

BACTERIOLOGICAL EFFECTIVENESS OF SOME TREATMENTS ON THE QUALITY OF BOREHOLE AND WELL WATER SOURCES

Oyawoye, Tomisin ^{*1}., Sule, Ismaila Olawale³., Ebere, Christian Ugochukwu¹., Ajadi, Fatimah Adenike¹., Onifade, Saheed Abayomi²., Sorunke, Taiwo Ayodeji¹., Ajadi, Ayobami Elias¹ and Bale, Sheriffdeen Issa¹

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¹Department of Microbiology; Faculty of Sciences, Federal University of Health Sciences, Ila-Orangun, P.M.B 204, Osun State, Nigeria.

²Department of Biotechnology and Molecular Biology, Faculty of Sciences, Federal University of Health Sciences, Ila-Orangun, P.M.B 204, Osun State, Nigeria.

³Department of Microbiology, Faculty of Life Sciences, University of Ilorin, Ilorin 1515, Nigeria

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ABSTRACT

A significant portion of the African population relies on well and borehole water for drinking and other domestic purposes. This study evaluated the effectiveness of four treatment methods; treatment with 'watercare' disinfectant, alum coagulation, *Bridelia ferruginea* leaf extract, and filtration sedimentation on the quality of these water sources, with treatment contact times of 30 minutes and 1 hour. Physicochemical and bacteriological analyses were conducted on both well and borehole water samples before and after treatment. The results indicated that disinfection with 'watercare' and alum treatment met the World Health Organization (WHO) and Nigerian Standard for Drinking Water Quality (NSDWQ) guidelines, except for the alum treatment, which lowered the pH to an acidic level. Bacteriological assessments, using the pour plate technique for enumerating total bacterial count and coliforms, revealed that none of the samples met the WHO and NSDWQ bacteriological standards. Total bacterial counts ranged from 1.5×10^2 CFU/ml in borehole water to 8.9×10^2 to 1.6×10^3 CFU/ml in well water. The identified microorganisms were predominantly Gram-negative rods, including *Acinetobacter baumannii*, *Serratia maltophilia*, *Myroides odoratus*, and *Burkholderia cepacia*. Gram-positive organisms identified included *Micrococcus* sp., *Corynebacterium* sp., and *Bacillus* sp. While 'watercare' disinfection and alum treatment achieved significant reductions in bacterial and coliform loads, the *Bridelia* extract had no significant effect. The study concludes that the water samples are unsafe for drinking, emphasizing the necessity of combining filtration, sedimentation, and disinfection for adequate water treatment.

Keywords: Coliform, Physicochemical, Treatment, Water source

Introduction

Water is essential to life, but many people do not have access to clean and safe drinking water

and many die of water-borne bacterial infections which prompted the need for treatment in order to make it free of contaminant especially where there is presence of coliform (Cabral, 2010).

Corresponding Author: Oyawoye, T.

Department of Microbiology; Faculty of Sciences, Federal University of Health Sciences, Ila-Orangun. oyawoyetomisin@gmail.com



Water possesses unique chemical and physical properties that are essential for sustaining life on Earth, distinguishing it from other planets. Unlike most liquids, which contract as they cool and reach maximum density when solidified, water behaves differently. It contracts as it cools until it reaches 4°C, after which it expands until freezing at 0°C. This anomaly results in ice being less dense than liquid water, allowing ice cubes to float in beverages, icebergs to remain buoyant in oceans, and ponds and lakes to freeze from the top down, preserving the aquatic life below (Pouliquen, 2024; Rajagopal *et al.*, 2016). Water acts not only as a solvent but also as a medium for biochemical reactions. Its unique hydrogen bonding capabilities contribute to its high specific heat, surface tension, and solvent properties, which are essential for cellular processes (Pouliquen, 2024). The safe drinking water for all is one of the major challenges of the 21st century and that microbiological control of drinking water should be the norm everywhere. Microbiological water analysis will be mainly based on the concept of fecal indicator bacteria (Cabral, 2010).

Globally, water covers approximately 71% of the Earth's surface. However, 97.5% of this is saline, leaving only 2.5% as freshwater. Potable water is available in most populated regions, though its cost and sustainability may vary (Mulay *et al.*, 2021). Water sources include groundwater, springs, hyporheic zones, aquifers, precipitation (such as rain, hail, snow, fog), surface water (rivers, streams, glaciers), biological sources (plants), and desalinated seawater. Precipitation, defined as any product of atmospheric water vapor condensation falling under gravity, includes drizzle, rain, sleet, snow, and hail. It occurs when atmospheric saturation causes water vapor to condense and precipitate (Scott, 2015).

