Open Access Research Journal of
Science and Technology
Journals home page: https://oarjst.com/
ISSN: 2782-9960 (Online)

OARJ OPEN ACCESS RESEARCH JOURNALS

(RESEARCH ARTICLE)

Effects of Artisanal Crude Oil Refining Activities on Surface Water Quality Indices in Rumuodogo Community, Niger Delta, Nigeria

Gift Kiisi Nkin [1,*], Jack Gbenenee Tetam [2] and Ndidi Nwachoko [2]

[1] *Department of Chemistry, Faculty of Science, Rivers State University, P.M.B 5080 Nkpolu-Oroworukwo Port Harcourt, Rivers State, Nigeria.*
[2] *Department of Biochemistry, Faculty of Science, Rivers State University, P.M.B 5080 Nkpolu-Oroworukwo Port Harcourt, Rivers State, Nigeria.*

Open Access Research Journal of Science and Technology, 2025, 15(02), 180-194

Publication history: Received 11 November 2025; revised on 16 December 2025; accepted on 18 December 2025

Article DOI: https://doi.org/10.53022/oarjst.2025.15.2.0148

Abstract

Artisanal crude oil refining, also referred to as illegal crude oil refining, *Kpo-fire* or oil bunkering, involves the unauthorized and unregulated processing of crude oil and is prevalent in oil-producing areas of the Niger Delta, Nigeria. This study assessed the effects of illegal crude oil refining activities on selected surface water quality indices in Mini-Ogboro, Rumuodogo Community, Rivers State, Nigeria. Surface water samples were collected from three sampling stations (upstream, midstream, and downstream) and analyzed for physicochemical parameters, as well as microbial quality. physicochemical parameters such as temperature, turbidity, and phosphate concentrations were above recommended WHO limits. Bacteriological analysis revealed total heterotrophic bacteria (THB) counts of 9.0×10^3, 15.2×10^3, and 8.4×10^3 CFU/mL; total coliform counts (TCC) of 2.0×10^3, 3.7×10^3, and 1.3×10^3 CFU/mL; and total faecal coliform counts (TFC) of 7.0×10^2, 5.0×10^2, and 2.0×10^2 CFU/mL at the respective sampling locations. These values exceeded World Health Organization (WHO) permissible limits for surface water quality. The findings indicate significant deterioration of surface water quality associated with illegal crude oil refining activities. Consequently, immediate interventions, including the installation of appropriate water treatment and filtration systems, are recommended to reduce turbidity and microbial contamination and protect public health.

Keywords: Artisanal oil refining; Surface water pollution; Water quality indices (WQI); Physicochemical and microbial parameters; Niger Delta

1. Introduction

Clean, safe, and adequate freshwater is fundamental to human survival and the sustainability of all living components within the ecosystem. Access to safe and potable drinking water is not only a basic human necessity but also a recognized human right that underpins health, food security, and socio-economic development. This importance is reflected in the United Nations Sustainable Development Goal (SDG) 6, which aims to ensure universal access to clean water and sanitation by 2030. Central to this goal is the improvement of water quality through the reduction of pollution, elimination of indiscriminate waste disposal, and minimization of the release of hazardous chemical substances into water bodies, thereby promoting the safe use and reuse of water resources globally [1]. From the earliest periods of human civilization to the present day, water has been indispensable for domestic, agricultural, and industrial purposes, including drinking, bathing, livestock rearing, and irrigation. Despite its essential role, freshwater remains a limited natural resource that is increasingly under threat, primarily due to anthropogenic activities [2, 37, 38]. Water is often described as the "universal solvent" because of its exceptional capacity to dissolve substances, a property that makes it vital for regulating body temperature, transporting nutrients, facilitating digestion, and eliminating metabolic wastes and toxins from the human body. Consequently, water use transcends nationality, culture, religion, and socio-economic status, making it one of the most critical determinants of human health and development

*Corresponding author: Gift Kiisi Nkin

worldwide [3]. However, the quality of available freshwater resources has been severely compromised by increasing pressures from population growth, intensified agricultural practices, natural resource exploitation, oil exploration, mining activities, urbanization, and industrial development [4, 37, 38]. Anthropogenic activities such as indiscriminate dumping of solid and liquid wastes, onshore and offshore hydrocarbon spillages, and open defecation contribute significantly to the introduction of potentially toxic elements (PTEs) and pathogenic microorganisms into surface and groundwater systems [4, 39]. Given that water quality is a major determinant of public health, the consumption of chemically or biologically contaminated water poses serious risks to human populations.

Globally, waterborne diseases remain a major public health concern, with an estimated 2.3 billion people affected and approximately 485,000 deaths annually attributed to diarrhoeal diseases resulting from contaminated drinking water [1]. The World Health Organization further reports that water contamination contributes to about 70% of various diseases and nearly 20% of cancer cases worldwide. Water quality challenges are complex and multifaceted, necessitating urgent global attention and coordinated action [5]. The continuous decline in water quality is particularly concerning due to its potential to disrupt the hydrological cycle and degrade aquatic ecosystems. In many rural communities, drinking water sources are increasingly contaminated with organic pollutants and heavy metals, with elevated concentrations often linked to illegal oil refining activities [6, 37, 38, 39, 40, 41].

In the Niger Delta region of Nigeria, illegal crude oil refining, commonly referred to as artisanal refining, has emerged as a major environmental challenge with far-reaching implications for water quality, ecosystem health, and human well-being [41]. The region is characterized by widespread environmental degradation, including soil erosion, frequent oil spills, water pollution, gas flaring, loss of biodiversity, inadequate socio-economic development, and persistent poverty. Illegal oil refining and exploitation have evolved over several decades and involve the unauthorized extraction and processing of crude oil using rudimentary and unsafe technological methods [41]. According to the Nigerian Environmental Study/Action Team (1991), as cited by Mba (1995), mineral resources are broadly categorized into fuel minerals, metallic minerals, and industrial minerals, each associated with different forms of illegal exploitation and refining processes [7]. The global recognition of environmental sustainability as a cornerstone of national development is reflected in the Sustainable Development Goals, which emphasize the reduction of environmental degradation and pollution. In several parts of the world, oil spills and related activities from illegal refining have resulted in long-term environmental damage, including contamination of surface and groundwater resources, destruction of farmlands, and loss of vegetation [8]. The negative impacts of oil refining activities include oil spillage, gas flaring, and the discharge of toxic chemicals used during crude oil processing, all of which pose significant threats to water quality and public health [9, 40, 41].

