Journal of
Food Chemistry & Nanotechnology

https://doi.org/10.17756/jfcn.2025-213

Research Article

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Evaluating and Comparing Drying and Extraction Techniques for Citrus (Mosambi and Kinnow) Peel Powder with Antimicrobial Effects

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Received: August 04, 2025
Accepted: September 18, 2025
Published: September 23, 2025

Citation: Sorout S, Sahil, Yadav K, Janghu S, Pandey AK. 2025. 2025. Evaluating and Comparing Drying and Extraction Techniques for Citrus (Mosambi and Kinnow) Peel Powder with Antimicrobial Effects. *J Food Chem Nanotechnol* 11(3): 139-149.

Published by United Scientific Group

Abstract

This study investigates the impact of two different drying techniques and two extraction protocols on the recovery and bioactivity of phytochemicals present in kinnow (*Citrus reticulata*) and mosambi (*Citrus limetta*) peels. The fruit peels were collected from Jaipur, Rajasthan, and were processed using sun drying and tray drying methods, then subjected to conventional solvent extraction and ultrasonication using various solvents including 50% methanol, 50% ethanol, water, and a 1:1 methanol-ethanol mixture. The extracts were evaluated for total phenolic content (TPC), total flavonoid content (TFC), antioxidant potential via 2,2-diphenyl-1-picrylhydrazyl (DPPH) radical scavenging assay, and antimicrobial activity against *Escherichia coli*, *Staphylococcus aureus* and yeast. The results indicate that tray drying followed by ultrasonication with a 50% methanol produced the most potent extracts in terms of both antioxidant and antimicrobial efficacy. This work supports the use of citrus peel waste as a low-cost, sustainable source of natural bioactive compounds for potential application in food preservation and nutraceuticals.

Keywords

Sun drying, Tray drying, Solvent extraction, Bioactive analysis, Antimicrobial

Introduction

Fruits are an essential part of the human diet, offering a variety of nutrients including various vitamins, minerals, antioxidants, and dietary fiber. Among the wide variety of fruits consumed by humans, citrus fruits are highly valued for their taste, health benefits, and functional diversity. In India, fruits like mosambi (sweet lime) and kinnow (a citrus hybrid) are cultivated in large volumes, contributing significantly to both domestic consumption and agro-industry. However, one of the less discussed challenges in the fruit industry is the generation of massive quantities of fruit waste such as peels, seeds, and pulp residues.

Fruits play a pivotal role in the human diet, demonstrating a rich offering of essential nutrients, including vitamins, minerals, antioxidants, and dietary fiber. Among the diverse selection of fruits, citrus varieties are particularly esteemed for their flavor, health advantages, and functional uses. In India, fruits such as mosambi and kinnow are produced in substantial quantities, significantly impacting both domestic markets and the agro-industry. Nevertheless, an often-overlooked concern within the fruit sector is the substantial generation of waste, comprising peels, seeds, and pulp residues, which poses environmental challenges.

About 30 - 50% of the total fruit weight is discarded as waste during industrial processing [1]. This waste is typically rich in bioactive compounds, yet it is often treated as garbage or animal feed thereby contributing to environmental stress, pollution and loss of potential value-added products. Fruit peels are a rich

source of phytochemicals such as polyphenols, flavonoids, essential oils, dietary fiber, and organic acids, all of which have documented benefits including antioxidants, antimicrobial, and anti-inflammatory activities [2].

During industrial fruit processing, it is estimated that approximately 30 - 50% of the total weight is discarded as waste [1]. This byproduct, commonly dismissed as refuse or animal feed, is often rich in bioactive compounds, thus presenting considerable environmental issues associated with pollution and the loss of potential value-added products. In particular, fruit peels are recognized as a substantial source of phytochemicals, including polyphenols, flavonoids, essential oils, dietary fiber, and organic acids. These compounds have been shown to exhibit a range of benefits, such as antioxidants, antimicrobial, and anti-inflammatory properties [2].

Given the current emphasis on sustainable food systems and circular bioeconomy, fruit peels are now being investigated not merely as waste but as valuable raw materials. Their recovery and utilization could offer economic, environmental, and health benefits. This has led researchers to explore various processing methods-especially drying and extraction techniques-to retain and isolate these compounds effectively [3].

In light of the growing focus on sustainable food systems and circular bioeconomy, fruit peels are increasingly recognized as valuable resources rather than mere waste. Their recovery and utilization present significant economic, environmental, and health advantages. Consequently, researchers are examining different processing methods, particularly drying and extraction techniques, to effectively preserve and isolate bioactive compounds [3].

Historically the use of fruit by-products plays a significant role in various traditional medicinal systems. Ancient Ayurvedic texts mention the therapeutic use of citrus peels in treating digestive problems, infections, and inflammation [4]. While fruit by-products have been used for generations in traditional medicine and home remedies, their industrial use has only started gaining real momentum in the last 20 years. In the beginning, sun drying was the only and most commonly used method because it was simple and cost-effective. However, with time more advanced techniques like tray drying, vacuum drying, and freeze-drying were introduced. These modern techniques offer better control over temperature and airflow, which is important for protecting bioactive compounds that are sensitive to heat.

Similarly, like drying methods, extraction techniques have also evolved. Traditional solvent extraction - using solvents like ethanol, methanol, or acetone-was commonly used because it was straightforward and easy to implement. However, it comes with several drawbacks, such as longer extraction times and the potential for harmful solvent residues in the final product. In the past few years, more advanced techniques like ultrasound-assisted extraction and several others have gained popularity for being faster, more efficient, and more environmentally friendly. Ultra-sonification assisted extraction has shown particularly promising results. Ultra-sonification assisted extraction uses sound waves to form tiny bubbles that collapse near plant tissues-a phenomenon called acoustic cavitation. This helps break open cell walls, making it easier to extract useful compounds more quickly and in greater amounts [5].

These advancements have sparked a fresh surge of investigation aimed at refining the parameters for drying and extraction to enhance the recovery of precious bioactive compounds from fruit rinds. These developments have fueled a new wave of research focused on optimizing the conditions for drying and extraction to maximize the recovery of valuable bioactive from fruit peels.

Numerous studies have examined the impact of various drying and extraction methods on the phytochemical profiles of fruit peels. For instance, tray drying has been shown to be more effective than sun drying in preserving the TPC and enhancing the antioxidant activity of orange peels [6]. In a related exploration, Tiwari et al. [7] indicated that ultrasound-assisted extraction markedly enhances the recovery of flavonoids and polyphenols from plant matrices.

Furthermore, specific investigations into citrus peels reveal that drying methods significantly affect both the quantity and quality of extracted compounds. Notably, kinnow peel is rich in hesperidin, naringin, and ascorbic acid, all of which are susceptible to degradation under high temperature drying. Conversely, these compounds are more effectively preserved at low to moderate drying temperatures, such as those employed in tray drying [8].

Specific studies on citrus peels have shown that drying methods influence not just the quantity but also the quality of extracted compounds. Kinnow peel, for example, has been found to contain high levels of hesperidin, naringin, and ascorbic acid-all of which degrade quickly under high-temperature drying but are better preserved in low to moderate temperatures like those used in tray drying [8].

Although largely underutilized mosambi peels have been reported to contain bioactive compounds similar to other citrus fruits, yet not much research has explored the impact of drying and extraction on its phytochemical composition [9].