The deterioration of potable water has become a global concern, primarily due to human activities. While water can be made potable through filtration, distillation, or various other methods, water that is unsafe for drinking may still be used for activities such as swimming or bathing,

provided it is not harmful to humans (Uddin *et al.*, 2021). Natural factors like lithology, topography, climate, atmospheric conditions, and hydrology, along with human activities such as waste disposal, industrial production, livestock farming, and mining, significantly influence water quality (Uddin *et al.*, 2018; Wieczorek *et al.*, 2023).

Chlorine, often used to disinfect water for drinking or bathing, is a skin and mucous membrane irritant and is regulated by government standards, typically at 1 part per million (ppm) for drinking water. Bathing water may also be kept in acceptable microbiological condition using chemical disinfectants such as chlorine or ozone, or by ultraviolet light treatment (US EPA, 2016a). This study aimed to assess the bacteriological quality of borehole and well water sources and also evaluated effectiveness of four treatment methods; treatment with 'watercare' disinfectant, alum coagulation, *Bridelia ferruginea* leaf extract, and filtration sedimentation on the quality of these water sources.

Materials and Methods

Collection of 'watercare' Disinfectant, Alum, and Plant Leaf Extraction

'Watercare' water purifier was purchased from a pharmaceutical store along Unity Road, Ilorin, and transported to the laboratory for water sample purification.

Alum was obtained from Ipata Market, Ilorin, and transported to the laboratory. The Alum was ground into a fine powder using a sterile crucible and stored until further use.

Bridelia ferruginea bark was collected from the *Bridelia ferruginea* tree of the family *Euphorbiaceae*, located within the residential quarters of the University of Ilorin, Nigeria. The sample was authenticated and deposited at the Plant Biology Herbarium of the University of Ilorin. The bark was air-dried at ambient laboratory temperature for approximately one

week, pulverized using a mechanical grinder, and the resulting powder was stored until needed. Fifty grams of the ground bark was dissolved in ethanol and placed on a shaker for 48 hours. The suspension was then decanted and filtered using sterile Whatman Paper No. 1. The filtrate was concentrated to dryness at 45°C using a rotary evaporator, and the residue obtained served as the leaf extract (Kolawole *et al.*, 2007).

Collection of Water Samples

Sampling of Dug Well Water

A clean stone was attached to one end of a long rope, with the other end attached to the bottle's string. The bottle was carefully lowered into the well, ensuring it did not touch the sides. The bottle was submerged just below the surface without hitting the bottom or disturbing any sediment. Once the bottle was judged to be full, it was retrieved by pulling up the rope, leaving some air space to allow for thorough shaking before analysis. The sample was immediately transported to the laboratory for analysis.

Sampling of Borehole Water

The borehole nozzle was first cleaned with cotton wool soaked in 70% ethanol. The borehole was then pumped, and water was allowed to run for 1-2 minutes. A sterilized bottle was carefully opened and filled with water, leaving a small air space for easier shaking before analysis. The bottle was securely covered and immediately transported to the laboratory for analysis.

Procedure for the Treatment of Water Samples

***Bridelia ferruginea* Leaf Extract**

One gram of the extract was introduced into a sterile test tube containing 9 ml of sterile distilled water, thoroughly shaken, and allowed to settle. One ml of the supernatant was drawn using a sterile pipette and added to 100 ml of the water sample. Microbial analysis was conducted at 30-minute and 1-hour intervals by drawing 1 ml of the mixture into sterile Petri dishes using a sterile

pipette and applying the pour plate method with Nutrient agar and MacConkey agar.

'Watercare' Disinfectant

Aseptically, 0.5 ml of 'watercare' disinfectant was added to 100 ml of the water sample. The mixture was thoroughly mixed as per the manufacturer's instructions and allowed to settle. Microbial analysis was performed at 30-minute and 1-hour intervals by drawing 1 ml of the mixture into sterile Petri dishes using a sterile pipette and applying the pour plate method with Nutrient agar and MacConkey agar.