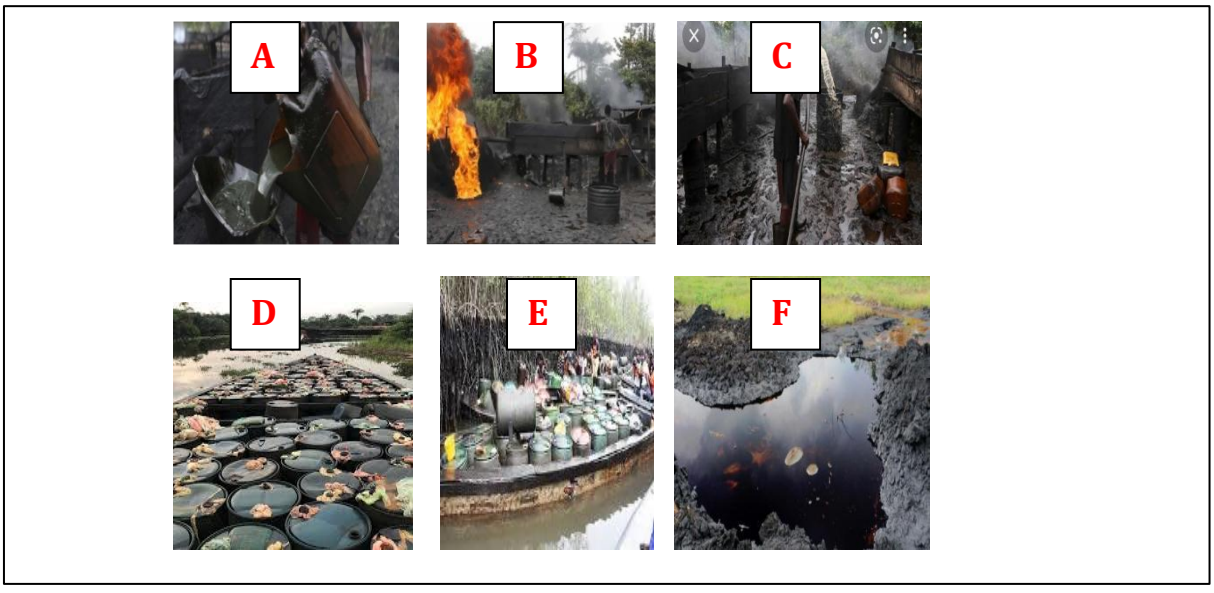

Figure 1 Illustrates the sequential stages involved in illegal artisanal crude oil refining at Rumuodogo Community in the Niger Delta region: (A) crude oil is sourced and transferred into an artisanal refining pot; (B) thermal processing (local heating) of the crude oil; (C) collection of refined products following heating; (D) storage of the collected products in drums; (E) transportation of the products; and (F) accidental discharge of products into surface water [36]

In Rumuodogo Community of the Niger Delta, the increasing prevalence of illegal crude oil refining activities is suspected to have significantly compromised surface water quality, thereby threatening the health and food security of residents living in close proximity to refining sites [10]. Prolonged exposure to contaminated water has been associated with serious health outcomes, including immune suppression, cancer, reproductive disorders, and acute poisoning [11, 12]. In view of these concerns and the absence of adequate water quality monitoring in the area, there is a compelling need to assess surface water quality using the Water Quality Index (WQI) approach [13]. This study therefore evaluates the effects of illegal crude oil refining activities on selected surface water quality indices in a Niger Delta community, with the aim of providing scientific evidence to inform public health protection, environmental management, and policy interventions [10, 13].

2. Materials and methods

2.1. Study Area Description

Mini-Ogboro is a riverine settlement located within the Rumuodogo community in Emohua Local Government Area (LGA) of Rivers State, southern Nigeria [14]. The area lies in the central part of the Niger Delta, a region characterized by extensive wetlands, mangrove swamps, back-swamp floodplains, and interlinked creeks that discharge into major rivers [15]. Mini-Ogboro is situated on low-lying coastal terrain dominated by alluvial and organic-rich sediments, with elevations generally less than 10 m above sea level [16]. The hydrology of the area is controlled by tidal fluctuations, seasonal flooding, and surface runoff, creating a dynamic aquatic environment where water quality is highly responsive to anthropogenic disturbances [17]. The climate is humid tropical, marked by high annual rainfall (most intense between April and October), elevated humidity, and consistently warm temperatures [18]. These conditions promote vigorous biological productivity but also enhance the mobility and dispersion of contaminants in aquatic environments [19]. Vegetation in and around Mini-Ogboro consists predominantly of freshwater swamp species, fringing mangroves, marsh grasses, and patches of secondary forest interspersed with small-scale agricultural fields [20]. Human activities in Mini-Ogboro are typical of rural Niger Delta communities, including artisanal fishing, subsistence farming, and local trading [21]. However, the area is also impacted by widespread artisanal crude oil refining ("illegal refining" or *Kpo-fire*). These activities involve crude oil tapping, makeshift refining facilities along creek lines, frequent spills, and open-burning of petroleum residues [22]. Such practices introduce hydrocarbons, soot, heavy fractions, and combustion by-products into surrounding water bodies, making Mini-Ogboro a relevant location for assessing the degradation of water quality and associated ecological risks [23]. Access to the study area is mainly through narrow laterite roads and navigable creeks, many of which are bordered by abandoned or active artisanal refining sites. Sampling points for this study were selected along surface water bodies receiving direct or diffuse inputs from illegal refining operations. The combination of hydrological connectivity, petroleum-related disturbances, and community dependence on natural waterways makes Mini-Ogboro an important case study for understanding the environmental consequences of illegal oil refining in the Niger Delta.

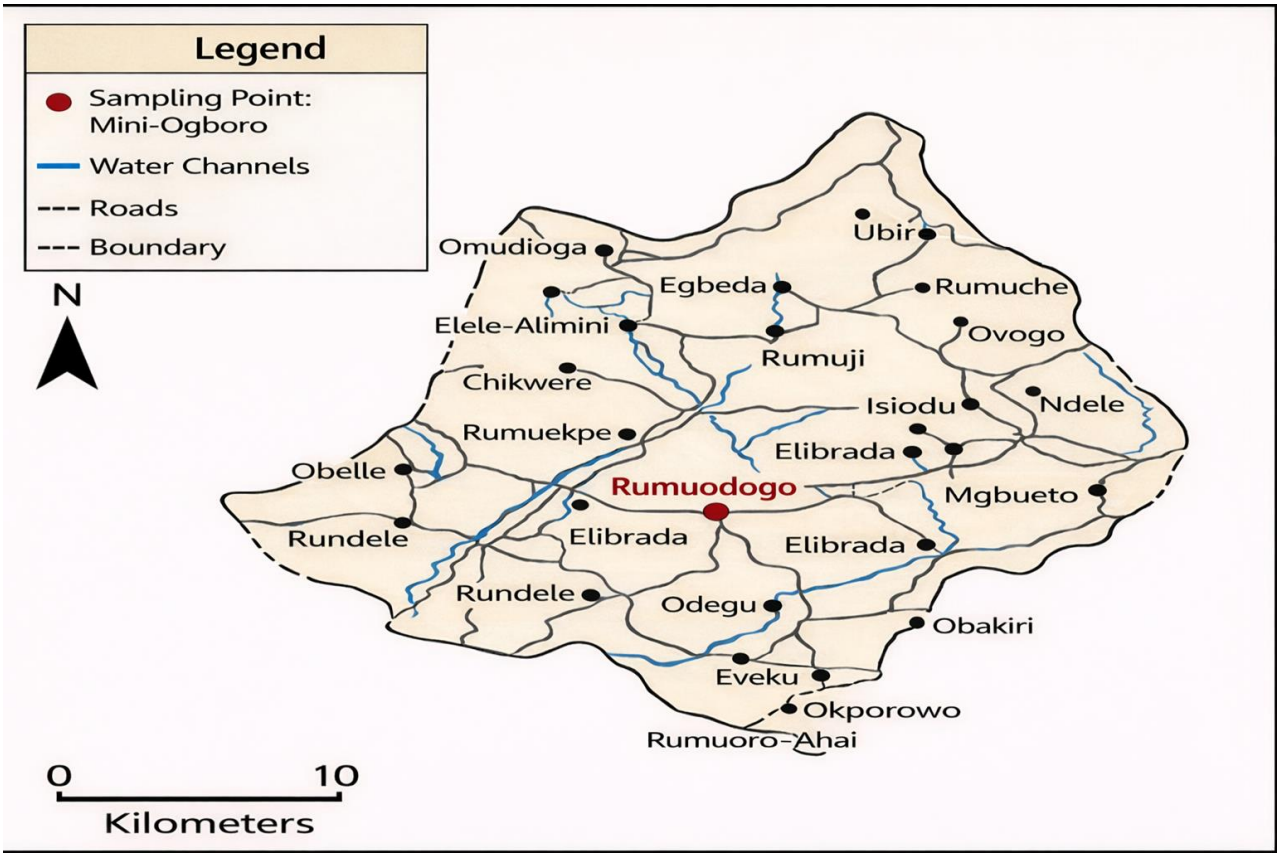

Figure 2 Map of the study area showing nearby communities and sampling points [42]

