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Conservation of palm wine using ultraviolet radiation

Ikechukwu Nnanwube[1], Mabel Keke[2]
Madonna University, Nigeria[1]
Delta State University of Science and Technology, Ozoro, Delta State, Nigeria[2]
ik.nnanwube@gmail.com[1], elohocute@yahoo.com[2]

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Abstract
Purpose: This study aimed to explore the effectiveness of ultraviolet (UV) radiation in the conservation of palm wine by reducing its microbial load, specifically mold, yeast, and bacteria.
Research Methodology: Palm wine was conserved by sterilization with ultraviolet rays. The time of contact of the rays and the intensity of the rays with palm wine were varied. The microorganisms considered include molds, yeasts, and bacteria.
Results: The results showed that the microbial load of sterilized palm wine decreased with time of contact with the rays. The microbial load of the wine before and after sterilization was determined. Moreover, the microbial load further decreased during the storage of the sterilized samples. In fact, sterilization time of 240 seconds (4 minutes) reduced the mould and yeast from 3.25×10^6 to 0.01×10^6 and then to zero after 10 days of preservation.
Conclusions: UV radiation is an effective method for reducing microbial contamination in palm wine, with an optimal sterilization time of 240 s providing the best results. This sterilization process not only reduced microbial presence immediately but also extended the shelf life of palm wine during storage.
Limitations: This study did not examine the impact of stirring palm wine during sterilization on microbial load reduction, which could potentially influence the treatment's effectiveness.
Contribution: This study established an ideal sterilization time of 240 s (4 min) for palm wine, providing an effective method for its preservation and ensuring its safety for consumption.

Keywords: *Bacteria, Microbial Load, Mould, Palm Wine, Ultraviolet Radiation*

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1. Introduction

Drinkable palm wine is widely consumed in Asia, sub-Saharan Africa, and South America. Palm sap is fermented to generate palm wine. The beverage is made from a variety of palm trees that are grown all over the sub-tropical and tropical regions, with a few breeds coming from climatic regions because of the illiberality of seedling freezing (Reichgelt, West, & Greenwood, 2018). This beverage is popular and known by various aliases worldwide. Significant uptick in the microbiological analysis of the beverage have led to increased discussion of the variety of Saccharomyces cerevisiae strains that predominate in the beverage Tapsoba, Legras, Savadogo, Dequin, and Traore (2015) and newly identified microorganisms from the drink (Ouoba et al., 2015).

According to numerous reports (Nwaiwu and Itumoh (2017), the method of hurling beverages has been established. Palm sap varies according to the location of the palm trees, as determined in various reports. Palm wine is normally a sweet, white liquid that fizzes because of the active microorganisms it contains. In the southeastern region of Nigeria, the Igbo ethnic group highly values palm wine as their primary alcoholic beverage during ceremonial occasions. Its pale appearance is due to the yeast suspension

(Tiimub et al., 2023). Palm wine has replaced human and animal blood in purification and expiatory ceremonies due to its potency in the development of African traditional religion (Okolo et al., 2019). The beverage is now bottled as industrial production has improved the process. In Nigeria, the product is typically packaged in beer containers, and most distributors sell the beverage in repurposed table water containers with a top that is not too tightly fitted to prevent foaming (Nwaiwu & Itumoh, 2017).

The maintenance of stable chemical and physical properties of palm wine depends heavily on its preservation. Many traditional preservative methods only render spoilage agents (microorganisms) inactive; therefore, over time, these organisms may regain their bioactivity and cause palm wine to spoil (Sirait, Dewi, & Rumengan, 2024). However, the use of chemical preservation methods has the disadvantage of interfering with the wine's active components, changing the texture, flavor, viscosity, and other characteristics, which defeats the goal of uniqueness (Akissi et al., 2024; Latunusa, Timuneno, & Fanggidae, 2023). Therefore, there is a critical need to investigate alternative wine preservation methods that combine effectiveness and creativity while eliminating deterioration agents and preserving the wine's excipients. Therefore, in an effort to discover the ideal preservation method with a low economic cost and a high success rate, this study was conducted.

