 Dissolved Oxygen (DO)

Dissolved oxygen is one of the most critical water quality parameters in tilapia culture. It directly affects fish respiration, growth, and overall health. In the case of the Nile tilapia a range of DO >5 mg/L is recommended. A DO value <2 mg/L is critical and the fish become stressed, while a level <1 mg/L is lethal and could result in death. DO concentrations below the critical level result in stress, reduced feed intake, and slowed growth. Maintaining aeration systems and proper water flow helps prevent low oxygen conditions, especially in intensive culture systems (Boyd, 2018).

2.5.3 pH

pH, one of the physicochemical parameters required for ecosystem health and support for ecosystem drive has specific ranges for aquatic organisms. For instance, the Nile tilapia can tolerate a broad pH range but perform best within a specific range that ensures the proper functioning of their physiological processes. The optimal range is 6.5–8.5, whiles below 6.0 or above 9.0, the fish becomes stressed. Extreme pH levels outside the optimal range can damage gill tissues, reduce fish immune function, and hinder growth. Acidic water (pH <6) can lead to toxic metal accumulation, while alkaline water (pH >9) reduces the efficiency of oxygen uptake (El-Sayed, 2006).

2.5.4 Ammonia (NH_3)

Ammonia is a toxic nitrogenous waste product produced by fish metabolism and the decomposition of organic matter. Ammonia exists in two forms in water: un-ionized ammonia (NH_3), which is highly toxic, and ammonium (NH_4^+), which is less toxic. The proportion of toxic NH_3 increases with higher pH and temperature. According to Boyd and Tucker (2012), ammonia toxicity impairs gill function, reduces oxygen uptake, and can lead to death if levels remain high. Effective biological filtration, regular water changes, and maintaining low stocking densities are necessary to manage ammonia levels. The optimal level is <0.05 mg/L, the stressful level is 0.05–0.1 mg/L while the lethal range is >0.1 mg/L.

2.5.5 Nitrite (NO_2^-)

Nitrite can be toxic to fish if accumulated in significant concentrations. High nitrite levels can cause "brown blood disease" (methemoglobinemia) in fish, reducing the blood’s ability to transport oxygen. This is particularly dangerous in intensive recirculating aquaculture systems (RAS) where proper biological filtration is critical to converting nitrite to less harmful nitrate (Boyd and Tucker, 2012). An optimal level of <0.1 mg/L is recommended while a range of 0.5–1 mg/L stresses the fish and any level above 1 mg/L may be lethal.

2.5.6 Nitrate (NO_3^-)

Nitrate is the final product of the nitrogen cycle, formed from the oxidation of nitrite by nitrifying bacteria. It is far less toxic than ammonia and nitrite but can still cause issues at high concentrations. El-Sayed (2006) noted that high nitrate concentrations over time can negatively affect fish growth and reproduction. Effective water management practices such as water exchanges, and proper filtration help keep nitrate levels within safe limits. The maximum acceptable level is 100 mg/L while the optimal level is <50 mg/L.

2.5.7 Total Alkalinity and Hardness

The recommended optimal range for total alkalinity is 50–200 mg/L as $CaCO_3$ and 100–250 mg/L as $CaCO_3$ for hardness. Alkalinity measures the buffering capacity of water, which helps stabilize pH and prevents rapid fluctuations. Adequate alkalinity is important for maintaining a stable pH, especially in systems with intensive fish production. Low alkalinity can lead to large pH swings, which stress the fish and harm their health. Lime ($CaCO_3$) can be added to ponds to adjust alkalinity if levels are too low (Boyd, 2018). Water hardness on the other hand refers to the concentration of dissolved calcium and magnesium ions. Hard water is typically better for Nile tilapia culture as it provides necessary minerals for bone development and metabolic processes. Hard water helps reduce stress and improves the overall health and growth of fish. Low water hardness can be corrected by adding lime or other mineral supplements (Boyd, 2018).

2.5.8 Turbidity

Turbidity refers to the cloudiness of water caused by suspended particles. Moderate turbidity can be beneficial for reducing predation and preventing excessive algal growth, but very high turbidity can clog gills and interfere with fish feeding. The optimal level is 30–60 cm Secchi disk depth and becomes high when <20 cm Secchi disk depth. Excessive turbidity, especially from organic waste or algae, can negatively impact fish health by reducing light penetration and affecting dissolved oxygen levels (Boyd and Tucker, 2012).

2.5.9 Salinity

Total dissolved solids and other compound been organic or inorganic ions contributes to the salinity levels or saltines of water body that play critical roles in the physiological conditions of aquatic organisms. Example, the Nile tilapia is euryhaline, hence they can tolerate a wide range of salinity levels, but their optimal growth occurs in freshwater 0–15 ppt with a tolerance range of 0–35 ppt. While tilapia can survive in brackish or saline environments, growth rates are typically higher in freshwater conditions. High salinity can stress the fish and reduce growth efficiency (El-Sayed, 2006). Some key water quality parameters and their acceptable limits for aquaculture production are as shown in Table 1.

Table 1: Key indicators of water quality and the acceptable limits for aquaculture production.

Water quality parameter	**Suitable range/acceptable limit**
Dissolved oxygen (DO)	>5 mg/L
Ph	6.5-9.0
Water temperature	25-30^{o}C
Ammonia (NH_3)	<0.02-0.05 mg/L
Nitrite (NO_2^-)	<0.1 mg/L
Nitrate (NO_3^-)	<50 mg/L
Turbidity	<25- 80 NTU
Salinity	0-0.5%

Source: (FAO and WHO 2023)

2.6 Treated Wastewater: Potential and Risks

Treated wastewater has the potential to supplement the water demand necessary for aquaculture sector, especially in regions where there is freshwater scarcity. Various wastewater treatment processes, such as activated sludge, biological filtration, and membrane filtration, remove pathogens, organic matter, and suspended solids. However, treated wastewater may still contain trace pollutants like heavy metals (cadmium, lead, and mercury), residual pharmaceuticals, and microplastics, which pose risks to aquaculture species and, eventually, to human consumers (Xu *et al.*, 2024).

Thus, comprehensive water quality assessments must be conducted before treated wastewater can be utilized. Furthermore, wastewater that has undergone tertiary treatment is considered more suitable for aquaculture because advanced processes like ultraviolet (UV) irradiation and reverse osmosis remove additional pollutants (Kumar *et al.*, 2015).

2.7 Plankton community and Water Quality as Indicators in Aquaculture Systems

Plankton communities, including phytoplankton and zooplankton, are essential components of aquaculture systems as they play a vital role in nutrient cycling and serve as food sources for certain species of fish and shellfish. Plankton can also act as sensitive bioindicators of water quality, responding rapidly to changes in nutrient levels, pollution, and other environmental factors (Bláha *et al.*, 2009).

Phytoplankton are primary producers that respond directly to nutrient availability, particularly nitrogen and phosphorus. Excessive nutrients, especially in treated wastewater, can lead to eutrophication and harmful algal blooms (HABs) production which are mostly dominated by toxic cyanobacteria. These conditions (eutrophication and HAB) can negatively impact fish health by depleting oxygen levels and producing toxins (Paerl,2023). Thus, monitoring phytoplankton composition, diversity, and abundance is essential to prevent adverse impacts on aquaculture systems that utilize treated wastewater.

According to Paerl (2010), a diverse phytoplankton community with balanced species representation indicates stable water conditions, whereas dominance by a few species such as *Microcystis* or *Anabaena* suggests nutrient imbalances. Therefore, phytoplankton monitoring serves as an early warning system for nutrient pollution and the potential for eutrophication in aquaculture systems.

Zooplankton are secondary consumers that feed on phytoplankton and serve as a food source for higher trophic levels in aquaculture systems. Their abundance, diversity, and community structure are influenced by water quality, particularly nutrient levels, pollution, and the presence of contaminants (Wen *et al.*, 2011). Zooplankton, such as rotifers and copepods, can thrive in moderate levels of organic waste, making them useful bioindicators of treated wastewater conditions. Zooplankton community shifts, such as a decline in diversity or an increase in pollution-tolerant species like rotifers, can signal poor water quality and the potential for adverse effects on fish health (Bláha *et al.*, 2009). Regular monitoring of zooplankton can thus provide insight into the biological suitability of treated wastewater for aquaculture.

Water quality parameters alone may not provide a complete assessment of treated wastewater's suitability for aquaculture. Biological indicators, such as the composition and health of the plankton community, complement physico-chemical assessments by providing insight into the ecological balance within aquaculture systems. For example, elevated nutrient levels may not immediately manifest in poor water quality readings but could lead to shifts in the plankton community that signal impending problems (Bláha *et al.*, 2009). The combination of water quality analysis and plankton monitoring offers a comprehensive approach to evaluating treated wastewater for aquaculture. Healthy plankton communities with high biodiversity and minimal pollutant-tolerant species indicate that treated wastewater has a low ecological impact and is suitable for aquaculture (Wen *et al.*, 2011).

2.8 Wastewater situation in Ghana

According to UNICEF, (2016) the sewage treatment system in Ghana is low and form about 4.5% of wastewater being treated before discharge, indicating significant gaps in sanitation infrastructure and posing risks to public health and the environment. Sewage treatment in Ghana is mainly under the Waste Management Department of the Municipal, Metropolitan and District Assemblies (Rooijen *et al.,*2010). Thus, the low waste management could be attributed to a lack of proper coordination at the district levels across Ghana. As a result of the nearly collapsed and ongoing rehabilitation of the current wastewater and sludge treatment facilities, domestic greywater is channeled into rivers and lagoons, like the heavily polluted Odaw River, which covers roughly 60% of Accra (not including Tema) (Amoah *et al*., 2023). Among the difficulties are dead river bodies. The "Odaw" River and Ghana's Korle Lagoon are two instances of such dead rivers. Additionally, according to UNICEF (2016), there are satellite sewage systems in place in a few Accra neighborhoods, including Dansoman, Teshie-Nungua, Burma Camp, Accra Sewage Treatment plant, UPSA, University of Ghana, Legon, Achimota School, 37 Military Hospital, and Ridge neighborhoods. The lack of ineffective wastewater treatment infrastructure is compromising the collection, treatment, and appropriate disposal/discharge of generated wastewater. The Accra Sewerage Systems Ghana Limited (SSGL) wastewater treatment plant at Lavender Hill and the Tema Central Sewerage Treatment Plant, both suffered significant operational breakdowns due to aging infrastructure and lack of maintenance. These facilities are presently undergoing renovation and capacity upgrades to restore functionality and improve wastewater management in the Greater

Accra Region (Amoah *et al*., 2023; Ghana News Agency, 2021). This has a significant impact on the amount of wastewater treated prior to release. When compared to wastewater from home sources, Qadir *et al.* (2010) state that industrial wastewater frequently has higher concentrations of metals, metalloids, and volatile or semi-volatile chemicals. It's interesting to note that the pathogenic and nutritional load of wastewater from household sources makes it very dangerous for its release into the environment. Wastewater treatment is required to remove the aforementioned in order to prevent contamination of the environment and dangers to human health when used. According to Amoah *et al.* (2009), an estimated 200,000 Accra residents eat "fast food" every day that includes raw lettuce leaves that are produced using waste contaminated irrigation water. Kumasi generates about 20,000 m³ of wastewater daily, but less than 10% is treated due to outdated infrastructure. While the Asafo and Ahensan facilities need major rehabilitation, a new treatment plant with a 1,000 m³/day capacity was built in 2021 to help address the city's wastewater issues (Amoah *et al*., 2023; Ghana News Agency, 2023; Construction Review Online, 2021). Apart from the amount of wastewater generated by about 8% of Kumasi's population that finds its way into the sewage system, grey water from restrooms and kitchens is dumped untreated into stormwater gutters and open drains. Wastewater treatment is necessary to stop untreated wastewater from indiscriminate discharge or disposal which can pollute the environment. This pertains to the particular goals of evaluating the safety of treated wastewater and its use as a means of irrigating crops, as mandated by the World Health Organization (2006). This also serve as scientific evidence to support the decision to use treated wastewater and in the end advocate for the practice of wastewater treatment in general.

2.9 Wastewater treatment facilities in Ghana

More than half of Ghana's treatment plants are located in the Greater Accra region, according to a monitoring assessment carried out by the Ghana Environmental Protection Agency (Dolo, 2015) on the country's sewage and faecal sludge treatment plant location, number, state, and treatment technologies. According to Adu Ahyiah and Anku (2003), there are no treatment plants in the Upper West and the Brong Ahafo area. The stabilization pond approach is the most widely utilized technique with nearly all faecal sludge and large-capacity sewage treatment plants (Mkali *et al.,* 2014). Most of the activated sludge facilities and trickling filters that have been documented are small-scale private businesses, such as large hotels, with limited capacity. The proportion of treatment facilities that fulfil EPA’s effluent criteria is unknown; however, it is less than 25% of all treatment plants that are in service. In addition to the Ghana EPA, other indications also showed that the treatment plants scarcely fulfill any international requirements (Marfo, 2014).

CHAPTER THREE

MATERIALS AND METHODS

3.1 Description of the study area

The study was conducted within the premises of the Water Research Institute (WRI) in Accra, Ghana. The Water Research Institute is one of the 13 institutions of the Council for Scientific and Industrial Research (CSIR). It was established in 1996 by the merger of the Institute of Aquatic Biology and the Water Resources Research Institute. Water Research Institute has a mandate to conduct research into water and related resources. Water Research Institute (WRI) generates and provides scientific information, strategies and services towards the rational development, utilization and management of the water resources of Ghana in support of the socio-economic advancement of the country, especially in the agriculture, health, industry, energy, transportation, education and tourism sectors. Water Research Institute is located in the Greater Accra Region of Ghana. Water research has shallow reservoirs that are fed with effluents from the Mudor Treatment Plant (MTP). The reservoir serves as a means of storing water to facilitate the operations of artisanal vegetable farmers with the enclaves of the Water Research Institute. The water is purposely used for the irrigation of vegetable farms. The Reservoir was constructed to serve as maturation ponds for the stabilization of liquid-treated water before discharging into the environment. The pond water has not been purposed for fish culture; however, efforts are being made for its secondary use for aquaculture production.