2.2. Sample collection

Water samples were collected from three designated sampling locations, upstream, midstream, and downstream, within the Mini-Ogboro area of Rumuodogo Community, Rivers State. These locations were strategically selected to capture spatial variations in water quality and to assess the extent of influence from illegal oil refining activities along the watercourse. At each site, samples were collected following standard procedures to prevent contamination, with bottles rinsed using the sample water prior to final collection. Immediately after sampling, all containers were sealed, properly labeled with the date, time, and sampling point identification, and placed in ice-packed coolers to maintain their physical and chemical integrity. The samples were then transported to the laboratory under controlled conditions for detailed physicochemical and organic pollutant analysis. All handling procedures adhered strictly to recommended guidelines to ensure the reliability and reproducibility of the analytical results.

2.3. Media Preparation

2.3.1. Nutrient Agar

Nutrient agar was employed for the isolation and enumeration of Total Heterotrophic Bacteria (THB) from the collected water samples. The medium was prepared according to the manufacturer's specifications and established microbiological procedures to ensure consistency and reliability of microbial growth. To prepare the medium, 28 g of commercially available nutrient agar powder was accurately weighed and transferred into a 1,000 mL Erlenmeyer flask containing an appropriate volume of distilled water. The mixture was gently swirled and heated over a Bunsen burner flame until the agar dissolved completely, producing a clear and homogeneous solution. The heating process lasted approximately 30 minutes and ensured that all components were fully solubilized. Following dissolution, the medium was sterilized in an autoclave at 121°C for 15 minutes under a pressure of 15 psi to eliminate all potential contaminants. After sterilization, the flask was removed and allowed to cool to approximately 45°C, a suitable temperature for pouring without compromising microbial viability or causing condensation. At this point, 15 mL of the molten nutrient agar was aseptically dispensed into sterile Petri dishes within a laminar flow hood to prevent contamination. The poured plates were allowed to solidify and subsequently dried in a hot-air oven to remove excess surface moisture, ensuring optimal

conditions for bacterial colony formation. All prepared plates were stored in sterile conditions and used promptly for microbiological analysis.

2.3.2. MacConkey Agar

MacConkey agar was prepared and used for the selective isolation and enumeration of Gram-negative enteric bacteria from the water samples. The medium was prepared following standard microbiological procedures to ensure accuracy and sterility throughout the process. To prepare the medium, 36 g of the agar powder was accurately weighed and transferred into a 1,000 mL Erlenmeyer flask containing an appropriate volume of distilled water. The mixture was swirled gently and then heated over a Bunsen burner flame until it reached a vigorous boil, ensuring complete dissolution of all components. This heating process lasted approximately 30 minutes, during which the medium became homogenous and free of particulate matter. After the agar had fully dissolved, the medium was sterilized in an autoclave at 121°C for 15 minutes under a pressure of 15 psi to eliminate any microbial contaminants. Upon completion of sterilization, the flask was removed and allowed to cool to approximately 45°C, a temperature suitable for pouring without premature solidification or thermal damage to the medium. Once cooled, 15 mL of the molten agar was aseptically dispensed into sterile Petri dishes inside a laminar flow cabinet to prevent airborne contamination. After pouring, the plates were allowed to solidify and subsequently dried in a hot-air oven to remove excess surface moisture, ensuring optimal conditions for bacterial growth. The prepared plates were then stored under sterile conditions and used promptly for microbial analysis.

2.3.3. Enumeration and Isolation of Microorganisms

For the microbiological evaluation, aliquots of the water samples were processed using standard plate count techniques to quantify the microbial populations present. An aliquot of 0.1 mL from each appropriately prepared sample was aseptically transferred onto sterile agar plates in duplicate to ensure accuracy and reproducibility of results. The aliquots were then evenly distributed across the surface of the agar using a sterile glass spreader, employing the spread plate method to facilitate the development of well-separated colonies. Following inoculation, the plates were incubated in an inverted position to prevent condensation from interfering with colony formation. Plates prepared for the enumeration of Total Heterotrophic Bacteria (THB) and Total Coliform Bacteria were incubated at 37°C, which represents the optimal growth temperature for most mesophilic bacteria of public health significance. For Total Fecal Coliforms, samples were inoculated onto Eosin Methylene Blue (EMB) agar and incubated at 44.5°C for 24 hours, a selective temperature that favors the growth of thermotolerant coliforms typically associated with fecal contamination. At the end of the incubation period, each plate was carefully examined, and the distinct colonies that developed were counted using a colony counter or visual assessment, depending on colony density. Colony counts from duplicate plates were recorded and expressed as colony-forming units per milliliter (CFU/mL). These data provided quantitative estimates of the microbial load present in the water samples and served as indicators of the sanitary quality of the study area.

2.4. Biochemical Analysis

2.4.1. Determination of Temperature

Water temperature was determined in accordance with standard analytical procedures. A calibrated mercury-in-glass thermometer was gently immersed into a 50 mL aliquot of each water sample contained in a clean sample bottle. Care was taken to ensure that the thermometer bulb was fully submerged and not in contact with the walls or bottom of the container to avoid measurement bias. The thermometer was allowed to equilibrate with the sample for approximately 5 minutes, after which the temperature was read directly from the scale and recorded in degrees Celsius (°C). All measurements were conducted at ambient laboratory conditions, and readings were taken promptly to minimize temperature fluctuations resulting from environmental exposure.

2.4.2. Determination of pH

The pH of the water samples was measured using a digital pH meter equipped with a glass electrode, following standard analytical procedures. Prior to measurement, the pH meter was calibrated using standard buffer solutions at pH 4.0 and pH 7.0 to ensure accuracy and reliability of readings. For each measurement, 50 mL of the water sample was transferred into a clean, dry beaker. The glass electrode was carefully immersed into the water sample, ensuring that the electrode was fully submerged but not in contact with the walls or bottom of the beaker to avoid erroneous readings. The sample was allowed to equilibrate with the electrode for a few minutes until a stable pH reading was obtained, which was then recorded in pH units. Between successive measurements, the electrode was thoroughly rinsed with distilled water to prevent cross-contamination between samples. All measurements were conducted at ambient laboratory temperature, and readings were taken in triplicate to improve precision and reproducibility.

2.4.3. Determination of Turbidity

Turbidity of the water samples was determined using a calibrated colorimeter, following standard analytical procedures. Prior to analysis, the instrument was calibrated with distilled water to ensure accurate baseline readings. Each sample bottle was thoroughly rinsed first with distilled water and then with the water sample to be analyzed to minimize contamination and carry-over effects. Subsequently, 50 mL of the water sample was poured into the cleaned sample bottle, which was securely placed in the colorimeter compartment. The instrument was allowed to stabilize for a few minutes to ensure consistent measurements. Using the control panel, the appropriate mode was selected, and turbidity was measured in Nephelometric Turbidity Units (NTU). The turbidity values were recorded directly from the instrument display. All measurements were performed in triplicate to ensure precision, and the instrument was recalibrated periodically to maintain accuracy throughout the analysis.

