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PRODUCTION OF ETHANOL GEL FROM CASSAVA AND BANANA PEELS

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Abstract

Fossil fuels, sometimes known as conventional fuels, are becoming increasingly scarce. To meet the fuel demand, we need an alternative source, which bioethanol can provide. Due to the abundance of agricultural wastes, they are the most viable source of commercial ethanol generation. In this study, we utilized the TMS 30572 cassava and burro/baby banana peels varieties sourced from the Owode local market in Osun State and processed into powder samples as sources of bioethanol. The acid hydrolysis was prepared by adding 4% sulphuric acid, and the biological hydrolysis was accomplished by inoculating gelatinized peel paste. Prior to the fermentation, a glucose analysis was performed; the Benedict's test confirms that both reducing and non-reducing sugars exist. Aspergillus niger was added to the hydrolyzed samples in conical flasks twenty-four (24) hours before the saccharomyces cerevisiae to breakdown the cellulose into simple sugar, agitated and left to incubate for four (4) days at a temperature of 28 °C to produce ethanol with cotton wool serving as a stopper. After the distillates were collected at 78 °C, the bioethanol gel was produced by extracting calcium acetate from dried blended snail shells in 5% acetic acid (vinegar), 5 g of CMC (carboxyl methylcellulose), and clean tap water, which were mixed together in a 250 ml glass beaker. The results from the biochemical and physicochemical analysis of the acid and biologically hydrolyzed cassava and banana ethanol gel samples were compared with the control (kay's ethanol gel). The results indicated that, apart from the control (kay's ethanol gel), the banana acid hydrolyzed ethanol gel has the highest quantity of ethanol produced, sugar content, alcohol percentage, pH, alcohol content, proof spirit, ash content, heating value, and calorific value of 49.50 g/mL, 16.5%, 49.15%, 4.9, 48.42%, 88.35%, 0.845%, 30575.66 kJ/g, and 2164.88 kJ/g, respectively. The cassava biologically hydrolyzed ethanol gel has the highest specific gravity of 0.9783 and flash point of 28.2%, while the cassava acidly hydrolyzed ethanol gel has the highest viscosity of 2.17 Pa.s and moisture content of 6.86%. The banana biologically hydrolyzed ethanol gel has the highest acidity of 10%. The results indicate that the addition of sulphuric acid to acid hydrolysis progressively increased ethanol yields, whereas yield decreased in the samples without sulphuric acid. The flammability test was also carried out by adding two (2) drops of the cassava, banana, and Kay's (control) ethanol gels in a glass beaker and lighting up to examine the nature of the flame produced. From the flame test, the cassava ethanol gel burns with a pale blue flame, the banana ethanol gel burns with a pale blue flame with a slight yellowish colour and Kay's (control) ethanol gel burns with a pale blue flame that is almost invisible in daylight, all evidence that ethanol is present. The ethanol gels will help to reduce the environmental risk associated with kerosene and charcoal sources of cooking fuel and help to consume waste, preserve the environment free of agricultural trash, and protect the environment from pollution.

Keywords: *Ethanol gel, Agricultural waste, Fermentation, Hydrolysis, Biochemical analysis, Physiochemical analysis*

Introduction

With the goal of resolving the energy dilemma (access, equity, affordability, storage, sustainability, infrastructure and technology) in the future, renewable technology is continuing to advance quickly year after year. Bioethanol is one of the renewable energy sources that can effectively replace fossil fuels. With little emissions, bioethanol can be used as gasoline or as a fuel additive (Mustafa *et al.,* 2014). In comparison to solid fuels (biomass, charcoal, and coal), ethanol is considered one of the "cleaner-burning" household fuels. It is considered a "modern" fuel, alongside liquefied natural gas (LNG), compressed natural gas (CNG), biogas, kerosene, and electricity (Lam *et al.,* 2012; World Health Organization, 2014). The daily cooking of about three (3) billion people worldwide involves the use of solid fuels such as biomass, agricultural waste, and charcoal (Venkata *et al.,* 2015). This is a problem because the use of bad cooking fuels has negative effects on the environment, the economy, and public health (WHO, 2022). One of the biggest problems for global public health is the harmful gases that are emitted during burning of solids and liquid fossil fuels, which cause serious health challenges for the users. Its demand is increasing, and thus worsens the environment's impact (WHO, 2022). According to Stumpf's (2006) estimation, about 2.5 billion people cook their food over open flames using firewood or leftover plant material. Both the environment and the health of the users are endangered by the pollutants from the open fire. Due to a multitude of health-damaging chemicals (formaldehyde, acetaldehyde, acrolein, polycyclic aromatic hydrocarbon, benzene, and others) and minute soot particles that can enter the lungs deeply, such inefficient cooking fuels and technologies release substantial amounts of indoor air pollution. Particularly at risk are women and small children, who spend much of their time close to the home hearth (Zwick, 2011). With more urbanization, there may be a gradual switch from firewood to charcoal as a fuel source, and charcoal's effectiveness could be improved. However, using renewable fuels like gas, oil products, and bioethanol fuel made from agricultural waste that have great heating value and little emissions impact would be a more practical approach. Thus, the production of ethanol gel to power clean cook stoves can reduce these hazardous emissions and pollution (Vigresh *et al.,* 2023).

Banana peels have been used to produce bioethanol gel in a number of studies, including one by Madhumala *et al.* (2020) that studied the bioethanol production from waste banana peel, focused on using pretreatment and hydrolysis as essential procedures in the synthesis of bioethanol. H_2SO_4 was used in the hydrolysis method and came to conclusion that the pretreatment stage is crucial to the generation of bioethanol.