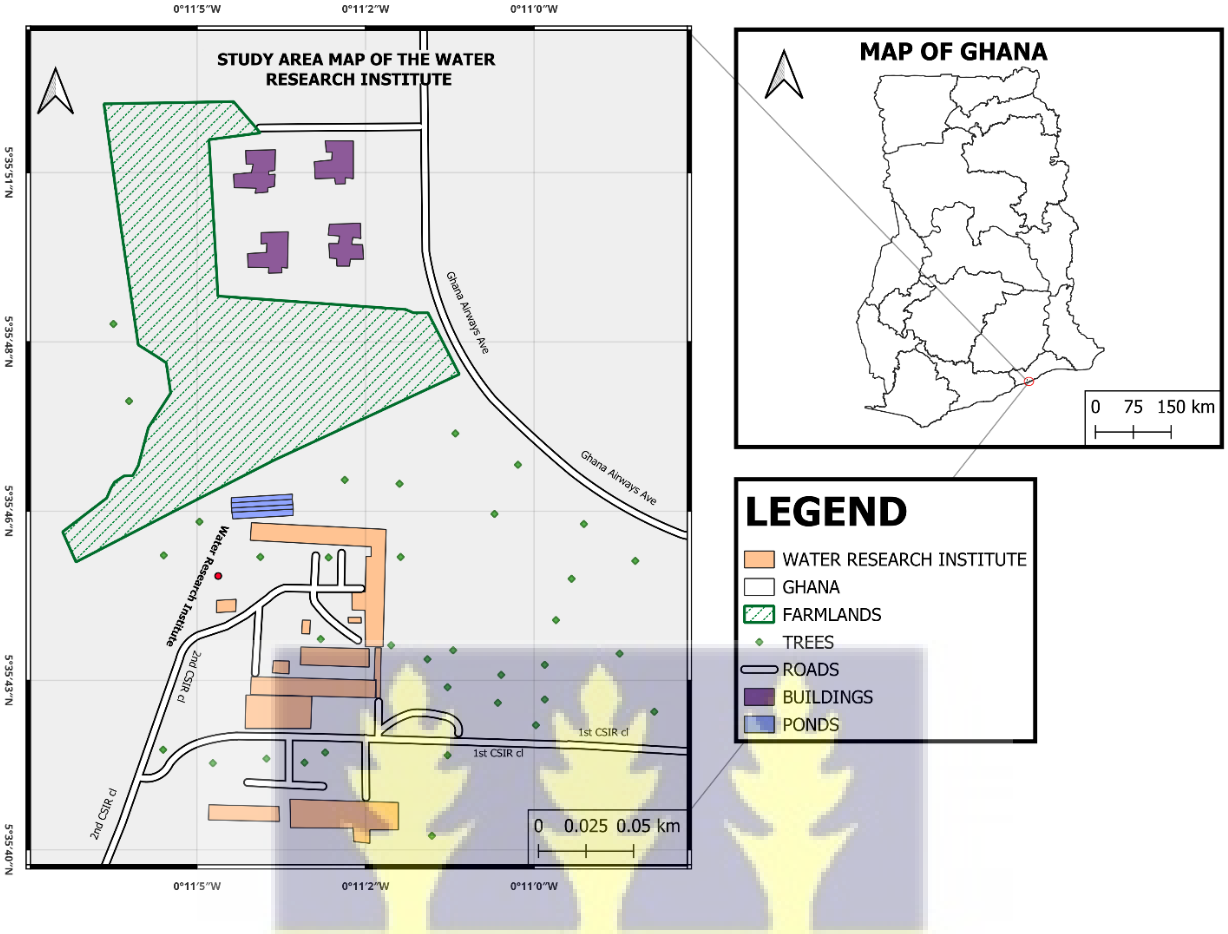


Plate 1: A map of study area, CSIR- Water Research Institute (WRI).

3.2 The Experimental Ponds

Four (4) ponds numbered A, B, D and D, were located behind the Water Research Institute (WRI) building and separated by a main drainage to the North. Along the drainage were vegetable farms. Observed vegetables watered using the treated wastewater were cabbage, spring onion, lettuce, cucumber and among others. The dimensions of the ponds are indicated in Table 2 and shown photographically in Plate 1. The 4 maturation ponds were interlinked where effluents from Pond A move to Pond B then to Pond C which also flow to the last Pond D and thereafter the effluent is channeled through pipes for irrigation of the vegetable farms.

Table 2: Dimensions of the four (4) experimental ponds

Pond Identity.	Length (m)	Breath (m)	Depth (m)
Pond A	32	2.4	0.9
Pond B	32	2.3	0.9
Pond C	32	2.1	0.9
Pond D	32	2.3	0.9

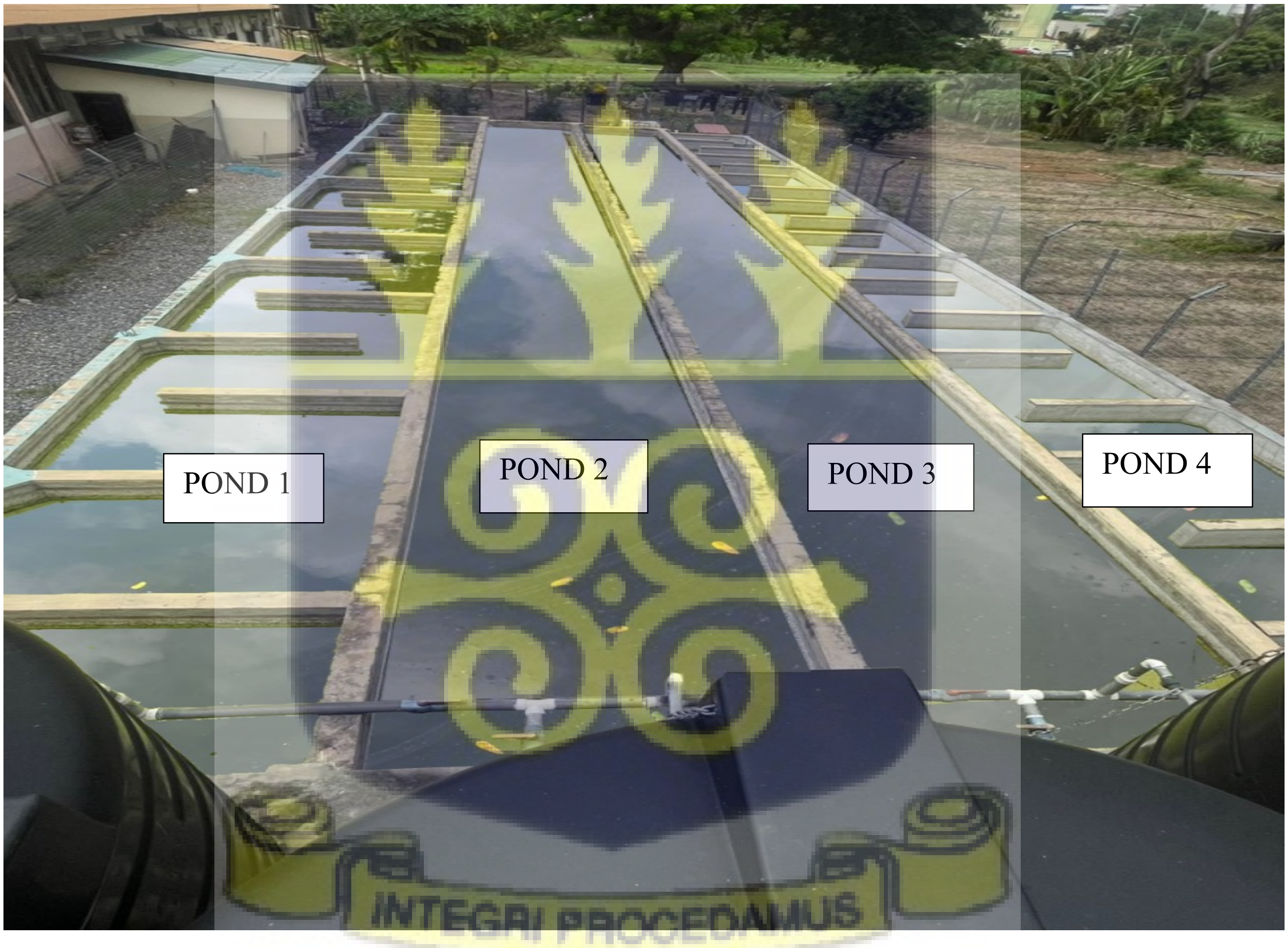


Plate 2: Pond layout for wastewater stabilization and storage for irrigation at WRI.

Photo credit: Ebenezer Botchway.

3.3 Measurement of Water Quality Parameters

Physico-chemical parameters, including pH, temperature, and dissolved oxygen, were measured in-situ using a multiparameter probe (Hanna handheld pH and temperature probe, and dissolved oxygen probe). Additionally, nutrient levels (ammonia and nitrite) were assessed ex situ using spectrophotometric methods. (UV-Visible Spectrophotometer, Agilent Technologies, Santa Clara, California, USA).

In-situ measurements consisted of continuous 24-hour monitoring of maturation pond water, with pH, temperature, and dissolved oxygen (DO) recorded at intervals of 6 am, 9am, 12am, 5pm, 8pm, 12am, 3am, and 6am, following the methodology outlined by Gonzalez and Mitchell (2023). This monitoring was conducted in September and October 2023.

In the case of the ex-situ measurements, 500 ml of water samples were collected from four (4) ponds during the day (12 noon) and at dawn (3 am) for the laboratory analysis of ammonia and nitrite levels. This was to ascertain the effects of photosynthetic activity during the daytime and the lack of photosynthesis at night. Also, it is expected that during the day the respiration is less and in the night respiration high (Johnson and Green, 2023). Water samples were collected into labelled bottles, kept on ice to keep the samples below at or below 4°C (39.2°F) and transported to the laboratory (Ghana Water Company Central Laboratory) around 37 military hospital which is approximately 2.5 km from the sampling site. Preserving samples on ice during transit helps to minimize biological activities of trapped organisms in the samples and to prevent the degradation of the nutrient in the water samples (Smith *et al*., 2023).

3.4 Plankton Data Collection

Composite water samples of 90L (3 x 30L) were collected from top, mid and bottom sections of each pond for phytoplankton and zooplankton/ insect larvae identification and abundance estimations, respectively. A total of eight (8) samples were collected for zooplankton (4) and phytoplankton (4) analyses. For the phytoplankton, a plankton net of mesh size 65µm was used to sieve 90L (30L from three spots/pond) of the pond water; backwashed and the retained organisms were concentrated into the collector. A composite sample from each pond comprised a mixture of three replicates. The same procedures were repeated for all the other ponds. The collected samples were transferred into labeled bottles and preserved with a few drops of concentrated formaldehyde. In the case of zooplankton, a similar technique was adopted using a bigger mesh size of 200µm plankton net. The preserved samples were then transported to the Department of Marine and Fisheries Sciences (DMFS), University of Ghana (UG) laboratory for analysis.

Plate 3: Collection of samples for plankton analysis. Photo credit: Dr. Mark Akrong

3.5. Biota Analysis

3.5.1 Phytoplankton analysis

In the laboratory, the preserved phytoplankton samples were washed and further concentrated using tap water and a sieve of mesh size 20µm. This was to reduce the toxicity of formalin as well as to standardize the total sample volume for identification and enumeration, respectively. One milliliter (1 ml) of a homogenized prepared sample was transferred into the Sedgewick Rafter counter chamber using a graduated Pasteur pipette. The covered chamber was mounted on the stage of a compound microscope (Leica Microscope). The phytoplankton cells were identified using the identification keys provided by Lawrence *et al*. (2020) and counted under the compound microscope of 100 magnifications (10 eyepiece x 10 objective lenses). For the phytoplankton enumeration, the identified individual species were counted in each cell of the counting chamber vertically. This procedure was then repeated and alternated for the column cells to achieve a random count of the phytoplankton cells. The assumption of this procedure is based on the hypothesis that the individual phytoplankton cells are evenly distributed across the entire chamber volume as such the abundance of the phytoplankton cells can be estimated based on the simple ration competition (Shen *et al*., 2021)

3.5.2 Zooplankton analysis

The laboratory Zooplankton analysis was similar to that of the phytoplankton identification and enumeration. The samples were similarly washed with tap water and concentrated using a sieve of mesh size 63 microns. Again, this was to reduce the formalin toxicity and also to standardize the total sample volume for identification and enumeration respectively. However, five millilitres (5 ml) of the homogenized plankton samples were transferred into a Bogorov tray using a ten-millilitre (10 ml) measuring cylinder. The prepared sample was mounted on the stage of a compound microscope (Leica Microscope). The organisms were observed, identified, and counted using a compound microscope at 40× magnification (10× eyepiece and 4× objective lenses). Identification was performed using the identification keys provided by Thompson *et al*. (2020). Zooplankton enumeration was done by counting all the individual species in the entire counting chamber.

3.6 Data Analysis

3.6.1 Phytoplankton

The abundance of the phytoplankton in each of the total volumes of the water filtered was estimated using a simple ratio based on the volume of the sample analyzed to that of the water sampled. This estimation is similar to the work done by Effendi *et al.* (2016) in Indonesia. The mathematical expression of the phytoplankton abundance is shown in the equation one below.

$$absi = \frac{n\ x\ CT\ x\ VT}{ct\ x\ vt} \ldots\ldots\ldots\ldots.1$$

where *Absi* is the abundance of species *i*, *n* is the total number of the individual phytoplankton counted in a number of the squares cell counted that is *ct,* CT is the total number of the squares in the Sedgwick Rafter counting chamber, *vt* is the volume of a liquid taken out of the standard volume concentrated that is *VT*. As such, the total phytoplankton abundance per pond is the summation of the individual species abundance as shown in equation one (1).

3.6.2 Zooplankton

A similar approach was adopted for the zooplankton abundance estimation as it was the case of the phytoplankton. Thus, the zooplankton abundance of each sample was determined by the number of the individual species counted in the sub-sample multiplied by the ratio of the standard volume and the sub-sample volume. This is expressed in equation two.

$$absi = \frac{n\ x\ VT}{vt} \ldots\ldots\ldots\ldots.2$$

where *Absi* is the abundance of species i, *n* is the total number of the individual zooplankton counted and *vt* is the volume of the subsample taken out of the standard volume concentrated that is *VT*. This gives us the abundance of zooplankton per pond as the summation of the individual species abundance as expressed in equation one (2).

3.6.3 Diversity indices

Diversity indices were also computed for the biota data using Shannon – Wiener (H') indices as shown in equation three (3).

$$H' = \sum_{I=1}^{S}(pi)(In\ pi)\ldots\ldots\ldots\ldots3$$

here H' is the Shannon-Wiener indices, *pi* is the estimated number of the individual species divided by the summation of the total count of the species, *In pi* is the natural log of the individual *pi* and the summation sign is the addition of the products of *pi* and *In pi* (Chao, A *et al.*2003)

The bota data was also subjected to species richness in accordance with Margalefs indices as shown in equation four (4).

$$D = \frac{(s-1)}{\log N} \ldots\ldots\ldots\ldots 4$$

Where *D* is the Margalefs indices, *s* is the total number of species in the ponds and *N* is the total individuals in the ponds (Miller & Smith, 2023).

The evenness of the biodata distributions among the ponds was also computed using the Pielous evenness indices as shown in Equation 5. (Wang & Lee, 2023)

$$j' = \frac{H\prime}{\log(s)}\ldots\ldots\ldots\ldots 5$$

j' is the Pielous indices, *H'* is the Shannon Weiner indices as expressed in equation three (3) above and *log(s)* is the natural log of total species estimated in the sample as indicated in species abundance estimation (equation one and two for the phytoplankton and the zooplankton, respectively).