2.4.4. Determination of Electrical Conductivity (EC)

Electrical conductivity (EC) of the water samples was determined using a digital conductivity meter equipped with a suitable probe, following standard analytical procedures. Prior to measurement, the conductivity meter was calibrated according to the manufacturer's instructions using a standard conductivity solution to ensure accuracy of readings. For each analysis, 50 mL of the water sample was transferred into a clean, dry beaker. The conductivity probe was then gently immersed into the sample, ensuring that the electrode was fully submerged and not in contact with the sides or bottom of the beaker to avoid measurement interference. The sample was allowed to equilibrate, and readings were taken once a stable value was displayed on the instrument. Electrical conductivity values were recorded in microsiemens per centimeter (μS/cm). Between successive measurements, the probe was rinsed thoroughly with distilled water to prevent cross-contamination between samples. All measurements were conducted at ambient laboratory temperature, and analyses were performed in triplicate to enhance precision and reproducibility.

2.4.5. Determination of Total Hardness (TH)

Total hardness of the water samples was determined using the ethylenediaminetetraacetic acid (EDTA) complexometric titration method in accordance with standard analytical procedures. For each analysis, 25 mL of the water sample was accurately measured into a clean conical flask. Subsequently, 1 mL of ammonium buffer solution was added to adjust the pH of the sample to approximately 10.0, which is optimal for calcium and magnesium ion complexation. A small quantity of Eriochrome Black T indicator was then added using a clean spatula, resulting in the formation of a wine-red or purple-colored complex. The sample was titrated with standardized 0.01 N EDTA solution, which was added gradually from a burette with continuous swirling until a distinct color change from purple to blue was observed, indicating the end point of the titration. The volume of EDTA consumed at the end point was carefully recorded. All titrations were carried out in triplicate to ensure accuracy and reproducibility, and reagent blanks were analyzed alongside samples to correct for potential interferences. Total hardness was calculated and expressed as calcium carbonate ($CaCO_3$) concentration in parts per million (ppm) using the following equation:

$$\text{Total Hardness (as CaCO3, ppm)} = \frac{\text{Tv} \times \text{M} \times 100 \times 1000}{V}$$

Where:

Tv = Volume (mL) of EDTA titrant used at the end point,

M = Molarity of the EDTA solution, and

V = Volume (mL) of the water sample analyzed.

2.4.6. Determination of Total Alkalinity

Total alkalinity of the water samples was determined using the acid-base titration method in accordance with standard analytical procedures. For each analysis, 50 mL of the water sample was accurately measured and transferred into a clean conical flask. Two to three drops of methyl orange indicator were added to the sample, resulting in an orange coloration indicative of alkaline conditions. The sample was then titrated with standardized 0.02 N Sulfuric acid (H_2SO_4), which was added gradually from a burette with continuous swirling to ensure thorough mixing. Titration was continued until a distinct and permanent color change from orange to pink was observed, indicating the end point. The volume of sulfuric acid consumed at the end point was carefully recorded. All determinations were carried out in triplicate to improve accuracy and reproducibility, and reagent blanks were analyzed where necessary to account for potential interferences. Total alkalinity was calculated and expressed as calcium carbonate ($CaCO_3$) concentration in parts per million (ppm) using the equation:

$$\text{Total Alkalinity (as CaCO3, ppm)} = \frac{\text{Tv} \times \text{N} \times 50 \times 1000}{V}$$

Where:

Tv = Volume (mL) of 0.02 N H_2SO_4 used at the end point,

N = Normality of the sulfuric acid solution, and

V = Volume (mL) of the water sample analyzed.

2.4.7. Determination of Nitrate Content by Brucine Colourimetric Method

Nitrate concentration in the water samples was determined using the Brucine colourimetric method following standard analytical procedures. Prior to analysis, all glassware, including test tubes, was thoroughly washed and rinsed with distilled water to eliminate potential contamination. For each determination, 2 mL of the water sample was accurately measured using a graduated cylinder and transferred into a clean test tube. Subsequently, 2 mL of aqueous sulfuric acid solution (4:1, v/v) was carefully added to each test tube to facilitate the nitration reaction. The test tubes were gently warmed and then suspended in a beaker containing distilled water to allow controlled cooling. Thereafter, 0.2 mL of Brucine reagent was added to each sample, resulting in the development of a light yellow coloration. The reaction mixture was then heated for approximately 20 minutes until a distinct golden-yellow color developed, indicating the presence of nitrate ions. Following color development, the samples were allowed to cool to room temperature to stabilize the absorbance. The absorbance of each sample was measured using a UV-Visible spectrophotometer at a wavelength of 410 nm against a reagent blank prepared under identical conditions. All analyses were performed in triplicate to ensure accuracy and reproducibility. Nitrate concentration in the water samples was calculated using the calibration equation:

$$NO_3^- \text{ (mg/L)} = [(A - B) \times 1.8078] + 0.0354$$

Where:

A = Absorbance of the water sample, and

B = Absorbance of the reagent blank.

2.4.8. Determination of Chloride by Argentometric Method

Chloride concentration in the water samples was determined using the argentometric (Mohr) titration method in accordance with standard analytical procedures. For each analysis, 50 mL of the water sample was accurately measured using a graduated cylinder and transferred into a clean conical flask. Subsequently, 0.2 mL of potassium dichromate ($K_2Cr_2O_7$) indicator solution was added to the sample using a pipette. The indicator imparted a yellow coloration to the solution, facilitating endpoint detection. The sample was titrated with standardized silver nitrate ($AgNO_3$) solution, which was gradually added from a burette with continuous swirling to ensure uniform mixing. During the titration, silver ions reacted with chloride ions to form a white precipitate of silver chloride (AgCl). Titration was continued until a distinct brick-red coloration, accompanied by the appearance of reddish-brown silver chromate crystals, was observed, indicating the end point of the reaction. The volume of silver nitrate solution consumed at the endpoint was recorded as the titre value. The procedure was repeated for all water samples, and blank determinations were conducted using distilled water to account for reagent impurities. All titrations were performed in triplicate to enhance analytical precision and reproducibility. Chloride concentration was calculated and expressed in milligrams per liter (mg/L) using the standard argentometric equation:

$$\text{Chloride (mg/L)} = \frac{(\text{T} - \text{B}) \times \text{N} \times 35.45 \times 1000}{V}$$

Where:

T = Titre value (mL) of $AgNO_3$ used for the sample,

B = Titre value (mL) of $AgNO_3$ used for the blank,

N = Normality of the silver nitrate solution, and

V = Volume (mL) of the water sample analyzed.