Munfarida *et al.* (2021) looked into the possibility of using banana peels as a source material for bio-alcohol. This study set out to determine whether it would be possible to make bio-alcohol from organic waste, mostly banana peels. Bio-ethanol was produced through fermentation of three distinct types of banana peel waste: Raja, Agung, and Nangka banana peels. For the five-day fermentation process, strains of Saccharomyces cerevisiae at 1%, 3%, and 5% were employed. The results showed that Raja Nangka peel waste produced the highest amount of bio-ethanol, 1.70% (p-value 0.05), when Saccharomyces cerevisiae was added at a concentration of 5%. Gaddafi *et al.* (2016) worked on the production of bioethanol using banana peels. The study found that three pretreatment methods such as water, alkaline, and acidic pretreatments were all effective in the generation of bioethanol. According to the findings, Pretreatment with acid provided the maximum lowering sugar concentrations, while pretreatment with water alone produced the lowest concentrations. It became clear that even having a lot of reducing sugars after hydrolysis does not ensure the synthesis of bioethanol.

Mustafa et al. (2018) studied bio-ethanol production from cassava (Manihot esculenta) waste peels utilizing acid hydrolysis and fermentation techniques in an effort to maximize the yield of ethanol from cassava peel. In this work, fermentation and co-culture techniques were employed to produce ethanol from cassava peel waste as the only carbon source. Using a co-culture of *Aspergillus niger* and *Saccharomyces cerevisiae*, the production of bioethanol from cassava peels was studied. The substrates were hydrolyzed using sulphuric acid solutions at concentrations of 2%, 6%, and 10%, respectively. The sample produced 37.35 g/ml of ethanol at its maximum after 4 days of pretreatment with 10% concentrated H_2SO_4, with a pH of 4.55, 15.5% sugar, and 8.5% alcohol concentration. Heriyanti et al. (2020) used acid hydrolysis and straightforward fermentation to study the bioethanol synthesis from cassava-based industrial wastes. This is a preliminary experiment to determine how much ethanol is produced during the fermentation process of cassava pulp and peel. Different fermentation times, such as 2, 4, 6, 8, 10, 12, and 14 days, were used to carry out the fermentation process. After eight days of fermentation, the ethanol concentration that was at its peak was 6.2%. Hermansyah *et al*. (2018) investigated the bioethanol synthesis from cassava peel using yeast isolated from durian. The concentration of sulphuric acid produced by the hydrolysis process was fermented by yeast for 1,2,3,4,5,6,7 and 8 days using yeast isolated from Durian fruit. Amount of sugar consumed was determined using the DNS method, and ethanol using gas chromatography. The best hydrolysis process time was 45 minutes, and the glucose concentration was 11.189%. Efeovbokhan *et al.* (2019) investigated the synthesis of bioethanol from hybrid cassava pulp and peel via microbial and acid hydrolysis. Three different types of cassava pulp and peel (CPP) were investigated for their potential to be used in the production of bioethanol. By scattering 20% w/v CPP in 100 mL of hydrochloric acid, acid hydrolysis was carried out. By adding *Aspergillus niger* to gelatinized CPP paste, biological hydrolysis was carried out. The acid hydrolysis of three cassava types yielded 110.7 g/L, 100.4 g/L, and 96.7 g/L of reducing sugar, respectively, whereas the cassava peel produced 98.9 g/L of reducing sugar at 0.7 M in 50 minutes.

The demand for bio-ethanol as an alternative energy source is rising, and Nigeria currently imports ethanol to meet its domestic demand (Balat, 2008). The introduction of an effective and clean-burning ethanol gel will have significant potential to help society and the environment. As a result, the study focused on the production of an ethanol gel from cassava and banana peels and determine its biochemical and physiochemical characteristics, which is anticipated to be extremely suitable for a normal rural household and safe to use when compared to more traditional options

Materials and Method

The flow chart for of ethanol production from cassava and banana peels is presented in Figure 1

Production of the Bioethanol Gel from Cassava Peels

Sample Collection

Fresh cassava peels were gathered from a cassava local market trash disposal site in Ido-Osun area, Osogbo Osun State, Nigeria, while banana peels were sourced from the banana seller and local fruit juice market at Owode and Ifon markets in Osun State, Nigeria and, 10 kg of each samples were properly packed into a polythene bag for further processing.

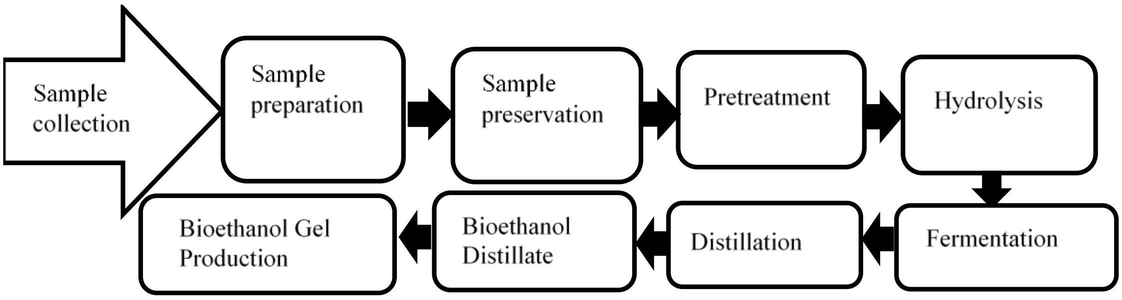

Figure 1: Flow chart for of ethanol production from cassava and banana peels

Sample Preparation and Preservation

The cassava peel sample was first washed with water and then distilled water to remove soil and dust. The peels were spread, sun-dried for a week at 13 % moisture content (wet basis), and then finely powdered with a pestle and mortar, followed by a grinding machine. The ground powdered cassava peels were passed through a 1 mm screen to achieve a standardized particle size range of 1 mm. The sample was maintained at room temperature (25°C) in airtight pails to prevent moisture absorption, and for further analysis before being transferred to the chemistry and microbiology laboratory at the Federal University of Technology Akure in Ondo State for further investigation.