These previous studies highlight the importance of method optimization in maximizing the potential of fruit peels as a functional ingredient. However, comparative studies using multiple fruits and a combination of drying and extraction methods under the same conditions are still limited.

This research paper aims to bridge that gap by conducting a comparative study of two drying techniques (sun drying and tray drying) and two extraction techniques (ultrasound-assisted extraction and conventional solvent extraction) on the peels of two different fruits named mosambi and kinnow. These fruits are chosen because of their commercial importance and the known presence of bioactive compounds in their peels which are often discarded as waste.

The study involves first drying the peels using above mentioned drying methods, followed by extraction of phytochemicals using both extraction techniques. The obtained extracts were then subjected to a series of analytical tests, including proximate composition (moisture, ash, protein, fat, and fiber), TPC, TFC, antioxidant activity (via DPPH

assay), and antimicrobial activity against selected bacterial, fungal and mold strains.

By comparing these parameters across different combinations of fruit types, drying methods, and extraction techniques, we aim to identify the most effective processing pathway for maximizing yield and bioactivity. This could provide valuable insights for industries looking to utilize fruit waste for the development of nutraceuticals, food additives, or functional ingredients.

In doing so, this research contributes to the growing body of knowledge in the field of waste value addition and sustainable food processing. It also supports the global movement toward zero-waste manufacturing and eco-friendly product development.

Materials and Methods

Material

Fresh kinnow and mosambi fruits were procured from local market in Jaipur, Rajasthan. The fruits were washed thoroughly under clean tab running water to remove any contaminants on surface of fruits. After that the peels of both the fruits were manually separated into small pieces and divided into two portions for drying.

Drying methods

A portion of kinnow and mosambi fruit peels was dried using the tray dryer in a lab. The peels were arranged in a single layer on stainless steel trays and dried at a constant temperature of 50 °C for a duration of 24 h. Tray drying helps in achieving uniform drying and reducing microbial contamination risks [10].

Another portion of fruit peels were uniformly spread in a single layer over a clean newspaper and placed at a sunlight prone area for sun drying. The drying was conducted under natural sunlight for 2 - 3 days during the months of January and February, when the ambient temperature across the region of Jaipur ranged between 15 °C - 22 °C. To make sure even drying, the peels were manually turned at regular intervals throughout the drying period [11].

The moisture content of both kinnow and mosambi peel powders was analysed through the oven-drying method as described in Association of Official Analytical Chemists (AOAC) Official Method 930.15 [12]. A known weight of each peel sample was taken and placed in a preheated hot air oven maintained at a temperature of 105 °C. The samples were left in oven to dry until a constant weight was achieved. Constant weight means all the free moisture has been removed. The final moisture content was calculated based on the weight loss observed. The moisture content calculated using the below mentioned formula:

$$Moisture\ (\%) = \left[\frac{(Initial\ weight - Final\ weight)}{Initial\ weight}\right] \times 100$$

Where, initial weight is the weight of the sample before drying (g) and final weight is the weight of the sample after drying at a constant weight (g).

pH determination

The pH of the samples was determined following the AOAC Official Method 932.12 [12]. A digital pH meter was calibrated using standard buffer solutions at pH 4.00 and 7.00 was used for all measurements. For liquid samples, approximately 50 mL of the homogenized sample was placed in a clean beaker, and the pH electrode was immersed directly in the solution. For semi-solid or solid samples, a 1:1 (w/v) slurry was prepared using distilled water prior to measurement. The electrode was rinsed with distilled water and gently blotted dry between measurements to avoid cross-contamination. All measurements were performed at room temperature (22 ± 2 °C), and the pH was recorded to two decimal places after stabilization of the meter reading.

Determination of total soluble solids (TSS)

TSS content of the samples was determined in accordance with AOAC official Method 942.15 [12] using a digital refractometer. The instrument was calibrated with distilled water prior to analysis. For clear liquid samples, a few drops were placed directly on the refractometer prism. In the case of pulpy or semi-solid samples, the juice was extracted by filtration or centrifugation to obtain a clear supernatant suitable for measurement. The TSS value was expressed as degrees Brix (°Brix), indicating the percentage (w/w) of soluble solids—primarily sugars—present in the sample. All measurements were carried out at 20 °C with automatic temperature compensation enabled. Each sample was analyzed in triplicate, and mean values were reported.

Determination of titratable acidity

Titratable acidity was determined according to AOAC Official Method 925.10 [12]. A known weight or volume of the sample (typically 10 g or 10 mL) was diluted with an equal volume of distilled water and thoroughly homogenized. The mixture was then titrated with standardized 0.1 N sodium hydroxide solution using phenolphthalein as an indicator. Titration was carried out until a persistent pale pink endpoint was observed, indicating neutralization of the organic acids present in the sample. The titratable acidity was calculated and expressed as a percentage of the predominant acid, depending on the sample type. Each sample was analyzed in triplicate, and the mean value was reported.

Peel powder preparation

After drying, the kinnow and mosambi peels were allowed to cool down at room temperature. Once cooled, they were crushed into a fine powder using a laboratory grinder. To make sure the powder was even and smooth, powder was passed through a fine sieve of mesh size 80. The final obtained peel powders were then stored in clean, airtight containers at room temperature until they were needed for the extraction process.

Extraction techniques

To analyze the bioactive compounds, present in kinnow and mosambi peel powders, two types of extraction methods (conventional and non-conventional method) with 4 different solvents were employed. The same protocol was applied to both types of peel powders to allow for direct comparison.

Conventional solvent extraction: This was carried out using four different solvents, named: methanol-ethanol, a 1:1 mixture of 50% methanol, 50% ethanol, and distilled water [13]. For extraction from each type of solvent, an accurately weighed 1 g of dried peel powder either of kinnow or mosambi was taken and mixed with 20 ml of the selected solvent in separate conical flask. Same process is done with all the solvents, with both the dried sample powders. All 8 mixtures were then incubated on an orbital shaker for 18 h set at a temperature of around 25 °C to enhance the extraction of phenolics and other bioactive compounds [14].

After incubation, all 8 samples were centrifuged at 4500 revolution per min (rpm) for a period of 5 min to separate solid residues from the liquid extract. The resulting filtrates or centrifuged liquids were carefully collected and passed through Whatman no. 1 filter paper to ensure clarity. These filtered extracts were subsequently stored at a temperature of -18 °C in airtight plastic bags or containers until further analysis [15]. This extraction procedure was performed identically for both kinnow and mosambi peel samples to ensure consistency in comparative evaluation.

Non-conventional solvent extraction: Ultrasound-assisted extraction technique was performed on both kinnow and mosambi peel powders using the same four type of solvents utilized in the process of solvent extraction method: 50% methanol, 50% ethanol, a 1:1 mixture of methanol and ethanol, and distilled water [16]. For each extraction, 1 gram of dried peel powder was mixed with 20 ml of the selected solvent. The mixtures were then treated with ultrasonic waves in a bath sonicator set at a temperature of 45 °C for a period of 30 min.

The use of ultrasonic waves enhances the disruption of plant cell walls, enabling the solvent to penetrate more effectively and extract bioactive compounds more efficiently. This method enhances solvent diffusion and mass transfer, resulting in a quicker and more effective extraction of phytochemicals.

After sonication process, all 8 mixtures were centrifuged at 4500 rpm for 15 min to separate the solid particles from the liquid. The supernatants obtained were carefully collected and filtered to remove any remaining unwanted particles. These filtered extracts were then stored in airtight containers at a temperature of -18 °C until they were analyzed for their biochemical and antimicrobial properties [17].