2.4.9. Determination of Calcium by EDTA Titration Method

Calcium concentration in the water samples was determined using the ethylenediaminetetraacetic acid (EDTA) complexometric titration method in accordance with standard analytical procedures. For each determination, 25 mL of the water sample was accurately measured and transferred into a clean conical flask. To selectively precipitate magnesium and maintain an alkaline medium favorable for calcium determination, 0.5 mL of 1 N sodium hydroxide (NaOH) solution was added. A few crystals of murexide indicator were then introduced, and the solution was swirled thoroughly until a distinct blue coloration was obtained. The sample was subsequently titrated with standardized 0.01 N EDTA solution, which was added gradually from a burette with continuous swirling. EDTA formed a stable chelate complex with calcium ions according to the reaction:

$$\text{EDTA} + Ca^{2+} \rightarrow \text{Ca–EDTA}^{2-}$$

Titration was continued until a clear color change from blue to purple was observed, indicating the end point. The volume of EDTA consumed at the end point was recorded. All determinations were carried out in triplicate to ensure analytical accuracy and reproducibility, and reagent blanks were analyzed to correct for potential interferences. Calcium hardness was calculated and expressed as milligrams per liter (mg/L) using the equation:

$$\text{Calcium Hardness((mg/L)} = \frac{(\text{Volume of EDTA} \times \text{Molarity of EDTA}) \times 105}{\text{Volume of sample}}$$

The calcium concentration was then obtained from calcium hardness using the conversion factor:

$$\text{Calcium (mg/L)} = \text{Calcium Hardness} \times 0.4$$

2.4.10. Determination of Sulphate by Turbidimetric Method

Sulphate concentration in the water samples was determined using the turbidimetric method following standard analytical procedures. Prior to analysis, the UV-Visible spectrophotometer was switched on and allowed to warm up for approximately 15 minutes to ensure instrumental stability. All glassware, including conical flasks, was thoroughly washed and rinsed with distilled water and subsequently rinsed with the water sample to minimize contamination. For each determination, 10 mL of the water sample was accurately measured and transferred into a clean conical flask. A volume of 0.5 mL of conditioning reagent was then added to the sample to stabilize the reaction medium and promote uniform crystal formation. The initial absorbance of the sample (A_1) was measured at a wavelength of 420 nm using the spectrophotometer. Subsequently, a small quantity of barium chloride ($BaCl_2$) crystals was added to the sample, and the mixture was gently swirled to ensure thorough mixing. The reaction mixture was allowed to stand for approximately 5 minutes to enable complete precipitation and development of turbidity. During this period, sulphate ions in the sample reacted with barium ions in an acidic medium to form a fine suspension of barium sulphate ($BaSO_4$) crystals of uniform size, resulting in a milky appearance. The turbidity produced is directly proportional to the sulphate concentration in the water sample. After the reaction period, the final absorbance (A_2) of the turbid solution was measured at 420 nm against a reagent blank prepared under identical conditions. The net absorbance (A) due to sulphate was obtained by subtracting the initial absorbance from the final absorbance, as expressed below:

$$Concentration\ (mg/L) = \left[(A - B) \times 79.663\right] - 0.1523$$

Where:

$A = A_2 - A_1$

A_1 = Initial absorbance value before the addition of $BaCl_2$,

A_2 = Final absorbance value after the addition of $BaCl_2$, and

B = Absorbance of the blank.

All measurements were carried out in triplicate to ensure accuracy and reproducibility. Sulphate concentration was determined from a calibration curve prepared using standard sulphate solutions and expressed as parts per million (ppm) or milligrams per liter (mg/L) SO_4^{2-}.

2.4.11. Determination of Phosphate by Stannous Chloride Method

Phosphate concentration in the water samples was determined using the stannous chloride colorimetric method based on the formation of a phosphomolybdate complex. In an acidic medium, phosphate ions react with ammonium molybdate $(NH4)_2MoO_4$ to form ammonium phosphomolybdate, which is subsequently reduced by stannous chloride to produce a blue-colored molybdenum complex. The intensity of the blue coloration is directly proportional to the phosphate concentration in the sample. Prior to analysis, the UV-Visible spectrophotometer was switched on and allowed to warm up for approximately 15 minutes to ensure wavelength stability. All glassware, including test tubes and conical flasks, was thoroughly cleaned, rinsed with distilled water, and subsequently rinsed with the water sample to minimize contamination. For each determination, 25 mL of the water sample was accurately measured and transferred into a clean conical flask. Subsequently, 0.5 mL of ammonium molybdate reagent was added, and the solution was gently mixed to ensure uniform reaction. The initial absorbance of the reaction mixture (A_1) was measured at a wavelength of 690 nm using the spectrophotometer prior to the addition of the reducing agent. Thereafter, 0.2 mL of stannous chloride solution was added to the mixture, and the solution was thoroughly mixed. The reaction mixture was allowed to stand for approximately 12 minutes to enable complete color development, during which a characteristic blue coloration was observed. The final absorbance (A_2) of the developed color was then measured at 690 nm against a reagent blank prepared under identical conditions. The net absorbance attributable to phosphate was obtained by subtracting the initial absorbance from the final absorbance. All analyses were performed in triplicate to ensure accuracy and reproducibility. Phosphate concentration in the water samples was calculated and expressed in milligrams per liter (mg/L) using the equation:

$$\text{Phosphate (mg/L)} = [(A-B) \times 0.2939] + 0.0133$$

Where:

A = Absorbance of the sample, and

B = Absorbance of the reagent blank.

2.4.12. Determination of Magnesium

Magnesium concentration in the water samples was determined indirectly from measured total hardness and calcium hardness values using standard analytical relationships. Total hardness (TH), expressed as calcium carbonate ($CaCO_3$), was first determined by EDTA complexometric titration, while calcium hardness (CaH) was obtained separately using the murexide-EDTA titration method. Magnesium hardness (MgH) was then calculated as the difference between total hardness and calcium hardness, as expressed by the equation:

$$\text{MgH} = \text{TH} - \text{CaH}$$

Where:

TH = Total hardness (as $CaCO_3$, mg/L),

CaH = Calcium hardness (as $CaCO_3$, mg/L), and

MgH = Magnesium hardness (as $CaCO_3$, mg/L).

The concentration of magnesium was subsequently calculated by converting magnesium hardness to magnesium ion concentration using the standard conversion factor:

$$\text{Mg (mg/L)} = \text{MgH} \times 0.244$$

All calculations were performed for each sampling location, and analyses were conducted in triplicate to ensure accuracy and reproducibility. This indirect method is widely accepted for magnesium determination in water quality assessment and provides reliable estimates when total and calcium hardness values are accurately measured.

2.5. Quality Assurance and Quality Control (QA/QC)

Quality assurance and quality control measures were strictly implemented throughout the sampling, laboratory analysis, and data interpretation processes to ensure the accuracy, precision, and reliability of the results obtained in this study.

2.5.1. Calibration of Instruments

All analytical instruments used for physicochemical and microbial analyses were calibrated prior to use and at regular intervals in accordance with the manufacturers' specifications and standard analytical protocols. The pH meter was calibrated using standard buffer solutions (pH 4.0 and 7.0), while the conductivity meter was calibrated with standard conductivity solutions. The spectrophotometer was wavelength-calibrated and zeroed using distilled water or appropriate reagent blanks before absorbance measurements. Turbidity meters and colorimeters were standardized using distilled water and reference standards as applicable. Calibration checks were performed periodically during analyses to minimize instrumental drift and ensure consistency of measurements.

2.5.2. Use of Blanks and Replicates

Reagent blanks were prepared and analyzed alongside water samples to account for background interference, reagent impurities, and instrument noise. Blank values were subtracted from sample readings where applicable. All physicochemical and microbial analyses were conducted in triplicate to assess analytical precision and reproducibility. Mean values and standard deviations were calculated from replicate measurements, and any anomalous results were reanalyzed to confirm consistency. Standard solutions were also analyzed intermittently to verify analytical accuracy and method performance.