For the banana peels, to make them easier to handle, the peels were first weighed before being cut into small pieces. Sun dried and oven dried at 60 °C overnight. The dried substrate was ground into powder form, sieved with a 1 mm screen, packed in airtight pails, and stored at 28 °C for further examination. Plate 1 showing the (a) collected cassava peels, (b) sundried cassava peels, (c) pounding process of the cassava peels and (d) powdered cassava peel. Plate 2 depicts the (a) Collection of banana peels (b) Sun dried banana peels (c) over dried at 60oC (d) powdered banana peels.

Preparation of the Aspergilus niger spore suspension

Aspergillus niger and Saccharomyces cerevisiae were isolated from stock cultures at the Federal University of Technology's microbiology laboratory in Akure, Ondo State. Using the morphological and biochemical processes outlined by Holt et al. (1994) and Oyeleke et al. (2012). The mixture consisted of three drops of tween 80 (non-ionic surfactant and emulsifier) and 10 mL of sterile distilled water, which were gently mixed in a test tube. Then, a five-day-old plate of Aspergillus niger isolate obtained from sand and rotten tomatoes was added to the mixture. In order to ensure that the spores in the slurry are evenly distributed, a sterile pair of forceps was used to carefully scrape the spores onto the plate. After that, filter paper was used to separate the mixture.

Pretreatment of the Cassava and Banana Peels

Two hundred (200) grams of the cassava and banana samples (fibre) and 10% (wt/wt) NaOH were added separately into a 500 cm^3 beaker in a 6:1 ratio of base to fiber. Before releasing pressure into the atmosphere, the mixtures were heated to 120 °C for six (6) hours. The solid residues were removed from the filtrate portion by filtering the solutions. The solid residues were dried at 45 °C after being thoroughly rinsed with neutral-pH tap water.

Biological hydrolysis

The pretreated cassava and banana peel samples, were weighed 200 g each, and placed in two (2) separate beakers. To make slurries, 100 milliliters of

Plate 1: Preparation of cassava peel sample consisting of washing, drying and grinding processes

distilled water were added to each beaker and mixed thoroughly. In order to ensure proper swelling, the mixtures were allowed to stand for 30 minutes. The pH meter was used to determine the pH of the samples, which were then autoclaved for 30 minutes at 120 °C. A milliliter of spore suspension with spores was added to each beaker and stirred. 50 milliliters of basal salt medium were also added and then placed on a platform shaker, one after another, to be mixed for 30 minutes at 130 rpm. A daily measurement of the sample sugar content was taken at 540 nm using the DNS technique. After that, the samples were kept in storage to ferment. The preheated cassava biological hydrolyzed sample is depicted in Plate 3.

Acid hydrolysis

The pretreated cassava and banana peel samples, were weighed 200 g each, and placed separately on two (2) 500 cm^3 conical flasks. 100 mL of distilled water and 4% sulphuric acid were also added into the flasks in a 6:1 acid-to-fiber ratio. In order to

Plate 2: Preparation of banana peel sample consisting of Peeling, cutting, drying and grinding processes

Plate 3: Biological hydrolysis preheating of the cassava sample

avoid contamination, the flasks were filled to the mark of 200 cm^3, and sterile cotton wool wrapped in aluminum foil was used to cover the flasks. The cassava sample was heated in a water bath at a temperature of 98 °C for two (2) hours, while the banana sample was heated at 120 °C for approximately six (6) hours. They were then autoclaved for 15 minutes, cooled down, and filtered into 500 cm^3 beakers using No. 1 Whatman filter paper. The pH of the filtrate samples was decreased from 4.9 to 4.5 by adding 10% NaOH. To wash the residual samples and achieve pH balance, distilled water was utilized in all treatments. After being dried in an oven at 90 °C for twelve (12) hours, the sample underwent additional analysis. Plate 4 depicts the (a) preheated cassava peels (b) preheated banana peels (c) autoclaved hydrolysate.

Benedict test

The Benedict test was conducted as follows: 1 mL of the cassava and banana sample solutions and 2 mL of Benedict reagents were completely mixed in two (2) separate test tubes and placed on a pot of boiling water for about 15 minutes. Hue shifts were produced by Benedict's reaction, which enables a semi-quantitative or approximate evaluation of the amount of reducing sugars in the samples. The quantity of reducing sugar in a sample can be determined by observing the colour shift from blue (no sugar) to green (0.5% sugar), yellow (1% sugar), orange (1.5% sugar), red (2% sugar), and brown (maximum sugar content). The color moving from blue to green indicates the presence of reduced sugar.