Alum

One gram of alum was introduced into a sterile test tube containing 9 ml of sterile distilled water, thoroughly mixed, and allowed to settle. 1 ml of the supernatant was drawn using a sterile pipette and added to 100 ml of the water sample. The mixture was thoroughly shaken and allowed to settle. Microbial analysis was performed at 30-minute and 1-hour intervals by drawing 1 ml of the mixture into sterile Petri dishes using a sterile pipette and applying the pour plate method with Nutrient agar and MacConkey agar.

Filtration and Sedimentation

Approximately 100 ml of the water samples were passed through a sterile muslin cloth into a sterile conical flask and allowed to settle. Microbial analysis was performed at 30-minute and 1-hour intervals by drawing 1 ml of the mixture into sterile Petri dishes using a sterile pipette and applying the pour plate method with Nutrient agar and MacConkey agar.

Physicochemical Analysis of Water Samples

Determination of Temperature

The temperature of each collected water sample was measured on-site using a mercury bulb thermometer. The thermometer was cleaned with water, then immersed in the water samples until a stable reading was obtained. The temperature was recorded in degrees Celsius (°C) (APHA, 2005).

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Determination of pH

The pH of the water samples was measured using a sensitive digital-electrode pH meter (Metrohm 632). Before each measurement, the electrode meter was calibrated using buffer solutions with pH values of 4, 7, and 9. The electrode was then immersed in a beaker containing 30 ml of the sample and left for 50-60 seconds until a stable reading was achieved (Ademoroti, 1996).

Determination of Total Solids (TS)

The Total Solids (TS) content in the water samples was determined using the evaporation method as outlined by APHA (1981). Initially, a crucible was dried in an oven at 105°C for 20 minutes and then cooled in a desiccator. The crucible was weighed to obtain its initial weight (W1). A measured volume of 50 ml of the water sample was transferred into the pre-weighed crucible and heated in the oven at 105°C until evaporation was complete. The crucible was then cooled in a desiccator and weighed repeatedly until a constant weight (W2) was obtained.

The Total Solids content was calculated using the following formula:

Calculation:

$$\text{TS (mg/l)} = \frac{(W2 - W1) \times 1000}{\text{Sample volume (ml)}}$$

Where:

W1 = Weight of the empty crucible (g)

W2 = Weight of the crucible with the evaporated sample (g)
(APHA, 2005).

Determination of Total Suspended Solids (TSS)

Total Suspended Solids (TSS) were determined using the filtration method. A Whatman filter paper was dried in a hot air oven at 105°C, cooled in a desiccator for 1 hour, and weighed until a constant weight was achieved (Wi). A

measured volume of 50 ml of the water sample was filtered through the pre-weighed filter paper, which was then dried in an oven at 105°C, cooled in a desiccator, and weighed repeatedly until a constant weight (W2) was obtained.

The TSS content was calculated using the following formula:

$$\text{TSS (mg/l)} = \frac{(W2 - W1) \times 1000}{\text{Sample volume (ml)}}$$

Where:

W1 = Weight of the dried filter paper (g)

W2 = Weight of the filter paper with the retained solids (g)
(APHA, 2005).

Determination of Total Dissolved Solids (TDS)

Total Dissolved Solids (TDS) were calculated by subtracting the Total Suspended Solids (TSS) from the Total Solids (TS). This approach follows the method described by George and Edward (1985), where:

$$\text{TDS (mg/l)} = Y - X$$

Where:

Y = Total Solids content (mg/l)

X = Total Suspended Solids content (mg/l)

Calculation:

$$\text{TDS (mg/l)} = Y - X$$

Where,

Y = Total solids content in mg/l

X = Total suspended content in mg/l

Determination of Dissolved Oxygen Content (DO)

Twenty-five ml of Agba water were pipetted into a BOD bottle, followed by the addition of 5 ml of 10% MnCl₂ solution and 5 ml of alkaline iodide solution. The bottle was sealed with a stopper and mixed by inverting several times

until a dirty brown color developed. Then, 10 ml of 25% HCl solution was added and mixed thoroughly, resulting in a reddish-yellow solution. The entire content was transferred into a 25 ml conical flask, and three drops of starch indicator were added. The precipitate was titrated with 0.05 M $\text{Na}_2\text{S}_2\text{O}_3$ until a colorless solution was obtained at the endpoint (APHA, 2005).