2.5.3. Sterilization Procedures

Strict sterilization and aseptic techniques were employed during sample handling and microbial analyses to prevent contamination. Glassware and sampling containers were thoroughly washed with detergent, rinsed with distilled water, and sterilized by autoclaving at 121 °C for 15 minutes prior to use. Microbiological media, instruments, and accessories were sterilized following standard microbiological protocols. Sampling bottles were pre-sterilized and handled using aseptic techniques during collection, transportation, and analysis. Work surfaces were disinfected before and after laboratory procedures to maintain a contamination-free environment.

2.5.4. Statistical Analysis

All data obtained from physicochemical and microbial analyses were statistically analyzed using the Statistical Product and Service Solutions (SPSS) software package, version 25. Descriptive statistics were employed to summarize the data, and results were expressed as mean ± standard deviation (SD) of replicate measurements. Prior to inferential analysis, data were assessed for normality and homogeneity of variance where applicable. Differences among sampling locations were evaluated using appropriate statistical tests, and mean values with a probability level of $p \leq 0.05$ were considered statistically significant. All statistical analyses were conducted at a 95% confidence level to ensure the reliability and validity of the results.

3. Result

Table 1 Bacterial counts for water samples collected from Mini-Ogboro in Rumuodogo community, Rivers State

Parameters	Upstream	Midstream	Downstream	WHO Standard Limit (2011)
THB (CFU/ml)	9.0×10^3	15.2×10^3	8.4×10^3	1.0 X 10^2CFU/ml
TCC (CFU/ml)	2.0×10^3	3.7×10^3	1.3×10^3	0
TFC (CFU/ml)	7.0×10^2	5.0×10^2	2.0×10^2	0

Key: THB; Total heterotrophic bacteria, TCC; Total coliform count, TFC; Total fecal coliform

Table 2 Result of the selected physicochemical parameters of Mini-Ogboro in Rumuodogo Community.

Parameters	Upstream	Midstream	Downstream	WHO Limit (2009)
Temp (0C)	28.50±0.00	28.55±0.07	28.50±0.00	12.5°C
pH	5.05±0.07ad	5.10±0.00ad	5.40±0.00*bc	6.5 – 8.5
E.C.	24.00±0.00ad	24.00±0.00ad	32.00±0.00*bc	<1000
TDS	16.00±0.00ad	16.00±0.00ad	21.34±0.00*bc	600
Turbidity	8.79±0.09ad	9.78±0.35ad	17.58±1.03*bc	<5.0 NTU
Salinity	0.03±0.00	0.03±0.00	0.03±0.00	<600mg/L
Chloride	1.00±0.00	1.00±0.00	2.00±0.00	< 250mg/L
Total Alk	15.00±1.41	14.00±2.83	15.00±1.41	20-200mg/L
Total Hard	61.44±5.43ad	60.48±9.50ad	57.60±5.43*bc	>200mg/L
Ca	6.14±0.00ad	6.14±0.00ad	9.36±0.00*bc	75mg/L
Mg	9.68±3.26bd	10.83±2.28*ad	8.30±1.31bc	30mg/L
Nitrate	0.51±0.00bd	0.47±0.00*ac	0.47±0.00*ac	< 50
Sulphate	2.32±0.00bd	2.88±0.00*ad	11.32±0.00*bc	<250mg
Phosphate	0.07±0.00ad	0.08±0.00ad	0.02±0.00*bc	Below 0.5

Values in the table above are expressed as mean ± standard deviation. Values with superscript * differs significantly when comparing upstream with others. Values with superscript ab differs significantly when comparing midstream with others. Values with superscript cd differs significantly when comparing downstream with others.

4. Discussion

The bacteriological and physicochemical assessment of water samples from Mini-Ogboro, Rumuodogo community revealed substantial spatial variations across upstream, midstream, and downstream locations, reflecting the influence of anthropogenic activities on water quality. The results indicate significant microbial contamination alongside alterations in key physicochemical parameters, raising concerns about the suitability of these water sources for domestic use.

4.1. Microbiological Quality of Mini-Ogboro stream

Table 3.1 revealed the mean bacterial counts of water samples collected from three different sampling points (upstream, midstream, and downstream) in Mini-Ogboro, Rumuodogo community, Rivers State. The Total Heterotrophic Bacteria (THB) counts recorded across the sampling points, 9.0×10^3 CFU/g (upstream), 15.2×10^3 CFU/g (midstream), and 8.4×10^3 CFU/g (downstream), were markedly higher than levels generally associated with good microbial quality. Although the World Health Organization (WHO) does not prescribe a specific guideline value for THB in drinking water, counts exceeding 5.0×10^2 CFU/g are widely regarded as indicative of poor sanitary quality and excessive organic loading. The elevated THB counts observed in this study suggest intense microbial proliferation, likely driven by the availability of organic substrates introduced through anthropogenic inputs, including illegal oil refining and related hydrocarbon pollution. Hydrocarbons and associated organic matter can serve as carbon sources for heterotrophic microorganisms, thereby stimulating bacterial growth. Similar findings have been reported by Chikere et al. (2011), who attributed elevated THB levels in oil-impacted aquatic environments to chronic oil spills and leaks that compromise water quality [24].

High THB levels are also ecologically and clinically significant, as they may signal the presence of opportunistic pathogens and a general decline in water quality. Water sources with such microbial loads are unsuitable for drinking without adequate treatment and may pose health risks to exposed populations. The Total Coliform Count (TCC) values, 2.0×10^3 CFU/g (upstream), 3.7×10^3 CFU/g (midstream), and 1.3×10^3 CFU/g (downstream),were substantially above the WHO guideline of zero coliforms per 100 mL of drinking water. Coliform bacteria are widely used as indicators of sanitary quality, and their presence signifies potential contamination by pathogenic microorganisms. The high coliform counts observed suggest fecal contamination arising from surface runoff, sewage leakage, improper waste disposal, and

disturbances associated with oil-related activities. Ekhaise and Anyasi (2005) similarly documented elevated coliform levels in oil-impacted communities, underscoring the link between petroleum activities and microbial deterioration of water resources [25]. Furthermore, Total Fecal Coliform (TFC) counts of 7.0×10^2 CFU/g (upstream), 5.0×10^2 CFU/g (midstream), and 2.0×10^2 CFU/g (downstream) are particularly alarming, as WHO guidelines require complete absence of fecal coliforms in drinking water. The detection of fecal coliforms indicates direct contamination by human or animal waste and points to a high likelihood of pathogenic organisms entering the water supply. The relatively elevated midstream values may reflect intensified human activities and waste discharge in that zone. Comparable findings were reported by Nwidu et al. (2008), who observed widespread fecal contamination in water bodies within oil-producing areas, attributed to disrupted land surfaces and uncontrolled wastewater discharge [26]. The combined presence of high THB, TCC, and TFC levels constitutes a serious public health concern. Such conditions increase the risk of exposure to pathogens including *Escherichia coli*, *Salmonella*, and *Shigella*, which are responsible for gastrointestinal infections, diarrhea, typhoid fever, and other waterborne diseases. Prolonged consumption or use of contaminated water may weaken immune responses, particularly among children, the elderly, and individuals with underlying health conditions, and may contribute to systemic complications affecting organs such as the liver and kidneys [26].