2. Literature Review

The primary organisms involved in the fermentation of palm sap are yeasts, which are part of the natural flora of palm palms. Regardless of the drink's origin, unless it is kept in cold storage, it always seems to get sour within 24 h. Palm wine is widely used in African traditional medicine, particularly in the treatment of measles, chronic yellow fever, and malaria (Oluwole et al., 2023; J. A. Santiago-Urbina, Arias-García, & Ruiz-Terán, 2015). In the Ivory Coast, palm wine is said to alleviate the pain caused by poisonous bites. Many nursing mothers who drink palm wine believe that it helps the flow of breast milk for their babies and has a diuretic effect. The distinctive qualities of palm wine have sparked interest in its potential industrial applications and practical uses (Nwachukwu, Ibekwe, Nwabueze, & Anyanwu, 2006).

In Nigeria, *Raphia hookeri* and *Elaeis guineensis* are the two trees most commonly used to make palm wine. The origin or provenance of these palm trees is a topic of great interest. The Raphia hookeri tree is the most widely known in the freshwater swamps of west and central Africa. It is produced in the tropical rain forests of India, Malaysia, Singapore, and Nigeria, where there are numerous regional variants. The *E. guineensis* oil palm variety is more prevalent globally (Forster et al., 2017; Rahu, Neolaka, & Djaha, 2023). According to a source, the E. guineensis palm tree evolved in the tropical rain forests of West Africa and is now widespread throughout the region, including in Cameroon, Côte d'Ivoire, Ghana, Liberia, Nigeria, Sierra Leone, Togo, Angola, and the Congo. According to Legras, Merdinoglu, Cornuet, and Karst (2007) and Pakiam (2020), some palm fruits were transported to the Americas and then to the Far East during the fourteenth and seventeenth centuries, where they flourished. Given that yeast strains are known to reflect human history (Legras et al. (2007), it is conceivable that the yeast strains identified in palm wine were transported to other countries via plant materials that were brought to those areas.

Naturally occurring yeasts of the genus Saccharomyces ferment the sugar-rich saps that comprise palm wine. In the absence of oxygen, sugar is used during fermentation, a metabolic process that yields alcohol and carbon (IV) oxide. The overall reaction equation is given by Equation (1).

$$C_6H_{12}O_6 \rightarrow 2\ C_2H_5OH + 2\ CO_2. \quad (1)$$

Anaerobic bacteria, such as Clostridium and Acetobacterium, can directly convert carbohydrates to acetic acid during anaerobic fermentation, eliminating the requirement for ethanol as an intermediate. The reaction is expressed in Equation (2).

$$C_6H_{12}O_6 \rightarrow 3\ CH_3COOH \quad (2)$$

In addition to cells lacking oxygen, yeast and bacteria also exhibit this phenomenon. Zymology is the field that studies fermentation. This calls for the application of a trustworthy preservation technique that can maintain the flavor and components of wine while also having a favorable impact on health. According to statistics, approximately ten million Africans enjoy palm wine, according to J. Santiago-Urbina and Ruíz-Terán (2014). Due to its all-natural constituents, it is a popular drink.

In addition, pathogenic microorganisms such as *Lactobacillus, Lieuconostoc, Klebsiella, Streptococcus, Racillus, Zymononas, Brevibacterium, Acetobacter, and Sereratia* are prevalent in Nigerian palm wine, which are thought to have originated from the water used by the tappers to dilute the beverage (Amoa-Awua, Sampson, & Tano-Debrah, 2007). The unique flavor of fresh palm wine has been attributed to lactic acid bacteria (Kouamé, Aké, Assohoun, Djè, & Djéni, 2020; Okafor, 2007). The tapper uses a palm wine keg to collect sap; other sources of yeast and bacterial microflora are the air, knife, and palm tree. As a result of the yeast's fermentative action, the alcohol content increases over time.

Due to the uncontrolled metabolic activities of yeast and bacteria, the beverage has a limited shelf life, which is a key issue in its handling. The food and beverage sectors have employed ultraviolet light to successfully clean surfaces and the air (Muliyanto, Indrayani, Satriawan, Ngaliman, & Catrayasa, 2023). Nucleic acid bases are the target organelles of microbes (pyrimidine and thymine bases) (Hakguder Taze, Unluturk, Buzrul, & Alpas, 2015). Radiation damages DNA, leading to lethality and death of the organism. The successful application of UV light to regulate the metabolic processes of these creatures will be a particularly welcome outcome (Dibofori-Orji & Ali, 2019; Nwaiwu & Itumoh, 2017). To our knowledge, there is no reported work on the preservation of palm wine using ultraviolet radiation. Therefore, this study aimed to establish standard conditions for palm wine preservation using ultraviolet radiation.