Determination of Biochemical Oxygen Demand (BOD)

The same procedure used for the dissolved oxygen determination was applied to a stored water sample kept in a dark cupboard for five days. The BOD was calculated by subtracting the dissolved oxygen content on day five from the initial dissolved oxygen content on the first day, as follows:

$$\text{BOD (mg/l)} = \text{DO}_1 - \text{DO}_5$$

Determination of Conductivity

Conductivity, the ability of water to conduct an electric current, depends on the concentration of ions in the solution and is measured in millisiemens per meter (mS/m). Measurements were taken immediately after water sampling, as conductivity can change over time and is temperature-dependent. The temperature of each sample was also recorded. Conductivity was measured using the Wagtech International conductivity kit (Wag WT 3020) (APHA, 2005).

Determination of Turbidity

The turbidity of water samples was measured using a spectrophotometer set to a wavelength of 460 nm. The spectrophotometer was first calibrated with a cuvette containing distilled water. After calibration, the turbidity of each sample was measured by pouring the water sample into a cuvette, placing it in the spectrophotometer, and recording the resultant values displayed on the screen (Fawole and Oso, 2007).

Determination of Nitrate (NO_3^-)

Nitrate levels were determined using the spectrophotometric method described by Naicker and Sithole, 2016 and Booth (1983).

Reagents:

Stock Nitrate Solution: 0.7218 g of anhydrous potassium nitrate was diluted to 1 liter with nitrate-free water, yielding a 100 mg/l solution.

Standard Nitrate Solution: 20 ml of stock solution was diluted to 1 liter with distilled water. One ml of this solution equals 8.86 ng NO_3^- .

1% Sodium Salicylate Solution

Mercury (II) Chloride

Concentrated H_2SO_4

50% NaOH

Sodium Hydroxide-Potassium Tartrate Solution: 1 kg of NaOH was dissolved in 1 liter, and 120 g of Potassium Sodium Tartrate ($\text{KNaC}_4\text{H}_4\text{O}_6$) was added, cooled, and diluted to 2 liters.

Sample Preparation: Fifty ml of water sample were measured into a 250 ml Erlenmeyer flask. Mercury (II) chloride was added to achieve a concentration of 1000 mg/l. The sample was mixed well, and its pH was adjusted to between 11 and 11.5 using 50% NaOH. The sample was then stirred for 5 minutes, allowed to settle, and filtered. Two ml of the filtrate were pipetted into a 50 ml cooperating disc, followed by 1 ml of 1% sodium salicylate solution. The mixture was evaporated to dryness in a drying oven at 105°C for 45 minutes. After cooling, 2 ml of concentrated H_2SO_4 was added and mixed thoroughly by swirling to dissolve all solids. Once cooled, 15 ml of nitrate-free distilled water was added to the sample residue, followed by 15 ml of sodium hydroxide-potassium tartrate solution. A yellow color developed immediately and remained stable for several hours, allowing the sample to cool to room temperature (Ademoroti, 1999).

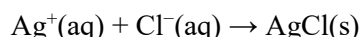
Spectrophotometry: The percentage transmittance/absorbance of the standard was read using a spectrophotometer at the specified wavelength, and a calibration graph was plotted. The percentage transmittance/absorbance of the water samples was then measured, and nitrate concentrations were extrapolated from the calibration graph.

Determination of Total Hardness

Total hardness, due to the presence of both calcium and magnesium ions, was determined using Erichrome Black-T as an indicator. The direct titration method involved buffering 100 ml of water sample to pH 10 using 5 ml of ammoniacal buffer and then titrating against 0.01 M EDTA. The endpoint was noted when the solution changed from purple to pure blue (APHA, 2005).

Determination of Chloride Ion Concentration

This method determines the chloride ion concentration in a solution through titration with silver nitrate. As silver nitrate is added, silver chloride precipitates:



The endpoint occurs when all chloride ions are precipitated, and additional silver ions react with chromate to form a red-brown precipitate of silver chromate.