4.2. Physicochemical Characteristics of Mini-Ogboro stream

The physicochemical parameters further reflect compromised water quality. The temperature across all sampling points ranged from 28.50 ± 0.00°C to 28.55 ± 0.07°C, remaining relatively uniform along the stream. Although these values exceed the WHO reference temperature of 12.5°C, they reflect typical tropical surface water conditions rather than direct thermal pollution. Nevertheless, elevated temperatures are known to enhance microbial metabolism and proliferation, thereby exacerbating bacterial contamination. Vermeulen et al. (2010) demonstrated a positive correlation between increased water temperature and higher concentrations of *E. coli* and *Vibrio cholerae*, highlighting the heightened risk of waterborne diseases in warmer aquatic environments [27]. The pH values recorded (5.05-5.40) were consistently below the WHO recommended range of 6.5-8.5, indicating acidic conditions across the study area. Acidic water can increase the solubility of toxic metals such as lead and copper, thereby posing additional health risks when such water is consumed. Low pH can also disrupt aquatic ecosystems and reduce the buffering capacity of water bodies, making them more vulnerable to chemical fluctuations.

Salinity levels were below WHO permissible limits, which is generally favorable for drinking water. However, persistently low salinity may reflect reduced mineral content, potentially leading to inadequate intake of essential electrolytes over time. Kumar et al. (2012) noted that low-salinity drinking water may contribute to mineral deficiencies, particularly in vulnerable populations [28]. Total alkalinity values (14.00-15.00 mg/L) were below the recommended range of 20-200 mg/L, suggesting limited buffering capacity. Low alkalinity makes water more susceptible to pH changes, which can destabilize water quality and adversely affect both aquatic organisms and human consumers. Oladapo et al. (2010) reported associations between low alkalinity in drinking water and increased health complaints, emphasizing the importance of balanced alkalinity [29]. Similarly, low total hardness and reduced calcium (Ca^{2+}) concentrations, although often perceived as advantageous, may have negative long-term health implications. Calcium and magnesium are essential minerals required for bone development and cardiovascular health. Chronic consumption of soft water deficient in these minerals has been linked to increased risks of osteoporosis and cardiovascular diseases (Mizuno et al., 2015), particularly among children and elderly individuals [30].

Nitrate (NO_3^-), sulphate (SO_4^{2-}), and phosphate (PO_4^{3-}) concentrations were all below WHO permissible limits, indicating minimal risk from these anions. Low nitrate levels reduce the likelihood of methemoglobinemia and other nitrate-related disorders [31]. Likewise, low sulphate levels minimize gastrointestinal irritation, although extremely low concentrations may affect water taste and palatability [32]. Low phosphate levels are not directly harmful to human health but may influence aquatic productivity and ecosystem balance. Electrical conductivity values were also below WHO limits, reflecting low concentrations of dissolved ions. While this suggests reduced salinity and contamination, it may also imply limited mineral availability, potentially contributing to nutrient deficiencies among consumers, as observed by [33].

Conversely, turbidity, chloride, and temperature exceeded WHO guideline values. High turbidity reduces water clarity and can shield microorganisms from disinfection, increasing the risk of waterborne diseases. Bamgbose et al. (2011) reported higher disease incidence in communities relying on turbid water sources [34]. Elevated chloride concentrations can impart an unpleasant taste and have been associated with increased hypertension, particularly in sensitive populations such as infants and the elderly [35]. Generally, the findings demonstrate that water sources in Mini-Ogboro are microbiologically unsafe and physicochemically unstable, largely due to anthropogenic pressures, including oil-related activities and inadequate waste management. The combined microbial contamination and

physicochemical imbalances underscore the urgent need for appropriate water treatment, continuous monitoring, and regulatory enforcement to safeguard public health and environmental sustainability in the study area.

5. Conclusion

Water from the Mini-Ogboro stream constitutes an important natural resource for the Rumuodogo community, where it is utilized for drinking and various domestic purposes. However, the findings of this study demonstrate that the quality of this water source is substantially compromised. Comparative analysis of the microbiological and physicochemical parameters against the World Health Organization (WHO, 2019) guidelines for drinking water revealed significant deviations in several key indicators. The markedly elevated levels of total heterotrophic bacteria, total coliforms, and fecal coliforms clearly indicate microbial contamination, suggesting the possible presence of pathogenic microorganisms and rendering the water unsafe for direct consumption. These microbial loads are indicative of poor sanitary conditions and are likely exacerbated by anthropogenic activities within the study area, including improper waste disposal and oil-related operations. In addition to microbial risks, exceedances in parameters such as turbidity, temperature, and chloride further degrade the aesthetic and chemical quality of the water, potentially reducing its acceptability and increasing the likelihood of adverse health outcomes. The combined effect of microbial contamination and physicochemical imbalance poses a considerable public health risk, particularly to vulnerable populations such as children, the elderly, and individuals with pre-existing health conditions. Prolonged exposure or consumption of untreated water from Mini-Ogboro may predispose residents to waterborne diseases, gastrointestinal infections, and other chronic health complications. Although Mini-Ogboro water remains a vital resource for the Rumuodogo community, its current quality status falls below internationally accepted standards for potable water. There is an urgent need for appropriate water treatment prior to consumption, continuous water quality monitoring, and the implementation of effective environmental management and regulatory measures. Such interventions are essential to safeguard public health, improve water safety, and ensure the sustainable use of this critical water resource.

Compliance with ethical standards

Disclosure of conflict of interest

No conflict of interest to be disclosed.

Authors' contributions

This work was carried out in collaboration among all authors. All authors read and approved the final manuscript.

References

[1] World Health Organization (WHO) (2019). National systems to support drinking-water: Sanitation and hygiene: Global status report: UNWater global analysis and assessment of sanitation and drinking water: GLAAS 2019 report. World Health Organization

[2] UNESCO. (2019). The United Nations World Water Development Report 2019: Leaving no one behind. United Nations Educational, Scientific and Cultural Organization, Paris.

[3] Li, P.& Wu, J. (2019). Drinking water quality and public health. Exposure and Health Journal,11:73-79.

[4] Naeem, K., Syed, T. H., Abdus, S., Nargis, J.&Kyong, S.K. (2013). Physicochemical investigation of the drinking water sources from Mardan, Khyber Pakhtunkhwa, Pakistan. International Journal of Physical Sciences; 8(33):161 167

[5] World Health Organization. (2017). Guidelines for drinking-water quality (4th ed., incorporating the 1st addendum). WHO Press, Geneva.

[6] Sam, K., Onyena, A. P, Eriegha, O. J &Eze, F. (2023). Water Quality Evaluation Using Water Quality Index and Pollution Model in Selected Communities in Gbaramatu Kingdom, Niger Delta, Nigeria. African Journal of Environmental Science and Technology,17(6): 118-134.

[7] Bayode, O. J., Adewunmo, E. A., & Odunwole, S. (2011). Environmental Implications of Oil Exploration and Exploitation in the Coastal Region of Ondo State, Nigeria: A Regional Planning Appraisal. Journal of Geography and Regional Planning, 4(3):110-121.

[8] Nwachoko, N., Jack, G. T.& Dume, B. B. (2021). The Impact of Oil Exploration Activities on Soil, Water and Shrimps of Maa-Or Gbor in Korokoro Community, Rivers State ,Nigeria. Chemical Science International Journal; 30(5): 44-50.

[9] Onyegeme-Okerenta, B.M., Oharisi, A.O &Wegwu, M.O. (2017). Impact of crude oil spillage on water and African catfish (Clariasgariepinus) in Uzere, Isoko South LGA of Delta State. European Journal of earth and Environment; 4(1),7-18.