3.7 Statistical analysis

The pool data of the physico-chemical parameters were put together and analyzed for the mean values and standard deviation for each parameter. The mean values were also compared using analysis of variance (ANOVA) to ascertain any significant differences.

CHAPTER FOUR

RESULTS AND DISCUSSION

4.1 Water Quality

4.1.1 Temperature

The mean water temperatures recorded at specific intervals across four experimental ponds during September and October 2023 (Figures 1 and 2) exhibited a typical diurnal pattern consistent with thermal behavior in tropical freshwater systems. Mean temperature levels were lowest at 6:00 AM and progressively increased to peak around 5:00 PM before declining through the night. This diurnal variation aligns with findings from García *et al*. (2021), who reported similar trends in small tropical reservoirs, where solar radiation is the dominant driver of daytime warming and nighttime cooling.

In September, early morning temperatures ranged between 25.70 ± 0.10°C and 26.47 ± 0.10°C, with afternoon peaks between 27.00 ± 0.46°C and 28.23 ± 0.31°C. These values are indicative of tropical climatic conditions, as described by Köppen (1936) and supported by Giannini *et al*. (2008), where ambient air temperatures remain elevated throughout the day due to prolonged solar exposure. The gradual rise from morning to afternoon is characteristic of tropical systems with high solar radiation and minimal shading (Wetzel, 2001).

Analysis of variance (ANOVA) revealed statistically significant differences among ponds (p = 0.012), suggesting spatial heterogeneity in thermal dynamics. This variability may be linked to differing physical features (e.g., pond depth, partitioning, and water surface area), shading, or biological processes such as respiration and photosynthesis. These findings are consistent with Boyd and Tucker (1998), who demonstrated that even minor variations in pond structure and biological activity can influence thermal regimes.

October temperatures followed a similar diurnal trend but with marginally reduced ranges: morning lows between 25.06 ± 0.26°C and 26.11 ± 0.04°C (6am B), and afternoon highs between 26.29 ± 0.23°C and

27.37 ± 0.14°C (5pm). The cooler average temperatures in October are likely attributable to seasonal transitions in atmospheric conditions and reduced solar insolation, as described by Imberger and Patterson (1981). However, ANOVA results indicated no significant differences among ponds ($p = 0.364$), suggesting more uniform weather conditions or stabilized biological processes during this month.

These observations correlate with work by Azaza *et al*. (2008), who noted that water temperature in aquaculture systems is closely aligned with ambient climatic fluctuations and becomes more consistent when meteorological variables such as wind speed and humidity are stable.

Notably, lower temperatures recorded in Pond 1 and Pond 4 may be attributed to structural features such as semi-partition walls, which reduce direct solar exposure and increase surface reflectivity. Such modifications have been shown to influence water heat retention, as confirmed by Wurts and Durborow (1992), who observed that partitioning and shading in aquaculture ponds can suppress temperature peaks and modulate diel fluctuations.

Additionally, the evening thermal plateau observed in both months (from 5:00 PM to midnight) could be explained by the thermal inertia of water and sustained biological respiration from planktonic communities. Kromkamp and Peene (1995) reported that microbial and phytoplanktonic respiration can contribute to minor thermal retention during the night, particularly in nutrient-enriched environments.

Water temperature is a critical ecological variable influencing aquatic metabolism, dissolved oxygen availability, and susceptibility to pathogens (Brett, 1971; Boyd, 2017). The recorded temperatures range for September and October 2023 (25.06°C–28.23°C) falls within the optimal thermal range for key cultured species such as *Oreochromis niloticus* (25–30°C) and *Clarias gariepinus* (23–30°C), as reported by Bardach *et al*. (1972) and Coyle et al. (2004), respectively. These results support the suitability of the observed thermal conditions for sustaining optimal aquaculture performance in the study location.

Moreover, the World Health Organization (WHO) and Food and Agriculture Organization (FAO) emphasized the importance of maintaining pond temperatures within species-specific thresholds to enhance fish growth, reduce stress, and optimize feed conversion ratios (FAO & WHO, 2023).

The observed wide error bars between midnight and early morning (Figure 1) may result from differential nighttime cooling across ponds, influenced by pond orientation, depth, and heat absorption properties. Similar variability was noted by Boyd and Tucker (1998), who linked overnight cooling rates to pond morphology and surrounding vegetation.

Comparative analysis between the two months further reveals subtle seasonal effects. The decline in October Pond temperatures corresponds with expected tropical weather cycles, such as reduced day length and early transitional rains, which influence surface heat accumulation (Hepher, 1988). These findings corroborate results from tropical aquaculture studies in West Africa, Southeast Asia, and South America, where small but significant month-to-month thermal shifts impact growth and feeding behavior of cultured fish (FAO, 2020).

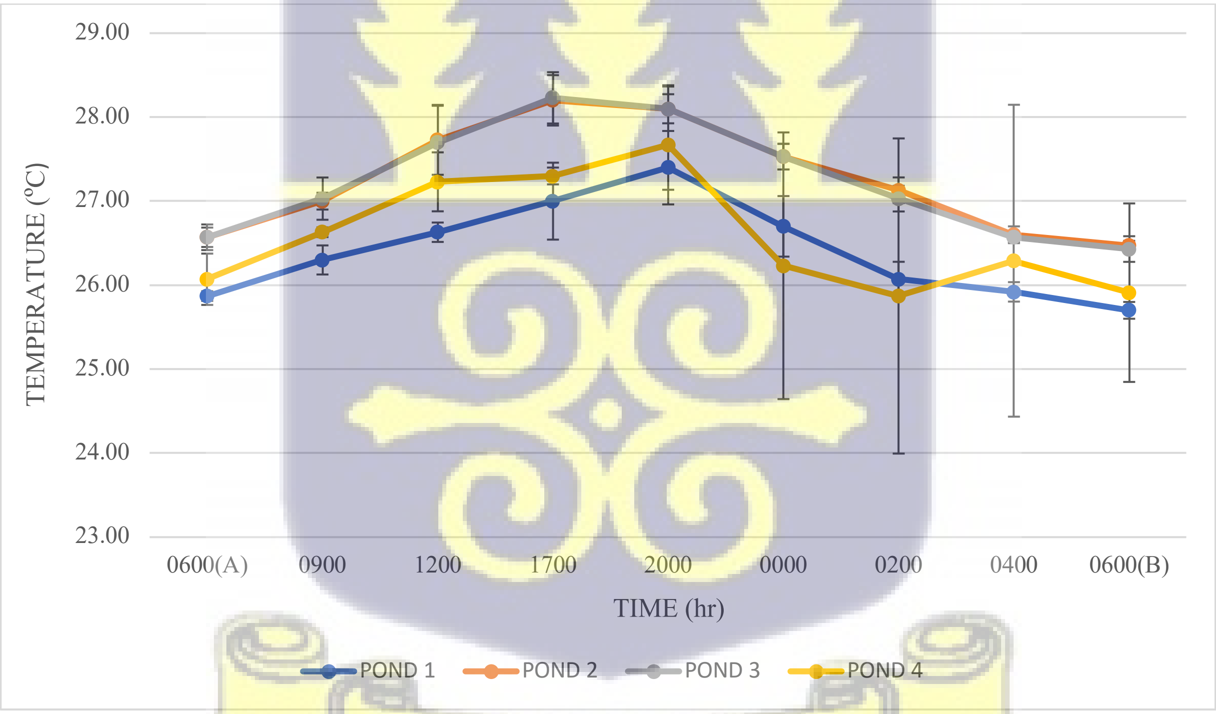


Figure 1: Mean temperature variations across the ponds over a 24hr period in September 2023.

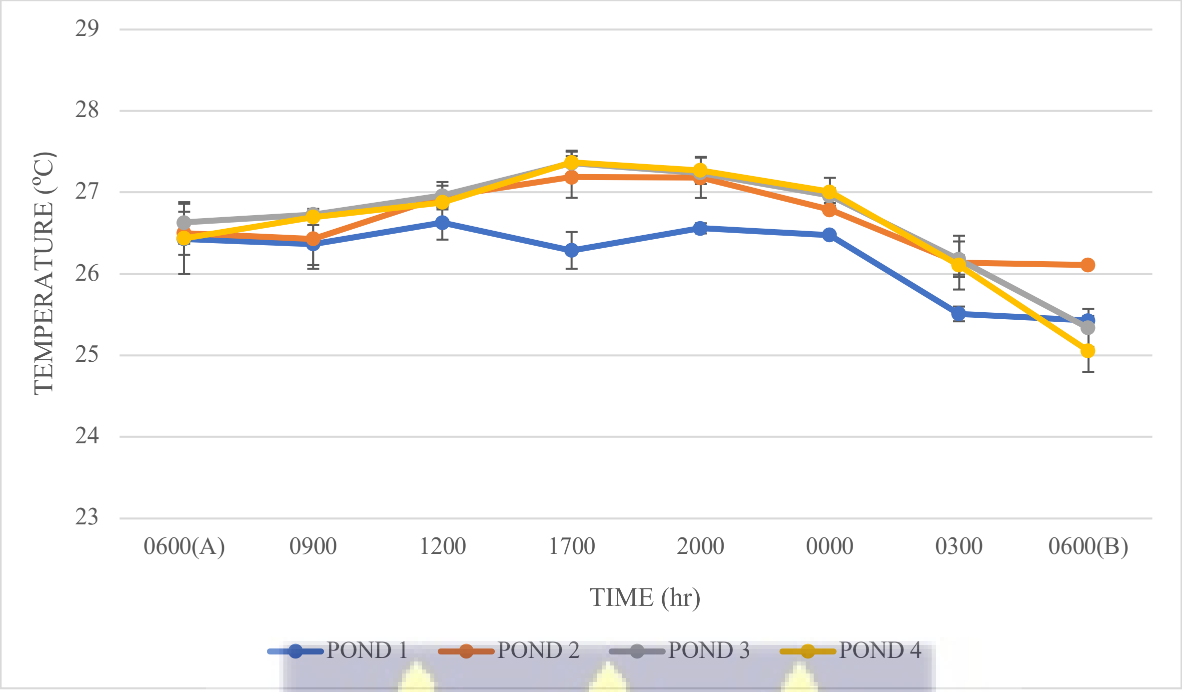


Figure 2: Mean temperature variations across the ponds over a 24hr period in October 2023.

4.1.2 pH

The diurnal fluctuations of pH levels observed in the four study ponds during September and October 2023 are presented in Figures 3 and 4, respectively. Across both months, pH values exhibited a distinct daily cycle: a rise from early morning, peaking in the late afternoon, followed by a decline during nighttime hours. This pattern reflects the dynamic interplay of biological processes; primarily photosynthesis and respiration in aquatic systems.

In September, pH values rose from early morning readings (7.75 ± 0.14 to 9.15 ± 0.10 at 6:00 AM) to peak values by 5:00 pm (9.48 ± 0.04 to 10.63 ± 0.05), before declining again by the next morning (8.31 ± 0.57 to 9.71 ± 0.23 at 6:00 am). This cyclic pattern is consistent with the diel metabolic activity of phytoplankton, whereby CO_2 uptake during photosynthesis reduces carbonic acid concentrations and thus increases pH (Baker *et al*., 2020; Harris *et al*., 2016). Similar diel trends have been observed in tropical aquaculture ponds and freshwater systems (Zhou *et al.*, 2018; Wetzel, 2001), reinforcing the relationship between pH and photosynthetic activity under solar influence.

October followed a comparable diurnal trend. Early morning values ranged from 8.05 ± 0.24 (6am) to 8.48 ± 0.35, rising to 9.29 ± 0.04 to 9.70 ± 0.15 by 5:00 PM, before again declining by the next day's morning 6am (8.12 ± 0.27 to 8.31 ± 0.10). Although the overall pH range remained alkaline, values in October were slightly lower than those recorded in September. This decrease may be attributed to seasonal factors such as reduced sunlight intensity, shorter photoperiods, or lower water temperatures, which can suppress the rate of photosynthesis and subsequently reduce daytime alkalinity (Azaza *et al.*, 2008; García *et al.*, 2021).

This is further supported by previous research that established seasonal shifts in water temperature and solar radiation directly influence aquatic photosynthesis, thereby altering diurnal pH levels (Imberger & Patterson, 1981; Zhou *et al.*, 2018). Moreover, the convergence of temperature and pH trends suggests a strong coupling between thermal and biogeochemical processes in these pond ecosystems.

The observed increase in pH during daylight hours across both months can be explained by the photosynthetic removal of CO_2 by autotrophic organisms such as phytoplankton and cyanobacteria, which reduces the concentration of carbonic acid and elevates pH (Baker *et al.*, 2020; Wetzel, 2001). Conversely, the nighttime decline in pH is due to the accumulation of CO_2 released through respiration by aquatic organisms including fish, invertebrates, and microbial communities (Zhou *et al.*, 2018). These findings are consistent with the metabolic models described by Harris *et al*. (2016), where pH is directly modulated by diurnal photosynthesis-respiration cycles.

Statistical analysis using ANOVA revealed a significant difference in pH among the study ponds in September ($p = 0.002$), indicating spatial variability, likely influenced by differences in primary productivity, nutrient loading, or shading. In contrast, no statistically significant differences were observed in October ($p = 0.905$), suggesting more uniform environmental conditions or a stabilization of biological activity across the ponds.

The variability in pH between ponds may also be a function of pond morphology and biotic composition. As supported by Boyd and Tucker (1998), small-scale differences in phytoplankton biomass, sediment resuspension, or aeration regimes can contribute to differential pH behavior in aquaculture ponds.

The observed pH values for September (7.75–10.63) and October (8.05–9.70) largely fall within the alkaline range, occasionally exceeding the upper optimal thresholds for freshwater aquaculture. According to Maltby *et al*. (2016), the ideal pH range for most cultured freshwater fish lies between 6.5 and 8.5, with optimal growth typically occurring between pH 7.0 and 8.0. Prolonged exposure to pH levels above 9.0 can induce physiological stress, reduce feed intake, impair gill function, and increase disease susceptibility in sensitive species (López *et al*., 2017).

Furthermore, nutrient dynamics—particularly nitrogen and phosphorus availability—are strongly pH-dependent. Wang *et al*. (2021) observed that optimal pH enhances nutrient solubility and promotes phytoplankton productivity, which, although beneficial to primary production, can lead to eutrophication and subsequent oxygen depletion if not managed properly.