Fermentation process

The fermentation was performed utilizing the simultaneous saccharification and fermentation procedure (SSF) described by Krumov *et al. (*2006) and Oghgren *et al. (*2006). Aspergillus niger was introduced to each flask that contained the hydrolyzed samples twenty-four (24) hours before Saccharomyces *cerevisiae* was added for fermentation. Glucose-containing yeast cells were introduced to the hydrolyzed solution after starting the fermentation process and allowing it to continue for 4 days. The flasks were agitated and left to incubate for four (4) days at a temperature of 28 °C, with cotton wool serving as a stopper. The flasks were shaken occasionally to ensure that the organisms were evenly dispersed in the substrate mixture and that the solutions were homogeneous. The samples were finally centrifuged, and the fermented bioethanol was then extracted using distillation. Plate 5 depicts the (a) yeast added to the solution (b) fermentation of the cassava and banana peels substrate (c) bioethanol from fermented solution

Distillation process

Distillation was performed using distillation equipment. The four biologically and acidically hydrolyzed cassava and banana fermented liquids were transferred to four (4) separate round-bottom

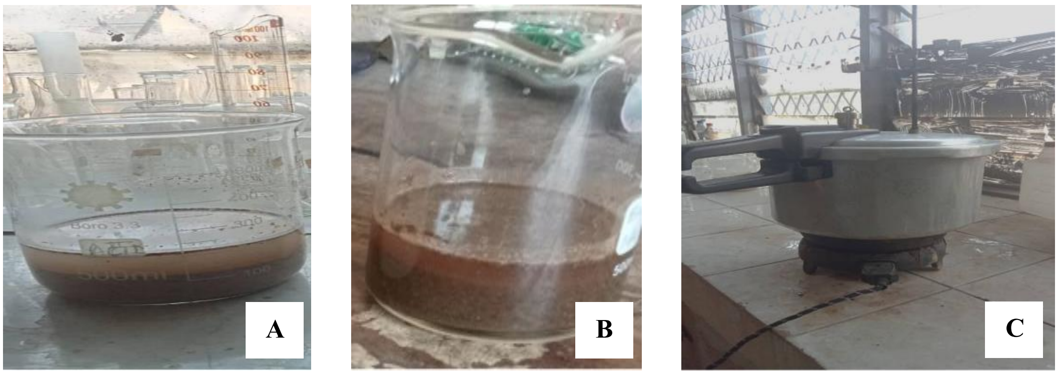

Plate 4: Acid hydrolysis of the cassava and banana samples and autoclaved hydrolysate

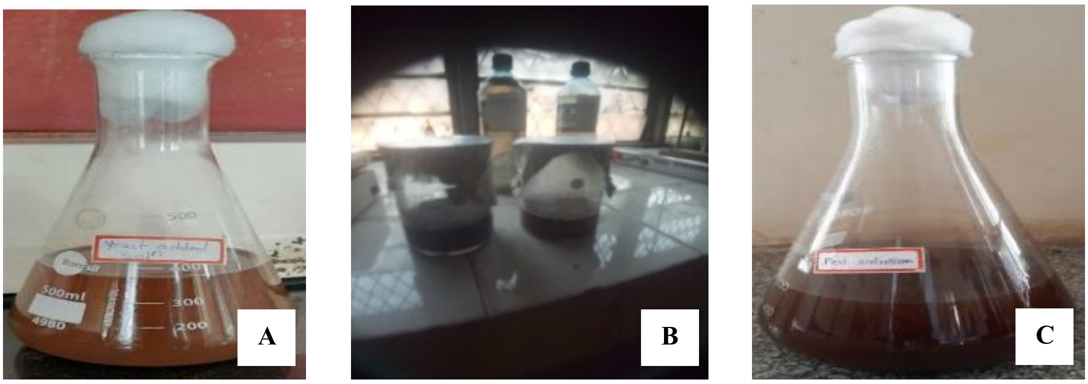

Plate 5: Yeast addition and fermentation of the cassava and banana substrate

flasks, each separately put on a heating mantle attached to a distillation column submerged in flowing water. The distillates were collected at 78 °C (the usual ethanol production temperature) in another four (4) separate flasks linked to the opposite end of the distillation column. This was done for each fermented broth using the procedure outlined in Oyeleke *et al.* (2012). Plate 6 depicts the (a) distillation setup (b) distilled bioethanol

Preparation of the calcium acetate used for the bio-ethanol gel

The calcium acetate used for the gelatinization of the banana and cassava bio-ethanol was produced by a process of extraction of the acetate from the snail shells in 5% acetic acid (vinegar). The snail shells were collected locally and sun-dried for 24 hours. The dried shells were broken down into smaller sizes using a wooden mortar and pestle. About 200 g of the crushed shells was weighed using a Mettler balance and blended in a 350 W electric blending machine to fine grain sizes. About 100 g of the blended shell was weighed and placed in a 1 liter Pyrex glass beaker. 500 mL of the 5% acetic acid was then measured and transferred into the content in the glass beaker. After 24 hours, the mixture was allowed to settle, and then the liquid was poured off and filtered through a fine muslin cloth with a small mesh size. The filtered solution was boiled to reduce the water content to about a quarter of the total volume. The solution was left to cool down to room temperature. The left-over is calcium acetate.

Production of the bio-ethanol gel

The cassava and banana bioethanol gels were separately produced by weighing 5 g of CMC (carboxyl methylcellulose) and 50 mL of clean tap water into a 350 W electric blending machine. The solution was blended to form a fine paste. The paste was then decanted into one of the five 250 mL glass beakers used for preparation. 10 g of the paste was weighed into the second and third 250 mL beakers, and 50 mL of the cassava and banana bioethanol distillates were measured using a measuring cylinder and added to the pastes in the second and third beakers, respectively. The contents of each of the beakers were properly mixed together using a glass rod to form slurry. 15 g of the prepared calcium acetate was weighed using a Mettler balance and poured inside the fourth and fifth 250 mL beakers, and 5 mL of clean water was added to form paste. The content of the second was added to the fourth beaker, while that of the third was added to the fifth beaker. Each mixture was stirred thoroughly to produce the cassava and banana bioethanol gels. Plate 7 depicts the (a) produced cassava bioethanol gel, and (b) produced banana bioethanol gel

Ethanol analysis

(i) Titratable acidity: 25 mL of the sample was titrated with a sodium hydroxide solution using a phenolphthalein indicator. The end point of the reaction was reached when the solution turned pink. Titrable acidity was calculated using Equation (1).