3. Research Methodology

3.1 Materials

All reagents used in this study were of analytical grade. Glassware were sterilized. Instruments were calibrated before use (Zairina, Wibisono, Ngaliman, Indrayani, & Satriawan, 2023). The glassware included glass test tubes, petri-dishes, pipettes, Bijou bottles, and McCartney bottles.

3.2 Methods

3.2.1 Microbial Analysis

3.2.1.1 Agar Nutrient

One liter of distilled water was used to dissolve a 28-gram dried nutritional agar. The mixture was then poured into 250-ml bottles, corked, and placed in an autoclave for 15 min at 121 $^{\circ}C$. Sterile nutrient agar was placed in a sterile Petri dish while still liquid, allowed to set, and then dried in an incubator for 15 min at 50 °C. For perfect sterility before use, it was cultured for 24 h (Zairina et al., 2023).

3.2.1.2 Malt Extract Agar

Mycological media differ from bacterial media because of the differences in the requirements of yeast and mold for growth, with fungi having an optimum pH much lower than that of most bacteria (Bansfield, Spilling, Mikola, & Piiparinen, 2023). Malt extract agar contains 10 % citric acid, which masks the growth of bacteria. Agar was prepared using the same method as that used for the preparation of nutrient agar.

3.2.1.3 Total Viable Count (TVC)

The pour-plate method was used to determine the total viable count, which accounted for the number of entire living organisms or clamps of organisms (colonies) in the palm wine sample. Nine milliliters of quarter-strength Ringer's solution (1/4R) was used as the diluent, and dilution factor of 10^{-1}, 10^{-2} were used for the preserved sample and 10^{-4} for the fresh, unsterilized sample.

Some molten agar medium (0.34) and 2% glucose solution were mixed with the above diluents in a sterile petri-dish, and then incubated at 35 ºC for 25 hours for bacterial organisms and at 37 ºC for 3

days in the case of yeast and mould. Colony counts were limited only to those–30-300 colonies. Duplicate sets of plates were incubated at each temperature, and the average results were obtained.

3.2.2 Sample Determination Physiochemical Analysis
The physicochemical properties examined included titrable acidity, volatile acidity, alcoholic content, sugar content, pH, moisture content, relative density, total dissolved solids, suspended solids, and vitamin C content.

3.2.2.1 Determination of the Titrable Acidity
Titratable acidity refers to the amount of organic acids in the wine sample being examined. Titration determines the quantity of organic acid that can be titrated to a pink endpoint with a dilute alkali solution (Volmer et al., 2017).

In a sterile 500 ml conical flask, 200 ml of distilled water was added and heated. An indicator solution containing 1% aqueous alcohol and phenolphthalein was added to 1 ml. To produce this light-pink tint, a 0.1 M NaOH solution was titrated. The boiling neutralized solution was then pipetted with 5 ml of the "must," which was then titrated once again to the end point with the same 0.1 M NaOH solution. The formula used to determine the titratable acidity was tartaric acid, as shown in Equation (3).

$$Tartaric\ acid\ g/100ml = \frac{V \times M \times 75 \times 100}{100 \times V} \quad (3)$$

where V=Volume of NaOH (Final reading - Initial reading)
M= Molarity of NaOH

3.2.2.2 Determination of Volatile Acidity
Volatile acidity measures the acetic acid content of the sample by determining the volume of base (NaOH) that completely neutralized 10 ml of the sample to the end-point coloration of pink lasting for approximately 15 s.

Boiling deionized water (600 ml) was injected into the outer chamber of the distillation column. An accurate volume of 10 ml of the sample was measured in the inner chamber. The sample container was thoroughly rinsed with deionized water in the same inner chamber. The outer chamber was brought to a slow boil, and approximately 40 ml of distillate was collected in the receiving flask. The distillate was quickly titrated with 0.1 M NaOH to a pink coloration end point in 15 s using approximately 1 ml of phenolphthalein indicator, or 2-3 drops. The amount of NaOH used was noted, and the volatile acidity was calculated. Volatile acidity was determined using Equation (4).

$$\text{Acetic acid g/100ml} = \frac{\text{V} \times \text{M} \times 60 \times 100}{100 \times \text{V}} \quad (4)$$

3.2.2.3 Determination of Alcoholic Content
Following the distillation of the alcohol in the sample, the specific gravity of the ethanol-to-water distillate was calculated (Isaac-Lam, 2016; Pappas et al., 2016).