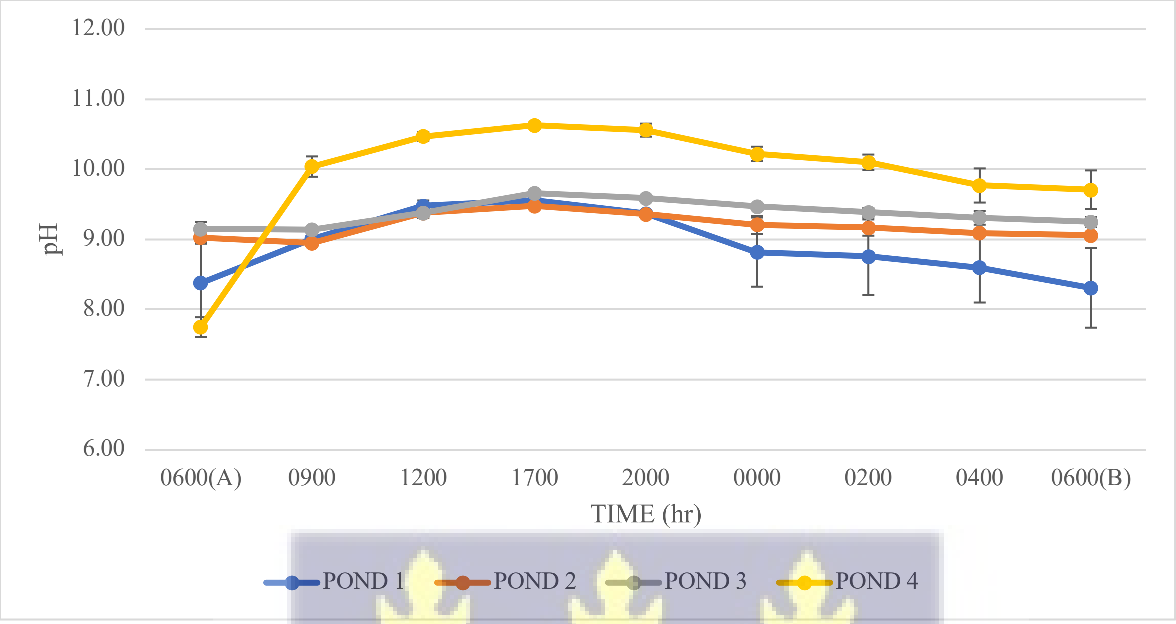


Figure 3: Mean levels of pH across the ponds over a 24hr period in September 2023

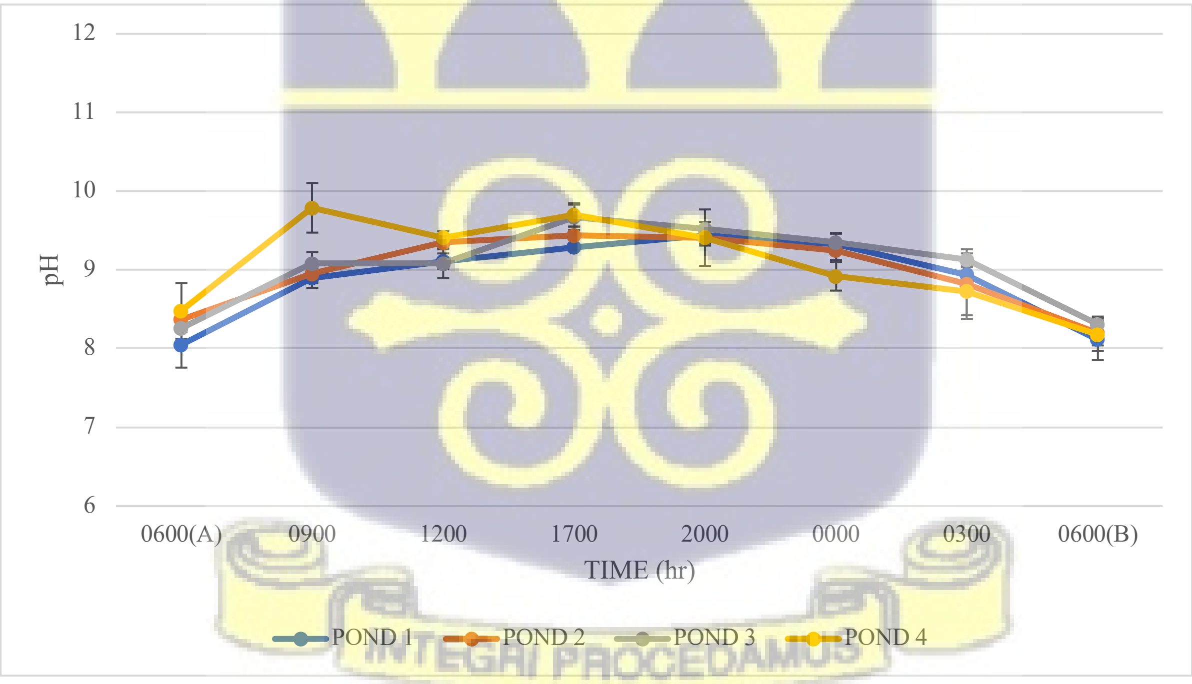


Figure 4: Mean levels of pH across the ponds over a 24hr period in October 2023

4.1.3 Dissolved Oxygen

Figures 5 and 6 present the diurnal fluctuations of dissolved oxygen (DO) levels recorded in the four study ponds during September and October 2023, respectively. Across both months, the data revealed distinct diurnal trends in DO concentrations, characterized by morning lows and evening peaks, followed by subsequent nocturnal declines.

In September, DO levels showed irregular patterns across the ponds, with Pond 4 maintaining relatively elevated concentrations compared to the others. A general increase in DO levels was observed from 6:00 am (1.21 ± 0.19 – 6.50 ± 1.97 mg/L) to 8:00 pm (3.04 ± 3.00 – 9.89 ± 2.00 mg/L), followed by a decline toward the following morning. The considerable variability, especially the large standard deviations in some ponds, may be attributed to differences in phytoplankton biomass, shading effects, and aeration levels among the ponds.

This diel pattern aligns with typical oxygen dynamics in shallow eutrophic systems, where photosynthetic activity during daylight hours elevates DO levels, and respiration dominates during the night, depleting oxygen (Baker *et al.*, 2020; Zhou *et al.*, 2018). These findings are consistent with those of Morris and Hogg (2004), who observed similar daily fluctuations in tropical aquaculture systems, driven primarily by phytoplankton photosynthesis during the day and community respiration at night.

DO measurements in October followed a comparable diel pattern but with overall lower values. DO levels rose from early morning (0.52 ± 0.22 – 0.77 ± 0.04 mg/L at 6:00 am) to a peak at 8:00 pm (4.44 ± 0.47 – 6.40 ± 0.92 mg/L), before declining again by the next morning (0.32 – 0.74 mg/L). The early morning values were particularly concerning, often falling below the critical threshold of 2.0 mg/L.

The reduction in DO values in October compared to September may be explained by seasonal shifts, reduced solar radiation and possibly increased organic matter accumulation leading to greater microbial oxygen demand. These findings support the conclusions of Reddy and DeLaune (2008), who reported that organic load and seasonal temperature changes are key drivers of DO variability in pond systems.

The patterns observed across both months highlight the dual role of photosynthesis and respiration in regulating DO. During daylight hours, phytoplankton and aquatic macrophytes use solar energy to produce oxygen via photosynthesis, elevating DO concentrations. As light diminishes, oxygen production halts, and respiration by fish, invertebrates, and microorganisms leads to a decrease in DO levels (Harris *et al*., 2016; Zhou *et al*., 2018).

Temperature further modulates these processes by influencing oxygen solubility and metabolic rates. While warmer water holds less oxygen, it simultaneously increases metabolic and microbial respiration rates (Morris & Hogg, 2004). Thus, although cooler nighttime temperatures can increase oxygen solubility, the overall DO tends to decline due to sustained respiratory oxygen consumption.

ANOVA results for September revealed statistically significant differences in mean DO levels among the ponds ($p = 0.0043$), indicating spatial heterogeneity likely resulting from differences in primary productivity, nutrient levels, or shading. Conversely, no statistically significant differences were observed in October ($p = 0.551$), suggesting uniform environmental conditions or lower biological productivity across ponds.

These findings are comparable to those reported by Boyd and Tucker (1998), who highlighted that pond design, depth, and internal biological communities can cause significant spatial variations in DO levels even within small aquaculture systems.

From an aquaculture perspective, maintaining appropriate DO levels is critical for fish health, feed conversion efficiency, and overall production. Graham *et al*. (2018) and Liu *et al*. (2020) reported that tilapia and catfish exhibit optimal performance at DO concentrations of 5–7 mg/L and 6–8 mg/L, respectively. Prolonged exposure to suboptimal levels (< 4 mg/L) can induce metabolic stress, suppress growth, impair immune function, and increase mortality rates (Boyd, 2015).

In the current study, DO levels in September ranged from 1.21 to 9.89 mg/L, while October levels were even more concerning, ranging from 0.32 to 6.40 mg/L. These results indicate that, particularly during early morning hours, DO levels frequently fell below critical thresholds, posing a potential risk to cultured species. Consistent exposure to DO levels below 2 mg/L, as recorded in some October measurements, may result in acute hypoxic stress, which is known to impair osmoregulation and gill function in tilapia and catfish (Baker *et al*., 2020; Boyd, 2015).

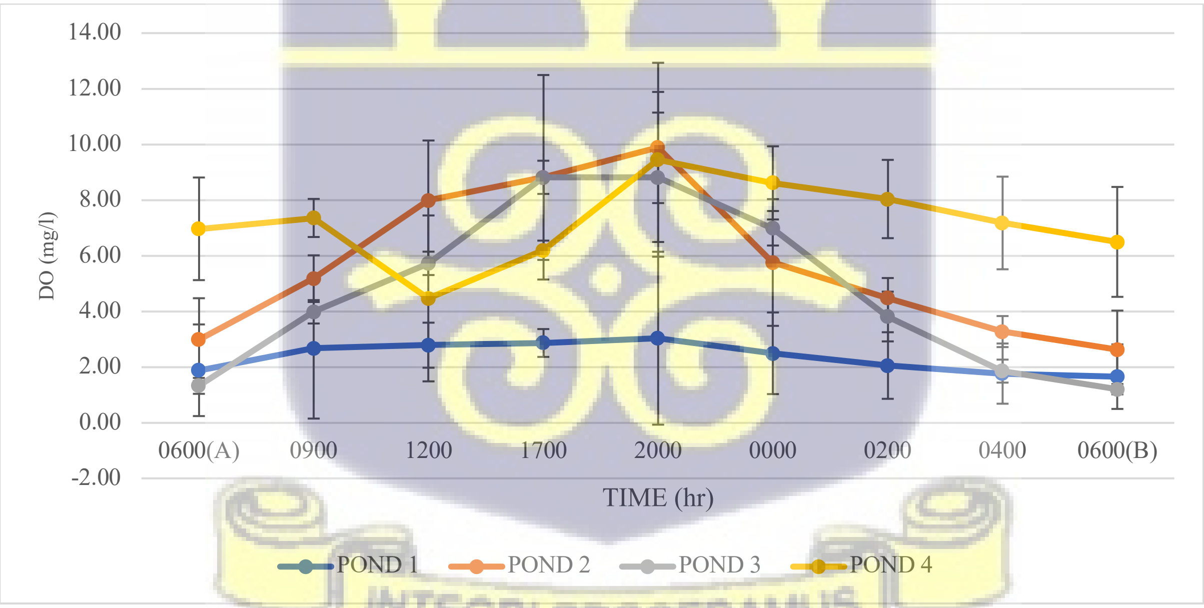


Figure 5: Mean levels of DO across the ponds over a 24hr period in September 2023

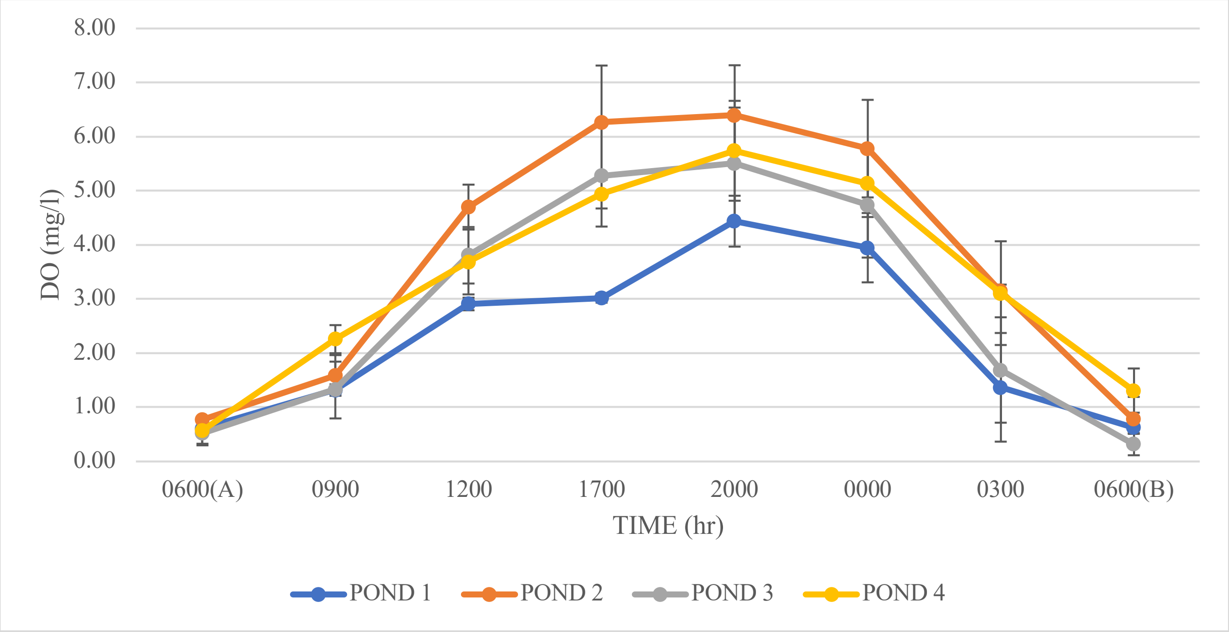


Figure 6: Mean levels of DO across the ponds over a 24hr period in October 2023

4.1.4 Ammonia

Ammonia concentrations recorded at 12:00 noon and 3:00 am during the study period are presented in Figure 7. The results indicate relatively high ammonia levels across Ponds 1, 2, and 3 (ranging from 2.44 to 2.84 mg/L), while Pond 4 displayed significantly lower values (0.50 to 0.56 mg/L). Despite these observable differences, analysis of variance (ANOVA) revealed no statistically significant variation in ammonia concentrations across the ponds (*p* = 0.960).

Ammonia levels in aquatic systems are known to fluctuate in response to diurnal biological processes. During daylight hours, phytoplankton and aquatic macrophytes actively assimilate ammonia for growth, leading to reduced concentrations in the water column (Davis *et al*., 2017). This explains the comparatively lower readings observed at noon in this study. However, during the night, particularly around 3:00 am, photosynthesis ceases and microbial and animal respiration continue unabated. This results in the accumulation of ammonia, as biological uptake slows while nitrogenous waste excretion and organic matter decomposition persist (Hoffmann *et al*., 2018). The pattern observed in this study is consistent with these documented diel ammonia dynamics. The notably low ammonia levels in Pond 4, compared to the other ponds, may be attributed to its better oxygenation profile and potentially lower organic matter load, as discussed earlier with respect to dissolved oxygen levels (Figure 5). Ammonia buildup is commonly associated with organic waste accumulation, particularly uneaten feed and fecal matter (Boyd, 2015). The consistently higher dissolved oxygen levels observed in Pond 4 suggest more efficient microbial nitrification, where ammonia is oxidized to nitrite and subsequently to nitrate by aerobic bacteria, thus reducing ammonia concentrations (Zhou *et al*., 2018).