Selection of best drying and extraction method

For the selection of the best drying and extraction technique multiple factors were taken into account such as TPC, TFC, antioxidant capacity (measured by DPPH assay), where all the experiments were performed in triplicate, antimicrobial activity (using two bacterial strain Gram negative (*E. coli*), Gram positive (*S. aureus*), and yeast.

TPC

Determination of the TPC was done using the Folin–Ciocalteu colorimetric method [18]. A measured quantity of 1 g of dried kinnow peel powder was thoroughly mixed with 20 ml of the chosen solvent. Then, a measured 1 ml volume of reagent Folin–Ciocalteu was added to the mixture. After an interval of 6 min, a measured volume of 10 ml of 7% sodium carbonate (Na_2CO_3) solution was mixed into the mixture. Distilled water was added to make up the volume of the mixture to 25 ml. Then the solution was incubated for a period of 90 min at room temperature in the dark to allow full color development. Absorbance of the mixture was recorded at a wavelength of 750 nm, using a ultraviolet-visible (UV-Vis) spectrophotometer. TPC was calculated using mg/100 g of sample [1]. Formula used to calculate TPC is mentioned below.

$$TPC\ (mg/100\ g) = \frac{c \times v}{M}$$

Where, c is concentration from the calibration curve (mg/mL), v is volume of extract (mL) and M is mass of dried extract (g).

TFC

Determination of the TFC was done using a method called aluminum chloride colorimetric [19]. An accurately measured volume of 0.5 ml of the extract was taken and mixed with a volume of 0.3 ml of 5% sodium nitrite solution. After a reaction time of 5 min, a measured volume of 0.3 ml of 10% aluminum chloride solution was added to the mixture. After an additional 6 min of reaction time, a measured volume of 2 ml of 1 N sodium hydroxide was added to the mixture. Distilled water was added to the final mixture to make up the volume to 10 ml. A well stir was given to solution and then its absorbance was determined at a wavelength of 510 nm using a UV–Vis spectrophotometer. TFC was calculated using mg/100 g of sample. Formula used to calculate TFC is mentioned below.

$$TFC\ (mg/100\ g) = \frac{c \times v}{M}$$

Where, c is concentration obtained from quercetin standard curve (mg/mL), v is total volume of extract (mL), and M is weight of the extract sample (g).

Antioxidant activity

Antioxidant activity was assessed using the DPPH free radical scavenging assay [20]. A measured volume of 1 mL of extract was mixed with 3 mL of a 0.1 mM DPPH methanolic solution and incubated in the no light condition for a period of 30 min at room temperature. The absorbance of the sample and blank was measured at a wavelength of 517 nm. The percentage inhibition of the DPPH radical was calculated to estimate the antioxidant capacity of the extract using the below mentioned formula.

$$Inhibition\ (\%) = \frac{A_0 - A_1}{A_0} \times 100$$

Where, A_0 is absorbance of control (DPPH without sample) and A_1 is absorbance of sample (DPPH with extract)

Antimicrobial activity

The antimicrobial activity of the extracts was studied using the 96-well microdilution method. The test organisms included *E. coli* (a Gram negative bacterium), *S. aureus* (Gram positive bacterium) and yeast, naturally occurring bacteria typically responsible for food spoilage. Each well of the microplate was

filled with a specific concentration of extract and inoculated with the microbial strain. The plates were then incubated under optimal conditions suited to each organism. After incubation, microbial growth was evaluated either visually or by measuring optical density, depending on the organism tested. The degree of inhibition observed helped determine the antimicrobial potential of each extract, providing insights into their possible application in food preservation or natural antimicrobial formulations.

Statistical analysis

All the experiments were carried out in triplicate, and two-way Analysis of Variance (ANOVA) was performed to analyze the effects of moisture content in sun drying and tray drying peel powder of (kinnow and mosambi). In contrast, students T-test was performed for kinnow and mosambi, estimation experiments. The analysis included two independent variables: name of analysis (a row factor) and name of sample (a column factor), and their interaction. A percentage contribution of each factor as a source of variation was calculated, and a significance level was checked. All statistical analyses were performed using GraphPad Prism, and the effect size was reported as mean ± standard deviation of at least three biological replicates.

Results and Discussion

The moisture content of citrus peel powders varies depending on the drying method used. As shown in figure 1, tray drying led to significantly lower moisture content compared to sun drying. For kinnow peel, tray drying reduced moisture while sun drying retained and for mosambi fruit peel, tray drying resulted increased whereas sun drying remained. These results confirm that tray drying is more efficient for moisture removal, which is essential for enhancing shelf life and reducing microbial spoilage (Table 1 and table 2).

TPC, TFC, and antioxidant activity

The results for TPC, TFC, and antioxidant activity are summarized in figure 2 and figure 3. The findings clearly show that both the drying method and extraction technique have a significant impact on the yield of bioactive compounds from citrus peel powders.

In the case of TPC, the highest value was recorded in tray-dried kinnow peel when extracted with 50% methanol using ultrasonication, measuring 0.866 ± 0.006 mg GAE/100 g. A similarly high value of 0.869 ± 0.013 mg GAE/100 g was observed using 50% methanol in solvent extraction under the same drying method. This indicates that tray drying helps preserve phenolic compounds, and the use of ultrasonic waves further enhances their release by effectively disrupting the plant cell structure. In contrast, samples extracted with water or ethanol showed considerably lower TPC values, suggesting that solvent polarity and interaction with the sample matrix also influence phenolic recovery.

For TFC, the best results were obtained from tray-dried mosambi peel extracted with a 50% methanol under both techniques, yielding 0.859 mg QE/100 g. Kinnow samples also showed good flavonoid content when sun-dried and extracted with the same solvent mixture via conventional extraction, 0.63 mg QE/100 g. These observations highlight that mixed solvents and ultrasound-assisted extraction together improve flavonoid release, likely due to increased solvent penetration and enhanced mass transfer.

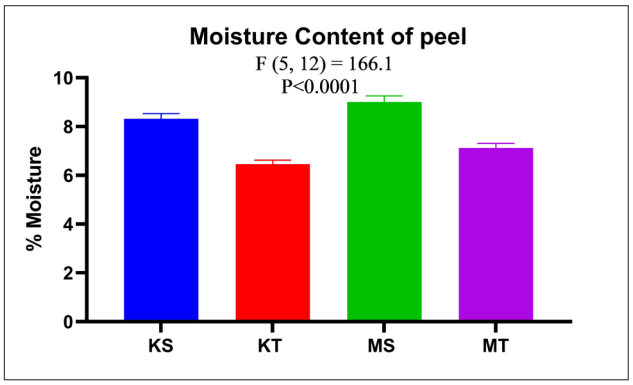

Figure 1: Moisture content of peel (p < 0.0001).

Table 1: Comparative evaluation of bioactive compounds and quality attributes in kinnow powder produced using different drying techniques (sun drying and tray drying).

Parameter	Sun drying	Tray drying
pH	4.79 ± 0.02	5.09 ± 0.03
TSS (˚Brix)	2.70 ± 0.04	1.60 ± 0.03
Titratable acidity (TA) (%)	1.70 ± 0.04	0.90 ± 0.02

Table 2: Comparative evaluation of bioactive compounds and quality attributes in mosambi powder produced using different drying techniques (sun drying and tray drying).