Plate 6: Distillation of the fermented cassava and banana substrate

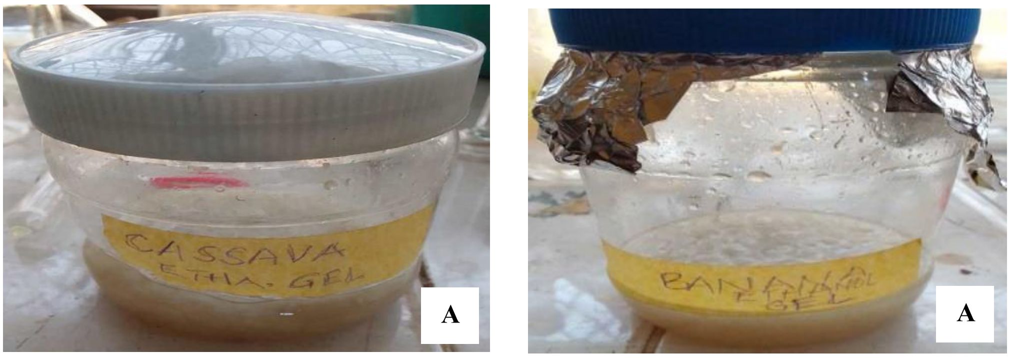

Plate 7: Cassava and banana bioethanol gel

$$TA = \frac{T \times 0.9 \times 100}{25} \qquad (1)$$

where TA is the titratable acidity in % and T is the end point of the reaction.

(ii) Specific gravity: The sample's specific gravity was calculated using a 25-mL density container. The bottle underwent a thorough cleaning process using a detergent solution, followed by rinsing with distilled water. Subsequently, it was dried in an oven at a temperature of 105 °C for duration of 30 minutes. Before measuring, the bottle was placed in a desiccator, and its weight was noted. Then, the bottle was filled with distilled water and weighed using a Mettler balance. The weight was also recorded. After that, the bottle was cleaned, dried in an oven, cooled in a desiccant, and finally filled with the sample. This was then weighed, and the result was documented. The sample solution's specific gravity was then calculated from Equation (2).

$$S.G = \frac{W_S}{W_{H_2O}} \qquad (2)$$

where $S.G$ is the specific gravity, W_S is weight of sample, and W_{H_2O} is the weight of water

(iii) Total sugars: The Abbe refractometer equipment was used to measure the total sugar content of the sample at a temperature of 30 °C. The equipment was powered on by switching the start button. The sample cell chamber was then opened by releasing the fastening knob, and a drop of the sample solution was placed on the cell compartment using a dropping pipette. The cell chamber was then closed, and the reading was taken directly from the scale.

(iv) pH: The sample's pH was measured using a pH meter equipped with a glass-calomel electrode.

(v) Determination of viscosity: The viscosity test was performed using a clean, dry Hoffman-type viscometer. To charge the viscometer, the thinner arm of the tube was inverted into the liquid sample, and the fluid was pulled up using suction force until it filled the viscometer's top tank mark. The instrument was then returned to its normal vertical position. The viscometer was then secured with a retort stand and clamp. The flow time was determined by monitoring the sample's flow between the upper and lower tank markings while applying gravitational force. It was calculated using Equation (3).

$$KV = \frac{FT \times 1.003}{FTW} \qquad (3)$$

where KV is the kinematic viscosity in $(Pa.s)$, FT is the flow time, and FTW is the flow time of water

(vi) Determination of flash point: About 5 g of the sample was placed in a 50 mL glass beaker, and the opening was carefully closed with foil paper, creating a small space for the volatilized vapour to pass out through. The beaker and contents were then held up with a clamp holder and hung on the retort stand, and the probe of a digital thermometer was dipped within the sample while ensuring that it did not touch the bottom of the beaker, which was placed on a hot plate. The sample was heated until a sufficient amount of vapor was produced when a flame was used. The temperature at which the sample flashed was then recorded.

(vii) Alcohol content: The specific gravity method was used to measure the alcohol content. The procedure for the specific gravity was followed, and the alcohol content was read off from the standard conversion table for converting ethyl alcohols to volume (ASTM D5599).

(viii) Determination of quantity of ethanol produced: Using a measuring cylinder, the distillate from banana acid hydrolyzed ethanol, banana biological hydrolyzed ethanol, cassava acid hydrolyzed ethanol, and cassava biological hydrolyzed ethanol was measured. The ethanol production amount was calculated by multiplying the volume of distillate by the density of ethanol (0.8033 g/cm^3) and expressed in g/l (Humphrey and Okafoagu, 2007).

(xi) Moisture content: The gravimetric method, as described in AOAC (1990), was used for this procedure. The initial step involved weighing a clean, dry evaporating dish using a Mettler balance (W_1). Following this, 10 g of the sample were carefully measured and added to the dish (W_2). The dish containing the sample was subsequently placed inside an oven and subjected to a drying process at a temperature of 105 °C for duration of 8 hours. After completing the drying process, the dish and sample were moved to a desiccator to cool down to room temperature before being reweighed. The process of drying, cooling, and weighing was repeated every 30 minutes until a stable weight (W_3) was obtained. The moisture content of the sample was then determined by calculating the percentage of the banana, cassava, and Kay's bioethanol gel samples total weight. The calculation was performed using Equation (4).

$$\%\,moisture\ content\ = \frac{W_2 - W_3}{W_2 - W_1} \times 100 \qquad (4)$$

where W_1 is the weight of empty evaporating dish, W_2 is the weight of sample + evaporating dish, and W_3 is the weight of sample + evaporating dish after drying at 105°C

(x) Heating value: The heating value was calculated using Equation (5).

$$HV\ =\ 2.326\,(147.60\,C)\ +\ 144.0\,V) \qquad (5)$$

where C is the % fixed carbon in the sample, and V is the % volatile matter (Bailey *et al.,* 1982)

(xi) Calorific value: The sample's caloric value was calculated by utilizing the bomb calorimeter. An established weight of the sample was placed into the calorimeter, and the necessary measurements were recorded.