Conversely, the elevated levels in Ponds 1, 2, and 3 may indicate higher organic loading, poor waste management, or insufficient nitrification—conditions often exacerbated under low-oxygen scenarios. These findings are in line with those of Avnimelech (2006), who reported that intensive aquaculture systems with poor aeration and high feeding rates tend to accumulate ammonia, especially during the night or under stratified conditions.

Ammonia, particularly its un-ionized form (NH_3), is toxic to aquatic organisms. The toxicity threshold varies with temperature and pH but is generally considered harmful at concentrations above 1–2 mg/L (Boyd, 2015). The Food and Agriculture Organization (FAO) and World Health Organization (WHO) (2023) recommend that ammonia levels in aquaculture systems remain below 2 mg/L to support optimal growth and minimize health risks.

In the present study, ammonia concentrations in Ponds 1, 2, and 3 exceeded these thresholds, ranging from 2.44 to 2.84 mg/L. This raises concern for fish health, as chronic exposure to elevated ammonia levels can impair gill function, suppress immune responses, and reduce feed efficiency (Tomasso, 1994; Ip *et al*., 2001). The findings are consistent with those of Wang *et al*. (2020), who observed growth suppression and increased mortality in *Oreochromis niloticus* exposed to ammonia levels exceeding 2.5 mg/L under tropical aquaculture conditions.

In contrast, the ammonia levels in Pond 4 remained well within the safe limits (0.50–0.56 mg/L), suggesting more favorable conditions for aquatic life. This reinforces the importance of pond design, aeration, and organic matter management in controlling ammonia concentrations, as also emphasized by Ebeling *et al*. (2006).

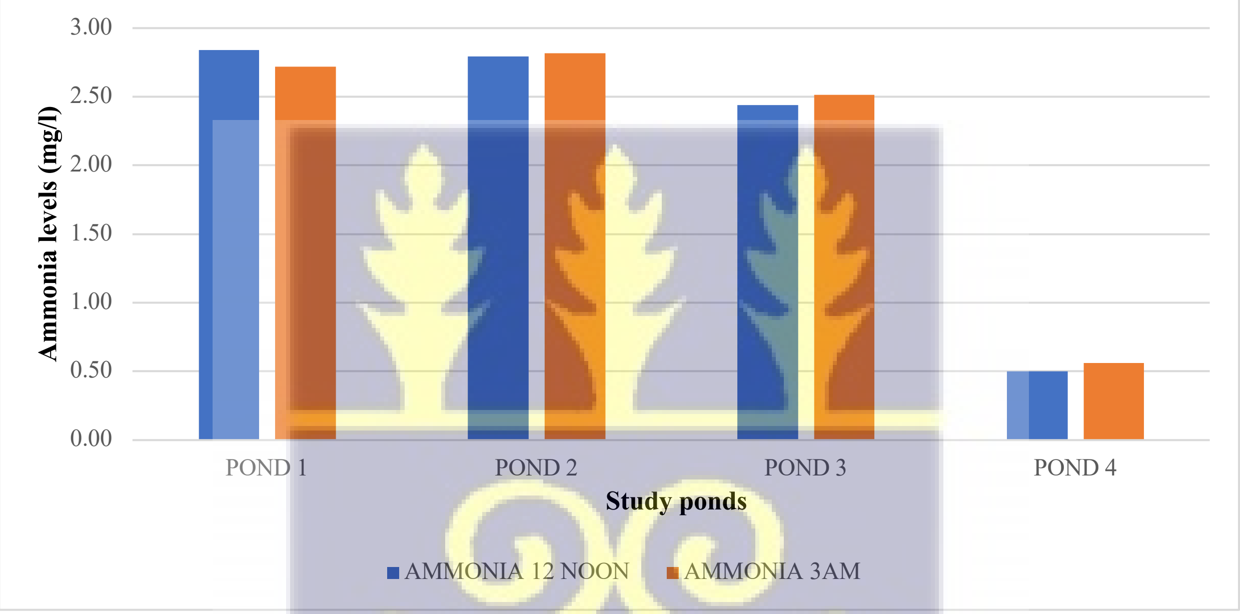


Figure 7: Ammonia levels across the ponds at 12 noon and 3am in September 2023

4.1.5 Nitrite

Nitrite concentrations measured at 12:00 noon and 3:00 am are presented in Figure 8. The data reveal relatively elevated nitrite levels in Ponds 1 and 2 (ranging from 0.216 to 0.237 mg/L), while considerably lower levels were recorded in Ponds 3 and 4 (0.002 to 0.109 mg/L). Despite these differences, analysis of variance (ANOVA) indicated that there were no statistically significant variations across the ponds ($p = 0.059$).

Nitrite (NO_2^-) is a critical intermediate in the nitrification process, where ammonia (NH_3) is first oxidized to nitrite by ammonia-oxidizing bacteria, and subsequently to nitrate (NO_3^-) by nitrite-oxidizing bacteria (Bock & Wagner, 2013). Diurnal variations in nitrite can occur due to changes in microbial activity, oxygen levels, and ammonia availability (Chen *et al.*, 2006). The slightly elevated levels observed during the 3 AM measurements in Ponds 1 and 2 may reflect a lag in nitrification due to reduced nighttime oxygen availability, which impairs microbial oxidation efficiency (Gao *et al*., 2014). In contrast, the lower concentrations observed in Ponds 3 and 4 could indicate more effective nitrification, potentially supported by better oxygen conditions or microbial community structure.

According to Hargreaves (2013), nitrite concentrations suitable for most freshwater aquaculture systems typically fall within the range of 0.1–0.5 mg/L, while values exceeding 1.0 mg/L are considered harmful. The Food and Agriculture Organization (FAO) and World Health Organization (WHO) (2023) have similarly recommended that nitrite concentrations should remain below 0.5 mg/L to avoid adverse health effects in aquaculture. In the current study, the observed nitrite levels across all ponds remained below this toxicity threshold, suggesting that the conditions were within the safe range for fish health and growth.

Nevertheless, chronic exposure to nitrite levels approaching the upper limit (e.g., >0.2 mg/L), as recorded in Ponds 1 and 2, has been associated with sub-lethal effects in several fish species. These include oxidative stress, reduced feed conversion efficiency, and altered gill function, as demonstrated by Naylor *et al*. (2000) and Sharma et al. (2020). Nitrite's toxic effect primarily stems from its interference with hemoglobin, forming methemoglobin and impairing oxygen transport in fish (Lewis & Morris, 1986).

The variations in nitrite concentrations between ponds may be linked to differences in ammonia input, organic matter accumulation, and dissolved oxygen availability—factors that influence microbial nitrification pathways (Tchobanoglous *et al*., 2003). The relatively low nitrite levels in Ponds 3 and 4 might also reflect more efficient nitrogen cycling, potentially aided by greater microbial diversity or improved pond management practices.

Importantly, the presence of low but detectable nitrite levels in all ponds highlights the need for continuous monitoring and control of nitrogenous waste. Regular water exchange, biofiltration, and aeration are crucial management strategies to ensure complete nitrification and avoid nitrite accumulation (Avnimelech, 2006).

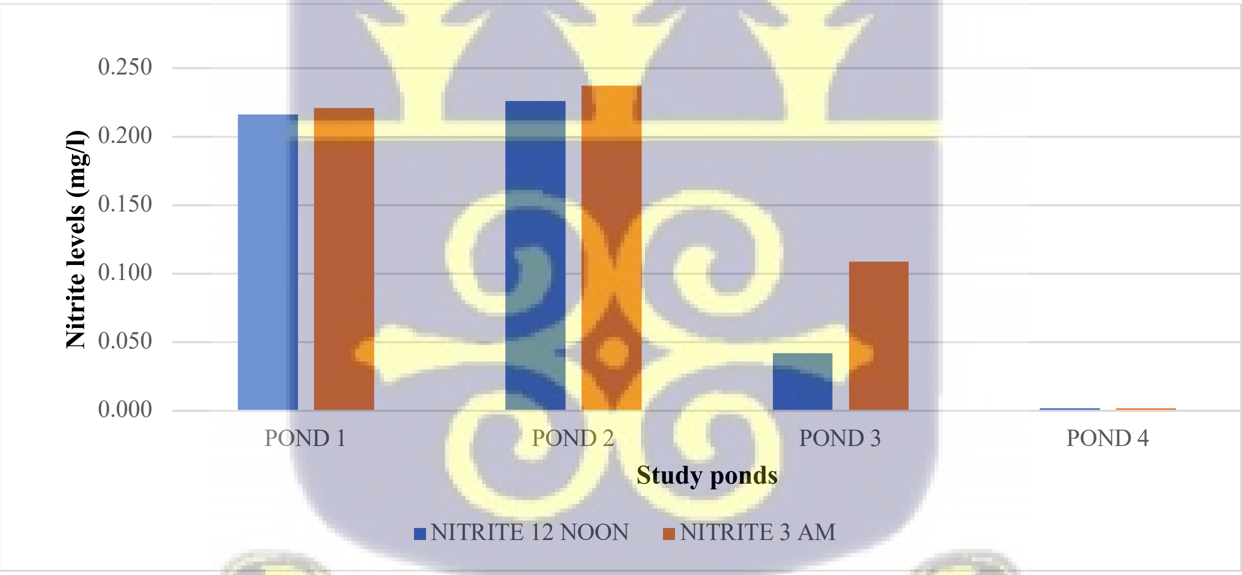


Figure 8: Nitrite levels across the ponds at 12 noon and 3 am in September 2023

4.1.6 Phytoplankton

Figure 11 displays the phytoplankton assemblage across the four study ponds, revealing eight taxa spanning Cyanobacteria (37.5%), Chlorophyceae (37.5%), Rotifera (12.5%), and Euglenoidea (12.5%). Dominant genera included *Gloeocapsa* in Pond 1, *Scenedesmus* in Pond 4, and rotifers in Pond 3. Ranking of phytoplankton abundance (individuals m^{-3}) followed the order: Pond 1 > Pond 3 > Pond 4 > Pond 2 (Figure 9).

Margalef's species richness index was highest in Pond 2, indicating a diverse community composition; conversely, Pielou's evenness and Shannon-Wiener diversity were greatest in Pond 3, suggesting a more evenly distributed phytoplankton community. Comparable studies in tropical aquaculture ponds report similar stratifications, often with elevated cyanobacterial and chlorophycean biomass heavily influencing richness and evenness metrics (Gong *et al*., 2014; Chen *et al.,* 2020).

The elevated abundance of *Gloeocapsa* in Pond 1 likely reflects high nutrient loading and favorable light conditions that drive rapid population turnovers (Whitton & Potts, 2012). In contrast, the impoverished community in Pond 2 may result from ammonia toxicity from degrading organic matter, which can inhibit algal proliferation (Davis *et al*., 2017). Ponds 3 and 4, displaying moderate abundance and high diversity, likely benefit from more balanced physicochemical conditions that support stable microbial ecosystems.

Emerging studies underscore the value of incorporating microalgae and cyanobacteria into tilapia diets. For instance, dietary supplementation with *Scenedesmus quadricauda* enhanced growth performance, digestive enzyme activity, antioxidant defenses, and immune response in Nile tilapia fingerlings (Abdel-Tawwab *et al*., 2022). Likewise, biofloc systems enriched with *Scenedesmus obliquus* yielded a 14% increase in tilapia biomass and improved survival rates (Silva *et al*., 2022).

Green algae like *Eudorina* and *Cladophora*, rich in lipids, proteins, and micronutrients, have been linked to improved body condition and feed conversion in tilapia (Li *et al*., 2020; Hasan *et al*., 2022). Although some cyanobacterial species (e.g., *Microcystis*) produce toxins, non-toxic strains have supported tilapia growth without negative health effects (Matsui *et al*., 2020). Similarly, *Euglena* and *Oscillatoria* are emerging as viable feed sources due to their omega-3, protein, and vitamin content, with preliminary studies indicating enhanced tilapia growth (Jiang *et al*., 2021; Hwang *et al*., 2019).

Rotifers, though classified as zooplankton, are crucial live feed in early larval stages, offering essential nutrients and fostering higher survival and growth in tilapia larvae (Lubzens *et al*., 2001; Rehana *et al*., 2023).

These findings align closely with previous research recommending phytoplankton-augmented feeds in tilapia culture: *Scenedesmus* inclusion improved tilapia feed efficiency and immunity (Abdel-Tawwab *et al*., 2022). Biofloc systems with *Scenedesmus* boosted tilapia survival and biomass yields (Silva *et al*., 2022). Enriched rotifer cultures enhanced larval tilapia performance (Rehana *et al*., 2023).

Together, these studies support our observation that ponds with richer algal communities (particularly those with balanced *Scenedesmus* and rotifer densities, such as Ponds 3 and 4) may offer nutritional advantages and stability conducive to aquaculture.