Parameter	Sun drying	Tray drying
pH	4.80 ± 0.03	4.02 ± 0.03
TSS (˚Brix)	1.40 ± 0.01	2.40 ± 0.03
TA (%)	1.60 ± 0.04	0.90 ± 0.02

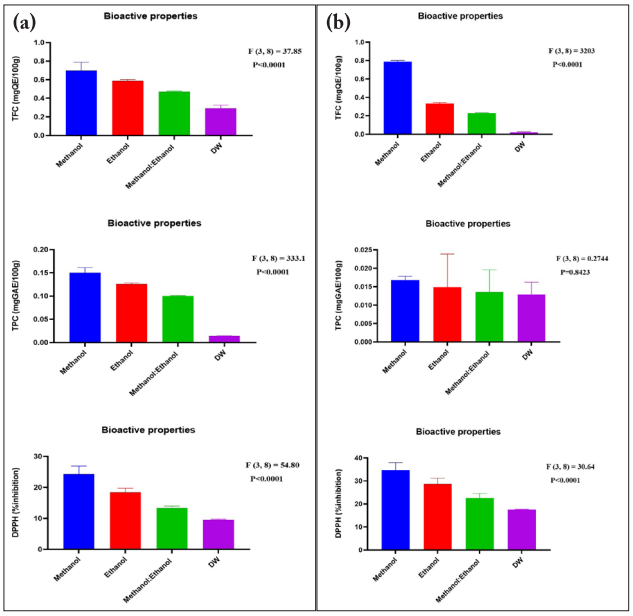

Figure 2: Bioactive properties of sun dried kinnow peel powder **(a)** solvents extraction and **(b)** ultrasonication.

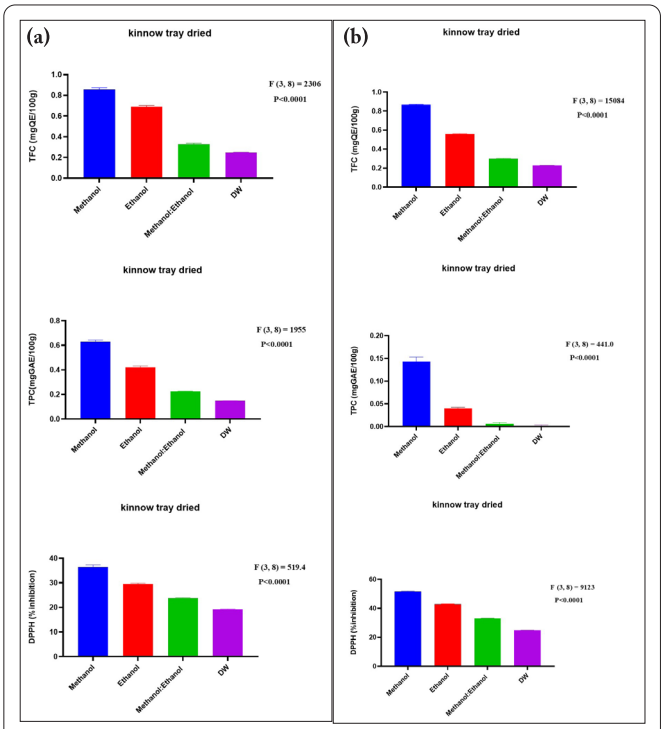

Figure 3: Bioactive properties of tray dried kinnow peel powder **(a)** solvent extraction and **(b)** ultrasonication.

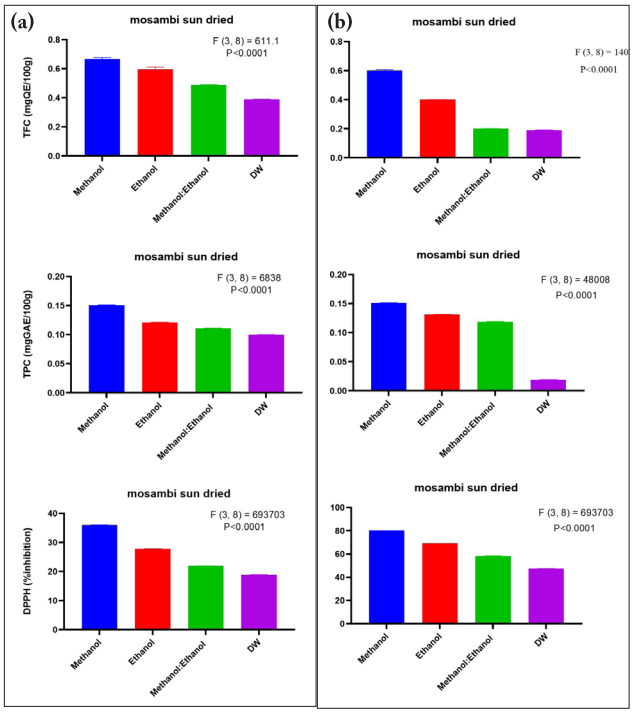

Figure 4: Bioactive properties of sun dried mosambi peel powder **(a)** solvent extraction and **(b)** ultrasonication.

The antioxidant activity, measured through the DPPH radical scavenging assay, further supports these trends. Among kinnow samples, the highest antioxidant activity was recorded in tray-dried peels extracted with 50% methanol using ultrasonication, showing a scavenging effect of 36.36%. Interestingly, the strongest response overall came from sun-dried mosambi peel, extracted with 50% methanol and processed using ultrasonication, reaching 51.58% inhibition. This suggests that antioxidant activity may also be influenced by other bioactive such as ascorbic acid or aromatic compounds, and not just polyphenols or flavonoids.

Taken together, these findings confirm that tray drying followed by ultrasonic extraction, particularly with methanol or a methanol–ethanol mixture, is the most effective combination for maximizing the recovery of phenolics, flavonoids, and antioxidant capacity in citrus peel powders.

Antimicrobial activity

The antimicrobial potential of citrus peel extracts was evaluated using *E. coli* as the test organism. Inhibition was measured by calculating the fold change in optical density ($OD_{18}h/OD_0h$), where a fold change value of 1.0 indicates no effect[18] on bacterial growth (neutral). Values less than 1.0 suggest inhibition, while values greater than 1.0 indicate stimulation of microbial growth. This interpretation was applied consistently across all treatment groups.

As illustrated in figure 4 to figure 28, the strongest antimicrobial activity was observed in treatments involving tray drying followed by ultrasonic extraction. Specifically:

- KTU = Kinnow tray drying + Ultrasonic extraction
- KTC = Kinnow tray drying + Conventional extraction
- KSU = Kinnow sun drying + Ultrasonic extraction

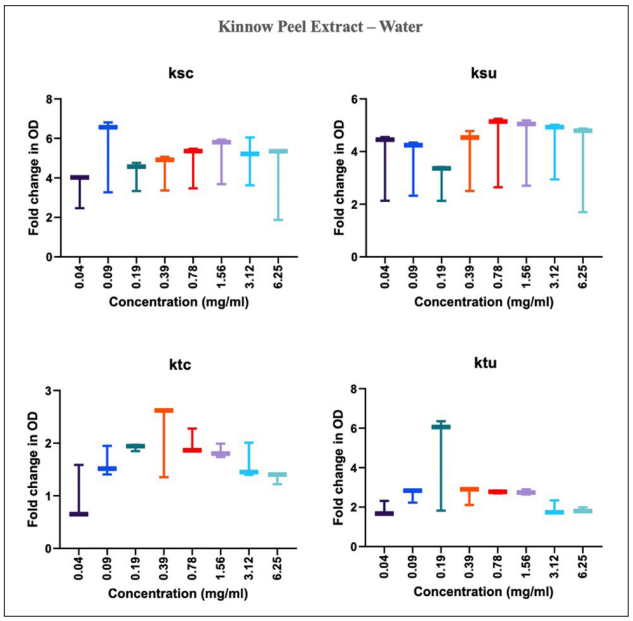

Figure 5: Effect of kinnow peel water extract on *E. coli* growth at varying concentrations. Fold change ($OD_{18}h/OD_0h$) is plotted for four drying and extraction conditions.

