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Anterior Skull Base Reconstruction: Does Fat Preparation Matter?

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Introduction

Autologous fat grafting for repair of skull base defects has a long history in otolaryngology and neurosurgery. In 1966 and 1969, Montgomery et al first described the use of fat grafts to prevent cerebrospinal fluid (CSF) leaks after sub-occipital and translabyrinthine approaches to resect vestibular schwannomas.1,2 Collins was then the first to describe using fat grafts after transsphenoidal hypophysectomies to fill the sella and sphenoid sinuses in 1973.3 Shortly thereafter, from 1975 to 1979, multiple accounts of using autologous fat grafts from the thigh and abdomen were reported.
after transtheroidal and transsphenoidal hypophysectomies. In current practice, autologous fat grafting has been ubiquitously adapted as a major component of skull base reconstruction.

Wigand first described the endoscopic repair of nasal CSF leaks in 1981.3 During the next three decades, alongside the advancement of endoscopic endonasal surgery, refinements in anterior skull base (ASB) reconstruction techniques have resulted in more successful endoscopic CSF leak repairs and radical tumor extirpations.6 Now, endoscopic endonasal repair is the standard of care for most ASB reconstructions.7 The goal of reconstruction is to create a watertight dural seal to effectively isolate the intracranial cavity from the sinonasal cavity and obliterate dead space.

The most commonly used free tissue grafts for ASB repair are fat, fascia, muscle, bone, and peristeum. Locoregional vascularized flaps and free tissue transfers are used for larger defects and persistent CSF leaks. Additionally, several biomaterials are often adjunctively utilized for preventing and treating CSF leaks.8–13 The choice of graft is based on the size, location, and character of the defect in addition to the presence of an intraoperative CSF leak, and other factors such as prior or future radiation treatment. Despite varying surgeon preferences and clinical scenarios, one of the most universal autologous grafts is adipose tissue, either alone or in combination with other tissues.

Although there are numerous publications reviewing methods of fat harvesting and fat processing,14–17 there is a paucity of literature in regards to fat harvesting and processing for ASB reconstruction and CSF leak repair. Based on current evidence, the majority of fat grafts for ASB repair are simply harvested and placed without further processing, either alone or as part of a multilayered closure.3,9,18 There are no studies validating whether processing of the fat prior to implantation makes any difference as compared with nonprocessed fat in ASB repair for CSF leak control.

This study was initiated to evaluate the postharvest processing of autologous fat grafts for ASB repair. After reviewing the processing techniques used for fat grafts in soft-tissue augmentation,14–17,19,20 we selected gauze sponge drying and compressing as a practical and proven method for enhancing fat graft survival. We then expanded this technique to the preparation of autologous fat grafts for ASB repair in an animal model—previously established by Nishihira and McCaffrey21—to determine whether processed fat would lead to lower rates of postoperative CSF leaks. Since autologous adipose tissue will remain an important element in skull base repair due to proven efficacy, low morbidity, and high availability, establishing whether processed fat is equally effective may result in quicker surgical times, less donor-site morbidity, and improved patient outcomes.

**Methods**

The protocol was reviewed and approved by the Institutional Animal Care and Use Committee at University of Tennessee Health Science Center. Adult male Sprague-Dawley rats were used for the study, and they were housed and cared for in the Animal Care Center. A minimum of 10 animals per group (control, wet fat, and dry fat) was anticipated to carry sufficient power in a power analysis.

For all three groups of animals, ASB defects were created in the same manner, using a protocol modified from that previously described by Nishihira and McCaffrey. Anesthesia was induced with intraperitoneal injection of ketamine and xylazine, dosed at 86/13 mg/kg. The rats were placed prone on a heating pad and under a dissecting microscope to facilitate lighting and magnification. A midline incision from the skull vertex to the nasion (≈20 mm) was performed, with subperiosteal elevation to expose the nasofrontal suture line (Fig. 1). A unilateral skull base defect was created at the nasofrontal suture line using a rotary tool (Dremel, Robert Bosch Tool Corp.) with a 2-mm burr, creating an ≈2 × 5 mm opening. The side of the defect, right or left, was alternated between animals. To ensure the skull base defect had been sufficiently created to allow passage of CSF from the cranial cavity to the nasal cavity, sinus mucosa was visualized at the anterior portion of the defect and the frontal lobe and olfactory bulb was seen at the posterior aspect.