Procedure:

A 100 ml water sample was pipetted into a conical flask, followed by the addition of 1 ml of 5% potassium chromate ($\text{K}_2\text{Cr}_2\text{O}_7$). The mixture was titrated against 0.05 M AgNO_3 until a pale pink color, indicating the endpoint, was achieved (Phiri *et al.*, 2006). Chloride concentration was then calculated using the formula:

$$((\text{mL AgNO}_3 \text{ for sample} - \text{mL AgNO}_3 \text{ for blank}) \times \text{N AgNO}_3 \times 3.545) / \text{g sample}$$

Enumeration of Total Bacterial Count

The pour plate technique was used to examine each water sample, following the standard methods outlined by the American Public Health Association (APHA). A sterile pipette was used to measure 1 ml of raw water, which was then aseptically introduced into a sterile Petri dish. Nutrient agar was poured into the Petri dish containing the water sample, swirled gently, and allowed to solidify. The plates were incubated at 37°C for 24 to 48 hours before enumeration.

Characterization and Identification of Bacterial Isolates

Bacterial isolates were characterized and identified based on their colonial and cellular morphology. Colonial morphology was assessed by observing the shape, size, optical characteristics, consistency, elevation, and pigmentation of colonies. Cellular morphology was determined through microscopic examination and staining techniques (Olutiola *et al.*, 2000). Biochemical tests, including catalase, oxidase, indole, and urease utilization, were performed, and tentative identification was conducted using Bergey's Manual of Determinative Bacteriology (Buchanan and Gibbon, 1974; Cheesbrough, 2007).

Pure cultures were obtained by transferring distinct colonies onto sterile solid EMB and NA agar plates using a sterile inoculating loop, followed by streaking. The plates were incubated and sub-cultured until satisfactory pure cultures were achieved. The pure cultures were then further inoculated onto nutrient agar plates and slants in McCartney bottles, incubated at 37°C, and stored in a refrigerator at 4°C.

Total Coliform Count

Presumptive Test

Coliform counts were determined using the three-tube assay of the Most Probable Number (MPN) method. The presumptive coliform test was performed using MacConkey broth. The first set of three tubes contained 10 ml of double-strength broth, while the second and third sets contained 10 ml of single-strength broth. Each tube was equipped with an inverted Durham's tube before sterilization. The three sets of tubes were inoculated with 10 ml, 1 ml, and 0.1 ml of the water samples, respectively, using sterile pipettes. The tubes were then incubated at 37°C for 24-48 hours and examined for acid and gas production. Acid production was indicated by a color change from purple to yellow, while gas production was detected by the presence of gas in the Durham tube. The MPN was estimated using the MPN table for the three-tube test (Fawole and Oso, 2007).

Confirmed Test

For the confirmed test, a loopful of the broth from tubes with a positive presumptive test was streaked onto a sterile Eosin Methylene Blue (EMB) agar plate. The plates were incubated at 37°C for 24 hours. Colonies exhibiting a pinkish or reddish color were considered positive for coliforms (Fawole and Oso, 2007).

Completed Test

A loopful of colonies from the confirmed test was inoculated into sterile MacConkey broth containing an inverted Durham's tube and incubated at 37°C for 24-48 hours. The production of acid and gas in the broth confirmed the presence of coliforms (Fawole and Oso, 2007).

Cellular Morphology

Gram Staining

A bacterial smear was prepared by placing a drop of sterile water on a clean slide. A bacterial colony was touched with a flamed loop, rubbed into the drop of water, and spread into a thin smear on the slide. After air-drying, the smear was heat-fixed by quickly passing the reverse side of the slide through a flame three times. The smear was then stained with Gram's crystal violet for 60 seconds, followed by rinsing with Lugol's iodine for 30 seconds. The slide was decolorized with 95% ethanol until it appeared free of violet stain, rinsed with water, and counterstained with safranin for 60 seconds. After rinsing with water, the slide was blotted, air-dried, and observed under a light microscope using the x100 objective lens with immersion oil. Gram-positive bacteria appeared blue to purple, while Gram-negative bacteria appeared pink to red (Fawole and Oso, 2007).

Motility Test

The motility of the bacterial isolates was assessed using the hanging drop technique with 24-hour-old cultures. A small amount of Vaseline jelly was applied around the edge of a cavity slide, and a speck of the organism was transferred onto a cover slip using an inoculating needle. The cavity slide was carefully placed over the cover slip in an inverted position,

forming a hanging drop. The motility was observed under a microscope using the x40 objective lens (Fawole and Oso, 2007).

Spore Staining

A bacterial smear was prepared by placing a drop of sterile water on a clean slide, inoculating with a sterile loop, and heat-fixing by passing it over a Bunsen flame several times. The smear was flooded with malachite green stain and heated over a beaker of boiling water for 10 minutes. After rinsing under the tap, the smear was counterstained with safranin for 20 seconds, washed under the tap, blotted dry, and examined under the oil immersion lens. The vegetative portion of the bacterium stained pink to red, while the spores stained green (Fawole and Oso, 2007).