- KSC = Kinnow sun drying + Conventional extraction
- MTU = Mosambi tray drying + Ultrasonic extraction
- MTC = Mosambi tray drying + Conventional extraction
- MSU = Mosambi sun drying + Ultrasonic extraction
- MSC = Mosambi sun drying + Conventional extraction

Among these, KTU and MTU treatments consistently showed the lowest fold change values, indicating the strongest inhibition of *E. coli* growth. This suggests that combining

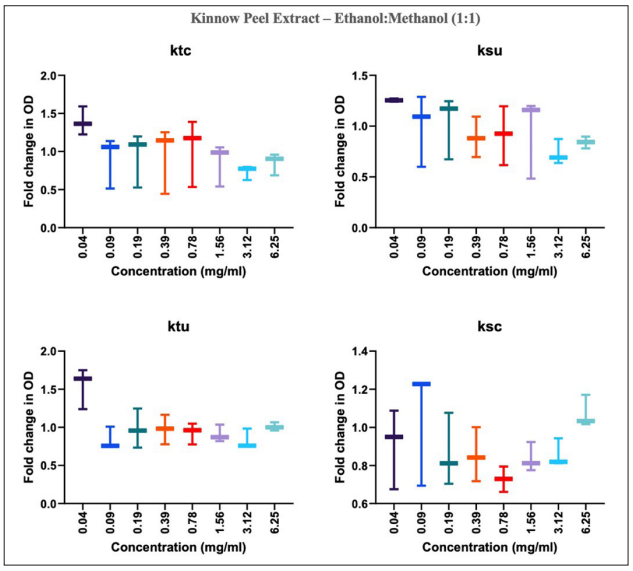

Figure 6: Effect of kinnow peel methanol-ethanol (1:1) extract on *E. coli* growth at varying concentrations. Fold change ($OD_{18}h/OD_0h$) is plotted for four drying and extraction conditions.

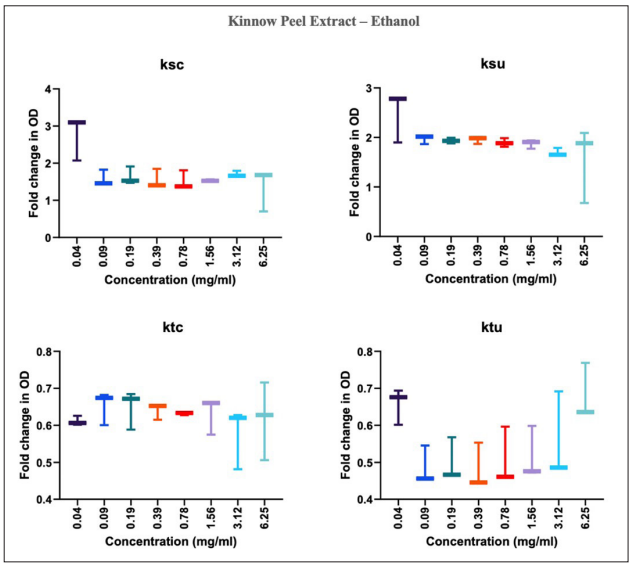

Figure 7: Effect of kinnow peel 50% ethanol extract on *E. coli* growth at varying concentrations. Fold change ($OD_{18}h/OD_0h$) is plotted for four drying and extraction conditions.

tray drying with ultrasonic-assisted extraction, particularly using methanol or methanol–ethanol mixtures, significantly enhances the antimicrobial properties of both kinnow and mosambi peels.

Notably, mosambi peel extracts, especially those extracted using 50% ethanol and water under ultrasonication, exhibited a broader antimicrobial spectrum, whereas kinnow extracts tended to show stronger inhibition overall. These results support the potential use of citrus peel extracts as natural antimicrobial agents for food preservation or therapeutic applications.

Conclusion

This study highlights how both drying methods and extraction techniques significantly impact the recovery of valuable bioactive compounds from kinnow and mosambi peels. Among all the tested combinations, tray drying followed

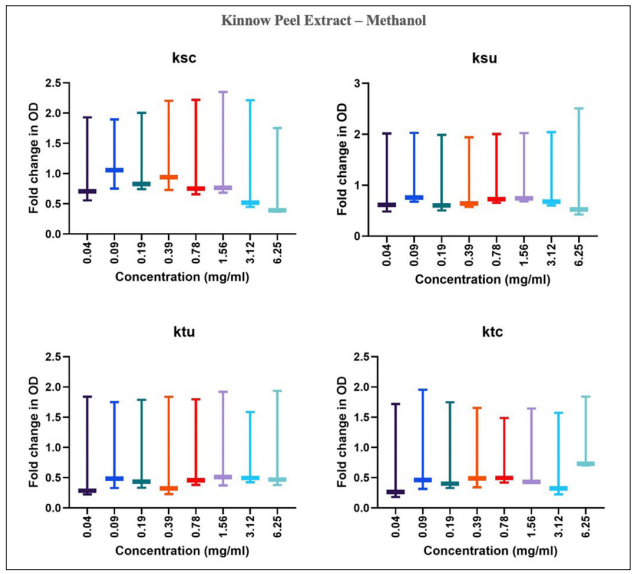

Figure 8: Effect of kinnow peel 50% methanol extract on *E. coli* growth at varying concentrations. Fold change ($OD_{18}h/OD_0h$) is plotted for four drying and extraction conditions.

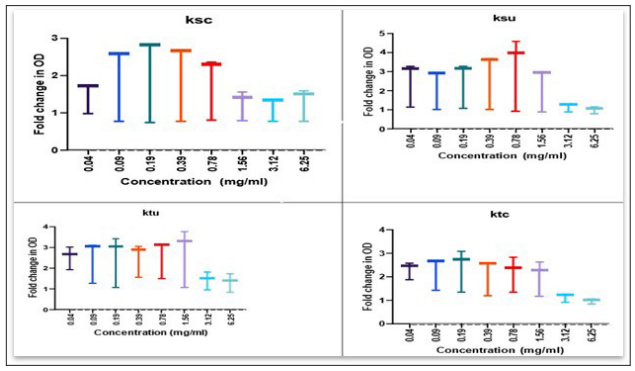

Figure 9: Effect of kinnow peel 50% methanol extract on *S. aureus* growth at varying concentrations. Fold change ($OD_{18}h/OD_0h$) is plotted for four drying and extraction conditions.