The animals were divided among three reconstruction arms. To minimize technical bias, the reconstruction groups were interspersed throughout surgery days and not performed sequentially. Right- and left-sided defects were assigned evenly among reconstruction groups. All the surgeries were performed by a single researcher. One-third of

![Fig. 1](image-url) The nasofrontal suture line (arrow) was identified to orient an anterior skull base defect, created using a Dremel rotary tool. Left- and right-sided defects were evenly created within each group.
the animals served as the control group, in which the scalp incision was closed with 4–0 nylon suture immediately after creation of the skull base defect, and allowed to emerge from anesthesia.

The second group was the unprepared, or wet fat group. In this group, an autologous, uncompressed wet fat graft was used to fill the defect. The fat was harvested by making a 1-cm incision in the intrascapular area to remove ~1 to 2 mm³ of adipose tissue. This area was chosen because it is accessible while the animal is still in position for skull base exposure, and eliminates the need to mobilize the animal while the skull base defect is open. Additionally, in the rat model, there is a relatively greater amount of adipose tissue in the intrascapular region than in the abdominal or thigh region, as would be selected in a clinical scenario. The fat harvest site was closed with 4–0 nylon suture. The harvested fat was packed tightly into the skull base defect, and fibrin sealant (Evicel, a product of Ethicon US, LLC) was placed over the reconstruction. The skin was closed as previously described for the control group, and the animal allowed to emerge from anesthesia.

The third group was the specially prepared fat, or dry fat group. Intrascapular fat was harvested as before but a greater volume (2–3 mm³) was procured to account for compression. The fat was dried and compressed by rolling it between gauze sponges. The prepared fat was used to tightly fill the defect, sealed over with fibrin sealant, with skin closure accomplished as in the wet fat and control groups, followed by emergence from anesthesia.

Animals in all groups received subcutaneous injection of 3 mL of normal saline for postoperative hydration, and an injection of buprenorphine 0.03 mg/kg for analgesia prior to awakening. The animals were monitored for 2 to 3 hours until fully awake and taking oral feeds, after which they were transported to the animal care facility. The daily monitoring and care were performed by staff in the animal care facility. The animals were allowed to recover and heal for a minimum of 21 days; the length of time averaged 26.7 days.

After this recovery time, anesthesia was again induced with intraperitoneal ketamine/xylazine mixture as described above. The incision over the skull was carefully opened as to not disturb the reconstruction. A burr hole was created in the parietal skull using the rotary tool, away from the skull base defect, and 0.1 mL of 5% fluorescein was injected into the subarachnoid space. The animals were monitored for 2 hours under anesthesia to allow circulation of the fluorescein in the CSF, after which the animals were humanely euthanized by asphyxiation and cervical dislocation. The nasal bones were removed (▶Fig. 2) and the nasal cavities inspected for the presence or absence of fluorescein under microscopic vision.

Results
A total of 47 male adult Sprague-Dawley rats were used, with 41 animals completing the protocol. Four animals expired during induction of anesthesia, and two from the control group expired during the postoperative period. ▶Table 1 shows the characteristics of the animals in each of the

<table>
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<th>Table 1 Characteristics of each treatment group</th>
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<tr>
<td>Control (no reconstruction)</td>
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<tr>
<td>Number of animals</td>
</tr>
<tr>
<td>Preoperative weight (g)</td>
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<td>Left:Right defects</td>
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<td>Healing time (d)</td>
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<td>Weight gain (g)</td>
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Note: There were no differences between the groups, $p = 0.16$. 