A volumetric flask was filled with 25 ml of the sample, and 2 M sodium hydroxide was used to neutralize the acid content of the sample. The solution was transferred into a distillation flask, with repeated rinsing to ensure complete transfer (45 ml of deionized water was used to avoid charring). Anti-foaming agents were added, and the flask was connected to the condenser with an airtight quick-fit end. The setup was distilled at a slow boil to obtain approximately 90 ml of distillate. The temperature of the distillate was adjusted to 200 $^{\circ}C$, and the volume was adjusted to 100 ml using deionized water. The distillate (50 ml) was accurately measured in a specific gravity bottle, and the weight was noted. Distilled water (50 ml) was carefully measured into the same bottle, and its weight was noted. The ratio of the weight of the distillate to the weight of an equal volume of distilled water was calculated. The equivalent alcohol percentage composition was calculated using the following equation:
0.9999 specific gravity= 0.01% alcohol

3.2.2.4 Determination of Sugar Content

This was determined using the direct reading engineering method (DREM) with a sachoremeter. The meter contains a glass bulb, one end of which is sugar saturated when dipped in a 10 ml of the sample embedded corresponding to sugar content, and the value is read out as calibrated in a chart attached to the wall of the bulb.

3.2.2.5 Determination of pH

The sample (10 ml of the sample was measured into a 10 ml conical flask. The electrode of the pH meter, cleaned with distilled water and dried, was dipped into the sample flask. The electrode was agitated to remove the air bubbles. The pH value was read as displayed on the liquid crystal display panel of the meter (Kalra, 1995; Zhao et al., 2024).

3.2.2.6 Determination of Moisture Content

A weighted evaporating plate was filled with 10 ml of the sample. (W_1).
The sample and dish were weighed (W_2). The evaporating dish containing the sample was placed on a ringed water bath, and the moisture evaporated. The dishes were dried in a glass desiccator to a constant weight (W_3). Moisture content was determined as the difference between wet and dry weights. The moisture content was calculated using Equation 5, as follows:

W_1 = Weight of an empty dish
W_2 = Dish weight + sample weight
W_2 -- W_1 = Weight of the sample
W_3 = Weight of the dish at a constant weight.
W_2 - W_3 = Weight of moisture evaporated

$$\% \text{ Moisture} = \frac{W_2 - W_3}{W_2 - W_1} \times \frac{100}{1} \quad (5)$$

3.2.2.7 Determination of Relative Density

The relative density of the sample was determined using the gravimetric method with a pycnometer. A pycnometer was prewashed with deionized water, dried in an oven, cooled in a desiccator, weighed, and its weight (W_1) was recorded. Sterilized water (5 ml) was accurately measured in the pycnometer and weighed (W_2). An amount of 5 ml of the sample was accurately measured into the pycnometer, and its weight (W_3) was recorded. The relative density was calculated using Eq. (6).
The relative density was calculated as the ratio of the weight of the sample to the weight of an equal volume of water.

$W_3 - W_1$ = Weight of sample
$W_2 - W_1$ = Weight of water

$$\text{Relative density} = \frac{W_3 - W_1}{W_2 - W_1} \quad (6)$$

3.2.2.8 Counting the Total Amount of Dissolved Solids (TDS)

A 50 ml glass beaker was filled with 10 ml of the sample. To remove air bubbles, the meter proof was dipped into the sample in the beaker and stirred gently. The reading was recorded exactly as it appeared on the liquid crystal display panel of the meter. The total dissolved solids were calculated using Equation (7).

$$\text{Total solids (mg/)} = \frac{W_1 - W_2 \times 1000}{V} \quad (7)$$

where
W_1 = Initial weight of dried residue + dish in mg
W_2 = Final weight of the dish in mg
V = Volume of sample used in ml

3.2.2.9 Determination of Suspended Solid

Similar to the measurement of moisture content, suspended solids were measured using the gravimetric method with an evaporating dish and a bath of water.
The weight of the dish (W_1) and the dish with 10 ml of the sample were recorded (W_2). In a water bath, the sample in the dish was evaporated to dryness, and its dry weight (W_3) was recorded. The relationship used to compute the suspended solids is given by Equation 8.
Weight of empty dish = W_1
Weight of empty dish + sample = W_2
Weight of sample = $W_2 - W_1$
Weight of dish + sample after evaporation to dryness = W_3
Weight of dry matter (solids) = $W_2 - W_3$

$$\text{Percentage of solids} = \frac{W_2 - W_3}{W_2 - W_1} \times \frac{100}{1} \quad (8)$$