Biochemical Tests

Microbact Identification for Gram-Negative Bacteria

The Microbact biochemical identification kit (24E set) was used for identifying Gram-negative bacteria, capable of identifying both oxidase-positive and oxidase-negative organisms. An 18-24-hour-old culture of the isolates was prepared on nutrient agar, and 0.85% normal saline solution was dispensed into McCartney bottles. One to three colonies from the young culture were picked and emulsified in 5 ml of sterile normal saline, forming a homogenous suspension. Using a sterile pipette, four drops of the suspension were added aseptically into each well of the Microbact kit. Wells 1, 2, 3, 20, and 24 were overlaid with mineral oil for oxidase-negative organisms, while wells 1, 2, 3, and 24 (excluding well 20) were overlaid for oxidase-positive organisms. The kit was then incubated for 18-24 hours, as per the Oxoid kit instructions. Results were read according to the kit manual, and the octal and nine-digit codes obtained were analyzed using identification software to determine the most probable identity of the Gram-negative bacteria.

Results

Physicochemical Parameters of Well Water

The physicochemical parameters of the well water sample prior to treatment in Table 1 showed the temperature ranged from 27.00 to 27.20°C, while the pH varied between 5.2 and 7.0. Chloride ion concentration was between 1.20 and 2.72 mg/L, and total hardness ranged from 27.60 to 30.00 mg/L. Electrical conductivity was recorded between 89.60 and 98.94 $\mu\text{S}/\text{cm}$. The total solids content ranged from 20.10 to 24.30 mg/L, with total suspended solids (TSS) ranging from 0.08 to 2.12 mg/L, and total dissolved solids (TDS) from 19.78 to 22.98 mg/L. Sulphate ion concentration was observed between 60.10 and 63.00 mg/L, nitrate ion concentration from 1.06 to 1.10 mg/L, and turbidity was noted between 4.46 and 5.50 NTU (Nephelometric Turbidity unit).

Physicochemical Parameters of Borehole Water

Table 2 outlined the physicochemical parameters of the borehole water sample prior to treatment. The temperature ranged from 30.00 to 30.20°C, with pH levels ranging from 5.1 to 7.5. Chloride ion concentration ranged from 0.52 to 1.16 mg/L, and total hardness from 15.20 to 19.20 mg/L. Electrical conductivity varied between 88.40 and 95.80 $\mu\text{S}/\text{cm}$. Total solids content ranged from 15.50 to 20.30 mg/L, TSS from 0.70 to 1.90 mg/L, and TDS from 13.60 to 19.60 mg/L. Sulphate ion concentration was between 58.70 and 62.40 mg/L, nitrate ion concentration from 1.04 to 1.09 mg/L, and turbidity ranged from 4.50 to 5.20 NTU.

Viable Bacterial Count and Identification of Bacterial Isolates

The viable bacterial count of the water samples prior to treatment presented in Table 3 showed

that the well water exhibited the highest bacterial count, ranging from 8.9×10^2 to 1.6×10^3 cfu/ml, while the borehole water had the lowest count, ranging from 1.5×10^2 to 6.4×10^2 cfu/ml. The bacteria isolated from the samples included Gram-negative organisms such as *Acinetobacter baumannii* (88.12%), *Serratia maltophilia* (99.22%), *Myroides odoratus* (98.98%), *Burkholderia cepacia* (99.99%), and *Enterobacter aerogenes*, identified using the Microbact kit and software. Gram-positive organisms identified were *Micrococcus sp.*, *Corynebacterium sp.*, and *Bacillus sp.*

Effect of Different Treatment Methods on Bacterial Count

Table 4 summarized the effect of various treatment methods on the bacterial count of the water samples. The 'watercare' disinfectant proved to be the most effective, reducing the bacterial count to a range of 0 to 0.7×10^1 cfu/ml after 30 minutes of contact time, and 0 to 1.3×10^1 cfu/ml after 1 hour of contact time. Alum was the second most effective treatment, reducing bacterial counts to a range of 0 to 5.2×10^1 cfu/ml after 30 minutes and 0 to 3.5×10^1 cfu/ml after 60 minutes. Filtration and sedimentation methods reduced bacterial counts to a range of 1.1×10^1 to 2.2×10^2 cfu/ml after 30 minutes and 1.0×10^1 to 1.72×10^2 cfu/ml after 60 minutes. *Bridelia ferruginea* leaf extract had the least effect, with bacterial counts ranging from 1.1×10^1 to 3.0×10^2 cfu/ml after 30 minutes of contact time, and 1.0×10^2 to 3.0×10^2 cfu/ml after 60 minutes.