Fig. 2 Fluorescein was introduced through a parietal skull burr hole (asterisk). After time for cerebrospinal fluid (CSF) circulation and subsequent euthanasia, nasal bones were removed (arrow) and the nasal cavities inspected for the presence or absence of fluorescein under microscopic vision.
reconstruction arms. The days between the creation of the
defect and fluorescein injection (healing time), averaged
25.5, 27.1, and 27.3 days, respectively, for the three
groups. Baseline weights and change in weight over the study period
were recorded. There were no differences among the groups
calculated by an analysis of variance (ANOVA).

The main outcome was the rate of persistent CSF leak, as
indicated by presence of visualized fluorescein in the nasal
cavity. The control group had a persistent CSF leak rate of
69%, the wet fat group had a leak rate of 50%, and the dry fat
group had a leak rate of 36% (►Fig. 3). The most striking
difference was observed between the dry compressed fat
animals and control nonreconstructed animals (64% vs. 31%
success, p < 0.05). While the wet fat group appeared
superior to no reconstruction (50% vs. 31% success), analysis did
not achieve significance (p = 0.15). Similarly, direct compar-
ison of wet versus dry preparation showed a higher success
rate for dry compressed fat (64% vs. 50% success), but analysis
did not achieve significance (p = 0.22).

We also looked at surgical times between the groups. As
anticipated, the average surgical times between the control
and reconstruction groups differed significantly at 15.3
minutes (control), 20.1 minutes (wet fat), and 22.6 minutes (dry
fat) (►Fig. 4). One-way ANOVA significant effect of group F(2,
40) = 35.14, p < 0.0001. Post hoc tests (Turkey) show that
each group is significantly different from one another.

Discussion

In analysis of surgical times, we found that there was a
statistically significant difference between no reconstruc-
tion, and the two types of fat reconstruction. In the clinical
setting, this difference may not occur as the fat graft harvest
can take place simultaneously. The clinically relevant graft
harvest donor sites, abdomen and thigh, are accessible
during the skull base exposure portion of the case. Simultaneous
harvest and preparation was not feasible in this animal
model.

The mechanism of skull base repair relies mainly on dead
space obliteration and mechanical reinforcement of the
primarily repaired dura against CSF pressure. For large skull
base defects, adipose tissue is often used as a bolster for a
multilayered closure. However, successful use of a fat graft
alone for large defects has been reported.14 The success of a
fat graft depends on adipocyte survival which in turn relies
on graft neovascularization and neoadipogenesis.15 A major
factor in graft survival and performance is contact with a
vascularized recipient site.16 Mashiko and Yoshimura
showed that grafted adipocytes and adipocyte-derived
stem cells (ASCs) within 100 to 300 μm of a surface exposed to
a vascularized recipient will survive. Within 2 to 4 weeks,
zones of survival and regeneration are demarcated.22 There-
fore, maximizing the number of adipocytes and ASCs avail-
able to survive and regenerate would seem to enhance
integrity of the graft over time.

In our study, we sought to determine if drying and
compressing the autologous fat graft led to a higher success
rate in ASB reconstruction. We hypothesized that by prepar-
ing the adipose tissue is such a way, it would allow the
grafting of a higher percentage of viable cells. This would in
turn lead to a higher rate of successful closure of the skull
base defect.

We defined success as no evidence of fluorescein in the
nasal cavities on final inspection after the animals had been
sacrificed. The rate of success in the control group with no
reconstruction was 31%. When wet, or unprepared fat, group
was used for the reconstruction, the success rate increased to
50%. However, when fat that had been prepared by drying
and compression was used for the reconstruction, the suc-
cess rate rose to 64%.

Conclusion

This study demonstrates that drying and compressing the fat
graft improves autologous fat reconstruction success for ASB
defects, and does not add significantly to surgical time over
nonprepared fat. Further, our results show that although wet
fat reconstruction trended toward higher success rates in the
closure of ASB reconstructions as compared with no recon-
struction, dried and compressed autologous fat grafts did
achieve statistically significant improvement. This study
validates the practice of compressing fat grafts prior to reconstruction of ASB defects.

Note
This was presented as an oral presentation at the North American Skull Base Society Annual Meeting in February 20–22, 2015.

Disclosures
None.

References