(xii) Flammability test: Each sample weighing 5.0 g was measured with an electronic Mettler balance and placed into a clean, dried boiling tube. The tube was then heated with a Bunsen burner for approximately 1 minute using a test tube tong. A spark lighter was then used to ignite the gas vapour emitted from the tube. The intensity and color of the flame were observed and noted for each sample.

(xiii) Ash content: The gravimetric method was utilized for this process, following the guidelines of AOAC (1990). The first step was to find the weight of a clean, dry, empty crucible using a Mettler balance (W_1). Then, 5 g of the sample was accurately weighed and added to the crucible (W_2). After the sample was placed in a crucible, it was heated in a muffle furnace at 550 °C for 4 hours. Once the ashing process was finished, the crucible and sample were moved to a desiccator to cool down to room temperature. After cooling, the crucible and sample were weighed again (W_3). The percentage of ash content in the banana, cassava, and Kay's bioethanol gel samples was calculated using Equation (6).

$$\%\,Ash\ = \frac{W_3 - W_1}{W_2 - W_1} \times 100 \qquad (6)$$

where W_1 is the weight of empty crucible, W_2 is the weight of sample + crucible, and W_3 is the weight of sample + crucible after ashing at 550°C.

Results and Discussion

Biochemical characteristic of the ethanol from the banana and cassava substrate

Table 1 depicts the quantity of ethanol, sugar contents, specific gravity and percentage of ethanol produced from the banana and cassava peels filtrate. Figure 2 depicts the quantity of ethanol, sugar contents, specific gravity and percentage of ethanol produced from the banana and cassava peels filtrate

The fermented acid and biological hydrolyzed cassava and banana substrate were compared with the control biofuel gel. The addition of sulphuric acid during acid hydrolysis resulted in a gradual increase in ethanol yield, whereas samples lacking sulphuric acid showed a decrease in yield. This suggests that the maximum ethanol yield was achieved at the given temperature (28°C), pH (4.5), and substrate weight (20 g). The maximum ethanol

Table 1: Biochemical characteristic of the ethanol from the banana and cassava substrate

Samples	Quantity of Ethanol (g/mL)	Sugar content (%)	Specific gravity	Percentage alcohol (%)
Banana Acid Hydrolyzed Ethanol Gel	49.50	16.50	0.9355	49.13
Banana Biological Hydrolyzed Ethanol Gel	40.50	12.50	0.9662	45.25
Cassava Acid Hydrolyzed Ethanol Gel	39.00	15.50	0.9647	45.71
Cassava Biological Hydrolyzed Ethanol Gel	32.00	15.00	0.9783	35.10
Biofuel Gel (Control)	80.00	17.80	0.8552	79.50

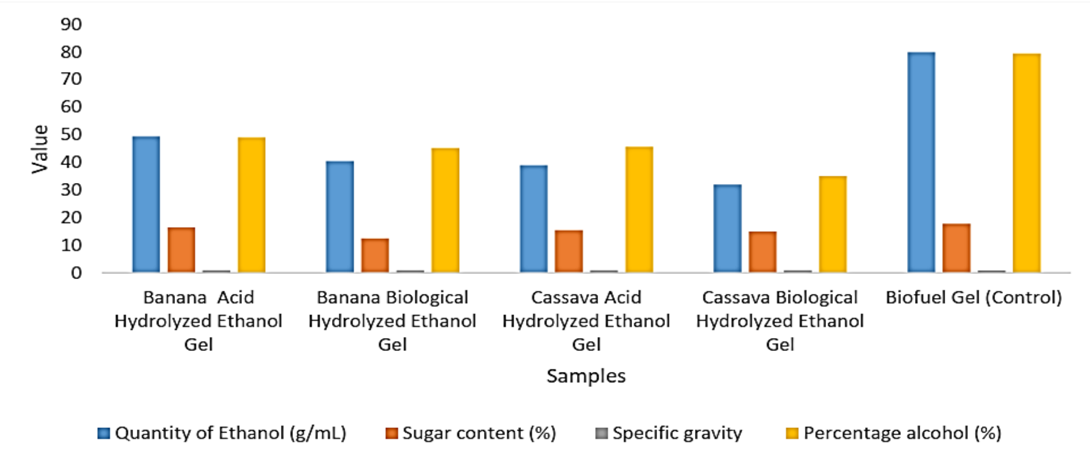

Figure 2: Biochemical characteristic of the acid and biological hydrolyzed banana and cassava substrate

yield are acid hydrolyzed banana ethanol, banana biological hydrolyzed ethanol, cassava acid hydrolyzed ethanol, and cassava biological hydrolyzed ethanol samples as shown in the biochemical characteristic of the ethanol substrate in Table 1. Figure 2 shows the graph of the biochemical characteristics of the hydrolyzed acid and biological samples, the sample with maximum ethanol was the one treated with 4% acid hydrolyzed banana ethanol gel apart from the control biofuel gel. The highest ethanol yield may be attributed to the concentration used; however, it begins to drop when the organism becomes starving and dies, resulting in a fall in metabolic activity and a subsequent decrease in ethanol yield. This study aligns with previous research by Shilpa *et* al. (2013) and Zainal *et al.* (2014) on ethanol hydrolysis, which found that the fourth day yielded the highest ethanol yield. The decrease in ethanol yield may be due to the yeast starving at no concentrations, resulting in lower productivity. Moreover, the availability of substrates that are quickly hydrolyzed to sugar by Aspergillus niger's amylolytic activity and subsequently transformed to ethanol by yeast cells in the medium may be the cause of an increase in ethanol yield (Stenberg *et al.,* 2000).