[10] Babatunde, A. (2010). The impact of oil exploitation on the socio-economic life of the Ilaje-Ugbo people of Ondo State, Nigeria. Journal of Sustainable Development in Africa, 12(5), 61–84.

[11] Nriagu, J. O., Udofia, E. A., Ekong, I., & Ebuk, G. (2016). Health risks associated with oil pollution in the Niger Delta, Nigeria. International Journal of Environmental Research and Public Health, 13(3), 346.

[12] Daniels-Azoma, A. E., Okeke, O. C., Ubechu, B. O., Amadi, C. C., & Ajayi, O. (2024). Assessment of water quality of Aba River in Aba area, southeastern Nigeria, using water quality index technique. IIARD International Journal of Geography & Environmental Management, 10(6), 128–163.

[13] Tyagi, S., Sharma, B., Singh, P., & Dobhal, R. (2013). Water quality assessment in terms of Water Quality Index. American Journal of Water Resources, 1(3), 34–38.

[14] National Population Commission (NPC). (2006). Population and housing census of the Federal Republic of Nigeria. NPC, Abuja, Nigeria.

[15] Niger Delta Development Commission (NDDC). (2010). Niger Delta regional development master plan. NDDC, Port Harcourt, Nigeria.

[16] Allen, J. R. L. (1965). Late Quaternary Niger Delta, and adjacent areas: Sedimentary environments and lithofacies. AAPG Bulletin, 49(5), 547–600.

[17] Nwankwoala, H. O., & Ngah, S. A. (2014). Groundwater resources of the Niger Delta: Quality implications and management considerations. International Journal of Water Resources and Environmental Engineering, 6, 155–163.

[18] Nigerian Meteorological Agency (NiMet). (2019). Climate review bulletin. NiMet, Abuja, Nigeria.

[19] Adeyemo, O. K., Adedokun, O. A., Yusuf, R. K., & Adeleye, E. A. (2008). Seasonal changes in physicochemical parameters and nutrient load of river sediments in Ibadan City, Nigeria. Global NEST Journal, 10(3), 326–336.

[20] Spalding, M., Kainuma, M., & Collins, L. (2010). World atlas of mangroves. Earthscan, London.

[21] Essoh, P. A., Umoh, G. S., Ekong, F. U., & Ubom, R. M. (2001). Rural livelihoods and resources extraction in the Niger Delta wetlands. A Journal of Environmental Analysis, 7, 580-681.

[22] Kadafa, A. A. (2012). Oil exploration and spillage in the Niger Delta of Nigeria. Civil and Environmental Research, 2(3), 38–51.

[23] Anejionu, O. C. D., Ahiarammunnah, P. N., & Nri-Ezedi, C. J. (2015). Hydrocarbon pollution in the Niger Delta: Geographies of impacts and appraisal of lapses in existing legal framework. Resources Policy, 45, 65–77.

[24] Chikere, C. B., Okpokwasili, G. C., & Chikere, B. O. (2011). Monitoring of microbial hydrocarbon remediation in the soil. 3 Biotech, 1(3), 117–138.

[25] Ekhaise, F. O., & Anyasi, C. C. (2005). Influence of breweries effluent discharge on the microbiological and physicochemical quality of Ikpoba River, Nigeria.African Journal of Biotechnology,4(10), 1062–1065.

[26] Nwidu, L. L., Oveh, B., Okoriye, T., & Vaikosen, N. A. (2008). Assessment of the water quality and prevalence of water-borne diseases in Amassoma, Niger Delta, Nigeria. African Journal of Biotechnology, 7(17), 2993–2997.

[27] Vermeulen, L. C., & Hofstra, N. (2014). Influence of climate variables on the concentration of Escherichia coli in the Rhine, Meuse, and Drentse Aa during 1985–2010. Regional Environmental Change, 14(1), 307-319.

[28] Shrivastava, P., & Kumar, R. (2015). Soil salinity: A serious environmental issue and plant growth-promoting bacteria as one of the tools for its alleviation. Saudi Journal of Biological Sciences, 22(2), 123–131.

[29] Ali, Q. A., & Ghareeb, O. A. (2023). Drinking water quality and its impact on public health: Review. International Scientific Research Journal, 4(9), 48–64.

[30] Helte, E., Säve-Söderbergh, M., Larsson, S. C., & Åkesson, A. (2022). Calcium and magnesium in drinking water and risk of myocardial infarction and stroke: A population-based cohort study. American Journal of Clinical Nutrition, 116(4), 1091–1100.

[31] World Health Organization (WHO). Guidelines for Drinking-water Quality (4th ed.). WHO, 2011.

[32] Levallois, P., & Villanueva, C. M. (2019). Drinking water quality and human health: An editorial. International Journal of Environmental Research and Public Health, 16(4), 631.

[33] Idomeh, J. E., Sam, K., Ekperusi, A. O., & Isokpehi, N. A. (2024). Potable drinking water in Nigeria's coastal communities: Insights from physico-chemical and bacteriological properties assessment of Kurutie's water and distribution systems. Ethiopian Journal of Environmental Studies & Management, 17(1), 16–28.

[34] Nwinyi, O. C., Uyi, O., Awosanya, E. J., Oyeyemi, I. T., Ugbenyen, A. M., Muhammad, A., Alabi, O. A., Ekwunife, O. I., Adetunji, C. O., & Omoruyi, I. M. (2020). Review of drinking water quality in Nigeria: Towards attaining the Sustainable Development Goal six. Annals of Science and Technology - A, 5(2), 58–77.

[35] Mondal, N. C., & Singh, V. P. (2012). Chloride migration in groundwater for a tannery belt in Southern India. Environmental Monitoring and Assessment, 184(5), 2857–2879.

[36] Energy Focus. (2022, May 1). Crude oil theft, illegal artisanal refining operations in the Niger Delta region of Nigeria and modular refining concept (Energy Focus Report: Analysis).

[37] Nkin, G. K. (2025). Remediation of per- and polyfluoroalkyl substances (PFAS)-contaminated environments: Emerging nanomaterials, electrochemical, and biological strategies. International Journal of Chemistry and Chemical Processes, 11(6), 68-97.

[38] Tetam, J. G., Nkin, G. K., & Mee-eeBari, K. R. (2025). Assessment of selected metals and polycyclic aromatic hydrocarbons in the gills and muscles of Oreochromis niloticus from Oyorokoto River, Andoni, Rivers State, Nigeria. International Journal of Chemistry and Chemical Processes, 11(6), 56–66.

[39] Tetam, J. G., Nkin, G. K., & Mee-eeBari, K. R. (2025). Selected metal and polycyclic aromatic hydrocarbon profiles in Eichhornia crassipes from Omuechi in Ikwerre Local Government Area, Nigeria. Open Access Research Journal of Chemistry and pharmacy 7 (02), 027-036.

[40] Nkin, G. K. (2025). Microplastics as silent invaders: A multiscale review of their toxicological effects and contaminant interactions in terrestrial and aquatic environments. Magna Scientia Advanced Research and Reviews, 14(2), 34–58.

[41] Nwachoko, N., Davies, B., & Tetam, J.G. (2020). Impact of illegal crude oil refining in Jike-ama River of Bille Kingdom, Rivers State, Nigeria. International Journal of Biochemistry Research & Review, 29(6), 46–51.

[42] Nkin, G. K. (2025, November 5). Map of the study area showing nearby communities and sampling points (Mini-Ogboro in Rumuodogo community in Emohua Local Government Area (LGA) of Rivers State, southern Nigeria) [Map]. Unpublished.