3.2.2.10 Determination of Vitamin C
This was performed using the titrimetric method with a standard indophenol solution.
Preparation and standardization of indophenol dye:

An indophenol solution was obtained by dissolving 0.05 g of 2.6-dichlorophenol. Indophenol dye was added to distilled water (100 ml). The filtrate was labeled after it was filtered. To standardize, 250 ml of water was added to 0.05 g of pure ascorbic acid, which was then dissolved in 10 ml of 20 % metaphosphoric acid. A 50 ml conical flask was filled with 10 ml of this solution, which was then titrated against an indophenol solution until a faint pink tint that lasted for 15 s was attained.

Procedure for the determination of ascorbic acid (vitamin C):
A 50 ml measuring cylinder was used to precisely measure 50 ml of the sample into a 100 ml volumetric flask. As a stabilizing agent, 25 ml of 20 % metaphosphoric acid was added. Distilled water was then used to fill the gap. To eliminate sulfur as acetone bisulfate and reduce interference with the titration endpoint, 2.5 ml of acetone was added to 10 ml of the solution before it was pipetted into a 50 ml conical flask. The sample was titrated against the indophenol solution until a 15-second pink colour was formed. This relationship was used to calculate the vitamin C content, as shown in Equation 9.

$$\text{100 ml sample} = \frac{B \times 10}{V} \text{ mg} \quad (9)$$

where B = volume of dye solution used
V =volume of dye solution in standardization

3.2.3 Physical Examination
3.2.3.1 Sensory Evaluation
This was done using a random sampling method. The fresh sample was given to a selected set of people consisting of six men aged 40-60, four men aged below 40 years. The same set of people was used to evaluate the preserved sample, and their reports were used as a measure of the physical sensory evaluation (Ishiwu, Nnanwube, Nkem, & Ezegbe, 2023; Mihnea, Aleixandre-Tudó, Kidd, & Du Toit, 2018).

4. Results and Discussion

4.1 Results

4.1.1 Analytical Results
The values for microbial analysis were obtained by multiplying the average number of colonies by the dilution factor. The microbial load of fresh unpreserved palm wine samples is shown in Table 1.

$$C_O = C_n \times \frac{1}{f}$$

where C_O = Original cell count
C_n = Number of colonies
f =Dilution factor

Table 1. Microbial load of fresh unpreserved palm wine sample

Average number of colonies		Total cell count	
Mould Yeast	**Bacterial**	**Yeast, Mould**	**Bacterial**
		F=10^{-4}	F=10^{-2}
325	25	3.25×10^{6}	2.5×10^{3}

The media used were malt agar for moulds and yeast, while standard plate agar fortified with malt extract agar and glucose was used for bacterial total visible count (TVC)

4.1.2 Preserved Sample

The microbial load was measured against the period of U.V radiation, and a table of count per time was created (Table 2). The variation in microbial count with time of exposure of palm wine sample to U.V. radiation for samples preserved for 10 days is shown in Table 3.

Table 2. Variation of microbial count with time of exposure of palm wine sample to U.V. radiation

Number of colonies			Total count	
Period of U.V exposure(seconds)	**Mould and Yeast F=10^{-4}**	**Bacteria F=10^{-2}**	**Mould and Yeast**	**Bacterial**
0	325	25	3.25×10^{6}	2.5×10^{3}
30	280	20	2.80×10^{6}	2.0×10^{3}
60	325	10	2.35×10^{6}	1.0×10^{3}
90	110	8	1.10×10^{6}	0.8×10^{3}
120	95	4	0.95×10^{6}	0.4×10^{3}
150	45	1.0	0.45×10^{6}	0.10×10^{3}
180	25	0.4	0.25×10^{6}	0.04×10^{3}
210	10	0.1	0.10×10^{6}	0.01×10^{3}
240	1.0	0.01	0.01×10^{6}	0.001×10^{3}

Table 3. Variation of microbial count with time of exposure of palm wine sample to U.V. radiation for samples preserved for 10 Days