This investigation validates the results of studies carried out by Wen *et al.* (2004) and Ado *et al.* (2009) which found that, VHG fermentation can significantly improve ethanol productivity and reduce the cost of ethanol production. And the researchers evaluated the effects of different parameters, such as cassava variety, fermentation temperature, and yeast concentration, on ethanol yield. The results of Jimoh *et al.* (2009) and Ajay *et al.* (2014), who found that ethanol yield increased with increasing substrate content, were also supported by this experiment. It was observed that the greatest sugar content used during the process was that of banana acid hydrolyzed ethanol gel. From the brix table (ASTM E100-20), it was observed that the greatest specific gravity was that of cassava biological hydrolyzed ethanol gel and the greatest alcohol percentage used during the process was that from banana acid hydrolyzed ethanol gel.

Table 2: Physiochemical characteristics of the ethanol gel from banana and cassava distillate

Samples	**Ph**	**Flash point(°C)**	**Acidity (% acetic acids)**	**Alcohol content by weight (%)**	**Proof Spirit (%)**	**Viscosity (Pas/Sec^2)**
Banana Acid Hydrolyzed Ethanol Gel	4.900	26.000	6.000	48.420	88.350	2.134
Banana Biological Hydrolyzed Ethanol Gel	3.800	27.000	10.000	40.280	73.340	2.156
Cassava Acid Hydrolyzed Ethanol Gel	3.600	26.300	9.000	37.250	77.730	2.177
Cassava Biological Hydrolyzed Ethanol Gel	4.000	28.200	5.600	32.650	67.270	1.702
Biofuel Gel (Control)	7.800	23.300	8.000	77.800	146.760	3770.411

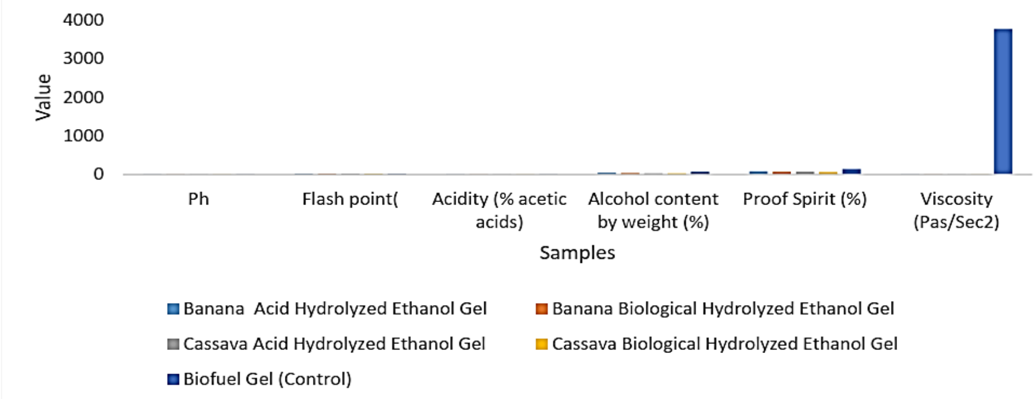

Figure 3: Physiochemical characteristic of the acid and biological hydrolyzed banana and cassava substrate

Physiochemical characteristic of the ethanol gel from the banana and cassava distillate

Table 2 depicts the ph, flash point, acidity, alcohol content by weight, proof spirit, and viscosity. Table 2 displays the physiochemical characteristics of the banana and cassava ethanol filtrate compared with the control biofuel gel after analysis. These findings demonstrate the safety, environmental friendliness, and nontoxic nature of the produced banana and cassava ethanol gel. According to the results of the pre-treatment procedures on the banana and cassava ethanol gel filtrate, the ethanols produced are very acceptable and environmentally friendly when utilized. Figure 3 provide graphical representations of the physiochemical characteristics of the banana and cassava ethanol filtrate to compare with the control biofuel gel.

Other physicochemical characteristic of the ethanol gel from the banana and cassava distillate compared with control (kay's ethanol gel)

Table 3 depicts the moisture content, ash content, heating value and calorific value of the banana and cassava ethanol gel compared with control (kay's ethanol gel)

Figure 4 depicts the moisture content, ash content, heating value and calorific value of the banana and cassava ethanol gel compared with control (kay's ethanol gel)

Table 3: Other physiochemical analysis of the banana and cassava ethanol gel compared with control (kay's ethanol gel)

Samples	Moisture content (%)	Ash content (%)	Heating value (kJ/g)	Calorific value (kJ/g)
Banana Ethanol Gel	5.461	0.845	30575.656	2164.877
Cassava Ethanol Gel	6.861	0.649	29615.664	1758.387
Kay's Ethanol Gel (Control)	0.800	0.174	33724.346	3184.158