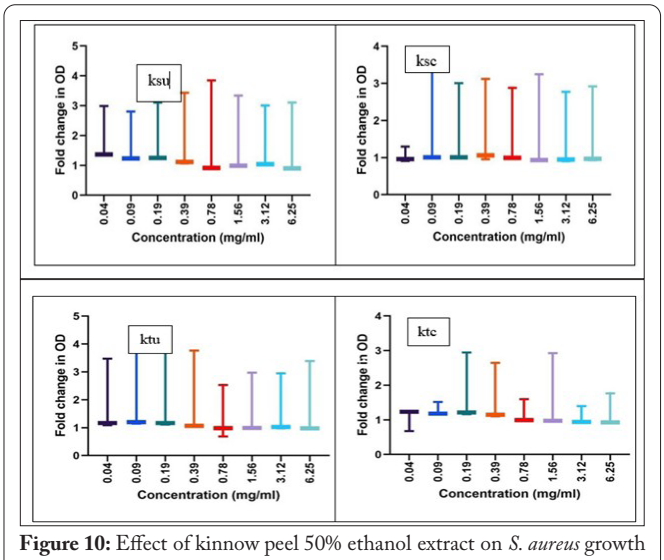

Figure 10: Effect of kinnow peel 50% ethanol extract on *S. aureus* growth at varying concentrations. Fold change ($OD_{18}h/OD_0h$) is plotted for four drying and extraction conditions.

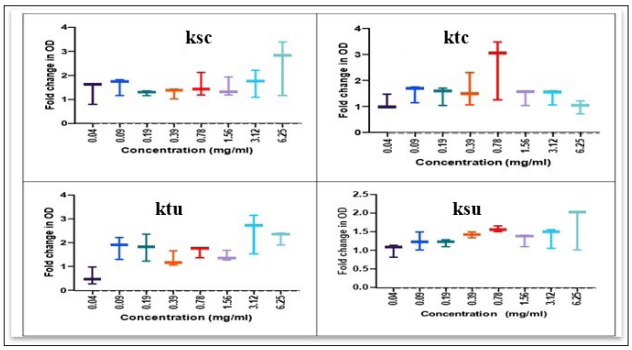

Figure 11: Effect of kinnow peel water extract on *S. aureus* growth at varying concentrations. Fold change ($OD_{18}h/OD_0h$) is plotted for four drying and extraction conditions.

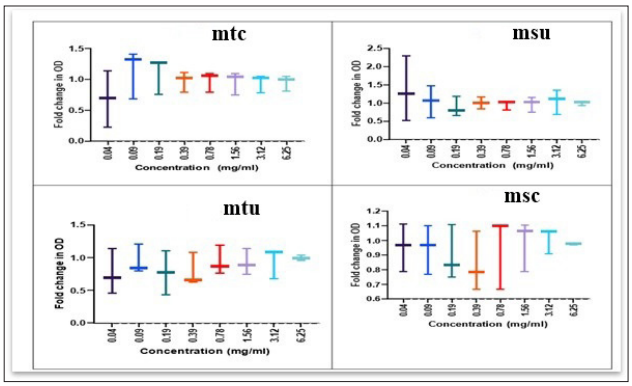

Figure 12: -Effect of kinnow peel methanol:ethanol extract on *S. aureus* growth at varying concentrations. Fold change ($OD_{18}h/OD_0h$) is plotted for four drying and extraction conditions

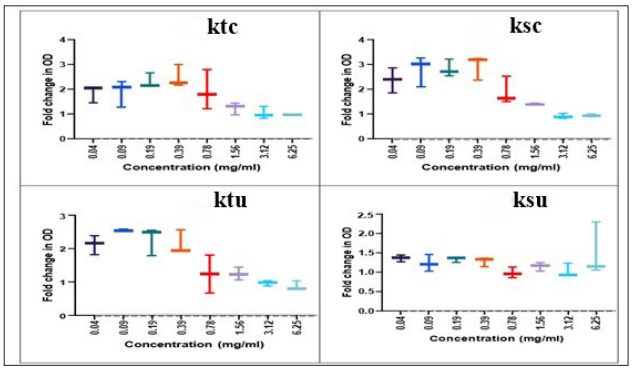

Figure 13: Effect of kinnow peel methanol:ethanol extract on yeast growth at varying concentrations. Fold change ($OD_{18}h/OD_0h$) is plotted for four drying and extraction conditions.

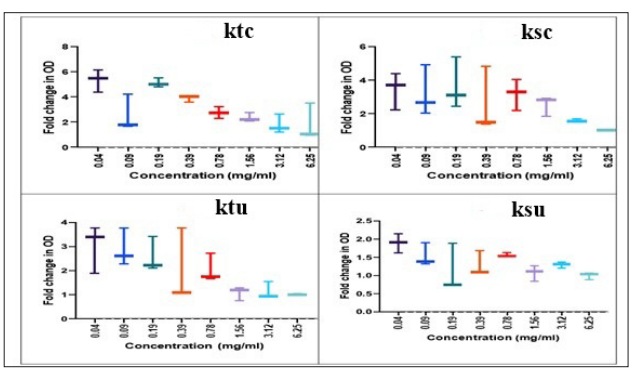

Figure 14: Effect of kinnow peel 50% methanol extract on yeast growth at varying concentrations. Fold change ($OD_{18}h/OD_0h$) is plotted for four drying and extraction conditions.

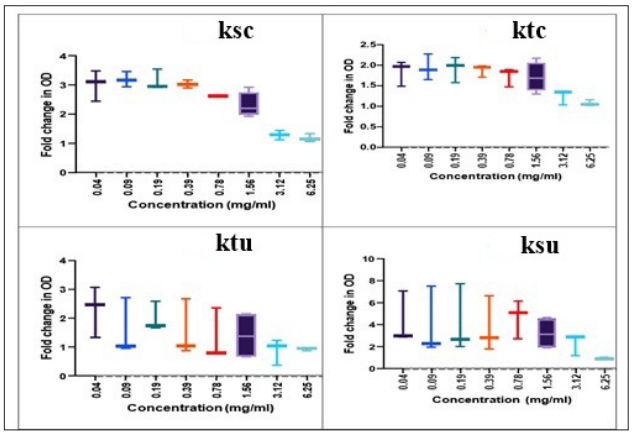

Figure 15: Effect of kinnow peel 50% ethanol extract on yeast growth at varying concentrations. Fold change ($OD_{18}h/OD_0h$) is plotted for four drying and extraction conditions.

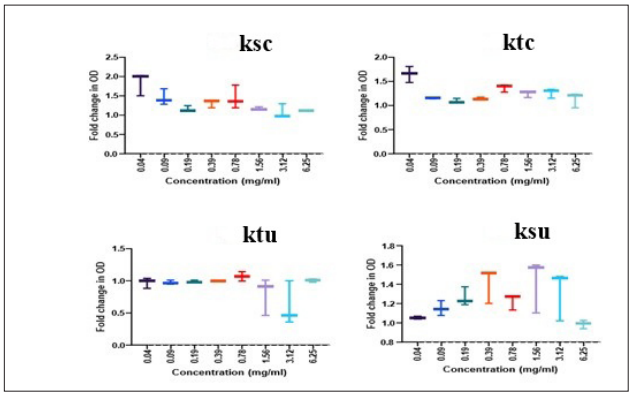

Figure 16: Effect of kinnow peel water extract on yeast growth at varying concentrations. Fold change ($OD_{18}h/OD_0h$) is plotted for four drying and extraction conditions.