Coliform Count of Water Samples

The coliform count of the water samples prior to treatment showed that the well water sample ranged from 0.5×10^1 to 3.0×10^2 cfu/ml, while the borehole water had a coliform count ranging from 0.9×10^1 to 3.6×10^1 cfu/ml.

Table 1: Physicochemical parameters of well water sample prior and after treatment

Parameters	Sample	'watercare'	Alum	Bridelia extract	Filtration and sedimentation
Temperature	27.00	27.00	27.20	27.00	27.00
pH	6.68	7.00	5.20	6.60	6.70
Chloride ion (mg/l)	1.20	1.40	2.72	1.92	1.28
Total hardness (mg/l)	28.00	28.80	27.20	30.00	27.60
Electrical conductivity (us/cm ³)	89.60	89.86	98.60	98.94	96.80
Total solids (mg/l)	24.30	20.10	22.90	25.10	21.70
Total suspended solids (mg/l)	1.84	0.08	1.84	2.12	1.92
Total dissolved solids (mg/l)	22.46	20.02	21.06	22.98	19.78
Sulphate ion (mg/l)	60.40	61.30	60.10	63.00	60.30
Nitrate ion (mg/l)	1.08	1.06	1.07	1.10	1.09
Turbidity (NTU)	5.30	4.46	4.90	5.50	4.58

Table 2: Physicochemical parameters of borehole water sample prior and after treatment

Parameters	Sample	'watercare'	Alum	Bridelia extract	Filtration and sedimentation
Temperature	30.00	30.00	30.20	30.00	30.00
pH	7.20	7.50	5.10	6.90	7.20
Chloride ion (mg/l)	0.72	0.52	1.16	0.88	0.68
Total hardness (mg/l)	19.20	15.20	16.60	18.60	16.80
Electrical conductivity (us/cm ³)	88.40	88.50	95.45	95.80	95.60
Total solids (mg/l)	15.50	19.90	19.60	20.30	20.30
Total suspended solids (mg/l)	1.90	1.06	1.74	1.82	0.70
Total dissolved solids (mg/l)	13.60	18.84	17.86	18.48	19.60
Sulphate ion (mg/l)	58.90	60.60	58.70	62.40	58.90
Nitrate ion (mg/l)	1.07	1.04	1.05	1.09	1.07
Turbidity (NTU)	5.10	4.50	4.90	5.20	4.80

Table 3: Comparison of Treatment Methods on Bacterial Count

S/N	Water source	Untreated water (cfu/ml)	Treated water							
			‘watercare’		Alum		Filtration and sedimentation		Bridelia leaf extract	
			30 mins	60 mins	30 mins	60 mins	30 mins	60 mins	30 mins	60 mins
1	B1	150	0	0	10	9	15	10	15	14
2	B2	450	0	0	0	0	28	20	38	30
3	B3	520	1	1	0	0	32	25	42	36
4	B4	250	1	4	20	16	40	30	54	34
5	B5	640	0	2	6	4	35	28	45	40
6	W1	1500	1	2	27	5	105	70	200	180
7	W2	1600	3	2	20	12	150	120	150	120
8	W3	250	0	0	10	2	150	110	160	150
9	W4	890	1	1	20	15	120	55	145	55
10	W5	1050	2	1	22	20	131	62	148	66

Key: B: Borehole water sample; W: Well water sample

Table 4: Comparison of different treatments on the coliform count of the water samples

S/N	Water source	Untreated water (cfu/ml)	Treated water							
			‘watercare’		Alum		Filtration and sedimentation		Bridelia leaf extract	
			30 mins	60mins	30 mins	60 mins	30 mins	60 mins	30 mins	60 mins
1	B1	36	0	0	0	0	15	10	20	18
2	B2	9	0	0	0	0	6	5	8	6
3	B3	28	0	0	0	0	12	8	15	12
4	B4	20	0	0	0	0	5	4	6	5
5	B5	19	0	0	0	0	5	5	6	4
6	W1	85	6	5	12	9	35	30	40	35
7	W2	230	30	13	37	21	80	61	191	92
8	W3	250	25	9	33	18	77	57	180	80
9	W4	130	15	11	17	13	69	45	75	30
10	W5	70	6	3	10	6	32	28	42	32