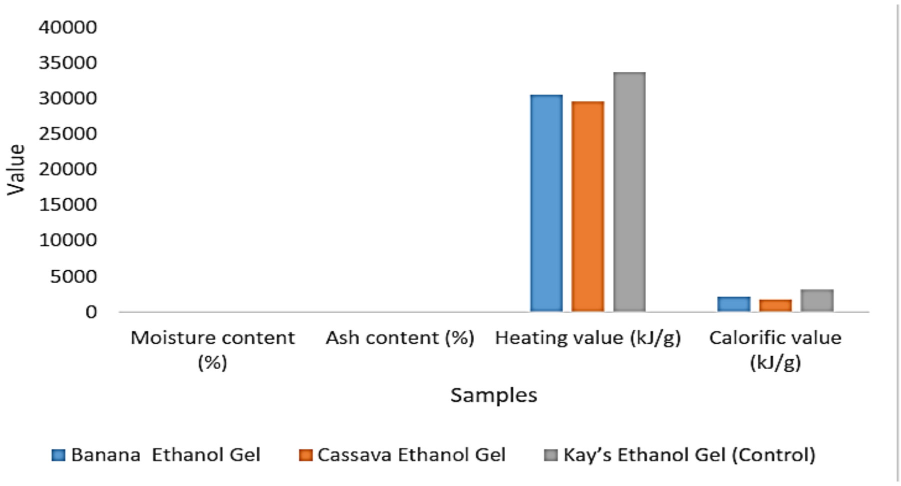

Figure 4: Other physiochemical analysis of the banana and cassava ethanol gel compared with control (kay's ethanol gel)

Table 4: Confirmatory test of the ethanol produced from the banana and cassava filtrate

Samples	Test	Observation	Inference
Banana Acid Hydrolyzed Ethanol Gel	Heat the distillate with 2 drops of potassium dichromate in a water bath for 30 minutes, using 5mL of distillate.	development of the color green	Ethanol present
Banana Biological Hydrolyzed Ethanol Gel	Heat the distillate with 2 drops of potassium dichromate in a water bath for 30 minutes, using 5mL of distillate.	development of the color green	Ethanol present
Cassava Acid Hydrolyzed Ethanol Gel	Heat the distillate with 2 drops of potassium dichromate in a water bath for 30 minutes, using 5mL of distillate.	development of the color green	Ethanol present
Cassava Biological Hydrolyzed Ethanol Gel	Heat the distillate with 2 drops of potassium dichromate in a water bath for 30 minutes, using 5mL of distillate.	development of the color green	Ethanol present
Control sample (Kay’s Ethanol Gel),	Heat the distillate with 2 drops of potassium dichromate in a water bath for 30 minutes, using 5mL of distillate.	development of the color green	Ethanol present

Table 5: Flammability test analysis of the banana and cassava ethanol gel compared with control (kay’s ethanol gel)

Samples	Banana Ethanol Gel	Cassava Ethanol Gel	Kay’s Ethanol Gel
flame test	Burns with a pale blue flame with a slight yellowish colour characteristic of ethanol with a presence of propyl alcohol	Burns with a pale blue flame characteristic of ethanol	Indicative of ethanol, it burns with a pale blue flame that is almost invisible in daylight.

Table 3 shows other physiochemical characteristics of the moisture content, ash content, heating value, and calorific value for the banana and cassava ethanol gels compared with the control"’s ethanol gel. The results as depicted in Figure 4 show that the highest moisture content, ash content, heating value, and calorific value are cassava ethanol gel, banana ethanol gel, control (Kay’s ethanol gel), and control (Kay’s ethanol gel), respectively.

Confirmatory test for ethanol production by co-culture from banana and cassava peels

The confirmatory test result is presented in Table 4.

Table 4 displays the confirmatory test results for the ethanol produced, and it was discovered that the crude distillate changed color from pink (dichromate) to green. A green tinge is a clear sign of the presence of ethanol in crude primary distillate.

Flammability test of the banana and cassava ethanol gel compared with control (kay’s ethanol gel)

The results of the flammability test from the banana and cassava peels compared with control (kay’s ethanol gel) are presented in Table 5.

Table 5 shows the flammability test analysis of the banana and cassava ethanol gels compared with the control’s ethanol gel. The presence of ethanol is indicated by the fact that both the banana and cassava gels burn with a pale blue flame, while the control gel burns with a pale blue flame that is almost invisible in daylight.

This study showed that acid hydrolysis of cassava and banana peels is more efficient than biological hydrolysis. The banana peel sample yielded the most reducing sugar, and hence the most ethanol. The acid and biological hydrolysis procedures yielded excellent results, indicating that TMS 30572 cassava and burro/babby banana peels may be utilized as a lignocellulosic material for ethanol production.

Conclusion

The production of an ethanol fuel gel from cassava and banana peels was successful. The results of the biochemical, physicochemical and flame test on the ethanol produced revealed that the banana ethanol gel has the acidity of 49.50 g/mL, 16.5%, 49.15%, 4.9, 48.42%, 88.35%, 0.845%, 30575.66 kJ/g,, 2164.88 kJ/g, and 10%, respectively. The cassava ethanol gel has the highest specific gravity, flash point, viscosity, and moisture content of 0.9783, 28.2%, 2.17 Pa.s and 6.86% respectively. Addition of sulphuric acid to acid hydrolysis progressively increased ethanol yields, whereas yield decreased in the samples without sulphuric acid. However, yield varies depending on temperature, pH, duration, and substrate concentration. Hence, these findings confirmed that, ethanol may be produced from cassava and banana peels that are safe, environmental friendliness, and nontoxic. The produced ethanol gel is most directly to lowering environmental and health risks for people, particularly in developing nations, and promote economic growth, enhance the lives of women and young people who are usually exposed.

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