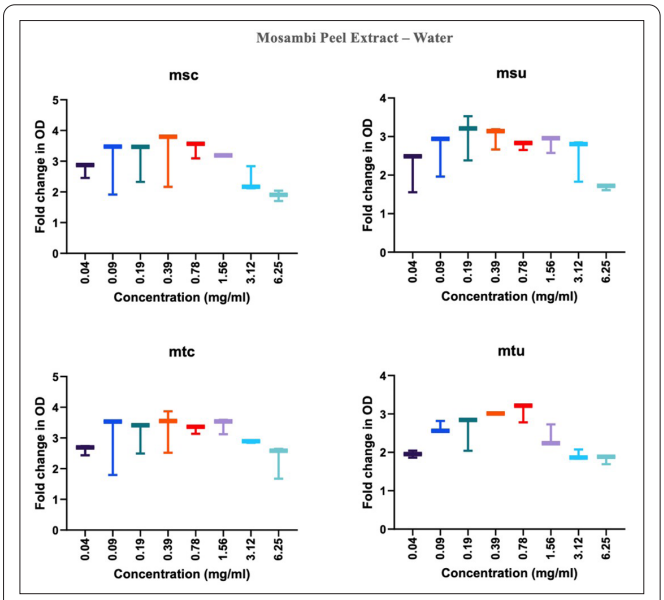

Figure 17: Effect of mosambi peel water extract on *E. coli* growth at varying concentrations. Fold change ($OD_{18}h/OD_0h$) is plotted for four drying and extraction conditions.

by ultrasonic extraction, especially using 50% methanol or a methanol–ethanol mixture, proved to be the most efficient for enhancing TPC, TFC, and antioxidant activity. Between the two fruits, kinnow peel was more effective in yielding phe-

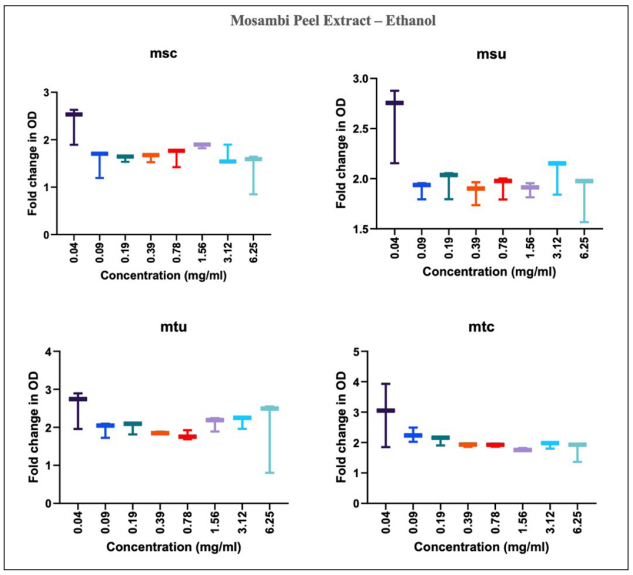

Figure 18: Effect of mosambi peel 50% ethanol extract on *E. coli* growth at varying concentrations. Fold change ($OD_{18}h/OD_0h$) is plotted for four drying and extraction conditions.

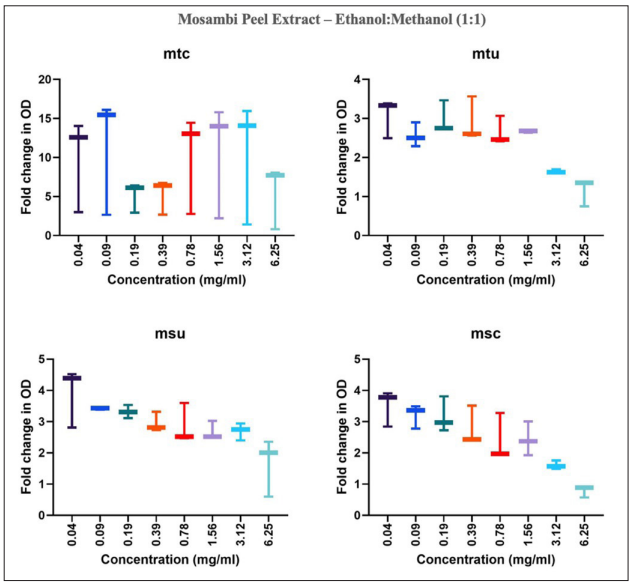

Figure 19: Effect of mosambi peel methanol:ethanol (1:1) extract on *E. coli* growth at varying concentrations. Fold change ($OD_{18}h/OD_0h$) is plotted for four drying and extraction conditions.

nolic compounds and antioxidant potential, while mosambi peel demonstrated stronger antimicrobial activity, particularly in ethanol-based ultrasonicated extracts. The findings confirm that citrus peels, which are typically discarded as waste, can be transformed into a valuable source of natural antioxidants and antimicrobials. With further optimization and validation, these extracts could find application in functional foods, natural preservatives, nutraceuticals, and cosmetic formulations, promoting sustainability and adding value to agro-industrial byproducts.

Acknowledgements

None.

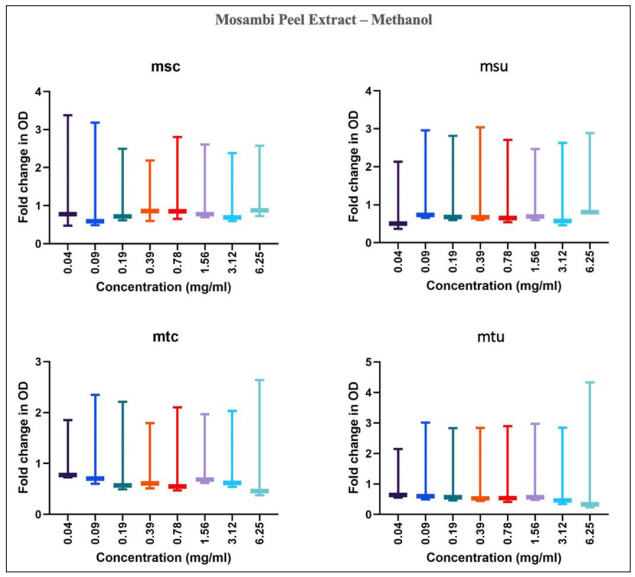

Figure 20: Effect of mosambi peel 50% methanol extract on *E. coli* growth at varying concentrations. Fold change ($OD_{18}h/OD_0h$) is plotted for four drying and extraction conditions.

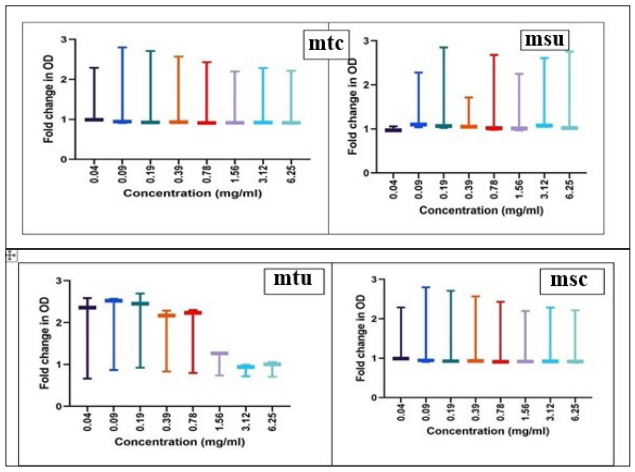

Figure 21: -Effect of mosambi peel 50% ethanol extract on *S. aureus* growth at varying concentrations. Fold change ($OD_{18}h/OD_0h$) is plotted for four drying and extraction conditions.