Key: B: Borehole water sample; W: Well water sample

Table 5: Occurrence of bacterial isolates in the raw water samples

Isolates	Samples	
	Well	Borehole
<i>Acinetobacter baumannii</i>	+	-
<i>Micrococcus</i> sp.	+	+
<i>Serratia maltophilia</i>	+	-
<i>Corynebacterium</i> sp.	-	+
<i>Myroides odoratus</i>	+	-
<i>Burkholderia cepacia</i>	-	+
<i>Bacillus</i> sp.	+	+
<i>Enterobacter aerogenes</i>	+	+

Key: +: present, -: Absent

Discussion

The bacteriological analysis revealed that the well water sample exhibited the highest bacterial count, while the borehole water sample had the lowest. The elevated bacterial count in the well water could be attributed to various factors, including improper use by residents, poor sanitary conditions around the well, and the depth of the well. The total coliform count mirrored this trend, with the well water containing the highest number of coliforms and the borehole water the least. These results exceed the limits set by both the WHO and NSDWQ standards, which specify a maximum coliform count of 0 cfu/ml.

The microbial contamination of the well water is likely influenced by both the well's depth and the surrounding environmental conditions, which may promote the introduction and survival of pathogenic organisms. This aligns with Ranchou-Peyruse *et al.* (2023). Similarly, the borehole water's microbial load could be related to the quality and depth of the borehole, as well as the maintenance practices employed.

Among the treatment methods evaluated, disinfection was the most effective in reducing both bacterial and coliform counts. No microbial growth was observed in the borehole samples after disinfection, although some growth persisted in the well samples, reflecting their higher contamination levels. Alum, used as a coagulant, was the second most effective treatment. Its coagulating properties significantly reduced the microbial load, corroborating the findings of Kolawole *et al.* (2007) and Deswal *et al.* (2023), who noted that the acidity of alum can diminish bacterial presence in water because it produces flocs when dissolved in water which eventually settled at the bottom and flocs collect suspended particles including microorganisms as they settle down, reducing the number of free microorganisms (Fasuan *et al.*, 2019).

On the other hand, *Bridelia ferruginea* leaf extract proved to be the least effective treatment.

The contact time between the water samples and the extract ranged from 30 to 60 minutes, which may have been insufficient for optimal efficacy. Previous studies, such as those by Kolawole *et al.* (2007), suggest that longer exposure periods, potentially up to 96 hours, might be necessary for the extract to achieve a significant antimicrobial effect. The study by Oluwagbamila (2023), also aligns with the general understanding that the efficacy of plant extracts often correlates with the duration of contact with the target microorganisms.

After treatment, the disinfectant ('watercare') remained the most effective method for reducing bacterial and coliform levels in the borehole water. However, its effectiveness was limited in well water, likely due to the high microbial load, indicating that the disinfectant's efficacy diminishes when contamination levels are too high possibly due to anthropogenic activities and environmental conditions such as presence of pollutants or contaminated aquifer (Twinomucunguzi *et al.*, 2021). Filtration and sedimentation were also effective in reducing microbial loads, though to a lesser extent compared to disinfection.

Conclusion

The critical importance of access to safe and clean water cannot be overstated. The findings from this study underscore the urgent need for public awareness regarding the current state of these water sources. 'Watercare' disinfectant and alum were highly effective in reducing bacterial contamination in well, borehole, and other water sources, with sedimentation and filtration offering supplementary but less efficient results. It is imperative to educate communities on the importance of treating water before it is considered safe for drinking and other domestic uses. Without these measures, the risks of waterborne diseases and related health concerns remain significant. Consistent monitoring and treatment of water sources are essential to protect public health.

Further research should explore optimizing natural treatments such as *Bridelia ferruginea*, by adjusting concentrations and contact times to enhance efficacy.

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Competing interest

The authors declare that they have no competing interest.

Conflict of Interest

The authors declare no conflict of interest. All contributors have been appropriately acknowledged either through authorship or in the acknowledgment section based on their level of contribution.

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