Period of colonies of exposure			Total count	
TEP(seconds)	**Mould and Yeast**	**Bacterial**	**Mould and Yeast F=10^{-4}**	**Bacterial F=10^{-2}**
30	130	18	1.30×10^{6}	1.8×10^{3}
60	85	11	8.5×10^{5}	1.1×10^{3}
90	50	6	5.0×10^{5}	0.6×10^{2}

120	18	3	1.8×10^5	0.3×10^2
150	8	NIL	0.8×10^3	NIL
180	6	NIL	0.6×10^3	NIL
210	2	NIL	0.2×10^3	NIL
240	NIL	NIL	NIL	NIL

4.1.3 Chemical Analysis

The chemical analysis of palm wine after exposure to U.V rays is shown in Table 4, while the variation in the properties of palm wine samples with time of exposure to U.V radiation is shown in Table 5.

Table 4. Chemical Analysis of Palmwine after Exposure to U.V rays

Exposure time(seconds)	pH	Turbidity	Ascorbic Acid (mg/l)	Titrable acidity (mg/l)	Relative Density	Volatile acidity (mg/l)	Alcohol content (%)	Total sugar (mg/l)
0	3.2	0.105	2.3	2.28	1.08	0.7	8.5	19.4
30	3.2	0.105	2.3	2.28	1.08	0.7	8.5	19.4
60	3.1	0.105	2.3	2.28	1.08	0.7	8.5	19.4
90	3.1	0.105	2.3	2.28	1.076	0.7	8.5	19.4
120	3.1	0.105	2.3	2.8	1.076	0.7	8.5	19.4
150	3.1	0.105	2.3	3.0	1.076	0.7	8.5	19.4
180	3.1	0.105	2.3	3.0	1.076	0.7	8.5	19.4
210	3.1	0.105	2.3	3.0	1.076	0.7	8.5	19.4
240	3.1	0.105	2.3	3.0	1.076	0.7	8.5	19.4

Table 5. Variation of Properties of Palm Wine Sample with Time of Exposure to U.V Radiation for Sample Preservation.

Time	pH	Turbidity	Ascorbic acid (mg/l)	Titrable Acidity (mg/l)	Relative Density	Volatile Acidity (mg/l)	Alcohol Content (%)	Total sugar (mg/l)
30	2.9	0.11	2.1	2.29	1.08	0.7	8.5	19.4
60	2.9	0.11	2.1	2.29	1.08	0.7	8.5	19.4
90	2.9	0.11	2.1	2.29	1.08	0.7	8.5	19.4
120	2.9	0.11	2.1	2.29	1.08	0.7	8.5	19.4
150	2.9	0.11	2.1	2.29	1.08	0.7	8.5	19.4
180	2.9	0.11	2.1	2.29	1.08	0.7	8.5	19.4
210	2.9	0.11	2.1	2.29	1.08	0.7	8.5	19.4
240	2.9	0.11	2.1	2.29	1.08	0.7	8.5	19.4

4.2 Discussion of Results

All electromagnetic rays have energy E = h f

Where f = frequency factor characteristic of any electromagnetic ray

h = plank's constant

This energy is an ionizing energy with high generating power

Table 1 shows the microbial loads prior to the exposure of palm wine to U.V radiation.

Table 2 shows the microbial load after exposure to U.V radiation. A comparison of the two tables shows a remarkable decrease in microbial load down the table. Within the first 30 seconds of exposure, the

microbial load decreased from 3.25×10^6 to 2.80×10^6 for bacterial representing 13.8 % reduction. After 120 seconds (2 minutes) of exposure, the load decreased extensively from 3.25×10^6 to 0.95×10^6 for moulds/yeast and 2.5×10^3 to 0.4×10^3 bacterial, representing 70.8 % to 84 % respectively. After 240 s (4 min), the microbial load was almost insignificant for moulds/yeast and zero for bacteria.

When the preserved palm wine (post-U. V.-bottled wine) was kept aside for 10 days and reanalyzed, a remarkable observation was made. There was a further decrease in microbial load. The sample of palm wine exposed for 30 seconds showed a further reduction in loads from 2.8×10^6 to 1.3×10^6 for moulds/yeast, approximately 50 % reduction. This could mean that the moulds/yeast that were not killed within the first 30 s were reasonably alternated so much that they could not grow on a medium, indicating bio-inactivity.