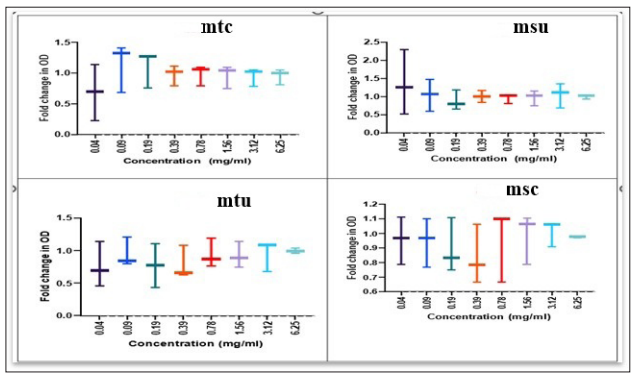

Figure 22: Effect of mosambi peel methanol:ethanol extract on *S. aureus* growth at varying concentrations. Fold change ($OD_{18}h/OD_0h$) is plotted for four drying and extraction conditions.

Conflict of Interest

None.

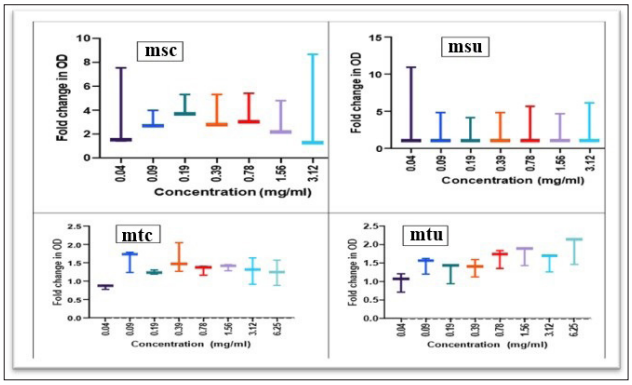

Figure 23: Effect of mosambi peel water extract on *S. aureus* growth at varying concentrations. Fold change ($OD_{18}h/OD_0h$) is plotted for four drying and extraction conditions.

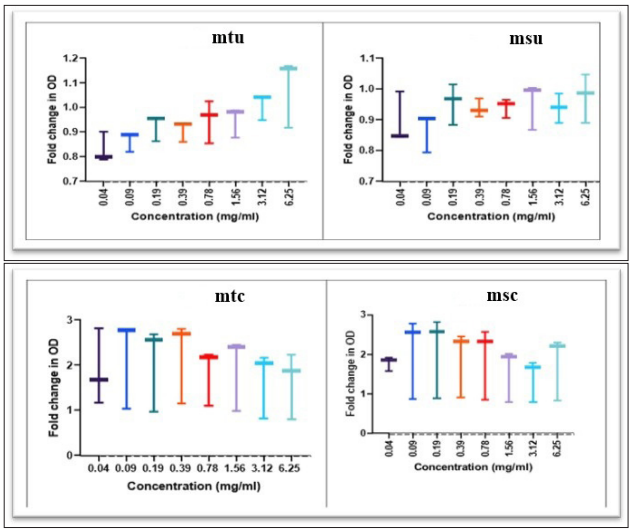

Figure 24: Effect of mosambi peel 50% methanol extract on *S. aureus* growth at varying concentrations. Fold change ($OD_{18}h/OD_0h$) is plotted for four drying and extraction conditions.

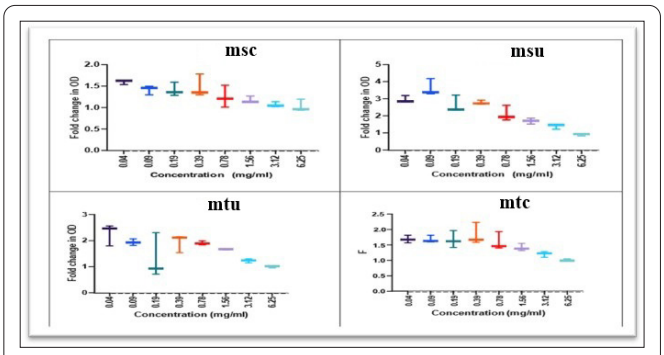

Figure 25: Effect of mosambi peel 50% methanol extract on yeast growth at varying concentrations. Fold change ($OD_{18}h/OD_0h$) is plotted for four drying and extraction conditions.

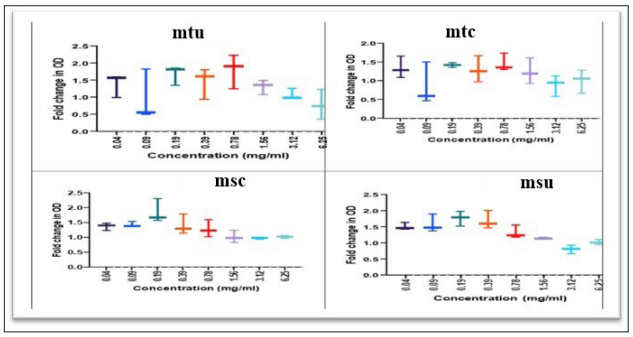

Figure 26: Effect of mosambi peel methanol:ethanol extract on yeast growth at varying concentrations. Fold change ($OD_{18}h/OD_0h$) is plotted for four drying and extraction conditions.

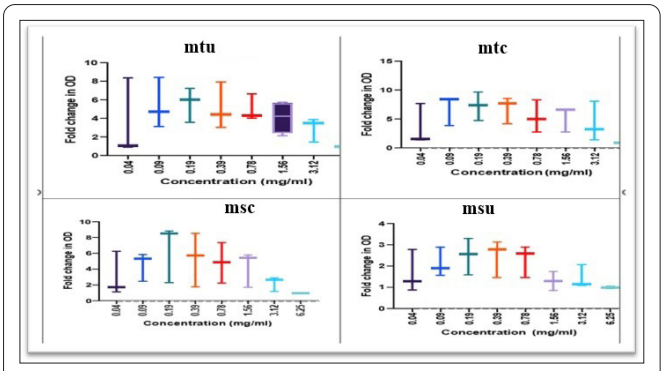

Figure 27: Effect of mosambi peel 50% ethanol extract on yeast growth at varying concentrations. Fold change ($OD_{18}h/OD_0h$) is plotted for four drying and extraction conditions.

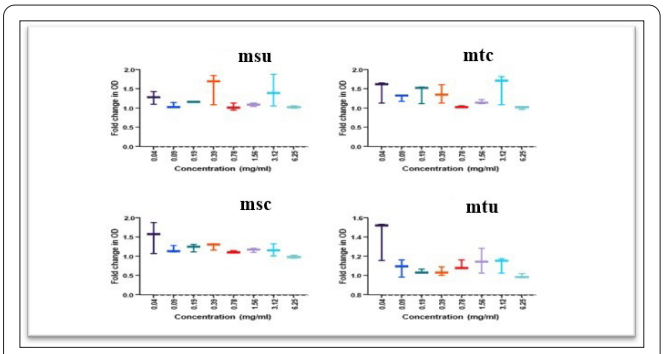

Figure 28: Effect of mosambi peel water extract on yeast growth at varying concentrations. Fold change ($OD_{18}h/OD_0h$) is plotted for four drying and extraction conditions.

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