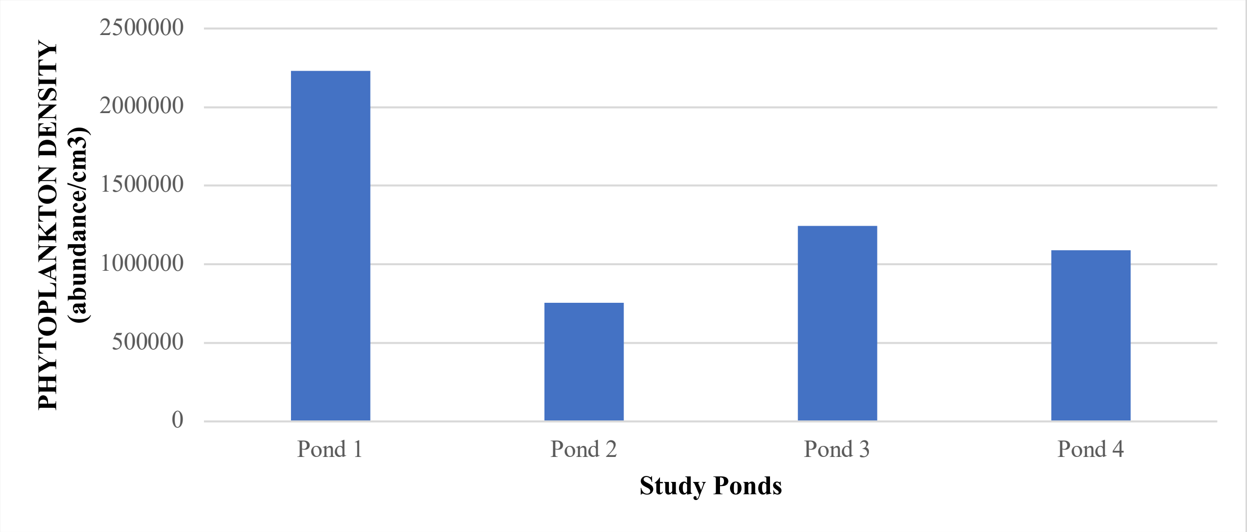


Figure 9: Phytoplankton density across the four ponds

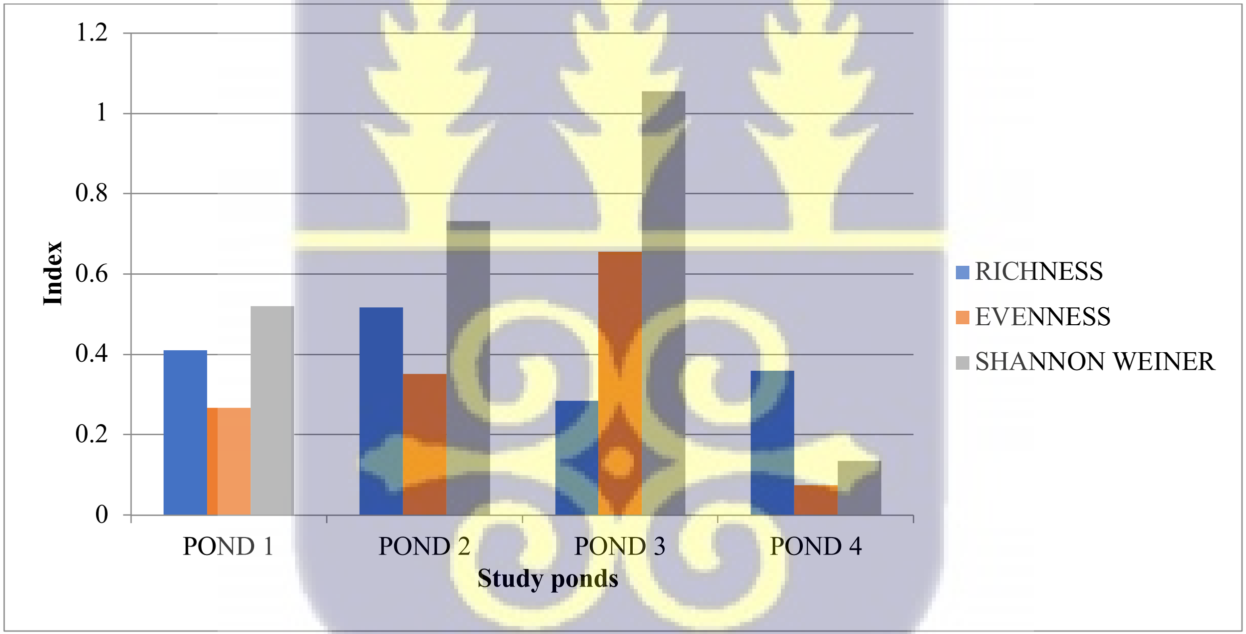


Figure 10: Diversity Indices of Phytoplankton in the four ponds

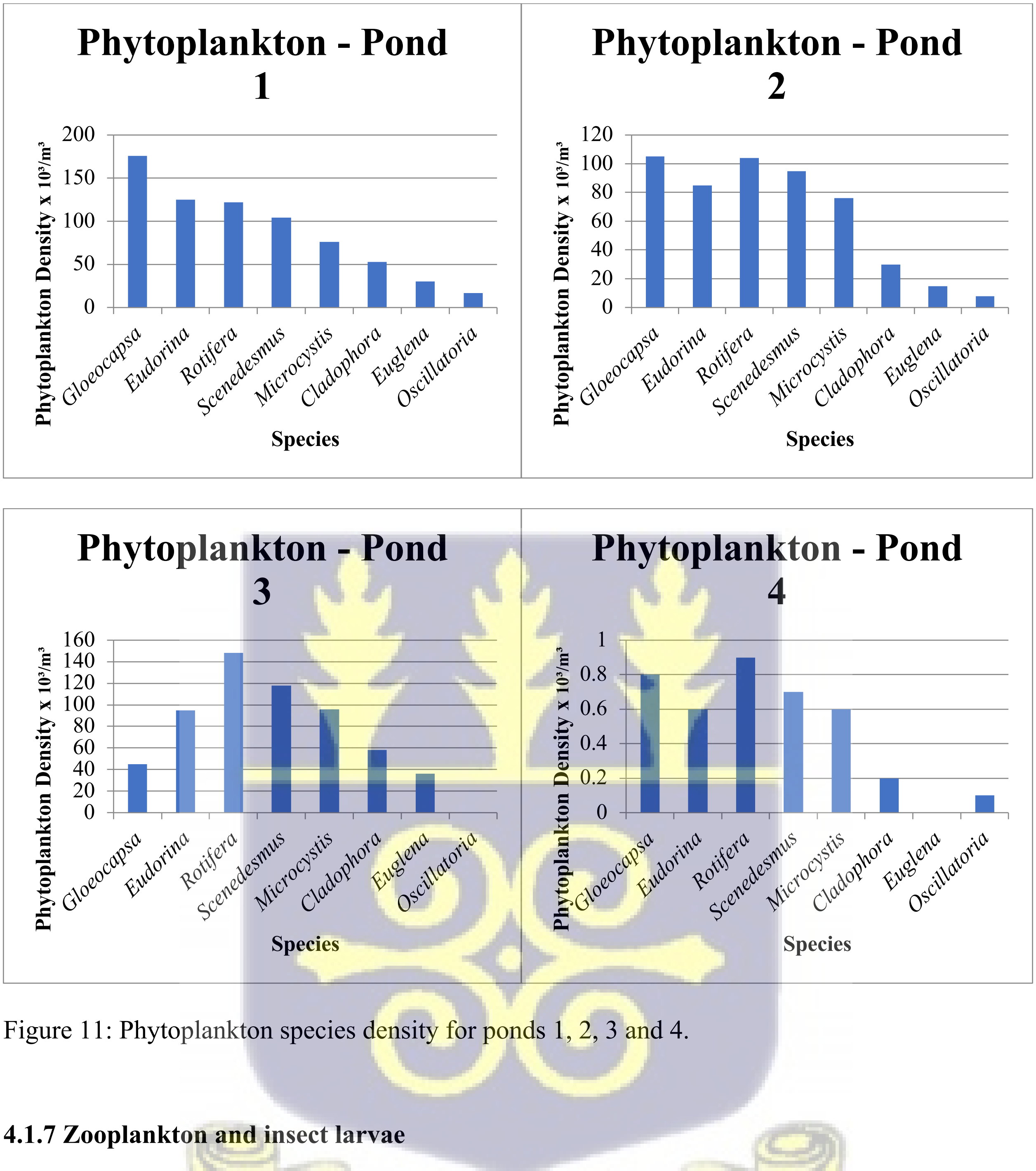


Figure 11: Phytoplankton species density for ponds 1, 2, 3 and 4.

4.1.7 Zooplankton and insect larvae

Figures 12 and 13 detail the zooplankton and insect larvae composition across four aquaculture ponds, with eight primary taxa identified. Rather than classical zooplankton groups like copepods and cladocerans, the study recorded dominant insect larvae such as chironomids, stoneflies, dragonfly nymphs, arachnids, *Dystacta* spp., ostracods, and polychaetes. Notably, dragonfly nymphs and ostracods were most abundant, especially in Pond 2, which also exhibited the highest overall zooplankton density.

The lack of traditional microzooplankton (e.g., *Daphnia*, *Cyclops*) is unusual but not unprecedented in nutrient-rich tropical ponds, where predation pressure, competition, and environmental variables (e.g., high ammonia or low dissolved oxygen) can reduce their populations (Paturej *et al*., 2017). Instead, the dominance of insect larvae suggests a shift toward macroinvertebrate assemblages, a phenomenon also noted in organically rich or sediment-disturbed aquaculture systems (Kumar *et al*., 2019). The findings align with growing interest in integrating invertebrate biomass into aquaculture nutrition:

Chironomids (non-biting midges), rich in protein and lipids, are already utilized as sustainable feed alternatives and have been shown to improve growth, feed conversion ratios (FCR), and immune response in tilapia (Béjar *et al*., 2019). Stoneflies, while less common in commercial feed, are natural prey in freshwater ecosystems and provide bioavailable protein and minerals (McKay *et al*., 2020).

Dragonfly nymphs, though primarily considered predators, are nutrient-dense and have potential as supplemental protein sources. López *et al*. (2021) report enhanced protein synthesis and antioxidant markers in tilapia fed with insect larvae mixtures that included dragonfly species.

Arachnids, especially aquatic mites, are poorly studied in aquaculture feed contexts but may contribute micronutrients and protein to larval fish stages (Jiang *et al*., 2018).

Dystacta spp**.**, a genus of predatory mantids, are known for high amino acid content. Although specific feeding trials in fish are lacking, insect-based proteins like this are increasingly studied as part of circular aquaculture systems (Smith *et al*., 2020).

Ostracods, commonly consumed by juvenile fish in natural systems, are known to improve gut health and omega-3 intake, with Zhou *et al*. (2022) showing measurable improvements in tilapia weight gain and FCR.

Polychaetes, despite their marine origin, are rich in DHA, EPA, and proteins and have been successfully tested in both marine and freshwater fish feeds. Adams *et al*. (2021) demonstrated that polychaete inclusion increased tilapia growth by 18% and improved feed digestibility.

The community structure observed dominated by larger macroinvertebrates suggests a benthic-driven food web influenced by organic loading, potentially from excess feed or detritus. Comparable work by Kadam *et al*. (2016) in Indian aquaculture ponds noted that chironomids and dragonfly nymphs flourished under similar high-nutrient, low-predation regimes. The presence of polychaetes, although more typical in estuarine or marine-influenced systems, might suggest possible contamination or temporary saline intrusion if found consistently in freshwater.

In contrast to conventional hatchery systems dominated by cladocerans and rotifers (Lubzens *et al*., 2001), the current system leans more toward semi-natural pond conditions, favoring insect larval stages as natural productivity indicators.

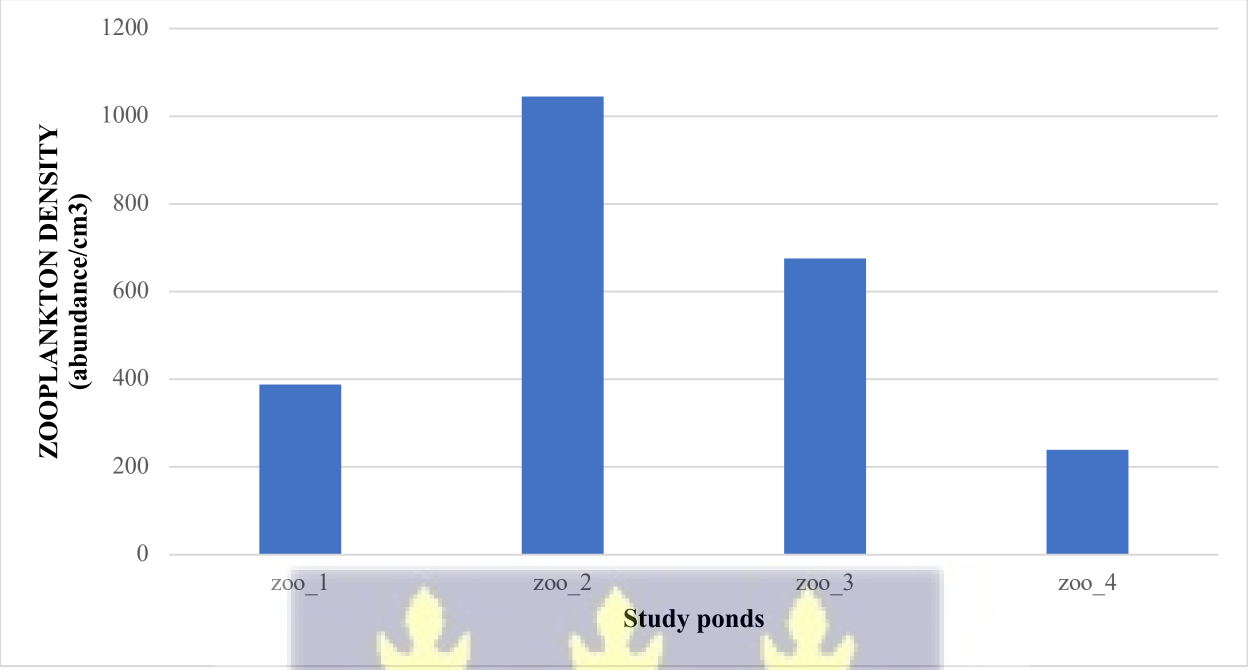


Figure 12: Zooplankton density across the four ponds.