The observed decline in alcohol content over time provides clear evidence that ultraviolet (UV) radiation effectively halted palm wine fermentation. This suggests that the microbial and enzymatic activities responsible for ethanol production were significantly impaired after UV exposure. The suppression of fermentation is further supported by the documented increase in pH levels over the storage period, which, when coupled with a reduction in free or unbound sulfur dioxide (SO_2), contributes to the heightened vulnerability of wine to biological degradation (Cvetkova et al., 2024). The absence of sulfur dioxide, a known antimicrobial and antioxidant agent, reduces the resistance of the beverage to microbial spoilage. Furthermore, environmental factors, particularly those influenced by UV exposure, play a critical role in this transformation. UV radiation interferes with enzymatic functionality by disrupting protein structures, thereby limiting the capacity of enzymes and microbial cells to catalyze essential biochemical reactions (Oktavia, Respationo, Erniyanti, Nofrial, & Tartib, 2023). It is also noted that UV treatment led to a measurable reduction in the available substrate content at the active sites of palm wine, possibly due to photodegradation or denaturation of organic components essential for microbial metabolism.

Karamoko, Deni, Moroh, Bouatenin, and Djè (2016) support this observation, having reported that during natural fermentation processes, a decrease in pH and sugar content is typically accompanied by an increase in titratable acidity and alcohol production. In contrast, the results of this study suggest a deviation from the normal fermentation pathway due to UV intervention. Notably, similar patterns of microbial load reduction were consistently observed across multiple bottles, reinforcing the hypothesis that UV irradiation had a sustained and positive impact on the suppression of microbial activity. These findings underscore the potential of UV sterilization as a nonthermal preservation technique for controlling fermentation and extending the shelf life of palm wine under ambient conditions.

Chemical analysis of the samples preserved, as shown in Tables 1, 2, 3, and 4, indicated that the values of chemical parameters were virtually the same for fresh and preserved palm wine. This means that while ultraviolet radiation monitored the microbial bioactivity, it did not affect the nutritional values of palm wine. This indicates that while UV radiation tracked the microbial activity, it had no impact on the nutritional content of palm alcohol. Because it eliminates endogenous microorganisms (the spoiling agent) and leaves the excipient components alone, ultraviolet light is a useful physical preservation technique for palm wine (the active ingredients).

5. Conclusion

5.1 Conclusion

This study confirms that ultraviolet (UV) radiation is an effective physical preservation method for palm wine, as it successfully inactivates endogenous spoilage microorganisms while preserving the functional and active components of palm wine. This study demonstrates that the contact time and intensity of UV exposure are critical parameters that significantly influence the microbial load and, consequently, the shelf life of palm wine. A sterilization time of 240 s was found to be optimal, resulting in a substantial reduction in microbial populations and delayed spoilage during storage. These findings provide a scientific basis for the application of UV sterilization in palm wine preservation, offering a

promising alternative to traditional methods that often allow for microbial reactivation. This approach supports improved product stability, safety, and extended usability, particularly in regions where refrigeration is limited or unavailable.

Furthermore, the application of UV radiation as a nonthermal preservation technique aligns with current global interests in developing minimally processed, safe, and shelf-stable beverages without the use of chemical additives. The effectiveness of this method in reducing the microbial load without altering the organoleptic or nutritional qualities of palm wine highlights its potential for broader industrial adoption. Given the traditional and cultural significance of palm wine, particularly in many African communities, integrating UV sterilization can help bridge the gap between traditional production methods and modern food-safety standards. Future studies are recommended to investigate the long-term biochemical stability of UV-treated palm wine and explore the scalability of this method in commercial settings.

5.2 Suggestions

Based on the findings of this study, further research should be conducted to optimize UV radiation parameters, particularly wavelength specificity, exposure duration, and dose uniformity, across different volumes and packaging conditions of palm wine. Additionally, future studies should explore the long-term effects of UV treatment on the physicochemical and sensory properties of palm wine during extended storage periods. For practical implementation, pilot- and industrial-scale trials are encouraged to assess the feasibility and cost-effectiveness of UV sterilization technology in rural and semi-urban settings where palm wine is widely produced and consumed. Stakeholders in the traditional beverage industry, including small-scale producers, cooperatives, and food safety authorities, should be engaged in developing guidelines for safe UV-based preservation. Moreover, regulatory frameworks and public awareness initiatives are needed to support the adoption of UV sterilization as a safe, sustainable, and culturally appropriate method for extending palm wine shelf life without compromising its traditional value.

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