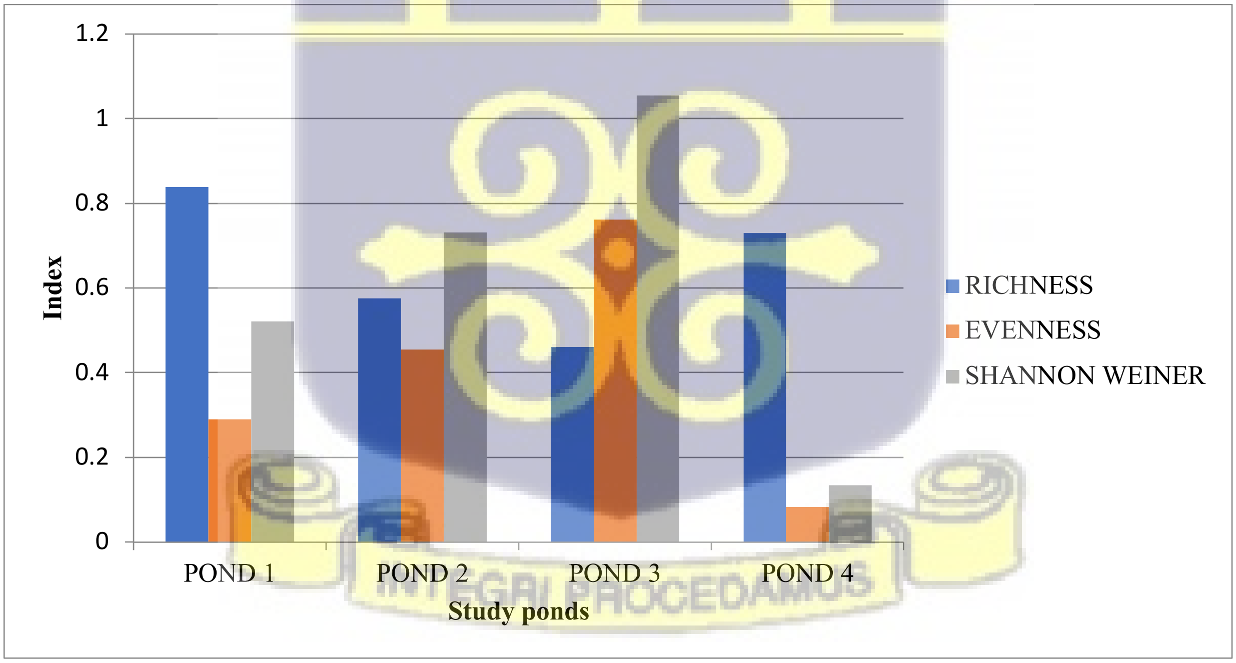
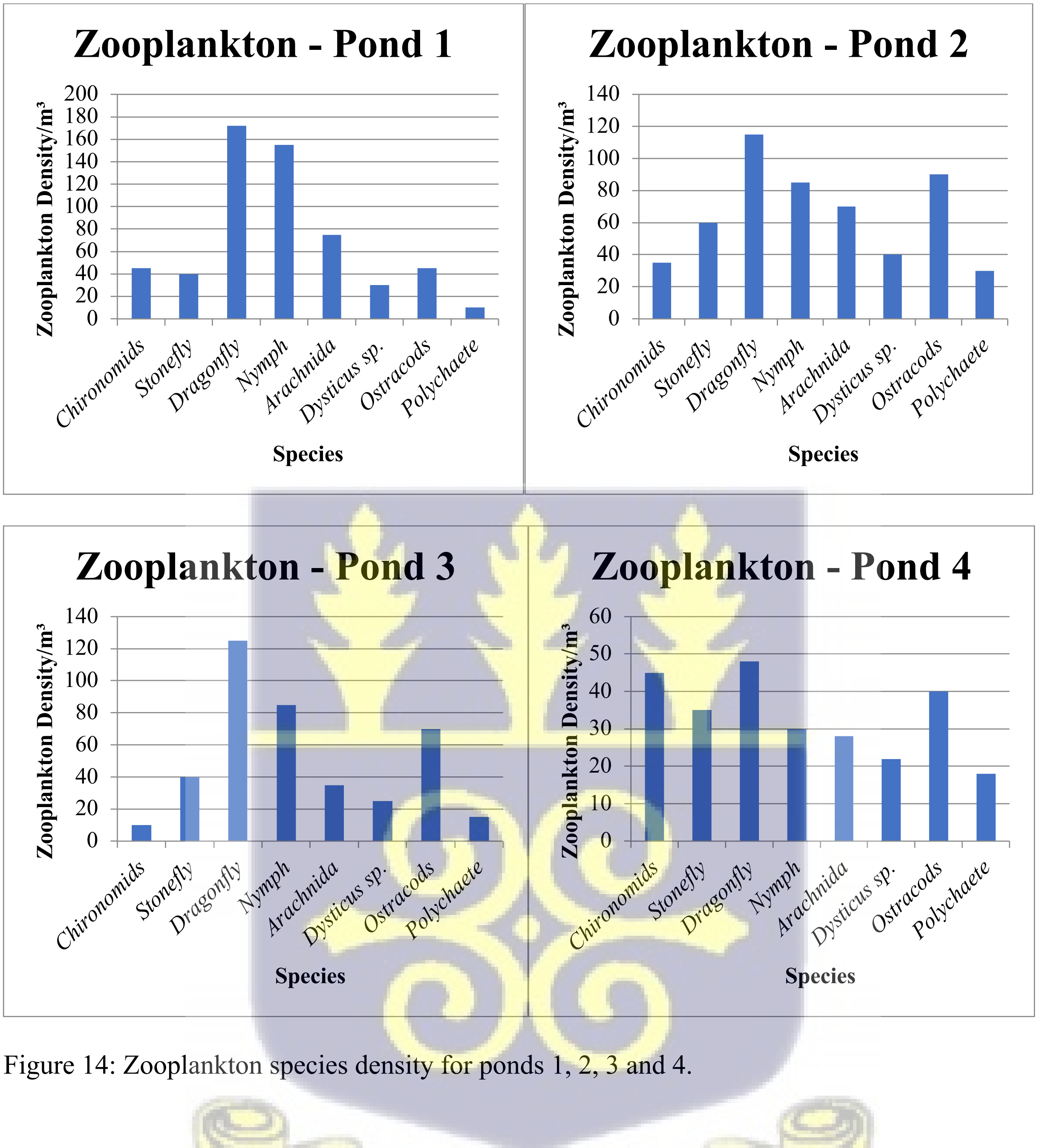


Figure 13: Diversity indices of zooplankton in the four ponds.


Zooplankton - Pond 1

Zooplankton - Pond 2

Zooplankton - Pond 3

Zooplankton - Pond 4

Figure 14: Zooplankton species density for ponds 1, 2, 3 and 4.

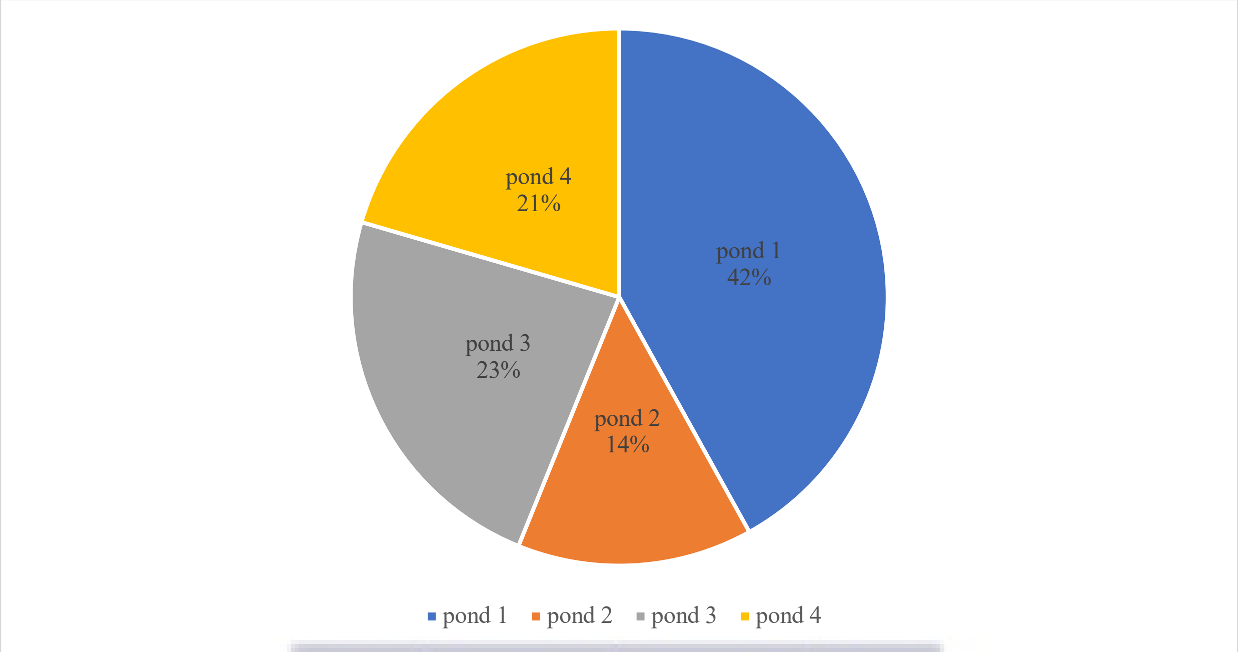


Figure 15: Percentage Contribution of phytoplankton in all the ponds.

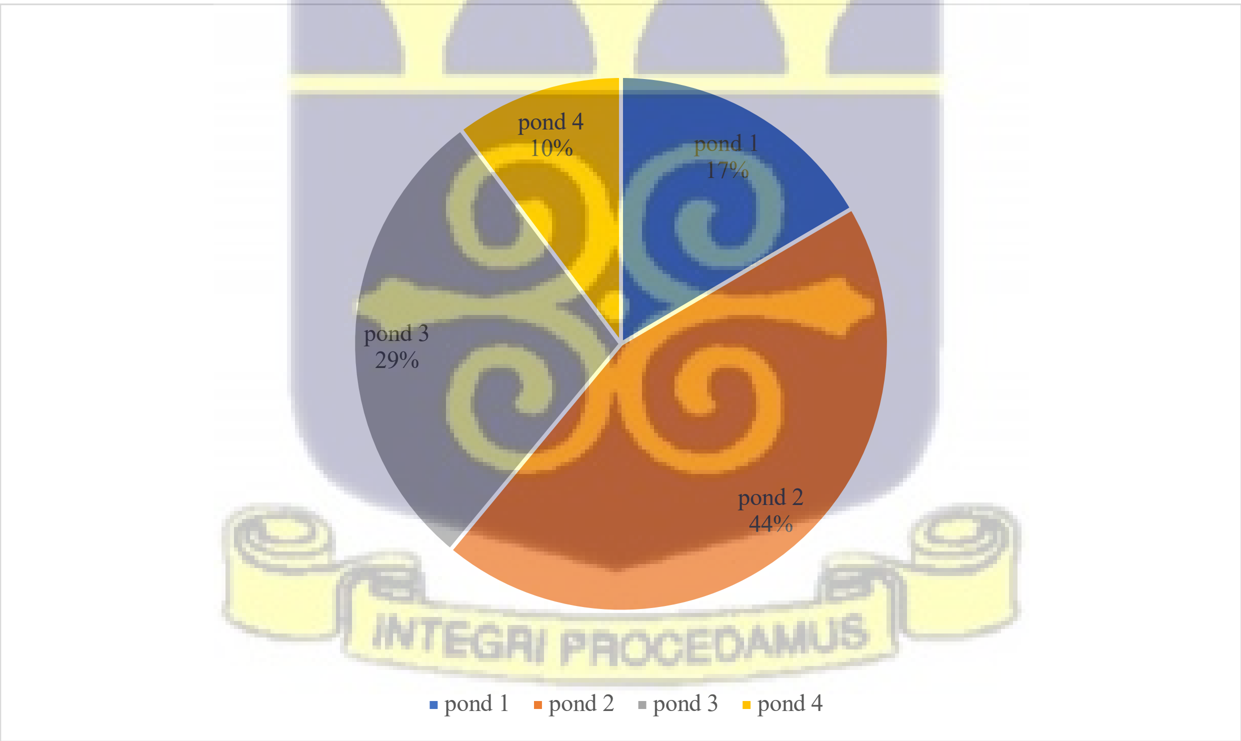


Figure 16: Percentage contribution of zooplankton in all the ponds.

4.2 Physico- chemical parameters measured in the ponds

Table 3: Observed parameter range in comparison to optimal range for aquaculture.

Parameters	Pond 1	Pond 2	Pond 3	Pond 4	Optimal range
Temperature (°C)	25.43-27.40	26.47-28.2	25.34-28.21	25.06-27.67	25- 30
pH	8.05-9.56	8.20-9.48	8.26-9.67	7.75-10.63	6.5-9.0
DO (mg/L)	0.63-4.44	0.78-9.89	0.32-8.82	0.57-9.45	>5
Ammonia (mg/L)	2.72-2.84	2.79-2.82	2.44-2.51	0.50-0.56	<0.05
Nitrite (mg/L)	0.216-0.221	0.226-0.237	0.042-0.109	0.002	<0.1

The temperature levels observed in all four ponds during the study fell within the optimal range of 25–32°C, aligning with the findings of Badiru (2005), who identified this temperature bracket as suitable for aquaculture. Minimal variation in temperature throughout the sampling period suggests environmental stability, a critical factor in sustaining aquaculture systems. This thermal consistency likely supported the diversity and abundance of phytoplankton in the ponds, as phytoplankton productivity typically peaks within a temperature range of 20–30°C (Tammi *et al.*, 2015). Furthermore, Sofarini (2012) noted that phytoplankton photosynthesis requires a minimum of 5°C and ceases at temperatures above 30°C. These observations collectively support the conclusion that the thermal conditions in the studied ponds were conducive to primary productivity and did not pose a limiting factor for aquaculture development.

Comparable studies reinforce this interpretation. Kausar and Salim (2006) demonstrated that warm water temperatures within this range enhance the metabolic and growth rates of *Labeo rohita*. Pörtner (2010) also emphasized that ectothermic organisms, such as fish, exhibit optimal physiological performance within species-specific thermal windows. These findings collectively validate the suitability of the recorded temperature range for sustaining aquaculture operations and maintaining a robust phytoplankton community.

The pH levels recorded in the ponds were predominantly within the 6.5–9.0 range, which is considered optimal for aquaculture (Gilmour *et al*., 2020). Although some readings approached the upper end of this range, none exceeded critical thresholds. This aligns with findings by Kocan *et al.* (2022), who reported that freshwater systems with slightly alkaline conditions support productive aquaculture operations. Conversely, Fashina-Bombata *et al*. (2021) emphasized the detrimental effects of pH levels below 6.5 on plankton productivity and overall aquatic ecosystem functioning.

Boyd (1990) and Lazur (2007) also noted that water pH significantly affects nutrient solubility, ammonia toxicity, and biological activity in fish ponds. As such, maintaining pH within the ideal range is crucial to preserving aquatic life. While the current study found that pH levels were largely supportive of aquaculture, the proximity to the upper threshold in some ponds suggests the need for periodic monitoring to prevent potential adverse effects.

Dissolved oxygen (DO) levels fluctuated considerably across sampling periods, with all ponds recording concentrations below 3 mg/L during early morning hours. These values fall short of the 5 mg/L recommended by Riche and Garling (2003) for optimal tilapia growth and the minimum of 3 mg/L reported by Ross (2002) for tilapia survival. These findings are consistent with those of Boyd (2010), who stated that DO levels in pond aquaculture are typically lowest at dawn due to overnight respiratory activity by aquatic organisms.

El-Shafai et al. (2004) similarly observed that wastewater-fed aquaculture systems often suffer from hypoxic conditions during the early morning, particularly under high organic loads. The low DO concentrations in this study suggest that oxygen availability could be a limiting factor for fish health and productivity, especially if such conditions persist for extended periods.

Ammonia concentrations in the studied ponds were found to exceed literature thresholds for safe aquaculture. Robinette (1976) identified unionized ammonia (NH_3) levels above 0.6 mg/L as toxic, with sub-lethal effects occurring at concentrations as low as 0.1 mg/L. Similarly, Meade (1985) highlighted interspecies variability in ammonia tolerance but agreed that elevated NH_3 is detrimental to fish health. The high ammonia levels observed may be attributed to ineffective wastewater treatment prior to pond discharge.

The process of ammonia oxidation, known as nitrification, was evident in the pond systems. According to Watson *et al*. (1981), autotrophic bacteria such as *Nitrosomonas* and *Nitrobacter* play key roles in converting ammonia to nitrite (NO_2^-) and then to nitrate (NO_3^-), which can be assimilated by phytoplankton. However, nitrite levels in the ponds were also found to be elevated, with some measurements exceeding the 0.1 mg NO_2-N/L threshold advised by Wickins (1982) for fish culture. This accumulation poses a potential toxicity risk and may explain variations in both phytoplankton abundance and zooplankton community structure across the ponds.

Tomasso (1994) further observed that high water temperatures and alkaline pH exacerbate ammonia toxicity, a dynamic likely at play in this study. Therefore, without effective management, the accumulation of nitrogenous waste could undermine the viability of wastewater-fed aquaculture.

The study found that insect larvae, rather than true zooplankton (e.g., *Daphnia*, *Bosmina*), dominated the samples. This deviates from typical freshwater systems where herbivorous zooplankton serve as primary consumers of phytoplankton (Wetzel, 2001). The lack of a clear relationship between phytoplankton abundance and zooplankton presence suggests disrupted trophic interactions, potentially due to elevated ammonia or imbalanced phytoplankton species composition.

Preferred phytoplankton species for zooplankton grazing, such as *Pediastrum spp.* and *Cosmarium spp.*, were not commonly observed. This may indicate a shift in phytoplankton assemblage under nutrient stress, reducing food availability for zooplankton. Jeppesen *et al.* (2007) documented similar findings in eutrophic or degraded aquatic systems, where high nutrient loads led to weak producer-consumer relationships.

These findings raise concerns about the sustainability of the current pond ecosystem for supporting fish populations, as a healthy zooplankton community is essential for early life stages of many fish species.

CHAPTER FIVE

CONCLUSIONS AND RECOMMENDATIONS

5.1 Conclusions

All parameters measured during the study fell within their respective limits for optimal fish production, with the notable exceptions of dissolved oxygen (DO), ammonia, and nitrite levels, which are critical for the health and survival of farmed fish. Temperature levels exhibited a consistent trend with DO levels throughout the study period, reflecting the breakdown of organic matter within the system. The fluctuations in dissolved oxygen levels indicated varying biological activities in the ponds at different times, suggesting a dynamic interaction between environmental conditions and aquatic life.

The pH levels recorded were predominantly at the upper boundary of the recommended range for aquaculture production, indicating persistently alkaline conditions. Such elevated pH levels can influence nutrient availability and overall fish health, potentially affecting growth rates and reproduction. Although the phytoplankton abundance was relatively low, it was still higher than that of the zooplankton, suggesting a possible imbalance in the ecosystem dynamics. The presence of Macrocystis spp. in the water samples indicated that the effluent may still contain fecal matter, pointing to concerns regarding water quality and potential contamination.

The zooplankton community was predominantly composed of insect larvae, with a noticeable absence of true zooplankton species such as copepods and calanoids. This absence may imply that the treated wastewater is not conducive to supporting the diverse zooplankton populations typically found in healthy aquaculture systems. True zooplankton play a crucial role in the aquatic food web, serving as a primary food source for many fish species. Therefore, their scarcity could limit the overall productivity and sustainability of fish farming in these ponds.

However, the prevalence of insect larvae may present an opportunity for preliminary experimental trials with fish species that feed on such organisms. Utilizing fish that consume insect larvae could provide insights into alternative aquaculture practices and potentially mitigate some of the challenges posed by the current water quality issues. Overall, while certain parameters were within acceptable limits for aquaculture, the significant presence of ammonia, nitrite, and the poor representation of true zooplankton suggest a need for further investigation and potential remediation strategies to enhance the viability of these ponds for sustainable fish farming.

5.2 Recommendations

- There must be continuous monitoring of water quality on the effluent (TWW) from the MTP to ascertain its conformity to standards for effluents from treatment plants to determine how suitable the effluent (TWW) can be employed in aquaculture production.
- Further studies should be carried out on the microbial loads in the TWW to confirm its suitability for the vegetable cultivation that is currently used for in the area.

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APPENDICES

Appendix 1. Mean pH values recorded during the study period in September

MEAN TEMPERATUE	POND 1	STDEV	POND 2	STDEV	POND 3	STDEV	POND 4	STDEV
6AM (A)	25.87	±0.06	26.57	±0.12	26.57	±0.15	26.07	±0.31
9AM	26.30	±0.17	27.00	±0.1	27.03	±0.25	26.63	±0.06
12 NOON	26.63	±0.12	27.73	±0.42	27.70	±0.44	27.23	±0.35
5PM	27.00	±0.46	28.20	±0.3	28.23	±0.31	27.30	±0.1
8PM	27.40	±0.26	28.10	±0.17	28.10	±0.26	27.67	±0.71
12 MID NIGHT	26.70	±0.36	27.53	±0.15	27.53	±0.15	26.23	±1.59
2AM	26.07	±0.21	27.13	±0.15	27.03	±0.15	25.87	±1.88
4AM	25.92	±0.12	26.60	±0.1	26.57	±0.06	26.29	±1.86
6AM (B)	25.70	±0.1	26.47	±0.06	26.43	±0.15	25.91	±1.06

Appendix 2. Mean pH values recorded during the study period in September

MEAN Ph	POND 1	STDEV	POND 2	STDEV	POND 3	STDEV	POND 4	STDEV
6AM (A)	3.38	±0.58	9.03	±0.09	9.15	±0.1	9.75	±0.14
9AM	9.01	±0.05	8.95	±0.01	9.14	±0.03	10.04	±0.14
12 NOON	9.48	±0.08	9.38	±0.08	9.38	±0.08	10.47	±0.06
5PM	9.56	±0.05	9.48	±0.04	9.66	±0.05	10.63	±0.05
8PM	9.37	±0.05	9.36	±0.05	9.59	±0.02	10.56	±0.09
12 MID NIGHT	8.82	±0.49	9.21	±0.13	9.47	±0.03	10.22	±0.1
2AM	8.76	±0.55	9.17	±0.12	9.39	±0.06	10.1	±0.11
4AM	8.6	±0.5	9.09	±0.01	9.31	±0.1	9.77	±0.27
6AM (B)	8.31	±0.57	9.06	±0.04	9.25	±0.07	9.71	±0.28

Appendix 3. Mean DO values recorded during the study period in September

MEAN DO	POND 1	STDEV	POND 2	STDEV	POND 3	STDEV	POND 4	STDEV
6AM (A)	1.89	±1.65	2.99	±1.49	1.33	±0.29	6.97	±1.84
9AM	2.68	±2.53	5.18	±0.84	3.99	±0.42	7.36	±0.68
12 NOON	2.79	±0.81	7.99	±2.15	5.73	±0.42	4.47	±2.98
5PM	2.87	±0.5	8.82	±3.67	8.82	±0.6	6.2	±0.35
8PM	3.04	±3.00	0.89	±2	8.82	±2.33	9.45	±3.48
12 MID NIGHT	2.5	±1.47	5.76	±2.28	6.99	±0.62	8.62	±1.31
2AM	2.06	±1.2	4.48	±0.72	3.82	±0.89	8.04	±1.41
4AM	1.77	±1.08	3.28	±0.56	1.86	±0.41	7.18	±1.66
6AM (B)	1.66	±1.16	2.63	±1.4	1.21	±0.19	6.5	±1.97

Appendix 4. Mean Temperature values recorded during the study period in October

MEAN TEMPERATUE	POND 1	STDEV	POND 2	STDEV	POND 3	STDEV	POND 4	STDEV
6AM (A)	26.43	±0.06	26.5	0.26	26.63	±0.23	26.44	±0.44
9AM	26.37	±0.31	26.43	0.32	26.73	±0.06	26.7	±0.10
12 NOON	26.63	±0.21	26.95	0.13	26.96	±0.17	26.88	±0.05
5PM	26.29	±0.23	27.19	±0.26	27.36	±0.14	27.37	±0.14
8PM	26.56	±0.06	27.18	±0.25	27.24	±0.05	27.27	±0.17
12 MID NIGHT	26.48	±0.02	26.79	±0.03	26.96	±0.09	27.01	±0.17
3AM	25.51	±0.09	26.14	±0.33	26.18	±0.22	26.11	±0.12
6AM(B)	25.43	±0.06	26.11	±0.04	25.34	±0.23	25.06	±0.26

Appendix 5. Mean pH values recorded during the study period in October

MEAN pH	POND 1	STDEV	POND 2	STDEV	POND 3	STDEV	POND 4	STDEV
6AM (A)	8.05	±0.29	8.37	±0.12	8.26	±0.05	8.48	±0.35
9AM	8.9	±0.13	8.95	±0.12	9.08	±0.15	9.79	±0.32
12 NOON	9.11	±0.10	9.35	±0.08	9.08	±0.18	9.41	±0.08
5PM	9.29	±0.04	9.44	±0.11	9.67	±0.16	9.7	±0.15
8PM	9.44	±0.11	9.41	±0.10	9.52	±0.09	9.41	±0.36
12 MID NIGHT	9.33	±0.13	9.25	±0.12	9.35	±0.12	8.92	±0.18
3AM	8.94	±0.15	8.82	±0.44	9.13	±0.08	8.73	±0.31
6AM(B)	8.12	±0.27	8.2	±0.16	8.31	±0.10	8.18	±0.21

Appendix 6. Mean DO values recorded during the study period in October

MEAN DO	POND 1	STDEV	POND 2	STDEV	POND 3	STDEV	POND 4	STDEV
6AM (A)	0.61	±0.06	0.77	±0.04	0.52	±0.22	0.57	±0.24
9AM	1.32	±0.53	1.59	±0.38	1.33	±0.10	2.26	±0.26
12 NOON	2.91	±0.12	4.70	±0.42	3.81	±0.52	3.69	±0.60
5PM	3.02	±0.09	6.27	±1.05	5.28	±0.94	4.94	±0.27
8PM	4.44	±0.47	6.40	±0.92	5.51	±1.03	5.74	±0.92
12 MID NIGHT	3.95	±0.64	5.78	±0.90	4.74	±0.97	5.14	±0.62
3AM	1.37	±1.00	3.15	±0.12	1.69	±0.97	3.11	±0.96
6AM(B)	0.63	±0.12	0.78	±0.41	0.32	±0.21	1.31	±0.41

Appendix 7. Abundance of plankton across ponds.

No. of Ponds	**Phyto (No/L)**	**Zoo (No/L)**	**Total plankton (No/L)**	**Phyto (%)**	**Zoo (%)**
Pond 1	**2232000**	**389**	**2232389**	**99.98**	**0.017**
Pond 2	**754560**	**1044**	**755604**	**99.86**	**0.138**
Pond 3	**1243840**	**676**	**1244516**	**99.95**	**0.054**

Appendix 8. Anova output for Temperature (September).

Anova: Single Factor

SUMMARY

Groups	*Count*	*Sum*	*Average*	*Variance*
Column 1	9	237.59	26.3988889	0.32788611
Column 2	9	245.33	27.2588889	0.43791111
Column 3	9	245.19	27.2433333	0.457175
Column 4	9	239.2	26.5777778	0.44389444

ANOVA

Source of Variation	*SS*	*Df*	*MS*	*F*	*P-value*	*F crit*
Between Groups	5.38156389	3	1.79385463	4.30473454	0.0116658	2.90111958
Within Groups	13.3349333	32	0.41671667			
Total	18.7164972	35				

Appendix 9. Anova output for Ph (September).

Anova: Single Factor

SUMMARY

Groups	*Count*	*Sum*	*Average*	*Variance*
Column 1	9	80.29	8.92111111	0.21693611
Column 2	9	82.73	9.19222222	0.03249444
Column 3	9	84.34	9.37111111	0.03278611
Column 4	9	89.25	9.91666667	0.76605

ANOVA

Source of Variation	*SS*	*df*	*MS*	*F*	*P-value*	*F crit*
Between Groups	4.77356389	3	1.59118796	6.07169154	0.00215921	2.90111958
Within Groups	8.38613333	32	0.26206667			
Total	13.1596972	35				

Appendix 10. Anova output for DO (September).

Anova: Single Factor

SUMMARY

Groups	*Count*	*Sum*	*Average*	*Variance*
Column 1	9	21.26	2.36222222	0.27229444
Column 2	9	51.02	5.66888889	7.11421111
Column 3	9	42.57	4.73	9.14085
Column 4	9	64.79	7.19888889	2.10078611

ANOVA

Source of Variation	*SS*	*df*	*MS*	*F*	*P-value*	*F crit*
Between Groups	110.816067	3	36.9386889	7.9318033	0.00042909	2.90111958
Within Groups	149.025133	32	4.65703542			
Total	259.8412	35				

Appendix 11. Anova output for Ammonia (September).

Anova: Single Factor

SUMMARY

Groups	*Count*	*Sum*	*Average*	*Variance*
Column 1	2	5.5632	2.7816	0.007442
Column 2	2	5.612	2.806	0.00029768
Column 3	2	4.9532	2.4766	0.00267912
Column 4	2	1.0614	0.5307	0.0018605

ANOVA

Source of Variation	*SS*	*df*	*MS*	*F*	*P-value*	*F crit*
Between Groups	7.11609622	3	2.37203207	772.692929	5.5606E-06	6.59138212
Within Groups	0.0122793	4	0.00306983			
Total	7.12837552	7				

Appendix 12. Anova output for Nitrite (September).

Anova: Single Factor

SUMMARY

Groups	*Count*	*Sum*	*Average*	*Variance*
Column 1	2	0.437	0.2185	0.0000125
Column 2	2	0.463	0.2315	6.05E-05
Column 3	2	0.151	0.0755	0.0022445
Column 4	2	0.004	0.002	0

ANOVA

Source of Variation	*SS*	*df*	*MS*	*F*	*P-value*	*F crit*
Between Groups	0.07494938	3	0.02498313	43.1208198	0.0016698	6.59138212
Within Groups	0.0023175	4	0.00057938			
Total	0.07726688	7				

Appendix 13. Anova output for Temperature (October).

Anova: Single Factor

SUMMARY

Groups	*Count*	*Sum*	*Average*	*Variance*
Column 1	8	209.7	26.2125	0.22150714
Column 2	8	213.29	26.66125	0.18624107
Column 3	8	213.4	26.675	0.42617143
Column 4	8	212.84	26.605	0.5622

ANOVA

Source of Variation	*SS*	*df*	*MS*	*F*	*P-value*	*F crit*
Between Groups	1.15518438	3	0.38506146	1.10323341	0.36429319	2.94668527
Within Groups	9.7728375	28	0.34902991			
Total	10.9280219	31				

Appendix 14. Anova output for Ph (October).

Anova: Single Factor

SUMMARY

Groups	*Count*	*Sum*	*Average*	*Variance*
Column 1	8	71.18	8.8975	0.28645
Column 2	8	71.79	8.97375	0.23042679
Column 3	8	72.4	9.05	0.26788571
Column 4	8	72.62	9.0775	0.34633571

ANOVA

Source of Variation	*SS*	*df*	*MS*	*F*	*P-value*	*F crit*
Between Groups	0.15760937	3	0.05253646	0.1857892	0.90516687	2.94668527
Within Groups	7.9176875	28	0.28277455			
Total	8.07529688	31				

Appendix 15. Anova output for DO (October).

Anova: Single Factor

SUMMARY

Groups	*Count*	*Sum*	*Average*	*Variance*
Column 1	8	18.25	2.28125	2.23715536
Column 2	8	29.44	3.68	5.86914286
Column 3	8	23.2	2.9	4.70571429
Column 4	8	26.76	3.345	3.52791429

ANOVA

Source of Variation	*SS*	*df*	*MS*	*F*	*P-value*	*F crit*
Between Groups	8.77913438	3	2.92637813	0.71637484	0.55052902	2.94668527
Within Groups	114.379488	28	4.0849817			
Total	123.158622	31				